IPM for Shrubs in Southeastern U.S. Nursery Production

Volume II

Eds. Matthew Chappell, Gary W. Knox, & Giselle Fernandez

IPM for Shrubs in Southeastern U.S. Nursery Production

Volume II

Editors:
Matthew Chappell, Gary W. Knox, Giselle Fernandez

Published by the Southern Nursery IPM working group
in cooperation with the Southern Region IPM Center

July, 2017

Copyright Information:

Disclaimer
This book was developed for nursery producers. Landscapers, students, arborists, and others may find the book useful, however, all readers should exercise caution in applying pesticides mentioned in this book. Pesticides listed in this book may not be legal to use in all situations or locations. In addition, while every effort will be made to keep the book up to date, label changes may occur between editions. The label is the law. It is the pesticide user's responsibility to read and follow the label.

Acknowledgment
The authors express gratitude to the Southern Region Integrated Pest Management Center for financial support and the Bugwood Network and its contributors for the use of pest images.

Check the Southern Nursery IPM Working Group website for the current edition and updates http://wiki.bugwood.org/SNIPM and via ebook vendors.

Printed in the United States of America, Martin Printing Co., Inc., Easley, SC

ISBN: 978-0-9854998-4-6

Contributing Authors:

Matthew R. Chappell
University of Georgia
Horticulture Department
211 Hoke Smith Building
Athens, GA 30602
hortprod@uga.edu

Juang-Horng (JC) Chong
Clemson University
School of Agricultural, Forest, & Environmental Sciences
Pee Dee Research and Education Center
2200 Pocket Road
Florence, SC 29506
juanghc@clemson.edu

Jeffrey F. Derr
Virginia Polytechnic Institute and State University
Department of Plant Pathology, Physiology, & Weed Science
Hampton Roads Agricultural Research and Extension Center
1444 Diamond Springs Road
Virginia Beach, VA 23455
jderr@vt.edu

Winston C. Dunwell
University of Kentucky, Research & Education Center
Nursery Crops Development Center
P.O. Box 469
1205 Hopkinsville Street
Princeton, KY 42445
wdunwell@uky.edu

Amy Fulcher
University of Tennessee
Department of Plant Sciences
252 Ellington Plant Sciences Bldg.
2431 Joe Johnson Drive
Knoxville, TN 37996
afulcher@utk.edu

Frank A. Hale
University of Tennessee
Department of Entomology and Plant Pathology
Soil, Plant and Pest Center
5201 Marchant Drive
Nashville, TN 37211
fahale@utk.edu

Francesca Peduto Hand
The Ohio State University
Department of Plant Pathology
475C Kottman Hall
Columbus, OH 43210
hand.81@osu.edu

William E. Klingeman
University of Tennessee
Department of Plant Sciences
252 Ellington Plant Sciences Bldg.
2431 Joe Johnson Drive
Knoxville, TN 37996
wklingem@utk.edu

Gary W. Knox
University of Florida
Department of Environmental Horticulture
North Florida Research & Education Center
155 Research Road
Quincy, FL 32351
gwknox@ufl.edu

Anthony V. LeBude
North Carolina State University
Mountain Horticultural Crops Research & Extension Center
Department of Horticultural Science
455 Research Drive, NC 28759
anthony_lebude@ncsu.edu

Chris Marble
University of Florida - IFAS
Mid-Florida Research & Education Center
2725 S. Binion Road
Apopka, FL 32703-8504
marblesc@ufl.edu

Joseph C. Neal
North Carolina State University
Department of Horticultural Science
262 Kilgore Hall, Box 7609
Raleigh, NC 27695
joe_neal@ncsu.edu

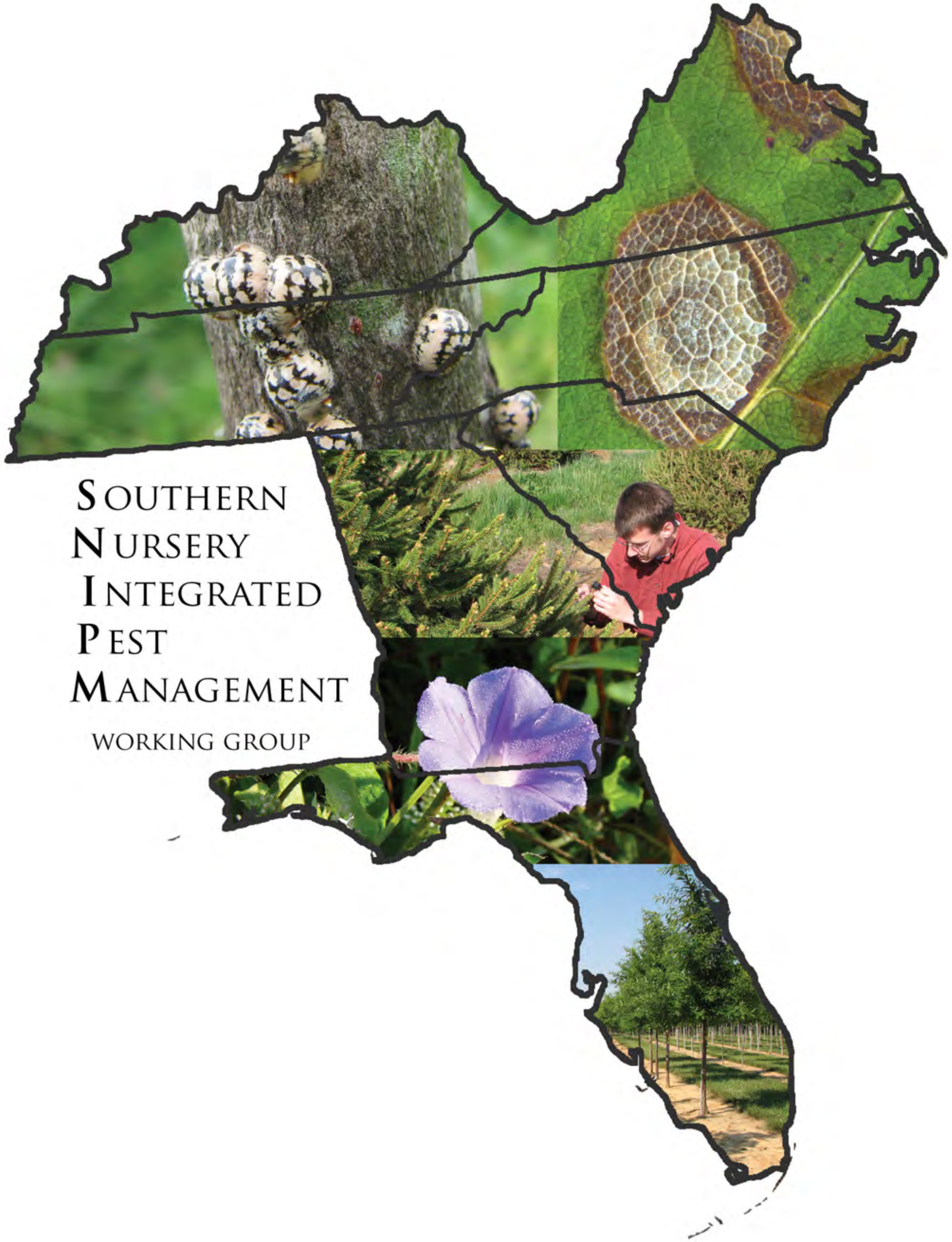
Angelia Rateike
University of Tennessee
Department of Plant Sciences
252 Ellington Plant Sciences Bldg.
2431 Joe Johnson Drive
Knoxville, TN 37996

Geoffrey M. Weaver
University of Georgia
Department of Horticulture
1111 Miller Plant Science Building
Athens, GA 30602
gmweaver@uga.edu

Sarah A. White
Clemson University
School of Agricultural, Forest, & Environmental Sciences
E-143 Poole Agricultural Center, Box 340310
Clemson, SC 29634
swhite4@clemson.edu

Jean L. Williams-Woodward
University of Georgia
Department of Plant Pathology
3313 Miller Plant Sciences Building
Athens, GA 30602
jwoodwar@uga.edu

Alan S. Windham
University of Tennessee
Department of Entomology and Plant Pathology
5201 Marchant Drive
Nashville, TN 37211
awindham@utk.edu



SOUTHERN NURSERY INTEGRATED PEST MANAGEMENT

WORKING GROUP

Preface:

IPM for Shrub in Southeastern US Nursery Production Volume II is the third book released by the Southern Nursery Integrated Pest Management Working Group (SNIPM). The first book *IPM of Select Deciduous Trees in Southeastern U.S. Nursery Production* was released in May 2012 and is available for download as chapter .pdf files at http://wiki.bugwood.org/SNIPM and as an eBook from the iTunes Bookstore https://itunes.apple.com/us/book/ipm-for-select-deciduous-trees/id541182125?mt=11. The second book *IPM for Shrub in Southeastern US Nursery Production Volume I* was released in June 2014 and can also be downloaded as chapter .pdf files at http://wiki.bugwood.org/SNIPM or from the iTunes Bookstore at https://itunes.apple.com/us/book/ipm-for-shrubs-in-southeastern/id903114207?mt=11.

The members of SNIPM gratefully acknowledge the Southern Region IPM Center in Raleigh, NC for partial support of this book. The authors are indebted to the Green Industry professionals who have served as an advisory board to SNIPM [Greg Ammon (Ammon Wholesale Nursery, KY), Jerry and Beth Blankenship (Blankenship Farms and Little Creek Nursery, TN), Pat Carey (Riverfarm Nursery, KY), Stewart Chandler (Monrovia Nursery, GA), Mark Gantt (Hefner's Nursery, NC), George Hackney (Hackney Nurseries, FL), Elliott Hallum (Mountain Creek Nursery, TN), James Hines (Hale and Hines Nursery, TN), Chris Ingle (Envirocare Landscape Management, Inc., SC), Tommy and Sharon King (King's Sunset Nursery, Inc., SC), Richard May and Lawson Taylor (May Nursery, FL), John McMakin, Jr. and Sr. (McMakin Farms, Inc., SC), Alex Neubauer (Hidden Hollow Nursery, TN), Kay Phelps (Clinton Nurseries, FL), Phillip Porter (Ray Bracken Nursery, SC), Ben Sanders (Griffith Propagation Nursery, Inc., GA), Jane Stanley (Saunders Brothers Nursery, VA), Tiffany Wells (Adcock's Nursery, NC), Gary Whitehurst (Worthington Farms, NC)]. The growers serving on this board have selflessly given their time, critical appraisal, and trust in the interest of strengthening the nursery industry in the southern United States. Growers from this advisory board as well as others provided valuable feedback on the most significant shrubs and pests on which to focus this book. The authors and editors also express their appreciation to those who provided images and plant expertise.

The SNIPM Working Group is multi-disciplinary group of Extension professionals that formed in 2008 in response to the elimination of state-designated IPM funding. It was formed to more efficiently and effectively develop and deliver educational programming to the southern U.S. nursery industry and the Extension personnel who serve the industry, as well as to better leverage competitive funding opportunities for applied nursery research and Extension initiatives. More information about SNIPM and its projects and accomplishments can be found at http://wiki.bugwood.org/SNIPM.

We hope that you, the reader, will find the information contained in this text useful and as we look forward to future products we ask that you not hesitate to contact us with suggestions. We appreciate your support.

CHAPTER 1

Hydrangea - *Hydrangea* spp.

Amy Fulcher, University of Tennessee
Jeff Derr, Virginia Tech
William Klingeman, University of Tennessee
Chris Marble, University of Florida
Joseph Neal, North Carolina State University
Alan Windham, University of Tennessee
Geoff Weaver, University of Georgia

Discussion of Japanese beetles (*Popillia japonica*) is largely reprinted from text prepared by Chappell et al. (2012); twospotted spider mite management is largely reprinted from text prepared by Fulcher et al. (2015); cottony camellia scale (*Pulvinaria floccifera*); and oystershell scale (*Lepidosaphes ulmi*) management is largely based on text prepared by Klingeman et al. (2014). Reprint of this content herein is made possible by permission of the original content authors.

SECTION 1

History, Culture and Management

Figure 1.1 Smooth hydrangea in bloom.

INTRODUCTION

1. Culture of Common Species
2. Propagation
3. Production: Fertility; Harvesting; and Flower Color Management
4. Irrigation
5. Pruning
6. Plant Growth Regulators to Control Size and Enhance Branching

Introduction

The genus Hydrangea includes evergreen or deciduous flowering shrubs for which there has been a tremendous surge in popularity in recent years. In 2007, hydrangea sales were $73,205,000 (USDA, 2009), which represents a threefold increase in value since 1997. Hydrangeas were the second most popular deciduous shrub following rose in the census. Hydrangeas commonly produced are deciduous and are native to the U.S. or Asia. The most popular species produced commercially in the southeastern U.S. are *H. arborescens, H. macrophylla, H. paniculata,* and *H. quercifolia*. There are hundreds of cultivars in existence and the number is increasing annually with new releases out of Europe and the southeastern U.S. The number of selections currently found in the U.S. trade includes *H. arborescens* (12), *H. macrophylla* (77), *H. paniculata* (32), and *H. quercifolia* (14) (Plant and Supply Locator, 2015).

The floral display of hydrangea is largely due to sterile florets, grouped into rounded or conical panicles or corymbs, that tend to be large and range from white to shades of pink, purple and blue. Conical panicles open from the base to the tip and continue to expand in length as they open. Flowers are prized for their ability to age to an antique look; both fresh cut stems and dried flowers are popular in the floral trade. Online floral companies advertise individual stems from $2.50 to $6.00 depending on cultivar and quantity. Use of several types of hydrangeas as well as remontant blooming selections of *H. macrophylla* in a landscape can create blooms for nearly the entire growing season. Most hydrangeas have yellow or no fall color, but *H. quercifolia* has striking fall color in a range of reds, oranges, and deep maroons.

Culture of Common Species

H. arborescens - Smooth Hydrangea

Smooth hydrangea is native to deciduous woodlands of New York, south to the Gulf States, and west to Iowa (USDA hardiness zones 3 to 9). Smooth hydrangea will grow well in full sun, especially in more northern parts of the U.S. with ample moisture; but does best in partial shade in the southeast. It is more tolerant of alkaline conditions than *H. macrophylla* and prefers moderate moisture levels (Dirr, 2004). It reliably flowers from Florida to Canada. Leaves (immature and mature) are more frost tolerant than those of *H. macrophylla* and *H. serrata* (Dirr, 2004). Fall color is often brown but some years a clear yellow. Smooth hydrangea bears large, white corymbs at the shoot terminals in mid-June for 6 to 8 weeks (Figure 1.1). Inflorescences of native populations consist primarily of small fertile flowers with a few large sterile flowers and thus

Figure 1.2 Mophead type *H. macrophylla* have an abundance of large, sterile flowers.

may not be ideal for breeding stock (Halcomb et al., 2013).

Figure 1.3 Lacecap type *H. macrophylla* have an outer ring of large sterile flowers and an inner ring of small fertile flowers.

H. macrophylla - Bigleaf Hydrangea
Bigleaf hydrangea is native to Japan and Korea. Plants usually grow to 3 to 6 feet (0.9 to 1.8 m) in height, but can grow to 10 feet (3 m) with width becoming equal to or greater than plant height. Bigleaf hydrangea is considered hardy in USDA Hardiness Zones 6 to 9. Many cultivars are not hardy, and thus, cultivar selection must be made carefully to match the location. Cold injury to the flower buds and improper pruning time (pruning after bud-set) are the usual reasons for flower failure in the landscape. Bigleaf hydrangeas form flower buds during the fall months. Because they bloom on the previous year's wood, they are very susceptible to low temperature injury. Flower color ranges from pink to lavender to blue depending on cultivar and soil chemical properties. There are also a few white-flowered cultivars whose flowers may turn very pale pink or blue as they age. Two horticultural groups of *H. macrophylla* are recognized. Mopheads or hortensias have large, round corymbs consisting primarily of large, sterile flowers (Figure 1.2). Lacecaps have an inflorescence consisting of an outer ring of large sterile flowers and an inner ring of small fertile flowers (Figure 1.3). Some selections are remontant and have a repeat bloom period. Bailey Nurseries' remontant selection, The Original Endless Summer™, and marketing efforts related to it are credited, at least partially, with the surge in hydrangea popularity.

Figure 1.4 Hardy hydrangea can have very large panicles up to 18 inches in length, or small ones such as these Little Lime™.

H. paniculata – Hardy Hydrangea
Hardy hydrangea is native to Japan and eastern and southern China. It is cold hardy to USDA hardiness zone 4, making it one of the most cold hardy hydrangea species. Hardy hydrangea can grow to 10 to 20 feet (3 to 6.1 m) in height and spread and can even be trained as a small tree. Some selections have a very coarse texture making them somewhat difficult to place in the landscape but very easy to grow. Most newer cultivars do not grow as large as the species and have a more refined texture. Beginning, in part, with the introduction of the cultivar 'Limelight' in 2002, hardy hydrangea's popularity surged.

Figure 1.5 Oakleaf hydrangea have panicles composed of sterile and fertile flowers.

Hardy hydrangea can grow under full sun to light shade, is pH adaptable, and prefers good drainage. It can tolerate urban environments and drier soil conditions (once established) better than most other hydrangeas in the trade but will wilt during drought and high temperatures (Dirr, 2004). Hardy hydrangea flowers in mid-to late-summer, beginning in mid-July and lasting until October. The inflorescences are large panicles, 6 to 18 inch (15.3 to 45.7 cm) long, that contain both fertile and sterile flowers (Figure 1.4). The flowers often cause the branches to bow over in a graceful arch when in container production (Dirr, 2004). Flowers open white and often age to a pale to medium pink. However, the change to pink is more consistent in cooler climates and cultivars that can age to pinkish purple colors may not do so in warmer areas.

H. quercifolia - Oakleaf Hydrangea
Oakleaf hydrangea is native to the southeastern U.S. and is hardy to USDA hardiness zone 5. Most cultivars reach 6 to 8 feet (1.8 to 2.7 m) in height. Plant spread is greater than height as plants sucker from roots. Like most hydrangeas, oakleaf hydrangea benefits from light shade in the landscape but will tolerate full sun if given ample moisture. They are subject to root rot if over-irrigated or placed in poorly drained soil. Oakleaf hydrangea flowers in early to mid-summer. Flowers are large, white to cream-colored panicles that turn pink to rose as they age (Figure 1.5). Inflorescences are composed of a mixture of large sterile and small fertile flowers.

Propagation
Hydrangea species are relatively easy to propagate by seed or by cuttings. Tissue culture is being used by some propagation nurseries to increase numbers of new cultivars. A detailed protocol for *H. macrophylla* tissue culture is available (Abou Dahab, 2007; Ruffoni et al., 2013). Seed propagation is desirable when looking for novel characteristics such as flower color or compact branching structure.

Seeds are collected in late fall and winter. No stratification is needed but a one to two-month cold period may increase germination uniformity and speed. Seeds are surface sown and misted to keep moist. Germination occurs within 30 days, often sooner. Specific notes on seed germination of select species follows. *Hydrangea arborescens* and *H. macrophylla* will germinate in two to three weeks after sowing on milled sphagnum. Seeds are small; take care not to sow too heavily (Dirr and Heuser, 2006). *Hydrangea paniculata* seeds require no pretreatment; plants will flower the same year seeds are sown. *Hydrangea quercifolia* seeds can be direct sown. Fresh *H. quercifolia* seeds germinate at high percentages two weeks after sowing. For detailed seed propagation information, consult Dirr, 2004.

Cutting propagation is commonly used to propagate hydrangeas for nursery production. In general, softwood cuttings collected earlier in the season (May) will yield more robust, faster growing cuttings than those collected later in the summer (July). But cuttings taken in either time frame root readily (Dirr, 2004). Single node cuttings are sufficient and allow for more cuttings per length of stem. Hydrangeas have large leaves and they should be cut in half, or more for *H. quercifolia*, to prevent them from blocking mist during rooting.

Figure 1.6 Hydrangeas root readily from cuttings (*H. macrophylla* shown).

Hydrangea arborescens is easy to propagate by softwood cuttings using 1,000 ppm indole-3-butyric acid (IBA), solution or talc (Dirr and Heuser, 2006). They often have 100% rooting within 2 to 3 weeks. Although *H. arborescens* roots readily through the summer months and into September (Raulston, 1995), it is recommended to root them early in the season to promote growth and establishment as a liner to increase overwintering survival (Dirr, 2004).

Hydrangea macrophylla is easy to propagate by softwood, semi-hardwood, or hardwood cuttings, although softwood cuttings are most common in the trade (Figure 1.6). A one part peat: three parts perlite substrate with intermittent mist is recommended for softwood cuttings. Terminal softwood cuttings treated with 1,000 ppm IBA, solution or talc, rooted in 3 to 5 weeks at near 100% (Dirr and Heuser, 2006). Success near 100% is reported for untreated softwood cuttings (Dirr, 2009). *Hydrangea macrophylla* can also be propagated by semi-hardwood cuttings using a 5 second dip of potassium salt of indole-3-butyric acid (K-IBA) at 500-1,500 ppm (Paul and Jhon, 1992) and by hardwood cuttings direct stuck outdoors in pine bark substrate (Dirr, 2004) but these are less commonly used methods. Hardwood cuttings rooted at approximately 50% with some remontant cultivars rooting at 80% by late spring the following year (Dirr, 2004). Rooting leaf bud cuttings and single bud cuttings successfully has also been reported (Dirr, 2004; Dirr and Heuser, 2006). For alternatives to K-IBA see Boyer et al. (2013).

Figure 1.7 Oakleaf hydrangea rooted cuttings should not be kept too wet or disturbed in order to increase survival.

Terminal softwood cuttings of *H. paniculata* will root in a well drained sand:peat substrate in approximately 4 weeks with 1,000 ppm IBA quick dip or talc (Dirr and Heuser, 2006). Untreated cuttings do not root in high percentages. This species can be rooted as late as September. Semi-hardwood and hardwood cuttings also root. Hardwood cuttings require a rooting hormone.

Hydrangea quercifolia is the most difficult of these species to root. Terminal cuttings collected as green wood or firm wood treated with 4,000 ppm IBA quick dip or talc root well in 5 to 6 weeks (Dirr and Heuser, 2006). Untreated cuttings do not root in high percentages (Dirr and Heuser, 2006). A well-drained propagation substrate such as 100% perlite or three parts perlite to one part peat and intermittent mist must be used. Following rooting, take precautions not to over-irrigate. Rooted cuttings should not be disturbed in order to increase survival (Dirr and Heuser, 2006) (Figure 1.7). Additionally, rooted liners must be overwintered in a cool environment, not a greenhouse, to increase the survival rate. Semi-hardwood cuttings can be treated with 2,500 ppm IBA.

Production

H. arborescens — Smooth Hydrangea

Rooted cuttings are typically potted into 1-gal (3.7 L) or 3-gal (11.4 L) containers. Containers should be well spaced to support branch development and lateral growth; 3-gal (11.4 L) containers are often placed on 2 ft (0.6 m) spacing. Plants in a 3-gal (11.4 L) container can be bumped to a 5-gal (18.9 L) container, which is the largest size of hydrangea that is typically marketed. Plants can be grown without shade, particularly in more northern environments. Stems are often not branched and can be stiff but weak and thus insufficient to support the large, heavy corymbs characteristic of

Figure 1.8 Smooth hydrangea often has weak stems and needs staked, pruned, and/or plant growth regulators to remain upright.

Figure 1.9 High temperatures can prevent *Hydrangea macrophylla* from blooming and cause distorted vegetative growth.

cultivated varieties, necessitating staking even during production (Figure 1.8). Inflorescences of native populations consist primarily of small fertile flowers with a few large sterile flowers.

H. macrophylla - Bigleaf Hydrangea
Typically, liners rooted in trays or 2-inch (5.1 cm) containers are transplanted into a pine bark-based substrate. Quart liners of 'Nikko Blue' and similar cultivars transplanted into 5-gal pots (18.9 L) can finish by fall (Bir, 2000). In a study that included pine bark, peat, and less traditional container substrate components, the optimum container substrate was three parts peat: one part bark followed by slag wool (Artetxe et al., 1997). Additionally, plant biomass increased as container size increased from 0.24 gal (900 cm^3) to 1.2 gal (4,600 cm^3). Bigleaf hydrangea is normally grown under shade in the southeastern U.S.

High summer temperatures, >86º F (30º C) during the daytime, are known to interfere with normal flower and vegetative development. Bailey and Hammer (1990) and before them Weiler and Lopes (1974) describe plants that predominately lack flowers, and when present, are malformed. Leaves are thick and elongated with a reduced width and internodes are compressed at the shoot tips (Figure 1.9). Additionally, Bailey (1989) describes high night temperatures at or above 80º F (27º C) as potentially damaging previously formed floral primordia, delaying, distorting or preventing new floral primordial development, and/or causing vegetative distortion. The severe southeastern U.S. heat wave of June 2015 appears to have been sufficient to prevent many *H. macrophylla* selections from blooming while in production and disrupted sales at least for some producers (Figure 1.10). Root restriction in container production can also reduce flower initiation (Yeh and Chiang, 2001).

Clark and Zheng (2015) found that in Ontario, Canada 1.3 to 2.3 pounds per yd^3 (0.75 to 1.35 $kg{\cdot}mL^{-3}$) N was the optimal rate of 19N-2.6P-10.8K plus minors, 8-9 month controlled release fertilizer (CRF) for bigleaf hydrangea in 2-gal (7.6 L) containers and 0.8 to 2.9 pounds per yd^3 (0.8 to 1.7 $kg{\cdot}mL^{-3}$) N was the optimal range of 16N-2.6P-10K (5-6 month longevity) for *H. paniculata* 'Bombshell' (Agro and Zheng, 2014). In a study with a variety of mulches (geotextile discs, coco discs, plastic discs, hazelnut shells, sawdust, Biotop, and crumb rubber) applied to container grown plants, hydrangea growth, quality and foliar color were better when fertilizer was placed under the mulch (Altland and Lanthier, 2007).

Low phosphorus levels have the potential to prevent excessive algae growth in reclaimed water and have the added benefit of conserving a natural resource. Phosphorus levels of just 0.29 pounds per yd^3 (0.17 $kg{\cdot}mL^{-3}$) P_2O_5 were sufficient to produce desired growth and quality (Halcomb et al., 2013). Rates as high as 0.53 pounds per yd^3 (0.31 $kg{\cdot}mL^{-3}$) P_2O_5 are commonly used and appear to be excessive (Halcomb et al., 2013). See Flower Color Management Section for further details.

Yellow foliage, chlorosis, and weak root systems commonly occur during nursery production of *H. macrophylla*, particularly in midsummer. This problem has been attributed to overwatering, root rot, and low iron uptake (Midcap, 2004; Midcap and Bilderback, 2002). A composted bark, mini nugget substrate with desirable physical properties (air space 39%, container capacity 53%, available water 21%, unavailable water 32%, bulk density 0.2 $g{\cdot}cm^3$, and cation exchange 8.1) produced the best growth of five different substrates tested for 1-gal (3.7 L) and 3-gal (11.4 L) containers (Midcap and Bilderback, 2002). The nine parts bark to one part kaolin clay substrate supported the least growth.

When being field produced, rooted cuttings should be at least 4 to 6 inches (10.2 to 15.2 cm) tall when lined out. Bigleaf hydrangea requires a site where water never stands, but can be grown on poorer soil than many woody nursery crops (Halcomb et al., 2013). *Hydrangea macrophylla* benefits from supplemental water during dry periods. When the temperature is in the upper 80s ºF or higher, plants in full sun can wilt, even when provided with adequate moisture.

Figure 1.10 The extreme heat of June 2015 appears to have damaged reblooming *Hydrangea macrophylla*, delaying blooming until fall and then blooming occurred sporadically and on a small percentage of plants. Temperatures across the southeast were above 100°F on June 29-30, 2015.

H. paniculata — Hardy Hydrangea
Hydrangea paniculata is a vigorous species even in container production. Medium rates of fertilizer are sufficient. A 2.25 inch (5.7 cm) liner will finish a 3-gal (11.4 L) container in 4 to 5 months. Minimal care is needed while in production. Pruning twice a season and/or plant growth regulators (PGRs) are typically used in an effort to increase branch number, flowers, and plant density. In a non-scientific study, consumers preferred more but smaller flowers when compared to plants with fewer but much larger inflorescences. Hardy hydrangea requires ample water while in container production. It can be produced in full sun, but partial shade is used further south.

H. quercifolia — Oakleaf Hydrangea
Oakleaf hydrangea (Figure 1.11) is not as easy to grow in a container as the previously mentioned hydrangea species. The following production schedule has been used successfully (adapted from Halcomb et al., 2013): Surface sow seeds into trays in September-November. Transplant seedlings into 36-cell flats when the seedlings develop the second pair of true leaves. Keep seedlings on the dry side. Transplant seedlings into jumbo 5.25-inch (13.3 cm) pots; pinching to leave two nodes to force branching. Transplant into 3-gal (11.4 L) containers when the roots fill the 5.25-inch (13.3 cm) containers. Prune any vigorous lateral branches that are stimulated after the pinching. Plants partially fill a 3-gal (11.4 L) container by mid-late April, but are not large enough to sell. If the seedlings go into 1-gal (3.7 L) containers from the 36-cell pack and they are grown with heat in a greenhouse all winter, 1-gal (3.7) containers can be ready to sell by late April. Transplanting the seedling straight into a 1-gal (3.7 L) container or the 36-cell pack straight to a 3-gal (11.4 L) container can lead to the small plants staying too wet and developing root rot.

Figure 1.11 Oakleaf hydrangea (*Hydrangea quercifolia*) have a distinctive leaf shape compared to other species in the genus.

In studies at the UT campus in Knoxville, individual 4-inch (10.2 cm) 'Alice' liners transplanted into 3-gal (11.4 L) containers were ready for retail sales in 4 months. Plants tolerate full sun in production if given adequate irrigation. Cole et al. (2013) reported dieback after shearing, and this has been observed in Tennessee. It was not clear if dieback was because the plants were using much less water than other plants in the irrigation zone and were thus over watered or some other cause. Oakleaf hydrangea appears to be the least "water loving" hydrangea species. Air pruning containers have been successfully employed in nurseries to reduce the chance that plants are too wet. This can be an especially helpful technique when oakleaf hydrangea must be grouped in an irrigation zone with other plants that require more water. To conserve water, plants are produced in the same irrigation zone as plants with similar water needs or in their own zone.

Fertility
Hydrangeas grow best with a soil pH of 5.0 to 6.5. A medium level of phosphorus and potassium is generally desirable. However, for *H. macrophylla*, lower phosphorus is desirable as it binds with aluminum even at lower pHs. Specialty fertilizers are available to aid in blueing of bigleaf hydrangeas (See Flower Color Management). For field production, test soil early enough so that any lime, phosphate, or potash can be broadcast prior to planting. Signs of iron, and to a lesser extent, boron deficiency may show at a higher pH. A typical recommendation for all shrubs is no more than 50 pounds (22.7 kg) of actual nitrogen per acre applied in late February and again in late June (agricultural grade nitrogen). It is not always economical to broadcast fertilizer after the crop is planted. The per acre rate can be used for side dressing, whether done by machine or hand. During container production, medium to high rates of fertilizer are often used but medium appears to be sufficient. Excessive nitrogen rates can lead to excessive foliage while reducing flower number. Fertilizer should be applied when foliage is dry so as to minimize the chance of damage to leaves, flowers, and buds.

Harvesting
Hydrangeas are commonly sold when they are 24 to 48-in (61-122 cm) tall. Field-grown hydrangeas are generally a 3-year crop, depending on species, soil type, fertility, moisture, growth rate, pruning, etc. (Figure 1.12).

Figure 1.12 Field grown oakleaf hydrangeas are typically a 3-year crop.

Flower Color Management
Bigleaf hydrangea is popular in part because some cultivars have blue flowers (sepals), and flower color can be manipulated to produce a range of colors from pink to lavender to blue. The color intensity is determined by a cultivar's inherent ability to produce blue pigment, aluminum availability, which in turn is based on the soil pH, and the cultivar's ability to take up aluminum. When the soil/substrate pH is acidic (4.5 to 5.5), the color can be expected to be blue because aluminum is generally highly available in a low soil pH. A soil/substrate pH of 6.5 or greater results in a pink flower color. When the soil/substrate pH ranges from 5.5 to 6.5, flowers may be pink, blue or lavender, or a mixture of pink and blue flowers may be present on the same plant. This is highly dependent upon cultivar. The flower color is not permanent. There are a few cultivars, such as 'Pia', 'Masja' (McNiel et al., 2007), 'Alpengluhen' ('Glowing Embers'), and 'Todi' that do not turn blue regardless of soil pH or have several colors on an individual inflorescence. At a low pH, these flowers may turn an unattractive muddy-red.

Flower Color Management in Containers
Nursery growers report they sell on average 10 blue hydrangeas for every pink one, so controlling the color is crucial (Kim Holden, Holden Nursery, personal communication). To produce a blue flowered bigleaf hydrangea in containers (Figure 1.13), the pH must be managed very closely to ensure aluminum remains available; as there are several variables that can influence substrate pH. These variables include substrate components, amount of lime in the substrate, how long ago the plants were potted, the quality of irrigation water, the amount of rainfall and irrigation to which the plants have been exposed, and the source and rate of fertilizer. Testing the water supply is crucial to gain the necessary information on water pH and alkalinity.

The addition of aluminum sulfate to the substrate is likely required as soilless substrates have little or no aluminum. The aluminum in aluminum sulfate lowers the pH, making the aluminum available to the plant. If the substrate pH is higher than 5.5, apply aluminum sulfate before the flower buds form in order to increase the blueness of the flower color. Too much phosphorus can make the aluminum less available so a low phosphorus fertilizer may be warranted. However, too much aluminum can damage or kill roots, resulting in plant stunting, leaves dropping, and smaller flowers aborting. Plant death can also occur, therefore use of aluminum sulfate must be done carefully and on a small scale until the grower becomes familiar with how the plants will respond in their individual production system. Not only can too much aluminum damage crops but so can too low of pH.

Figure 1.13 Soil pH, in addition to nutrient availability, can dictate color of bloom on some *Hydrangea macrophylla* cultivars. Color can range from pink to blue.

Blom and Piott (1992) examined flower color of *H. macrophylla* forced into bloom in greenhouse production and determined that about 0.5 ounces (13.3 g) of aluminum sulfate per 6-inch (15.2 cm) diameter container was needed. As a rough guide of application rates for the nursery, apply 1 tablespoon (11.4 g) of aluminum sulfate per 3-gal (11.3 L) container, record results, and modify as needed. Make applications when plants are about 4 inches (10.2 cm) tall and again 2 weeks later. Repeated drenches are a common strategy to deliver aluminum but are labor intensive. Drenches must be repeated because the container substrate will not retain the aluminum. Alternatives to drenching with aluminum sulfate exist and may be less labor intensive. Handrick (1997) found that including 10% (v/v) kaolite, a calcined clay mineral, in the potting substrate increased aluminum and sepal blueness. Plants grown in 10-20% zeolite (mined from volcanic rocks and ash layers reacted with alkaline groundwater) precharged with aluminum produced blue sepals when it was the only source of aluminum (Opena and Williams, 2002). Incorporating 30-40 percent zeolite reduced growth. Even at higher pH values, adding calcined clay and aluminum sulfate caused blue sepals. Increasing rates of thermally activated kaolin-clays (Pozzolan) and/or aluminum increased blue coloration and consumer desirability ratings, both of which were best when aluminum sulfate and Pozzolan were used together (Stoven and Owen, 2008).

Figure 1.14 Mineral soils, unlike soilless substrates used in container production, typically have sufficient levels of aluminum and require little to no amendment to achieve blue flower color in *Hydrangea macrophylla*. Sandy soils with a high pH may require acidification and/or the addition of aluminum to enhance blue flower color.

Flower Color Management in Field Production
As opposed to soilless substrates used in container production, mineral soils normally have sufficient levels of aluminum (Figure 1.14). Therefore, elemental sulfur rather than aluminum sulfate is recommended. The sulfur is necessary to lower the pH to 4.5-5.5, so that the naturally occurring aluminum is available to plants. Because aluminum can become toxic at a low pH, additional aluminum as would be supplied by aluminum sulfate is not desirable. Before planting any crop or adjusting the pH, a soil test is necessary. Field-grown bigleaf hydrangeas can be induced to bloom blue by lowering the soil pH with 90% sulfur several months in advance of bud set (6 months if the pH is high). However, lowering the pH rapidly with a large amount of sulfur can cause the soil pH to go below 4.0, which can damage or kill roots.

Irrigation
Hydrangeas are considered high water users, with *H. macrophylla* generally considered on the higher end, readily wilting when dry or heat stressed, and *H. quercifolia* on the lower end. Several experiments have been conducted in recent years that help quantify how much water hydrangea species need (Figure 1.15). The data from these studies can be helpful in refining irrigation scheduling for hydrangea.

Warsaw et al. (2009) found no growth differences for *H. arborescens* 'Dardom' regardless of whether partial or full daily water replacement was employed. However, plants had a substantial reduction in water use with partial replacement irrigation schedules. In a Michigan study, *H. arborescens* 'Abetwo' used approximately half the water that 'Limelight' used (Pershey, 2014). Pershey (2014) developed a seasonal crop coefficient for *H. arborescens* 'Abetwo', 3.51, which is approximately half the crop coefficient of *H. paniculata* 'Limelight'.

Hydrangea macrophylla is considered a heavy water user and wilts easily under water deficit, however differences exist among selections. Average daily water use of 'Fasan' [7.8 fl. oz. (231 mL) per day] varied from that of 'Pia' [7.0 fl. oz. (207 mL) per day] when grown in 1.8-gal (7 L) container outdoor under 40% shade (Chappell et al., 2011). However, both species used approximately 4.5-5.0 gal (17-19 L) of water over a 2.5-month period. Additionally, opportunities exist to refine irrigation applications, as the reputation of a heavy water user seems to promote over-irrigation of this species. 'Mini Penny' irrigated at 20% volumetric water content (VWC; considered 'dry' by many growers) used half or less the amount water of the nursery standard irrigation regime yet had no reduction in growth (van Iersel et al., 2009b). Nutrient leaching was also reduced by reducing irrigation volume. Water uptake of some selections does seem to be restricted to higher VWC compared at least to some species, which may partially explain its reputation as a heavy water user; water uptake by 'Fasan' decreased at 28% VWC and ceased at 16% VWC, a higher VWC than *Gardenia jasminoides* 'Radicans' (O'Meara et al., 2014). Daily light integral (the amount of light that a crop receives) is an accurate predictor of 'Pia' and 'Fasan' daily water use (O'Meara et al., 2013). This indicates those plants grown under shade will require less irrigation compared to plants grown in full sun.

Container substrate and plant moisture content can be regulated through irrigation management, in turn controlling growth and in some cases reducing or eliminating the need for PGRs. This has been most commonly reported in *H. macrophylla.* For 'Hermann Dienemann', 'Nymphe', and 'Renate Steiniger', cool morning temperatures, reduced irrigation [2.8 fl. oz. (82 ml) per 6-inch pot per day], and the PGR daminocide all reduced shoot length similarly (Roeber and Haas, 1997). Increasing the irrigation volume [irrigating with 4.4 fl. oz. (130 ml) or approximately 3.7 fl. oz. (110 ml) per 6-inch pot per day] earlier in production increased height, plant width, and leaf area. Conversely, increased irrigation at the end of the production cycle did not lead to greater shoot growth but did improve inflorescence development (Roeber and Hass, 1997). 'Leuchtfeuer' produced in white peat:composted bark 0.4 to 0.60 inch (10-15 mm), 2:3 mix and subjected to a water-limiting irrigation regime had reduced leaf area, height, and flower size, but quality and compactness were increased (Morel, 2001). In a study with a variety of mulches (geotextile discs, coco discs, plastic discs, hazelnut shells, sawdust, Biotop, and crumb rubber) applied to container grown plants, there was no appreciable difference in water use of mulched and non-mulched 'Fasan' and 'Endless Summer', suggesting plant transpiration, and not substrate evaporation, is the greater component of daily water use (Altland and Lanthier, 2007).

Figure 1.15 Research conducted by the University of Tennessee in partnership with Holden Nursery aims to determine optimal irrigation rates for container-produced *Hydrangea macrophylla*.

In an Oregon study, *H. macrophylla* 'Nikko Blue' used twice as much water as *Acer palmatum* var. *atropurpureum* and had the greatest increase in crop coefficient of all species in the study (Regan, 1994). 'Fasan', being a higher water user, had a greater crop coefficient than 'Pia' (O'Meara et al., 2013). Shade cloth decreased crop coefficients substantially.

Hydrangea macrophylla irrigated with reclaimed water with a salt level of 5.65 $dS \cdot m^{-1}$ had smaller flowers and leaves and were smaller overall, 70% by dry weight. Additionally, they had delayed flowering and altered flower color (Miralles et al., 2013). The reclaimed water also caused marginal necrosis that rendered the plants unsalable.

Hydrangea paniculata is considered more tolerant of dry conditions than *H. macrophylla* but is a high water user and will take advantage of abundant water, unlike *H. quercifolia*. Under 100% daily water replacement regimes, *H. paniculata* 'Limelight' used more water than any other irrigation treatment-species combination, approximately 0.7 in [(18 mm) per container per day) approximately twice as much as *H. arborescens* (Pershey, 2014). Irrigation regimes that supplied less than the daily water use led to smaller plants for 'Limelight' whereas other shrub species in the study were not smaller (Pershey, 2014). However, *H. paniculata* 'Unique' had higher water use efficiencies under irrigation regimes supplying partial daily water use and had no difference in growth compared to replacing 100% of water lost, suggesting irrigation volume could be further reduced without a growth reduction (Warsaw et al., 2009). Additionally, 'Unique' may be a viable selection where *H. paniculata* is desired but persistent water shortages exist. Pershey (2014) developed seasonal crop coefficients for *H. paniculata* 'Limelight' (6.07 in 2009 and 5.46 in 2010).

When in the same irrigation zone as more water loving plants, *H. quercifolia* can be potted in containers with porous sidewalls to help prevent the roots from staying too wet. In a study comparing daily water use with on-demand irrigation schedules, plants irrigated according to the on-demand schedule would receive 0.31 gal (1,197 mL) and 0.12 gal (463 mL) of water on average per day for 3-gal (11.4 L) and 1-gal (3.7 L) containers, respectively (Hagen et al., 2014). Both the on-demand and daily water use irrigation schedules used less water than a 1 inch (25.4 mm) per day conventional irrigation schedule. In 3-gal (11.4 L) containers, water use was reduced by 63 and 56% for on-demand and daily water use, respectively, and 57 and 36% for on-demand and daily water use, respectively, in 1-gal (3.7 L) containers.

Drenches of synthetic ABA increased the time between irrigation events for *H. macrophylla* before wilting, which increased with increasing concentration to 1,000 ppm (van Iersel et al., 2009a).

In a 2-year study including *H. arborescens* 'Annabelle', *H. quercifolia* 'Alice', and several *H. paniculata* cultivars, irrigated plants generally bloomed earlier or in a few cases at the same time as plants without irrigation (Dunwell et al., 2001a). Irrigation hastened flowering by about 10 days for *H. paniculata* 'Kyushu', 'Tardiva', and 'White Moth' (Dunwell et al., 2001).

Hydrangea paniculata 'Grandiflora' irrigated with water containing 2.4 ppm free chlorine had no damage until approximately day 60 of using this water source, when 5% of flowers and matured leaves exhibited some symptoms but were not rendered unmarketable (Cayanan et al., 2009). At approximately 75 days, new and more severe damage was present (Cayanan et al., 2009).

Pruning

Hydrangea arborescens (smooth hydrangea) will need to be pruned multiple times each season during production to produce full, densely branched plants that ultimately produce many blooms. Avoid pruning in the spring, the season of sale, as that will remove or at least delay significantly the floral display.

Hydrangea macrophylla growth can become rank and require multiple prunings per season (Midcap, 2004) (Figure 1.16). This species generally flowers on old wood, so it should not be pruned after bud set in late fall through bloom the following season, or the buds will be pruned away. If drastic pruning is needed, such as to reduce size, do it right after flowering. Remontant bloomers, those that bloom throughout the season on new growth, have more flexibility concerning pruning. In a study that examined pruning at 2 to 3 inches (5.1 to 7.6 cm) above substrate level (rejuvenation pruning), pruning halfway back, i.e., to the previous years' growth, and no pruning, bigleaf hydrangea in 3-gal (11.4 L) containers had more blooms when pruned halfway back, 29% more than when the plants weren't pruned (Conwell et al., 2002). Bloom number for rejuvenation pruning was not different from unpruned controls.

Figure 1.16 Often, *Hydrangea spp*. require several pruning events to develop a salable plant.

Hydrangea paniculata flowers on new wood so it can be pruned in winter or early spring. For field production, cut back to within 6 inches of the ground in February-March after first growing season and after second growing season if not sold. Pruning plants in container production can lead to different results than those for hydrangeas planted in the field. Plants can be pruned immediately following blooming and may bloom again (Bir, 2000). Renewal pruning, removing 1/3 of the oldest stems to ground level, can help stimulate new growth. Pruning practices can increase flower size (Dirr, 2004); although, in replicated studies hand pruned 'Limelight' and Little Lime in 3-gal (11.4 L) containers had fewer and smaller flowers than water and plant growth regulator-treated plants (Cochran and Fulcher, 2013; Cochran et al., 2013).

Hydrangea paniculata can also be trained to a tree form ("standard"), unlike other hydrangeas (Figure 1.17). Typically, large cultivars are selected for standard forms. This training should begin at an early age in the nursery and will require a stake. Allowing small branches to temporarily remain on the trunk will help develop caliper. Be sure to remove temporary branches regularly, before they exceed pencil size diameter. To develop the tree form, select and stake a single strong stem and remove all other competing growth. Once the stem reaches the desired height, cut it back using a heading back cut to stimulate branching. Prune out the tips of new growth repetitively to develop a dense, bushy canopy. This can be done by cutting all top growth back to the same height or pruning the topmost branches back to slightly different heights allowing the branches to emerge from a short section of the trunk rather than one point. While more time consuming, a stronger standard can be produced by the latter technique (Gilman, 2012). To maintain the tree form, remove

Figure 1.17 *Hydrangea paniculata* can be trained as a standard.

branches that develop from the trunk several times throughout the year and cut the canopy back periodically to preserve the dense, rounded shape.

Hydrangea quercifolia flowers on old wood so should be pruned after flowering, if necessary. Pruning late fall through bloom time the following year will remove the buds. *Hydrangea quercifolia* can have a very undesirable, asymmetrical growth habit, with most branches lying over the side of the container. However, newer releases such as the USDA releases 'Ruby Slippers' and 'Munchkin' are more compact and symmetrical in their growth habit. The typical branch architecture is highly undesirable as it takes up more space in production and transportation and doesn't display well in garden centers. In a study comparing tissue culture and cutting propagation, 'Alice' oakleaf hydrangea plants propagated by tissue culture generally had greater quality rating and more branches (either long or short branches) without pruning (Cochran et al., 2014). Cutting-propagated plants needed to be pruned to achieve the same quality. As with all hydrangeas, keep plants well spaced.

Plant Growth Regulators to Control Size and Enhance Branching

Commercially available plant growth regulators are typically used to induce branching among nursery crops and include ethephon, an ethylene generator, chemical pinching agents such as dikegulac sodium, a DNA synthesis inhibitor, or cytokinin, a synthetic compound that supplements naturally occurring growth hormones that promote branching. Other PGRs that function as growth retardants, i.e., gibberellic acid inhibitors, are used on *H. macrophylla*. In Europe, where significant hydrangea breeding occurs, many PGRs have been deemed unsafe for workers and are no longer used in the trade. Breeders are selecting for shorter and more compact plants in order to eliminate the need for PGRs.

To control growth of *H. arborescens* 'Invincibelle® Spirit', daminozide at 5,000-7,000 ppm is recommended during production (Anonymous, 2010). Recommendations follow: a minimum of three foliar applications approximately 10-20 days apart are applied beginning when the plants are small. Drenches are not recommended. Plants that are already at the market dictated height are not good candidates for PGRs. It is not advised to prune before application. When the leaves in the apical meristem begin to point upward, the plants are growing out of the application. When the leaves lay flat, the PGR is active. As with all PGR applications, environmental and cultural conditions such as fertilizer levels, light, humidity, and temperature can impact the efficacy of the PGR.

Plant growth regulators are often used in nursery production of *H. macrophylla* to control growth and are nearly always used in florist hydrangea production. Plant growth regulator efficacy can vary with production system and environmental conditions and often varies by plant selection, underscoring the need to conduct trials on a small scale with all selections before initiating a PGR program on a large number of plants. Care should be taken when using PGRs as residual effects may be seen on florist type hydrangea the following season. Bailey and Clark (1992) tested three PGRs and found that summer PGR applications the year before spring forcing reduced flower size and plant height. Applications of dikegulac sodium caused phytotoxicity initially but increased the number of branches and flowers of container grown *H. macrophylla* 'Merritt's Supreme' (Sun et al., 2015). Dikegulac sodium plus pinching increased branch number at 400 ppm in one location of the study while 800 ppm plus pinching increased the number of flowers compared with unpinched controls at both locations. Pinching plants was only successful at increasing branch number compared with untreated controls in one location of the study. Dikegulac sodium (800 and 1,600 ppm) did not significantly increase branching compared to untreated pots of *H. macrophylla* 'Nikko Blue' (Hester et al., 2013). Benzyladenine (300 ppm and 600 ppm) and ethephon (500 and 1,000 ppm) were not effective at inducing branching on either 'Nikko Blue' or 'Merritt's Supreme'.

In replicated studies, late spring pruning of *H. paniculata* 'Limelight' and Little Lime™ in 3-gal (11.4 L) containers had the same branch number compared to water controls and had fewer and smaller flowers than water and PGR-treated plants, calling into question the standard practice of pruning to stimulate branch development for this species (Cochran and Fulcher, 2013; Cochran et al., 2013). In fact, pruning Little Lime™ reduced flower number by at least 78% compared with PGR treatments: benzyladenine (300 or 600 ppm), ethephon (500 or 1,000 ppm), or dikegulac sodium (800 and 1,600 ppm). Dikegulac sodium (either rate) was the only treatment that increased branch number and plant quality without reducing floral display of Little Lime™ (Figure 1.18). Time to first flower was not recorded in these trials, however in a limited trial time to first flower for 'Phantom' grown in 3-gal (11.4 L) containers was not affected by PGR application (benzyladenine).

Hydrangea quercifolia has been reported as susceptible to dieback after shearing, especially when there are wet conditions (Cole et al., 2013). This has led to interest in

Figure 1.18 Little Lime™ hardy hydrangea treated with 800 ppm dikegulac sodium had about 3 times more branches than those that were hand pruned.

using PGRs to control plant size. Ancymidol (25, 50 and 100 ppm foliar and 1, 2, 4 ppm drench) and uniconazole (12.5, 25, 50 ppm foliar and 1, 2, 4 ppm substrate) applied to ‘Pee Wee’ or ‘Alice’ foliage or substrate did not reduce height or width at 12 to 16 weeks after treatment compared to pinched plants.

Gibson and Groninger (2007) found that two applications of cyclanilide (100 ppm), a chemical pinching agent, increased total branch number on seed propagated oakleaf hydrangea, but two applications of benzyladenine (100 ppm) did not, nor did one cyclanilide application. Two cyclanilide applications (100 ppm) increased total branch number on ‘Alice’ compared to non-treated plants (Cochran et al., 2014). ‘Ellen Huff’ oakleaf hydrangea had more branches with increasing concentrations of foliar-applied cyclanilide up to 200 ppm (Holland et al., 2007); quality was not consistently enhanced at any rate. Phytotoxicity following two cyclanilide applications is commonly reported, but was not observed by the end of the experiments when plants would be market ready (Cochran et al., 2014; Holland et al., 2007).

Arthropod Pest Management

Figure 1.19 Melon aphid, also called the cotton aphid, (*Aphis gossypii* Glover) can feed on plants of all ages, but younger plants may be more susceptible to their feeding.

COMMON ARTHROPOD PESTS

1. Aphids
2. Cottony Camellia Scale
3. Oystershell Scale
4. Redheaded Flea Beetles
5. Japanese Beetle
6. Rose Chafer
7. Black Twig Borer
8. Twospotted Spidermite

Hydrangea species and cultivars grown in the southeastern U.S. generally support very few insect and mite pests, few of which cause significant or lasting aesthetic injury (Johnson and Lyon, 1991).

Aphids

New growth of hydrangeas in indoor and outdoor plantings, nurseries and in greenhouses can support populations of several aphid species (Hemiptera: Aphididae). Feeding aphids produce large amounts of honeydew, on which sooty mold will grow. Feeding injury may also result in distortion of expanded foliage, as well as stunting and contortion of new leaves and buds. Damage on established shrubs is generally minor; however, the distortion of growing tissues may pose a problem for cuttings or young plants in greenhouses or nurseries.

Aphid species affecting hydrangeas include melon aphid (Figure 1.19), also called the cotton aphid, (*Aphis gossypii* Glover), bean aphid (*Aphis fabae* Scopoli) and green peach aphid (Figure 1.20) [*Myzus persicae* (Sulzer)] (Leonard, 1964; Johnson and Lyon, 1991). Aphids often do not have wings and are small [0.1 inch (2.5 mm) long or less], pear-shaped, soft-bodied insects. Color is generally not a good identifying characteristic for melon aphid and green peach aphid because these two species vary from pink to black in color, even within the same population. Aphids may be distinguished from other similarly sized insects by presence of a pair of cornicles (like "tailpipes") that protrude from the upper abdomen. Melon aphid has a pair of solid black or dark brown cornicles that helps distinguish this species from green peach aphid, which has only a black band on the cornicle tips. An individual aphid can complete development within a week and produce more than 60 offspring without mating via a reproductive strategy called parthenogenesis. Large populations comprised of overlapping generations of aphids may develop on the same shoots and stems.

Management

Because of the explosive growth potential of aphid populations, management should begin shortly after detection of an aphid infestation, particularly where natural enemy arthropods are limited. Aphid populations often first appear on hydrangeas as early as March and can persist until winter (Table 1.1). Aphid species infesting hydrangea can develop on a wide range of host plants, including common weeds and other crops in landscapes, nurseries and greenhouse operations. These alternative food resources can serve as refuge from crop-directed management actions and spot treatments. Therefore, a proactive approach to aphid management should include good weed

Figure 1.20 Green peach aphids (*Myzus persicae*) have red eyes and may vary in color from pale yellow to green to pinkish-red. As with other aphids, newer growth/younger plants are more susceptible to infestation.

Table 1.1 Seasonal activity of the major arthropod pests of *Hydrangea* in the mid-southern U.S., and unless otherwise noted, represent occurrence in USDA Plant Hardiness Zone 7[Z].

Arthropod Pest	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aphids												
Black twig borer												
Cottony camellia scale												
Japanese beetle												
Oystershell scale												
Redheaded flea beetle												
Rose chafer												
Twospotted spider mite												

[Z] Depicted activity may be early or later than shown depending on location. Activities represented in the table are scale insect crawler emergence, as well as adult or nymphal activity of the most common insect and mite pests.

management and cultural sanitation efforts in which weeds, crop debris, and infested plants are removed regularly. Aphids are generally weak fliers so wind and air currents facilitate dispersal. Susceptible plants downwind of infected plants should be aggressively scouted.

In warm and wet weather, naturally existing entomopathogenic fungi (e.g., *Beauveria bassiana*) can eliminate aphid populations on infested host plants. In naturalized and landscape settings, lady beetles, lacewings, syrphid flies and parasitoid wasps may also effectively control aphid populations. Broad-spectrum insecticide use should be limited to conserve these natural enemy populations. Ant activity can be used as a monitoring aid, because ants forage for aphid honeydew and interfere with the actions of natural enemies. Ant treatment may be necessary when releasing biological control organisms.

Insecticides registered for aphid management (Table 1.2) include carbaryl, acephate, pyrethroids, neonicotinoids (acetamiprid, dinotefuran, imidacloprid and thiamethoxam), pymetrozine, azadiractin, abamectin, spirotetramat, neem oil, horticultural oil and insecticidal soap. Systemic insecticides, such as neonicotinoids, could be applied as topical sprays, soil drenches, broadcast granules, or trunk sprays. The neonicotinoids may require days or weeks to move into the plant tissues but can provide long residual protection against aphid infestation; therefore, these chemicals should be applied before aphid populations increase. For existing populations, weekly or biweekly topical insecticide treatments can be made to prevent re-infestation of terminal shoots.

Commercial growers should understand that aphids are capable and efficient vectors of many plant viruses between susceptible hosts, including hydrangea. Foliar symptoms of virus infections can include chlorotic mottling, leaf deformation, chlorosis or other discolorations, and mosaic and ring spot patterns on leaves (Sinclair and Lyon, 2005). Hydrangea can host many different viruses, including *Arabis mosaic virus* (ArMV), *Cucumber mosaic virus* (CMV), *Hydrangea mosaic virus* (HdMV), *Hydrangea latent virus* (HdLV), *Hydrangea ringspot virus* (HdRSV), *Impatiens necrotic spot virus* (INSV), *Tobacco necrosis virus* (TNV), *Tobacco rattle virus* (TRV), *Tobacco ringspot virus* (TobRSV), *Tomato blackring virus* (TBRV), *Tomato ring spot virus* (TomRSV), and *Tomato spotted wilt virus* (TSWV) (Figure 1.21) (Allen et al., 1985; Machado Caballero et al., 2009). In 2005 and 2006, *H. macrophylla* 'Endless Summer' found in Minnesota retail outlets were diagnosed with a carlavirus named *Hydrangea chlorotic mottle virus* (HdCMV) that was successfully transmitted to healthy 'Endless Summer' and 'Nikko Blue' plants by *M. persicae* aphids. In those preliminary trials, HdCMV was not readily transmitted to *H. arborescens* or *H. paniculata* (Machado Caballero et al., 2009). *Alfalfa mosaic virus* (AMV), which also can be spread by aphids, was recently recovered from *H. macrophylla* in New York (Lockhart et al., 2013).

Figure 1.21 Symptoms of viral infection on *Hydrangea quercifolia*. Note the typical viral symptom of a unique "watermark" on foliage.

Cottony camellia scale

A soft scale and occasional pest of hydrangea, cottony camellia scale (Figure 1.22) [*Pulvinaria floccifera* (Westwood) (Hemiptera: Coccidae)], which is also called cottony yew/taxus scale, also feeds on camellia, holly, yew, maple, and other plant species representing 35 plant families (Johnson and Lyon, 1991; ScaleNet, 2013). The cottony camellia scale is distributed in the eastern and midwestern U.S. and west to Texas, with populations in California and the Pacific Northwest (ScaleNet, 2013). Before egg production, adult females are about 0.12 inches (3 mm) in length, elongated oval, yellowish brown in the back, with dark brown margin. There is only one generation per year. In May, adults produce hundreds of reddish or purplish eggs on the underside of leaves that are contained within white, waxy, fluffy ovisacs that are about twice the length of female. Crawlers emerge in June in Virginia (Day, 2009). Different from the armored scales, soft scales, use piercing-sucking mouthparts to feed on the phloem of plants. Although feeding by a large number of nymphs can cause slight yellowing of the leaves, the biggest problem is the large amount of honeydew and sooty mold associated with soft scale infestation.

Figure 1.22 Cottony camellia scale (*Pulvinaria floccifera*) is a soft scale that infests not only camellia, but also hydrangea and several other woody ornamentals. The adult scale is flat, 1/8-inch in diameter and yellowish-tan. As with other soft scales, cottony camellia scale produces large amounts of sugary honeydew that attracts ants and causes the leaves to become covered with black sooty mold.

Oystershell scale
Oystershell scale (Figure 1.23) [*Lepidosaphes ulmi* (L.)] is an armored scale that is an occasional pest on hydrangea species. Oystershell scale is broadly distributed across the U.S. and, in addition to hydrangea, feeds on twigs and branches of more than 100 plant species. Severe infestations may cause branch die-back. Because female oystershell scales are about 0.098 inch (2.5 mm) long and blend in with bark colors, detection is difficult. Oystershell scale crawlers emerge in April in Kentucky (Mussey and Potter, 1987), May in Michigan and Ohio (Herms, 2004), and May and July (two generations) in Virginia (Day, 2009). Emergence coincides with first flowering of Vanhoutte spirea (*Spiraea x vanhoutii*) (Herms, 2004).

Scale Management
Scale insect pests can be difficult to detect, especially at low population levels, and can be challenging to control. Good sanitation practices are important for preventing scale insect outbreaks. Prune infested stems from lightly infested plants and remove cut tissues and dead plants from the production area. Debris including senesced leaves and pruned stems should be destroyed to ensure scale insect crawlers will not emerge to re-infest host plants. Dead scales do not fall from plants; thus, to determine if pesticide treatments are necessary or were effective in controlling scales, crush the waxy covering. When crushed, gut contents will be extruded from live armored scale insects.

When scale insect pests are detected, management actions are best timed to coincide with visual confirmation of crawler emergence and activity. For example, degree-day models indicate that oystershell scale eggs hatch following accumulations of about 760 degree-days using a 40º F (4.4º C) (base temperature (Mussey and Potter, 1987) or about 360 degree-days using a 50º F (10º C) base temperature (Herms, 2004). Oystershell scale crawlers have been observed in Ohio and Kentucky at about the time that eastern redbud (*Cercis canadensis* L.), flowering dogwood (*Cornus florida* L.), Japanese flowering crabapple (*Malus floribunda* Siebold ex Van Houtte), Sargent's crabapple (*Malus sargentii* Rehder), lilac (*Syringa vulgaris* L.) and doublefile viburnum (*Viburnum plicatum* var. *tomentosum*) have begun to flower (Herms, 2004; Mussey and Potter, 1987). Pesticides used to manage scale insects are most effective when applied to crawlers shortly after peak crawler emergence (Table 1.2). Horticultural oils and insect growth regulators can help conserve natural enemy populations (Frank and Sadof, 2011; Raupp et al., 2006; Rebek and Sadof, 2003). Dormant-season horticultural oils can be applied to dormant hydrangea in winter and early spring. Control with oil is likely achieved through both impaired respiration and disruption of cellular membranes in treated arthropods. Follow-up applications of dormant oil, after leaf drop and before bud swell in fall and early winter, will also help reduce armored scales population growth by limiting impact of subsequent generations. Refined, or summer, horticultural oil treatments can be applied to control eggs, crawlers and immature instars on actively growing trees and shrubs.

Figure 1.23 Oystershell scale [*Lepidosaphes ulmi* (L.)] is an armored scale that is typically found on larger branches near the central leader of shrubs.

Redheaded flea beetle
Systena frontalis (F.), is a native flea beetle (Figure 1.24) that is widespread across the eastern U.S. and northward into Canada (Smith, 1970). In addition to commercially cultivated host plants like alfalfa (*Medicago sativa* L.), beans (e.g., *Phaseolus* sp.), blueberry and cranberry (*Vaccinium* sp.), grapes (*Vitis* sp.), and corn *Zea mays* L., *S. frontalis* also feeds on many weedy plants common across the southern U.S., including velvetleaf (*Abutilon theoprhasti* Medic.), pigweed (*Amaranthus* sp.), dogbane (*Apocynum cannabinum* L.), various asters, lambsquarters (*Chenopodium* sp.), Canada thistle (*Circium arvense* (L.) Scop., Joe Pye weed (*Eupatorium* sp.), jewelweed (*Impatiens* sp.), smartweed (*Polygonum* sp.), and giant foxtail [*Setaria faberi* (R.A.W. Herrm.)]. Ornamental host plants include evergreen azaleas (*Rhododendron* sp.), *Forsythia* sp., *Hibiscus* sp. (especially *H. paniculata* Siebold), *Ilex* sp. (especially *I. crenata* Thunb.), *Itea virginica* L., *Loropetalum* sp., *Physocarpus* sp., *Weigela* sp., and *Zinnia* sp. (Jacques and Peters, 1971; Maltais and Ouellette, 2000; Kunkel, 2012).

Adult beetles are 0.12 to 0.24 inches (3 to 6 mm) long, with a shiny black pronotum and elytra. The reddish-black to reddish-yellow head may be disguised by the pronotum, and

Figure 1.24 Redheaded flea beetle (*Systena frontalis*) affects a wide range of host plants in nursery production but is particularly damaging on hydrangea. Look for infestations beginning shortly after foliage emerges and monitor throughout the entire growing season.

may appear uniformly dark depending upon lighting. Brownish antennae extend outward about half as long as the body. Hind femora on *S. frontalis* legs are enlarged, which facilitates flea-like jumping when disturbed and to escape predation (Riley, 1983). Adult beetles feed on both upper and lower leaf surfaces, skeletonizing leaf tissues. Mobile adults can be captured using a sweep net and by vigorously shaking canopies above a white cloth (Dudek, 2011).

Only limited life history information is available. Pale yellow eggs are deposited singly and beetle larvae develop in soil. Larvae feed on organic matter and roots, and pass through three larval instars, growing to 0.2 to 0.39 inches (5 to 10 mm) long before pupation (Dudek, 2011). Adult beetles emerge and are active from late June through mid-September in Michigan, where adult emergence was reported at between 550-750 degree days [base 50º F (4.4º C)] (Dudek, 2011). Red headed flea beetles overwinter as eggs in soil in northern climates. Larval development may be possible in both soilless substrates and potting media.

Kunkel (2012) found that larvae from overwintering eggs were active in containers upon accumulations of 257– 481 growing degree-days (GDD) at a 50º F (4.4º C) base temperature threshold. In Delaware, this coincided with full flowering of black locust (*Robinia pseudoacacia* L.) and Chinese fringetree (*Chionanthus retusus* Lindl. & Paxton). First emergence of adult beetles was observed once 590– 785 GDD50 (base temperature 4.4º C), had accrued, which coincided with bud swell and flowering of Magnolia grandiflora (L). Larvae from the second generation were active at 1818– 1860 GDD50 (base temperature 4.4º C), about the time that (*Lagerstroemia* sp.), rose of sharon (*Hibiscus syriacus*), and plumbago (*Ceratostigma plumbaginoides* Bunge) were flowering. Overlapping life stages that were present between August and October confounded evidence of a third generation in Delaware (Kunkel, 2012). Three or more generations per year may be possible in the mid southern U.S.

Management

Efficacy of biological controls in the landscape and nurseries is unknown. Control via commercially available entomopathogenic nematodes have shown limited promise (Kunkel, 2012). Broad spectrum and contact insecticides labeled for beetle and weevil control (Table 1.2) have been effective at managing adult beetle populations.

Japanese beetle

Adult Japanese beetles (Figure 1.25) (*Popillia japonica* Newman) attack flowers, fruits and foliage of more than 300 species of plants including *H. paniculata* and *H. quercifolia*. Since its introduction in 1916 via infested nursery stock, Japanese beetles have become one of the most damaging pests in the eastern U.S. (Held, 2004). Adult beetles are 0.3 to 0.4 inches (8 to 11 mm) long and metallic green and copper-brown in color. They are active day fliers that disperse readily across long distances. Japanese beetles require one full year to complete development from egg to egg stadia. Larvae, or white grubs, feed on roots of turfgrasses and other susceptible plants. Up to five dozen eggs per female are deposited into moist soil, hatch and develop through three instars. Third instar larva overwinter and pupation occurs in the spring. Adult beetles emerge in early summer, usually following a rainfall event. They are highly mobile and gregarious, capable of rapidly defoliating susceptible plants. On hydrangea, adult beetles skeletonize leaves.

Figure 1.25 Adult Japanese beetles (*Popillia japonica* Newman) attack flowers, fruits and foliage of more than 300 species of plants, including *H. paniculata* and *H. quercifolia*. Emergence is often quick and damage can occur very quickly, therefore continual monitoring is necessary. A wide range of contact insecticides can prevent damage, yet scouting is key to minimize damage.

Adult rose chafer

The rose chafer (Figure 1.26), *Macrodactylus subspinosus* (Fabricius), may also occasionally feed on hydrangea foliage and will skeletonize leaves causing injury very similar to feeding by Japanese beetle adults. White grubs of rose chafer develop optimally in sandy soils and will overwinter as third instar larvae. There is one generation per year. Rose chafer adults, which emerge in May and are active through early June.

Japanese beetle & adult rose chafer management

Scout starting in May for adult rose chafer feeding on flowers of rose and peony, which are highly preferred. Traps used for monitoring Japanese beetles should be placed near susceptible species in late May and typically include both a floral lure and sex attractants that are intended to help track first flight of adults. Traps should be placed at least 200 ft (61 m) away from plants under protection. Because only a fraction of lured beetles are caught in traps, trapping is ineffective for managing beetles, but does serve as a monitoring tool (Chappell et al., 2012). Japanese beetle adults on susceptible plants can be controlled with foliar applications of short-residual insecticides that require repeated applications to maintain uninjured plants during adult flight periods. Systemic insecticides can provide longer residual control (Table 1.2). Follow the National Plant Board's U.S. Domestic

Japanese Beetle Harmonization Plan when shipping nursery stock from areas that may be infested with Japanese beetles to beetle-free areas (NPB, 2013).

Figure 1.26 Rose chafer (*Macrodactylus subspinosus*) is more known for infesting rose and grape, but also infests hydrangea. Use rose as an indicator crop for infestation and apply preventative insecticide applications accordingly.

Predation by birds, small mammals and generalist insect predators also can reduce Japanese beetle and rose chafer populations. Two wasp species (*Tiphia vernalis* Rohwer and *T. popilliavora* Rohwer) parasitize larvae underground, and a tachinid fly (*Hyperecteina aldrichi* Mesnil) attacks adult Japanese beetles. The bacteria *Bacillus popilliae* Dutky exclusively attacks Japanese beetle larvae, but it is best suited for large scale, regional application rather than individual site applications. Microscopic entomopathogenic nematodes occur naturally in the soil, and, together with a symbiotic bacterium, can ultimately kill larvae by means of septicemia. Nematodes that have been shown to be most effective against Japanese beetle larvae are *Steinernema glaseri* Steiner and *Heterorhabditis bacteriophora* Poinar. The latter is commercially available.

Black twig borer

The black twig borer (Figure 1.27), *Xylosandrus compactus* (Eichhoff), is an ambrosia beetle species introduced into Fort Lauderdale, Florida most probably from eastern Asia (Wood, 1982). This species has since spread to Alabama, Florida, Georgia, Louisiana, Mississippi, North Carolina, South Carolina, and Texas (Mangold et al., 1977; Chong et al., 2009).

Adult black twig borer females are dark brown to black and their bodies are stout, cylindrical, 5/100 to 7/100 inch (1.4-1.9 mm) long, about two times as long as wide, with the pronotum more than half the length of elytra. Adult males are half the size of females, flightless, and can only be found in the gallery, along with eggs, larvae and pupae (Wood, 1982). Eggs are oval, white and smooth, on average 2/100 inch long and 1/100 inch wide (0.55 x 0.28 mm). The legless larvae are about 8/100 inch (2 mm) in length when mature, with a pale brown head. Pupae are similar in body length to the adults.

Figure 1.27 Black twig borer (*Xylosandrus compactus*) infests a wide range of species, and causes considerable damage in all hydrangea species.

Black twig borer attacks small diameter twigs and branches of trees and shrubs stressed by difficult transplant establishment, poor plant maintenance, excessive pruning, improper landscape placement, and water strain. Hundreds of plant species are susceptible to *X. compactus* attack, including red maple (*Acer rubrum* L.), eastern redbud (*Cercis canadensis* L.), flowering dogwood (*Cornus florida* L.), southern magnolia (*Magnolia grandiflora* L.), oak (*Quercus* sp.) and *H. macrophylla* (Chong et al., 2009). An individual female *X. compactus* attacks smaller twigs about 4/100 to 28/100 inch (1-7 mm) diameter, while multiple females may work in concert to attack larger branches with diameters about 30/100 to 87/100 inch (9-22 mm) (Ngoan et al., 1976). Entry holes are about 3/100 inch (0.8 mm) diameter and lack frass tubes or other signs of tunneling. In addition to destruction of pith by tunneling beetles (Figure 1.28), plant injury may also occur in response to symbiotic fungal growth that impedes the vascular system. Wilted or flagging terminals are the most noticeable symptom of beetle attack, with death of stem portions above the entry hole. In South Carolina, flagging stems on southern magnolia were most prevalent below 6.5 ft (2 m) (Chong et al., 2009). Infested stems desiccate while leaves remain attached to the terminals. Attacks can also leave a 0.4 to 8 inch (1 to 21 cm) long canker on larger surviving branches (Chong et al., 2009).

Figure 1.28 Tunneling of black twig borer (*Xylosandrus compactus*) is very distinctive in hydrangea.

Females construct a brood gallery in the center of the pith or wood. Each female may lay up to 16 eggs during the 38 to 58-day adult lifespan. Larvae develop through two larval instars while feeding on an introduced ambrosia fungus, after which they pupate within the brood gallery. In flowering dogwood host tissues, durations spent in stadia as egg, larva, pupa and teneral adult lasted 5, 7.5, 7.5 and 8 days, respectively (Ngoan et al., 1976). Black twig borer overwinters as adults in the gallery and first emergence from stems is typically in February in Florida (Ngoan et al., 1976) and early March in South Carolina (Chong, unpublished data). *Xylosandrus compactus* populations remain active from February through October, thus determination of the number of generations across its U.S. distribution is difficult.

Management

For monitoring purposes, ethanol-baited traps should be deployed by the end of February. Traps should be hung about 1.5 ft (0.46 m) above the ground (Reding et al., 2010) and checked regularly. No effective biological control options for black twig beetle are currently available. Treatment (Table 1.2) with pyrethroids, particularly bifenthrin and permethrin, can repel ambrosia beetle attacks on other tree species for up to 10 days, depending upon air temperatures (Mizell and Riddle, 2004). Systemic neonicotinoids are not effective in preventing attacks by ambrosia beetles, nor will they kill beetles already in the gallery (Chong, unpublished data).

Twospotted spider mite

Twospotted spider mite (Figure 1.29) (*Tetranychus urticae*), is one of the most common and destructive mite species in both production and landscape settings and can attack over 200 ornamental plant species (Johnson and Lyon, 1991), including hydrangeas. Adult and immature twospotted spider mites are 0.02 inches (0.51 mm) long. Both life stages are pale-green to yellow or cream colored and have two dark green-black patches of spotting on the body. Eggs are spherical and translucent. Foliar feeding injury becomes evident on susceptible host plants as twospotted spider mites feed on the undersides of leaves, and empty leaf cell contents to affect a silvery stippling pattern of feeding injury. Twospotted spider mites spin silken threads that can aid in dispersal, and webbing can cover branches when infestations become severe.

Figure 1.29 Twospotted spider mite (*Tetranychus urticae*) damage on hydrangea is identified by bronzing of foliage and discoloration or deformation of flowers. Under a dissecting microscope, the female typically has two irregular dark blotches on the sides of its body.

Twospotted spider mites overwinter as adult females either in bark crevices or in the soil (Johnson and Lyon, 1991), and may persist throughout the season within protected nursery structures, provided that plant food resources, including weed species, remain present. Eggs of subsequent seasonal populations are deposited directly on leaf undersides. Spider mite populations begin to expand in April and pass through several generations per year, with peak activities occurring in June and July (Potter, 2008). Twospotted spider mites become increasingly active as localized conditions become hot and dry, and during warmer seasons of the year (Table 1.1). In tree fruit systems, twospotted spider mites migrate from groundcovers onto trees, which may help trigger higher levels of summer activity (Gotoh, 1997). Development of twospotted spider mite on raspberry (*Ribes idaeus*) can be completed in about 7 days at 86° F (30° C), 14 days at 77° F (25° C), 16 days at 68° F (20° C), and 25 days at 59° F (15° C) (Bounfour and Tanigoshi, 2001). Each female can produce 38 to 125 eggs (Bounfour and Tanigoshi, 2001). Populations remain active as long as environmental conditions and plant quality are favorable for reproduction and growth.

Management

Incipient populations of twospotted spider mites are easy to overlook due both to their small size and their overwintering habits. Scouts can monitor susceptible crops and weed refuges by looking for the characteristic stippling injury to leaf surfaces. In outdoor container operations and landscapes, overhead irrigation and hand watering can limit mite population growth (Drees, 2004). Susceptible plants can be repositioned away from dry, dusty roads and also away from doorways and exhaust fans within production structures. Many natural enemy organisms will feed on twospotted spider mites. In Oregon, commercially available *Neoseiulus fallacis* (no official common name) predatory mites suppressed twospotted spider mites in ornamental nurseries (Pratt, 1999). Once introduced, some predatory organisms can persist by foraging on prey present on alternate host plants [e.g., crabapple (*Malus* sp.)] including ground covers (Stanyard et al., 1997).

Several key factors should be considered when using chemical miticides for population management. Because twospotted spider mites reproduce rapidly and can quickly develop pesticide resistance, it is critical to rotate between different modes of action (Table 1.2). Select a miticide that is most effective against the life stages detected during scouting. Conserve beneficial arthropods, which can provide long-term biological control of twospotted spider mites, by selecting miticides that are compatible with the natural enemies that are already present. Pyrethroids that are registered for spider mite management are generally not effective.

Table 1.2 [1] Pest-directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Hydrangea* spp.

IRAC Code[2]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[3,4]	Use Site[5]	REI (hrs)[6]	Aphids	Cottony camellia scale	Oystershell scale	Flea beetles	Japanese beetles & rose chafer	Black twig beetle	Spider mite
1A	Acetylcholinesterase inhibitors	Carbamates	carbaryl[7]	Sevin SL	L, N, G	12	x	x	x	x	x		
			methiocarb	Mesurol 75W	N, G	24	x						x
1B[17]	Acetylcholinesterase inhibitors	Organophosphates	acephate[7, 8]	Orthene T&O[8]	L, N, G	24	x	x	x		x		
				Lepitect[9]	L, N, G	24	x	x	x		x		x
				Precise GN[8]	G, N	12	x						
			chlorpyrifos	Dursban 50W[8]	N	24	x	x	x	x	x	x	x
				DuraGuard ME[8]	N, G	24	x	x	x	x	x		x
			dicrotophos	Inject-A-Cide B[10]	L	N/A	x	x	x				x
			dimethoate	Dimethoate 4E[8], 4EC[8]	N	330	x	x	x				x
			malathion	Malathion 5EC[8,9]	L	12	x	x	x		x		x
			methidathion	Supracide 2E[8,16]	N	720		x	x				
			oxydemeton methyl	Harpoon[8,10]	L	0	x	x	x				x
				MSR Spray Concentrate[11,16]	N	240	x						x
			trichlorfon	Dylox 420 SL	L	N/A							
3A[17]	Sodium channel modulators	Pyrethroids / Pyrethrins	bifenthrin[7]	Attain TR	G	12	x	x	x				x
				Menace GC	L, N, G	12	x	x	x	x	x	x	x
				Onyx	L	N/A	x	x	x	x	x	x	x
				OnyxPro	L, N, I	12	x	x	x	x	x	x	x
				Talstar S Select	N, G	12	x	x	x	x	x		x
				Talstar Nursery G	N	12							
			cyfluthrin	Decathlon[12]	L, N, G, I	12	x	x	x	x	x		
			beta-cyfluthrin	Tempo Ultra WP[13]	L, I	N/A	x	x	x	x	x		
				Tempo SC Ultra[12]	L, I	N/A	x	x	x	x	x		
			lambda-cyhalothrin	Scimitar CS; Scimitar GC	L ; L, N, G	N/A; 24	x	x	x	x	x		x
				Demand	L	N/A	x	x		x	x		x
			cypermethrin	Demon WP	L, I	N/A	x						
			deltamethrin	DeltaGard G[12]	L, I	N/A				x	x		

Table 1.2 [1] (***continued***) Pest-directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Hydrangea* spp.

IRAC Code[2]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[3,4]	Use Site[5]	REI (hrs)[6]	Aphids	Cottony camellia scale	Oystershell scale	Flea beetles	Japanese beetles & rose chafer	Black twig beetle	Spider mite
3A[17]	Sodium channel modulators	Pyrethroids / Pyrethrins	fenpropathrin	Tame 2.4 EC[8]	N, G, I	24	x		x		x		x
			tau-fluvalinate	Mavrik Aquaflow	L, N, G, I	12	x			x			x
			permethrin	Astro	L, G, I	12	x				x		
				Permethrin Pro	L, I	N/A	x				x		
				Perm-Up 3.2 EC	L, N, G, I	12	x				x		
			pyrethrins	Tersus	N, G	12	x	x	x	x	x	x	x
				Pyganic	N, G	12	x	x	x	x			x
			pyrethrum	Pyrethrum TR	N, G	12	x	x	x	x			x
3A + 4A[17]	Sodium channel modulators	Pyrethroids + Neonicotinoids	bifenthrin + clothianidin	Aloft LC G, LC SC	L	N/A	x	x	x	x	x		
			bifenthrin + imidacloprid	Allectus SC	L, I	N/A	x	x	x	x	x		x
			cyfluthrin + imidacloprid	Discus N/G	N, G, I	12	x	x	x		x		
			lambda-cyhalothrin + thiamethoxam	Tandem	L	N/A	x	x	x	x	x		
			zeta-cypermethrin + bifenthrin + imidacloprid	Triple Crown T&O	L, I	N/A	x	x	x	x	x	x	x
3A + 27 [17]	Sodium channel modulators	Pyrethroids + Piperonyl-butoxide (PBO)	pyrethrins + piperonyl butoxide[8]	Pyreth-It	N, G	12	x			x			
4A[17]	Nicotinic acetylcholine receptor agonists	Neonicotinoids	acetamiprid	TriStar 8.5 SL[8,9,12]	L, N, G	12	x	x	x		x		
			clothianidin	Arena 0.25 G[9]	L, I	12	x						
				Arena 50 WDG[9]	L, I	12	x						
			dinotefuran	Safari 2G[9]	L, N, G, I	12		x	x				
				Safari 20 SG[9]	L, N, G, I	12	x	x	x		x		
				Zylam Liquid[9]	L	N/A	x	x	x				
				Transtect 70 WSP[17]	L	N/A	x	x	x		x		
			imidacloprid[10]	Xytect 75WSP; 2F[9]	L, N, G, I	12	x	x	x		x		
				Marathon II[9]	N, G, I	12	x	x	x		x		
				Marathon 60WP[9]	N, G, I	12	x	x	x		x		

Table 1.2 [1] (***continued***) Pest-directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Hydrangea* spp.

IRAC Code[2]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[3,4]	Use Site[5]	REI (hrs)[6]	Aphids	Cottony camellia scale	Oystershell scale	Flea beetles	Japanese beetles & rose chafer	Black twig beetle	Spider mite
4A[17]	Nicotinic acetylcholine receptor agonists	Neonicotinoids	thiamethoxam	Merit[9]	L, I	N/A	x	x	x		x		
				CoreTect[9]	L, I	N/A	x	x	x		x		
				Discus Tablets[9]	N, G, I	12	x	x	x		x		
				Flagship 25WG[9]	N, G, I	12	x	x					
				Meridian 0.33G[9]	L, I	N/A	x	x		x	x		
				Meridian 25WG[9]	L, I	N/A	x	x		x	x		
4C + 5 [17]	Nicotinic acetylcholine receptor agonists	Sulfoxaflor + Spinosyns	sulfoxaflor + spinetoram	XXpire[13,16]	L, N, G	12	x		x				x
5	Nicotinic acetylcholine receptor allosteric activators	Spinosyns	spinosad	Conserve; Entrust	L, N, G	4							x
6 [17]	Chloride channel activators	Avermectins, Milbemycins	abamectin[8]	Lucid, Avid[8]	L, N, G	12							x
				Aracinate TM[10]	L, N, G, I	N/A	x	x	x				x
			emamectin benzoate	Arbormectin	L	N/A	x		x		x	x	x
				Tree-äge	L	N/A			x				
				Enfold	N	12							x
			milbemectin	Ultiflora	N	12							x
6 + 20D[17]	Chloride channel activators	Avermectins, Milbemycins	abamectin + bifenazate	Sirocco[12]	L, N, G, I	12	x						x
7A	Juvenile hormone mimics	Juvenile hormone analogues	s-kinoprene	Enstar AQ	G, I	4	x	x	x				
7B	Juvenile hormone mimics	Fenoxycarb	fenoxycarb	Preclude TR	G	12	x	x	x				x
7C	Juvenile hormone mimics	Pyriproxyfen	pyriproxifen	Distance IGR[8]	L, N, G, I	12	x	x	x				
				Fulcrum	L, N, G, I	12	x	x	x				
8C	Misc. non-specific (multi-site) inhibitors	Fluorides	cryolite (sodium alumino-fluoride)	Kryocide	L	N/A				x			
8D	Misc. non-specific (multi-site) inhibitors	Borates	sodium tetraborohydrate decahydrate	Prev-AM Ultra	N, G	12	x	x	x				x
9B[17]	Selective homopteran feeding blockers	Pymetrozine	pymetrozine	Endeavor[8,12]	L, N, G, I	12	x						
		Pyrifluquinazon	pyrifluquinazon	Rycar	G	12	x						

Table 1.2 [1] **(*continued*)** Pest-directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Hydrangea* spp.

IRAC Code[2]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[3,4]	Use Site[5]	REI (hrs)[6]	Aphids	Cottony camellia scale	Oystershell scale	Flea beetles	Japanese beetles & rose chafer	Black twig beetle	Spider mite
10A[17]	Mite growth inhibitors	Clofentezine	clofentazine	Ovation SC[8]	N, G	12							x
		Hexythiazox	hexythiazox	Hexygon DF[8,12]	L, N, G, I	12							x
10B[17]	Mite growth inhibitors	Etoxazole	etoxazole	Beethoven TR	G	24							x
				TetraSan 5 WDG	L, N, G, I	12							x
12B	Inhibitors of mitochondrial ATP synthase	Organotin miticides	fenbutatin-oxide	Meraz, ProMite 50WP[8]	L, N, G	48							x
13	Uncouplers of oxidative phosphorylation via disruption of the proton gradient	Chlorfenapyr	chlorfenapyr	Pylon[8]	G	12							x
16	Inhibitors of chitin biosynthesis, type 1	Buprofezin	buprofezin	Talus 70DF	L, N, G	12		x	x				
20B[17]	Mitochondrial complex III electron transport inhibitors	Acequinocyl	acequinocyl	Shuttle 15 SC	L, I	12							x
				Shuttle-O[8]	N, G	12							x
20D[17]	Inhibition of complex III at the Qo site	Bifenazate	bifenazate	Floramite SC	L, N, G, I	12							x
				Floramite SC/LS	L	N/A							x
21A	Mitochondrial complex I electron transport inhibitors	METI acaricides and insecticides	fenazaquin	Magus[8]	L, N, G, I	12							x
			fenpyroximate	Akari 5SC	N, G, I	12							x
			pyridaben	Sanmite	N, G	12							x
			tolfenpyrad	Hachi-Hachi SC[8]	G	12	x	x					
23	Inhibitors of acetyl CoA carboxylase	Tetronic and Tetramic acid derivatives	spiromesifen	Forbid 4F	L	N/A							x
				Judo (=Savate)[8]	N, G	12							x
				Kontos[8]	N, G, I	24	x	x	x				x
25	Mitochondrial complex II electron transport inhibitors	Beta-ketonitrile derivatives	cyflumetofen	Sultan	L, N, G, I	12							x
28	Ryanodine receptor modulators	Diamides	chlorantraniliprole	Acelepryn	L, I	N/A	x	x[15]			x[15]		
				Acelepryn G	L, I	N/A	x	x					
			cyantraniliprole	Mainspring	L, G, I	4	x	x			x		
29 [17]	Chordotonal organ modulators – undefined target site	Flonicamid	flonicamid	Aria	L, N, G	12	x	x	x				

Table 1.2 [1] **(*continued*)** Pest-directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Hydrangea* spp.

IRAC Code[2]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[3,4]	Use Site[5]	REI (hrs)[6]	Aphids	Cottony camellia scale	Oystershell scale	Flea beetles	Japanese beetles & rose chafer	Black twig beetle	Spider mite
Unknown	Unknown	Azadirachtin	azadirachtin[7]	Azatin O	L, N, G, I	4	x	x	x	x	x		
				Azatin XL	N, G, I	4	x			x	x		
				Azatrol EC	L, N, G, I	4	x	x	x	x	x		x
				Ornazin EC	L, N, G, I	12	x	x	x	x	x		
Not classified	Various		*Beauveria bassiana*[7]	BotaniGard ES; Mycotrol ESO; Mycotrol WPO	L, N, G, I	4	x			x			x
				BotaniGard 22 WP	L, N, G, I	4	x						
				Naturalis-L	L, N, G	4	x			x			x
			Chromobacterium subtsugae	Grandevo PTO	L, N, G	4	x						x
			Isaria formosorosea	NoFly	G	12	x						
				Preferal	L, N, G	4	x						x
			horticultural oil[7,8]	Ultra-Pure Oil, TriTek	L, N, G, I	4	x	x	x				x
			insecticidal soap[7,8]	M-Pede[8]	L, N, G, I	12	x	x	x	x	x		x
			neem oil[7]	Trilogy	L, N	4	x	x					x
				Triact 70[8]	L, N, G, I	4	x	x	x				x
			capsicum extract, garlic oil, soybean oil	Captiva	L, N	4							x
			kaolin clay	Surround WP	L, N, G, I	4					x[12]		

Table 1.2 (***continued: footnotes***) Pest-directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Hydrangea* spp.

1. Pests listed on pesticide labels are subject to change. Within states and counties, products may have additional permitted uses by 2(ee) and Special Local Need allowances. Consult your County Extension Agent or State Agency to determine if other uses are allowable. The label should always be consulted to confirm that chart-listed pests still appear on the label. Check the product labels for specific site restriction information, notes on application, sensitive plant species and specific target pest species.
2. IRAC Code designations and Related Modes of Action are explained at the Insecticide Resistance Action Committee Database 2016. (IRAC 2016; http://www.irac-online.org/teams/mode-of-action/).
3. Trade names of products are provided as examples only. No endorsement of mentioned product nor criticism of unmentioned products is intended.
4. Products may not be registered or renewed for use in all southeastern U.S. states. Consult your state's Department of Agriculture to confirm legal use of products in your state.
5. Use site information is provided for reference only: L = landscape; N = nursery; G = greenhouse; I = interiorscape. t/s*Products listed for use in L, N, G or I sites may include active ingredients in differently labeled products that are designated for use in turfgrass or sod production use sites. Mention of those products is beyond the scope of this resource guide. Products listed herein may not be legal or appropriate for site uses in turfgrass or sod production.
6. Re-Entry Interval (REI) designations apply to agricultural (nursery) uses. Within REI column, 'N/A' is used to indicate products with Landscape and/or Interiorscape site uses and that present Non-Agricultural Use Requirements. Consult product labels for specific details required for compliance within those application conditions and use sites, including those for which REI listings do not apply. Additionally, some product labels may list site uses that are beyond the scope of this publication (e.g., sod farms, silvicultural & Christmas tree nurseries, pastures, rights-of-way, etc.). These site uses may involve Agricultural Use Requirements that list an REI not presented here.
7. Multiple formulations and trade names of the same active ingredients are available. Labels among these products often differ in legally allowable uses (regarding sites, pest, REI, etc.); representative example(s) presented.
8. See product label for information about potential crop phytotoxicity, known plant sensitivity, and how to test for phytotoxicity.
9. Check label for additional restrictions (e.g., on the number of times the product can be applied in a growing season or year, or additional application restrictions within permitted use sites; and crop types or flowering status that may not be legally treated.)
10. Product formulated for tree or shrub injection; specialized equipment may be required.
11. Use of handheld application equipment is prohibited. For use only on seedling trees and non-bearing fruit trees in commercial nurseries.
12. Intended for commercial/professional use only. Not intended for homeowner use.
13. Label-indicated efficacy against target pest group is primarily by population suppression.
14. Product labels may restrict landscape uses only to commercial landscapes. Residential landscapes are not a permitted application site.
15. On the Acelepryn Section 2ee label.
16. Product uses cancelled or in process of Federal cancellation or review. Use of existing stocks of any end-use product may be allowable beyond the cancellation date. Consult your extension agent or state agency for information about allowable use and conditions for proper disposal.
17. Applications of products from these footnoted IRAC groups are optimized when not preceded by/or followed by products in subsequently noted IRAC Group(s), particularly when managing the indicated pest. Suggested rotation combinations to avoid include serial treatments with a.i. products from: IRAC 1B, 3A, 6 (western flower thrips); IRAC 4A, 4B, 4C (green peach aphid); IRAC 4A and 9B, 9C (glasshouse and silverleaf whiteflies); IRAC 10A, 10B, 15 (Panonychus [red and citrus] spider mites); IRAC 20B and 20D bifenazate (twospotted spider mite).

Disease Management

COMMON DISEASE PESTS

1. Powdery Mildew
2. Cercospora Leaf Spot
3. Southern Blight
4. Bacterial Leaf Spot
5. Botrytis Blight
6. Phytophthora Root and Crown Rot
7. Anthracnose
8. Hydrangea Viruses

Powdery Mildew

Powdery mildew (Figure 1.30) is caused by the fungus *Erisyphe polygoni*. The first sign of infection is the appearance of small, white, powdery colonies of the fungus on leaves. As the infection progresses, these colonies expand and coalesce to cover the entire leaf surface, and purple to yellow discoloration may appear. The fungus will spread to other leaves, and stems may also become colonized. Heavily infected plants can develop stunted leaves and flowers and generally become unsalable (Hagan and Mullen, 2001).

Figure 1.30 Powdery mildew on *Hydrangea macrophylla*. Note the gray to white color along the veins.

Powdery mildew is primarily a problem for *H. macrophylla* but can also affect *H. arborescens* and *H. paniculata*. Oakleaf hydrangea (*H. quercifolia*) is rarely affected by powdery mildew (Hagan and Mullen, 2001). The *H. macrophylla* cultivar 'Veitchii' is known to be highly resistant to powdery mildew (Li et al., 2008; Windham et al., 2011), and several other cultivars have also been reported to have some resistance (Table 1.3).

High relative humidity and warm temperatures are conducive for infection; free water on leaves is not necessary for infection. The disease spreads rapidly under these conditions which occur frequently outdoors throughout much of the Southeast and are common in greenhouses and overwintering houses. Increasing the spacing among plants to allow sufficient air circulation and light penetration facilitates evaporation, thereby limiting humidity at the leaf surfaces and minimizing the potential severity of powdery mildew infection. Powdery mildew is primarily spread by wind-blown spores. Spores can be carried on tools or clothing.

Management

Removing infected plants and plant debris can help prevent the spread of powdery mildew. However, these measures alone cannot prevent the occurrence of powdery mildew where environmental conditions support infection. Disease resistance and fungicides are the primary means of minimizing infection. Several fungicides are approved for treating powdery mildew (Table 1.4). Repeated preventative applications should be used to protect greenhouse hydrangea crops (Hagan and Mullen, 2001; Hagan et al., 2004; Knox, 2013).

Table 1.3 Hydrangea cultivars with reported resistance to common diseases. Source indicates the plant patent or peer-reviewed paper which indicates that the cultivar is resistant.

Disease	Resistant Cultivars	Source	Disease	Resistant Cultivars	Source
Powdery Mildew	‘Bountiful Bouquets’	US 20140283252 P1	Powdery Mildew (*continued*)	‘Shirofuji’	
	‘Dear Dolores’	US PP25566 P3		‘Veitchii’	
	‘Harbits’	US PP211186 P2	Cercospora Leaf Spot	‘Midnight Duchess’	US PP18341 P3
	‘Horwack’	US 20130269074 P1	Botrytis Blight	‘Horwack’	US 20130269074 P1
	‘Lindsey Ann’	US PP26249 P2	Leaf Spot (Cercospora, Botrytis, and/ or bacterial)	‘Little Bo Peep’	US 20140283253 P1
	‘Little Bo Peep’	US 20140283253 P1		‘Ami Pasquier’	Mmbaga et al., 2012
	‘Lynn’	US PP20019 P2		‘Ayesha’	
	‘Magical Emerald’	US PP20464 P2		‘Blue Bird’	
	‘Magical Opal’	US PP20483 P2		‘Forever Pink’	
	‘Magical Pearl’	US PP20614 P2		‘Fuji Waterfall’ (‘Fujinotaki’)	
	‘Midnight Duchess’	US PP18341 P3		‘Miyama-yae-Murasaki’	
	‘Show Stopper’	US 20140283251 P1		‘Seafoam’	
	‘Twist-N-Shout’	US PP20176 P3		‘Taube’	
	‘White King’	US PP21065 P2		‘Tricolor’	
	‘Amaga Amachi’	Windham et al., 2011		‘Veitchii’	
	‘Diadem’				
	‘Komachi’				
	‘Omacha’				

Cercospora Leaf Spot

Cercospora leaf spot is a common disease of hydrangeas that affects all species and cultivars grown in the southeast. It is caused by the fungus *Cercospora hydrangeae*. Symptoms typically occur first on lower leaves and appear as small purple, brown, or black spots (Figure 1.31). As the infection progresses the spots grow larger, leaves turn yellow and fall off, and symptoms spread to upper leaves. Cercospora leaf spot rarely kills hydrangeas but can decrease plant vigor due to the repeated loss of leaves (Hagan and Mullen, 2001; Vann, 2009). Some cultivars are reported to be resistant to Cercospora leaf spot (Table 1.3).

Figure 1.31 *Hydrangea macrophylla* with advanced symptoms of Cercospora leaf spot. Note the "bull's-eye" appearance of symptoms.

Management

Leaf wetting encourages the development of Cercospora leaf spot, and splashed water allows the fungus to spread to upper leaves. Thus, it typically occurs after periods of frequent rainfall, usually in late summer or autumn. Light intensity also affects leaf spot development. Disease severity on *H. macrophylla* increases as light intensity increases (Li et al., 2008). Plants in full sun exposure may be severely infected with leaf spot, whereas those grown under 30% shade cloth or greater have fewer lesions. The most likely source of new infections is fallen leaves from previous years. To minimize damage; remove leaf litter, avoid planting *H. macrophylla* in full sun and use fungicide sprays if desired. Preventative fungicide treatments are the best option to prevent infection. (Hagan and Mullen, 2001; Hagan et al., 2004; 2005) (Table 1.4).

Southern Blight

Southern blight is a disease caused by the soil-borne fungus, *Sclerotium rolfsii*. Southern blight typically attacks lower stems at the soil-line (Figure 1.32). A lesion develops on the infected stem which then begins to rot and becomes girdled. Leaves wilt and eventually turn brown and fall off. Wilting is usually the first noticeable symptom of southern blight (Mullen, 2001). Southern blight also produces distinct signs of disease. White mycelium and tan-to-reddish-brown spherical sclerotia are often observed on infected stems. Southern blight occurs under high-moisture conditions when temperatures are between 86 and 95° F (30 and 35°C). While it is mostly a problem in mineral soil, southern blight has also been reported to attack container-grown crops.

Figure 1.32 Southern blight (*Sclerotium rolfsii*) on stem of *Hydrangea macrophylla* at the soil line. Note the directional webbing that moves from the soil line upward.

Management

To prevent contamination with southern blight, avoid allowing containers or substrates to come into contact with soil (Halcomb and Windham, 2011). Infected plants should be removed from production areas and destroyed to minimize the risk of future infections. Heavily infested soil can be sanitized though solarization, steam, or fumigation (Mullen, 2001). Untreated fields where southern blight has been a problem should not be replanted with hydrangea. Preventative fungicide sprays (Table 1.4) directed at the stem may help prevent southern blight infection (Halcomb and Windham, 2011).

Figure 1.33 Hydrangea bacterial leaf spot on *Hydrangea quercifolia*, the most susceptible species of the genus.

Bacterial Leaf Spot

Bacterial leaf spot is caused by the bacteria *Xanthomonas campestris*. It is most severe on oakleaf hydrangea (Figure 1.33) (*H. quercifolia*) but can also infect *H. macrophylla* and *H. arborescens* (Byrne, 2010; Uddin et al., 1996). The infection first appears as purple-red, often angular, spots. Typically, it occurs first on lower leaves and then progresses upwards through the canopy. *Xanthomonas campestris* thrives under moist conditions and survives in plant debris.

Management

Irrigation practices that minimize leaf wetness should be used to prevent the occurrence of bacterial leaf spot, while sanitizing tools between uses and removing infected leaves will help prevent its spread. Bactericides can be used as a preventative measure early in the growing season or applied at the first sign of bacterial leaf spot. However, they may have limited effect and can cause some phytotoxic symptoms (Byrne, 2010; Cleveland, 2012; Mmbaga and Oliver, 2007) (Table 1.4). Cuttings for propagation should be taken from healthy plants only.

Botrytis Blight

Caused by the fungus *Botrytis cinerea*, Botrytis blight can affect all species of hydrangea, but is most damaging in *H. macrophylla*. Botrytis blight first appears as brown spots (Figure 1.34), and masses of grayish-brown spores may be visible on affected parts (Hagan and Mullen, 2001). The flowers and buds are most commonly affected and quickly fade and wither. Infected leaves develop spots similar to those seen with Cercospora and bacterial leaf spot infections (Mmbaga et al., 2012). Leaves may have zonate spots similar to anthacnose. Stem infections may girdle the whole stem causing it to wilt and die. (Daughtery et al., 2000).

Management

Botrytis blight can develop and spread rapidly when conditions are cool, wet and humid. To minimize damage, avoid overhead watering and restrict watering to the earlier part of the day so that leaves have sufficient time to dry by nightfall. Increasing the spacing between plants and providing adequate ventilation and heat in greenhouses will help prevent Botrytis blight by reducing moisture on leaves. Fungicide applications can be effective at preventing this disease (Table 1.4). However, they will not eliminate it unless used in combination with good cultural practices. Since Botrytis spores survive for extended periods of time in debris, all fallen plant material should be removed and destroyed. Sanitation of all work surfaces and tools after use is a necessary component of Botrytis blight prevention because the spores are easily moved by wind, water, and normal nursery activities. Avoid damaging plants since wounded plant parts are highly susceptible to infection. Botrytis blight is persistent and can infect many plant parts. Thus, cuttings should never be taken from plants which have shown signs of Botrytis blight (Hagan and Mullen, 2001; Daughtrey et al., 2000).

Figure 1.34 Botrytis blight on *Hydrangea quercifolia* seedling. Note the highlighted border surrounding the necrotic tissue.

Phytophthora Root and Crown Rot

Phytophthora root rot and crown rot diseases are caused by fungal-like organisms in the genus *Phytophthora*. Root rot is commonly found in container-grown plants, but may be found in landscape plantings too. Symptoms first appear as wilted foliage, and leaves later turn yellow and senesce. Crown rot usually attacks above-ground plant parts such as stems near the soil-line; root rot is primarily a disease of the root system. Infected plants will be stunted and often die (Hagan and Mullen, 2000; 2001).

The most critical factor in preventing Phytophthora diseases is to avoid over-watering. Substrates and containers that allow sufficient drainage should be used, and irrigation should be scheduled such that over-watering is avoided by taking into account rainfall and weather conditions before irrigating. For field production, hydrangeas should be planted in well-drained soil, and areas prone to flooding or with a fragipan or hardpan should be avoided (Hagan and Mullen, 2001).

Management

Since *Phytophthora* spp. can survive in all infected plant parts and in substrate, diseased plants must be removed and destroyed to prevent future infections. Containers and substrate from infected plants should also be disinfected (containers) and/or discarded (substrate). All debris should be removed, and work surfaces and tools must be sanitized between crops. Container-grown plants should be kept on crowned gravel pads to ensure proper drainage and avoid transmission of propagules between containers. Store substrates and containers on concrete pads or asphalt, and avoid re-using containers or sanitize them between uses (Hagan and Mullen, 2001). Used substrate should be discarded, but can be sanitized by steaming or composting if necessary (Noble et al., 2011; Schweigkofler et al., 2014). Fungicides cannot cure Phytophthora root rot but can be used as a preventative measure against infections (Table 1.4). They are most effectively used as drenches or when incorporated into the potting substrate during propagation (Bailey et al., 2012; Hagan and Mullen, 2001).

Figure 1.35 *Hydrangea macrophylla* with anthracnose.

Anthracnose

Anthracnose is a disease caused by the fungus *Colletotrichum gloeosporioides*. It is most prevalent in dense plantings of bigleaf hydrangea (*H. macrophylla*) and develops under wet, hot conditions [75 to 95°F (24 to 35°C)]. Symptoms occur as large necrotic brown spots which appear simultaneously on leaves and flowers, especially during periods of heavy rainfall. The infected spots (Figure 1.35) are a lighter brown in the center, with darker discoloration on the outer edges and have a “bull’s-eye” appearance. Spores are spread by water.

Management

Remove infected leaves and flowers, and take cuttings from disease-free plants to avoid future infections. Preventative applications of fungicides can be used to avoid anthracnose infections, but repeated and frequent applications are required (Hagan and Mullen, 2001) (Table 1.4).

Hydrangea Viruses

At least 14 viruses (Figure 1.36) have been reported to infect hydrangeas (Pscheidt and Ocamb, 2015). Viruses are particularly damaging to bigleaf hydrangea (*H. macrophylla*) which is highly susceptible to infection (Lockhart et al., 2013; Tang et al., 2009; Veerakone et al., 2012). Plant viruses cannot be cured, and infected plants must be removed and destroyed to prevent viruses from spreading to other plants. Three of the most common and damaging hydrangea viruses are the

Figure 1.36 *Hydrangea quercifolia* with oak leaf pattern symptoms of a virus.

hydrangea ringspot virus, hydrangea chlorotic mottle virus, and tomato ringspot virus.

Hydrangea ringspot virus causes leaves to become rolled and crinkled with yellow or brown spots, and leads to stunted flowers and plant growth. This virus is only transmitted through contact with infected plants or tools (Pscheidt and Ocamb, 2015). Thus, removing infected plants and debris as well as sanitizing tools will help prevent future infection.

The leaves of hydrangeas infected with the hydrangea chlorotic mottling virus become red, blistered, mottled, and chlorotic. This virus is spread by the green peach aphid (*Myzus persicae*) so it can easily travel between plants and throughout a landscape or nursery (Machado Caballero et al., 2009). Since many plants can serve as hosts for these aphids, production areas should be kept free of weeds to avoid harboring aphid populations which could serve as sources of infection (Capinera, 2014).

Symptoms of tomato ringspot virus are indistinguishable from those caused by hydrangea ringspot virus. However, this virus is spread by nematodes (Pscheidt and Ocamb, 2015). Remove and destroy infected plants and avoid re-planting hydrangeas in that area. Avoid re-using substrate or containers from potentially infected plants or sanitize before re-use. Treatment with fumigants or solarization can be effective at eradicating nematodes from soil (Hagan, 2005; Krueger and McSorley, 2015).

Table 1.4 Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of hydrangea[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Cercospora leaf spot	Powdery mildew	Bacterial leaf spot	Botrytis blight	Phytophthora root rot, water molds[X]	Anthracnose
1: MBC Benzimidazoles: Upwardly systemic. Broad spectrum fungicide for various fungi. Fungicide resistance risk high. Tank mix with fungicides from a different fungicide group (FRAC) to prevent or delay resistance development. Do not mix with copper.						
thiophanate methyl: Cleary's 3336 F, EG; Allban 50 WSB; OHP 6672 50WP, 4.5F	x	x		x		x
1 + 2: MBC Benzimidazoles + Dicarboximides: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.						
thiphanate methyl + iprodione: 26/36[W]	x			x		
1 + 14: MBC Benzimidazoles + Aromatic Hydrocarbons: Systemic. Effective against water molds, or oomycetes. Long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.						
thiophanate methyl + etridiazole: Banrot 40WP[W]					x	
1 + M3: MBC Benzimidazoles + Multi-site inhibitor: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.						
thiophanate methyl + chlorothalonil: Spectro 90 WDG, Zyban[W]						x
1 + M5: MBC Benzimidazoles + Multi-site inhibitor: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.						
thiophanate methyl + chlorothalonil: Spectro 90 WDG[W]	x	x		x		x
2: Dicarboximides: Locally systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.						
iprodione: Chipco 26019 FLO, N/G; 26GT; Raven[V]	x			x		
3: DMI or SI Triazoles: This group was formerly known as De-Methylation Inhibitors (DMI) and are now classified as Sterol Biosynthesis Inhibitors (SBI or SI). Upwardly systemic. Rain-fast in 2 hours. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.						
fenarimol: Rubigan A.S.		x				
imazalil: Fungaflor TR		x		x		
metconazole: Tourney[W]	x	x				x
myclobutanil: Eagle, 20W; Siskin	x	x				
myclobutanil: Rally 40WSP	x	x				
propiconazole: Banner Maxx, Banner Maxx II	x	x				
tebuconazole: Torque	x	x				
triadimefon: Bayleton 50, Flo; Strike 50 WDG[V]						

Table 1.4 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of hydrangea[Y].

FRAC code: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Cercospora leaf spot	Powdery mildew	Bacterial leaf spot	Botrytis blight	Phytophthora root rot, water molds[X]	Anthracnose
3: DMI or SI Triazoles: This group was formerly known as De-Methylation Inhibitors (DMI) and are now classified as Sterol Biosynthesis Inhibitors (SBI or SI). Upwardly systemic. Rain-fast in 2 hours. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.						
triticonazole: Trinity, Trinity TR	x					
3 + 11: SI Triazole + QoI Q: Upwardly systemic. Rain-fast in 2 hours. Some curative activity. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance.						
triadimefon + trifloxystrobin: Trigo	x	x				x
3 + M5: SI Triazole + Chlorothalonil: Upwardly systemic. Rain-fast in 2 hours. Some curative activity. Broad spectrum fungicide for greenhouse and nursery use. Medium risk for resistance.						
propiconazole + chlorothalonil: Concert II	x	x				x
4: Phenylamides: Systemic. Effective against diseases caused by oomycetes, or water molds, including damping-off, root and stem rots, and foliar diseases. Use as soil drench or foliar application.						
mefenoxam: Subdue Maxx, Subdue GR					x	
4 + 12: Phenylamides + Phenylpyrroles/Osmotic signal transducers: Systemic. Broad spectrum drench-applied fungicide.						
mefenoxam + fludioxonil: Hurricane, Hurricane WDG[U,V,W]					x	
5: Morpholine: Inhibits sterol biosynthesis in membranes. For use only in greenhouse and similar enclosed structures. Do not use after flower buds visible.						
piperalin: Pipron		x				
7 + 11: Carboxamides/Succinate Dehydrogenase Inhibitors (SDHI) + QoI Q: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Spray apply for leaf disorders, soil drench for Phytophthora.						
pyraclostrobin + boscalid: Pageant, Pageant Intrinsic[W]	x	x		x	x	x
boscalid + pyraclostrobin: Orkestra Intrinsic	x	x		x		x
benzovindiflupyr + azoxystrobin: Mural	x			x		x
9 + 12: Anilino-Pyrimidines/Methionine biosynthesis inhibitors + Phenylpyrroles: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.						
cyprodinil + fludioxonil: Palladium[W]	x	x		x		x
11: Quinone outside Inhibitors (QoI): These fungicides are also known as strobilurins. Locally systemic. Effective on mildews, foliar pathogens, and most fungi. Some control of oomycetes, or water molds. Fungicide resistance risk high. Apply as a soil drench for Phytophthora root rot.						
azoxystrobin: Heritage	x	x		x	x	x
azoxystrobin: Strobe 50WG	x	x		x[U]		
fenamidone: FenStop	x	x			x	
fluoxastrobin: Disarm O	x	x			x	x
kresoxim methyl: Cygnus	x	x				x

Table 1.4 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of hydrangea[Y].

FRAC code: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Cercospora leaf spot	Powdery mildew	Bacterial leaf spot	Botrytis blight	Phytophthora root rot, water molds[X]	Anthracnose
11: Quinone outside Inhibitors (QoI): These fungicides are also known as strobilurins. Locally systemic. Effective on mildews, foliar pathogens, and most fungi. Some control of oomycetes, or water molds. Fungicide resistance risk high. Apply as a soil drench for Phytophthora root rot.						
pyraclostrobin: Insignia; Insignia SC	x	x			x	
pyraclostrobin: Empress Intrinsic					x	
trifloxystrobin: Compass; Compass O 50 EDG[V]	x	x			x	x
12: Phenylpyrroles/Osmotic signal transducers: Non-systemic but good residual protection. Broad spectrum fungicide, not effective against oomycetes, or water molds.						
fludioxonil: Emblem, Medallion; Medallion WDG, Mozart TR	x			x		
14: Aromatic Hydrocarbons: Locally systemic. Effective against water molds, or oomycetes.						
dicloran: Botran 75 W				x		
etridiazole: Terrazole 35% WP, CA, L; Truban 25EC, 30WP[V]					x	
17: Hydroxyanilides: Locally systemic. For use in outdoor and greenhouse nursery crops.						
fenhexamid: Decree 50 WDG[V]				x		
19: Polyoxins: Locally systemic chitin synthase inhibitor. For use in outdoor and greenhouse nursery crops. Resistance risk high.						
polyoxin D zinc salt: Affirm; Veranda O; Endorse WP [W]	x	x		x		x
21: Quinone inside Inhibitors: Locally systemic. Effective against water molds, or oomycetes. Resistance risk unknown but presumed to be medium to high. Apply as a soil drench for Phytophthora root rot.						
cyazofamid: Segway, Segway O, Segway SC					x	
25: Glucopyranosyl Antibiotics: Bacterial protectant. Broad spectrum bacterial control.						
streptomycin sulfate: Agri-Mycin 17						
28: Carbamates: Cell membrane permeability, fatty acid interruption (proposed). Low to medium resistance risk.						
propamocarb hydrochloride: Banol					x	
33: Phosphonates: Fully systemic; when applied to leaves, product can translocate to lower parts. Effective against water molds, or oomycetes, such as Phytophthora, Pythium, and downy mildew pathogens. Low risk for fungicide resistance development. Apply as a soil drench for Phytophthora root rot.						
phosphorous acid: Alude		x	x		x	
phosphorous acid: K-Phite T/O, 7LP		x	x		x	
potassium phosphite: Vital		x	x		x	
fosetyl-Al: Aliette WDG		x	x		x	

Table 1.4 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of hydrangea[Y].

FRAC code: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Cercospora leaf spot	Powdery mildew	Bacterial leaf spot	Botrytis blight	Phytophthora root rot, water molds[X]	Anthracnose
40: Carboxylic Acid Amides: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes. Apply as a soil drench for Phytophthora root rot.						
mandipropamid: Micora					x	
dimethomorph: Stature DM, SC					x	
43: Pyridinemethyl-benamides Delocalisation of spectrin-like proteins: Locally systemic, translaminar. Control of oomycetes, or water molds. Medium to high resistance risk. Apply as a soil drench for Phytophthora root rot.						
fluopicolide: Adorn					x	
44: *Bacillus subtilis*: Microbial disruptor of cellular membranes. Spray apply for leaf disorders, soil drench for Phytophthora.						
Bacillus subtilis: Cease	x	x	x	x	x	x
45 + 40: Quinone outside Stigmatellin-binders (QoS) + Carboxylic Acid Amides: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes. Apply as a soil drench for Phytophthora root rot.						
ametoctradin + dimethomorph: Orvego[W]					x	
49: Piperidinyl-thiazole- isoxazolines: Locally systemic. Control of oomycetes, or water molds. Rain fast. Medium to high resistance risk.						
oxathiapiprolin: Segovis					x	
M: Chemicals with multi-site activity: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.						
(M1) copper hydroxide: Champ DP, Champ Formula 2	x	x	x			x
(M1) copper hydroxide: Kocide 2000			x			
(M1) copper hydroxide: CuPro 5000, NuCop	x	x	x			x
(M1) copper octanoate: Camelot O	x	x	x	x		x
(M1) copper sulfate: Basicop						x
(M1) copper sulphate pentahydrate: Phyton 27, Phyton 35	x	x	x	x		
(M1) tribasic copper sulfate: Cuproxat	x	x	x			
(M1 + M3) copper hydroxide + mancozeb: Junction	x	x	x	x		x
(M3) mancozeb: Dithane 75 DF; Fore 80 WP[V]	x	x	x	x		x
(M3) mancozeb: Mancozeb 4 F, Flowable with Zinc	x	x	x	x		x
(M3) mancozeb: Pentathlon DF	x	x	x	x		x
(M3) mancozeb: Pentathlon LF	x	x	x	x		x
(M3) mancozeb: Protect DF	x	x	x	x		x

Table 1.4 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of hydrangea[Y].

FRAC code: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Cercospora leaf spot	Powdery mildew	Bacterial leaf spot	Botrytis blight	Phytophthora root rot, water molds[X]	Anthracnose
M: Chemicals with multi-site activity: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.						
(M4) captan: Captan 50WP	x					
(M5) chlorothalonil: Daconil Ultrex, Zn Flowable, Weather Stik [T]	x	x		x		x
(M5) chlorothalonil: Manicure 6FL, Manicure Ultra; Thalonil 90 DF, Thalonil 6L	x			x		x
M3 + 3: Multi site inhibitor + Sterol Biosynthesis Inhibitor: Effective as protectants (and upwardly systemic) on broad spectrum including most fungi and mildews. Fungicide resistance risk low.						
mancozeb: + myclobutanil: Clevis[W]	x	x				x
NC: Not a Classified substance: Contact fungicide for greenhouse and nursery use. Low risk for resistance.						
potassium bicarbonate: MilStop	x	x				x

[Z] (FRAC 2017).
[Y] Check current products for labeled pesticides, sites for control and plant safety and efficacy on fungal species. This table reports information on fungicide labels and does not necessarily reflect product efficacy. Refer to fungicide labels for rates and usage, specific host information, possible phytotoxicity, re-entry intervals and resistance management. Within columns, products indicated by "x" are labeled for use against the listed pathogen type. Always test product on a sample (small number) of plants before treating an entire crop, as some fungicides listed are for control of the disorder but not specifically labeled for use in *Hudrangea* spp.
[X] Including the causal agent of sudden oak death, *Phytophthora ramorum*.
[W] Chemical contains more than one active ingredient, thus product may be listed within more than one FRAC code designation (FRAC 2017).
[V] Not for use in residential landscapes; commercial use only.
[U] Only use as a suppressant, does not grant preventative control.
[T] Do not apply with mist blowers or high pressure spray equipment in greenhouses.

Section 4

Weed Management


© Joe Neal, NCSU

Weed Management in Hydrangea

Preemergence Herbicides in Container Production

Hydrangea macrophylla is a popular landscape plant and container nursery crop. The species is relatively easily propagated and produced in container culture. However, weed management is a challenge in production. Of the 25 (or more) preemergence herbicides labeled for use in container nursery crops, only six are labeled for use on *H. macrophylla*. These include granular prodiamine, dimethenamid-p + pendimethalin, oxyfluorfen + pendimethalin, pendimethalin, oxyfluorfen + oxadiazon, prodiamine and oryzalin. Napropamide and s-metolachlor are labeled for use around hydrangea in field plantings or landscape settings. Several herbicide labels specifically state that use on hydrangea may or will result in unacceptable crop injury. However, grower experiences and research have demonstrated that even labeled herbicides may injure *H. macrophylla* in container production. For example: we observed over 20% necrosis of the growing points (Figure 1.37) following applications of granular oxyflurofen + pendimethalin (Figure 1.38) or oxadiazon (a component of Regal OO). Even "non-burning" types of herbicides, such as dimethenamid-p + pendimethalin, caused bud damage when granules are caught in the growing points (Figure 1.39). The bud damage is visible within a few days after treatment. Sometimes, new sprouts emerge from below and plants recover within 6 to 8 weeks (Figure 1.40). However, this can prolong the time to market, thus increasing production costs.


© Joe Neal, NCSU

Figure 1.37 Herbicide granules are caught in the growing point of *Hydrangea macrophylla* (Left photo), resulting in necrosis of the growing point (Right photo).

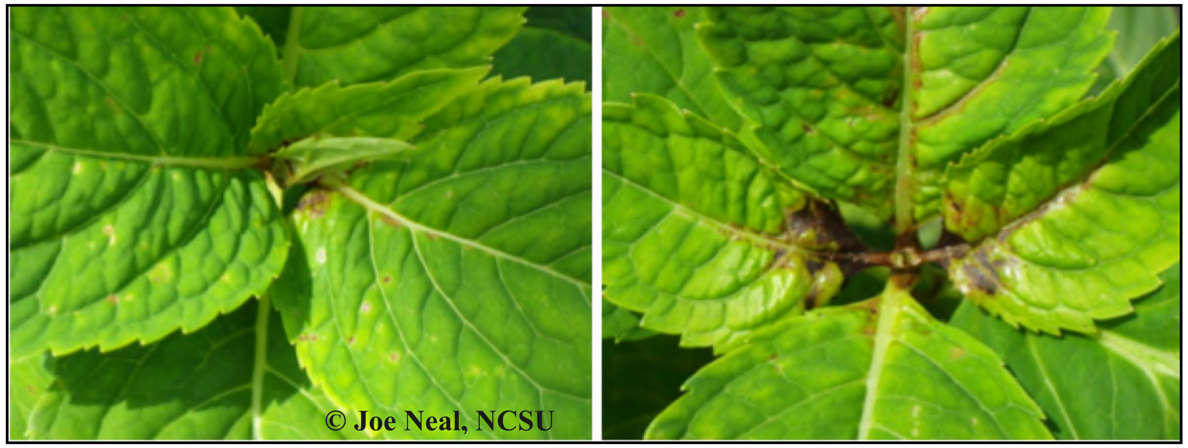

© Joe Neal, NCSU

Figure 1.38 Photos of bud necrosis in plants receiving overhead irrigation only (right photo) vs granules shaken off (left photo) - photos taken 1 day after treatment. Oxyfluorfen + pendimethalin (OH2) treatments resulted in injury. Injury was less severe when granules were shaken off by hand (Left) compared to plants that were irrigated to remove granules (Right).

Figure 1.39 Physically shaking plants to remove granules from the foliage prevented bud damage from dimethenamid-p + pendimethalin (Freehand). Plants on left had granules shaken off; plants on the right were irrigated to remove granules.

Alternatively, physically removing the granules by shaking the plants after treatment reduced or eliminated the tip damage (Figure 1.39). Following application of granular preemergence herbicides, plants were either shaken to remove granules or irrigated. All three granular herbicides caused chlorosis and necrosis of the growing points when granules were not physically removed before irrigation. This injury was visible as soon as one day after treatment. However, when granules were removed by manually shaking the plants, no bud damage was observed from dimethenamid-p + pendimethalin or isoxaben + trifluralin treatments. Injury from oxyfluorfen + pendimethalin was dramatically reduced when granules were removed by shaking but plants still exhibited foliar necrosis (Figure 1.39). So, the answer seems quite simple at this point – avoid "burning-type herbicides" and shake the plants, and all will be well…. *But, read on.*

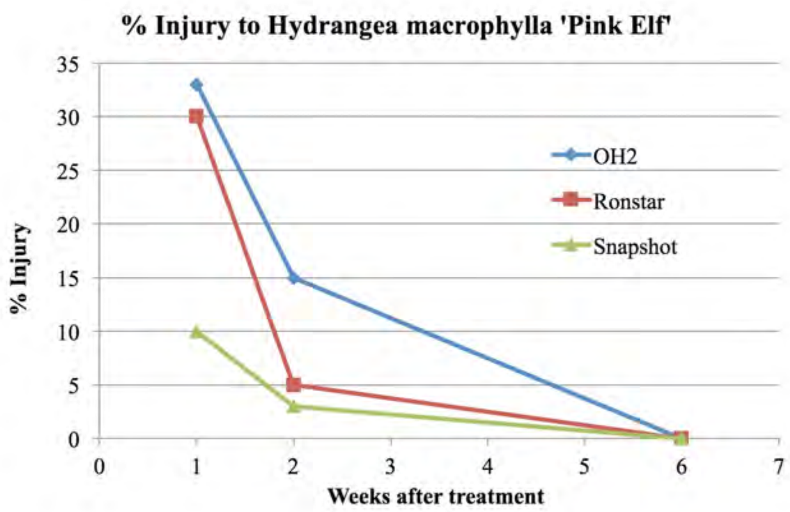


Figure 1.40 Percent injury to *H. macrophylla* 'Pink Elf' from preemergence herbicides including OH2 (oxyfluorfen + pendimethalin), Ronstar 2G (oxadiazon), and Snapshot (isoxaben + trifluralin). By 6 weeks after treatment, plants had recovered from the initial injury (NC State University, 2008).

Figure 1.41 *Hydrangea macrophylla* plants treated with dimethenamid-p + pendimethalin GR (Freehand) were similar in appearance to non-treated plants (non-treated on the left, treated on the right). However, fresh weight measurements revealed that treated plants were about 15% smaller.

Ten weeks after treatment, we observed that plants with early season bud damage had not completely recovered, and were smaller and lower in quality than the non-treated plants. This further emphasizes the necessity of avoiding this type of injury. Pruning plants to remove the damaged buds did not result in normal re-growth.

Plants treated with a "non-burning" type herbicide, dimethenamid-p + pendimethalin GR, then shaken to dislodge the granules looked comparable to the non-treated plants and were all considered "marketable" (Figure 1.41). However, even these plants were, on average, about 15% smaller than the non-treated plants, when above-ground fresh weights were measured. (Figure 1.42).

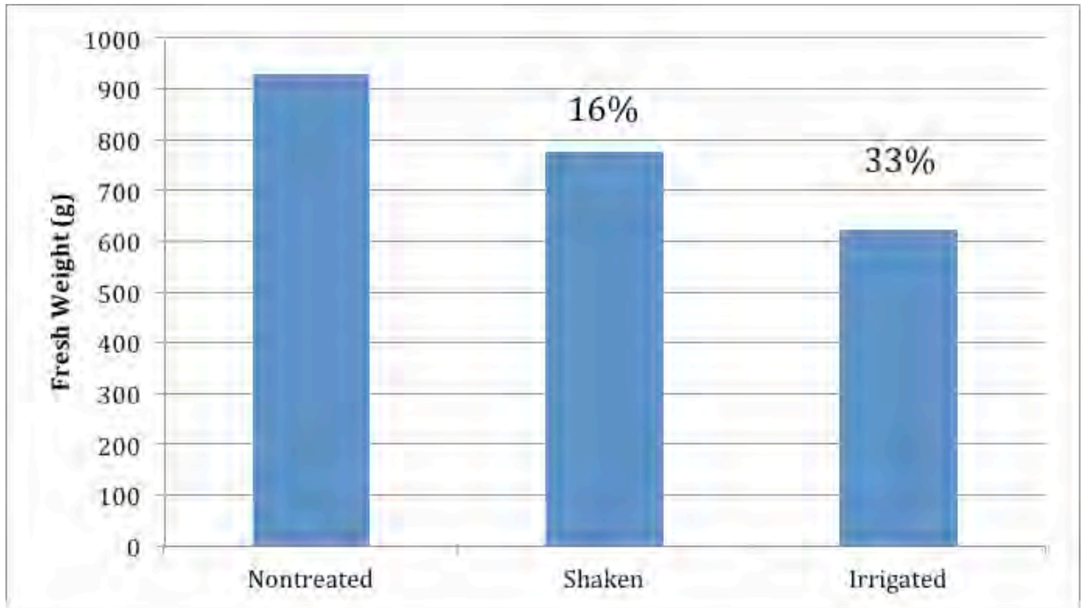


Figure 1.42 Removing herbicides by shaking plants resulted in about 16% reduction in growth compared to hand-weeded plants. Irrigating to remove granular herbicides reduced growth by 33%. These data are based on measuring hydrangea fresh weights 10 weeks after treatment. (data source: J. Neal 2013)

Granular Versus Spray Applied Herbicides

Figure 1.43 Isoxaben applied to hydrangea causes tip necrosis and mottled leaves. Non-treated plant is on the left, treated plant on the right. Photo taken 3 weeks after treatment.

There is growing interest in using spray applied herbicides for weed control in nursery crops. *Hydrangea macrophylla* is particularly sensitive to preemergence herbicides and is damaged when herbicide sprays are applied during active growth. Isoxaben + prodiamine is a popular preemergence herbicide spray in container nurseries. However, this should not be used over hydrangea plants. Isoxaben is not labeled for use on hydrangeas and, when used, can cause severe injury to the growing points (Figure 1.43). Treating during an irrigation cycle reduced the severity of injury, but did not eliminate the bud damage. Dimethenamid-p is labeled for use in hydrangea production. Dimethenamid-p treatments over actively growing hydrangea resulted in chlorosis and some necrosis of the growing points. However, treating plants during an irrigation cycle washed off the herbicide, preventing tip damage (Figure 1.44). This sort of bud injury can also be avoided if applications are done before bud break or as directed sprays to prevent contact with the foliage. Directed applications are generally not feasible in container production but may be feasible in field production.

Figure 1.44 Dimethenamid-p applied to actively growing plants can cause chlorosis in the growing points and some tip necrosis. This injury can be avoided if applications are made during an irrigation cycle or before bud-break. On the left dimethenamid-p was applied to dry foliage and was allowed to dry on the foliage for 2 hours. The plant on the right was treated with dimethenamid-p during an irrigation cycle.

To reiterate, when using preemergence herbicides, avoid bud damage by following these guidelines:

When using granular herbicides:
- Utilize “non-burning” granular herbicides (such as dimethenamid-p + pendimethalin);
- Be sure the foliage is dry before application;
- Physically remove granules from the growing points before irrigation is applied.

When using spray herbicides:
- Use only labeled products;
- Apply when foliage is wet and irrigate before spray can dry to minimize the risk of foliar injury.

Our research suggests that under the best of circumstances, when preemergence herbicides are used on *H. macrophylla*, growers should expect 10 to 15% growth reduction. The other alternative is to rely on hand weeding and / or mulches for weed control in *H. macrophylla* production.

Alternatives to Preemergence Herbicides?

To avoid crop injury, many hydrangea growers avoid using preemergence herbicides altogether or utilize mulches to reduce weeds. Previous research has shown that relying on hand-weeding alone can increase container nursery crop production costs by as much as $0.40 per pot (compared to an effective preemergence herbicide program supplemented with hand weeding). So, growers of hydrangea are left with balancing herbicide injury risks with the higher costs of hand weeding.

Mulches can be almost as effective as preemergence herbicide programs. Many mulching materials have been evaluated for weed control in container nursery crops, including hydrangea. A diversity of mulching materials are available including: pelleted wool byproducts, pelleted paper, coated fabric disks, wood chips, rice hulls (Figure 1.45), human hair disks, and many others. Bottom line -- they do reduce weed growth. They may also offer increased hydrangea growth by insulating the substrate and retaining moisture.

While mulches can be almost as effective as preemergence herbicides, weed seedlings can emerge around the edges or through slits in fabric mulches (Figure 1.45). When using mulches, maintain a sufficient depth for the purpose. In landscape plantings we recommend 2 to 3 inch (5.1 to 7.6 cm) of mulch, but this is not practical in small containers. Recent research with rice hulls suggests about 0.5 inch (1.3 cm) of mulch may be sufficient to control weeds when the seeds are introduced on top of the mulch (think bittercress or spurge which spread by dispersing seeds). However, a 1-inch (2.5 cm) layer of rice hulls was needed to prevent weeds germinating from below the mulch layer. Consequently, when using organic mulches in containers, a 1 to 1.5 inch (2.5 to 3.8 cm) thick layer is recommended.

Figure 1.45 Mulches such as the fabric disk impregnated with copper and rice hulls can be about as effective as preemergence herbicides but weeds can emerge along the edges or through "slits" in the fabric or through organic mulches. Emerged weeds need to be removed when small.

- Mulches offer advantages of weed control, enhanced moisture retention (which may lead to greater growth), and ease-of-use. However, mulches have traits that detract from their popularity and use.
- Higher cost. Our work suggests costs to be about 5 times the cost of an effective preemergence herbicide. Plus, they can be costly to apply to pots.
- Removing emerged weeds can be a challenge. Removing emerged weeds can disrupt the mulch layer, leading to more weed emergence.
- Blow-off. High wind events can blow these mulches (organic and fabric mulches) off of the pots. This results in replacement costs as well as weed emergence.
- Consumer acceptance. Customers may or may not accept crops with mulches on the pots. Removal of fabric disks allows for re-use on another crop but with the expense of labor to remove disks before shipment.
- Top dressed fertilizer may not penetrate the mulch layer as well as if it were placed directed on the substrate surface (also, consider "blow off" may remove top dressed fertilizer).

Postemergence Weed Control

Emerged weeds should be removed by hand when young. Hydrangea plants have a high demand for water. Weeds that compete effectively for water, such as eclipta or crabgrass, are likely to significantly reduce crop growth. Grasses can be controlled with selective postemergence herbicides. Fenoxaprop, fluazifop-p-butyl and sethoxydim are labeled for over-the-top applications in hydrangea production. These herbicides selectively control grass weeds with little if any activity on broadleaf plants. Follow label guidelines for dosing and recommended spray adjuvants.

Broadleaf weeds should be controlled by hand removal. Directed applications of non-selective herbicides like glyphosate, glufosinate, or diquat, risk damaging the tender green stems of hydrangea plants. Directed applications of such products may be possible in older, established field plantings or landscape beds. But, even in these situations the applicator should be very careful to avoid contacting hydrangea stems or foliage. Directed sprays of bentazon or sulfosulfuron may be used as directed sprays in field nurseries or landscape beds to control emerged sedges. Similarly, halosulfuron may be used as a directed spray to control emerged sedges in landscape plantings.

Table 1.5 Preemergence herbicides registered for use on *Hydrangea* spp. or that are prohibited for use due to observed phytotoxicity.

Genus and species	Common Name	Barricade	Biathlon	Broadstar	Dacthal	Freehand	Gallery	OH2	Pendulum (granule)	Pennant Magnum	Regalkade	Regal OO	Rout	Snapshot TG	Surflan	Sureguard	Tower
Hydrangea spp.	hydrangea		Ø	Ø	F		Ø			F		f/c		Ø		Ø	f/c
Hydrangea arborescens	hydrangea, smooth		Ø	Ø	F		Ø	Ø*		F		f/c	Ø*	Ø		Ø	
Hydrangea macrophylla	hydrangea, bigleaf	f/c	Ø	Ø	F	f/c	Ø	f/c*	f/c	F	f/c	f/c		Ø	f/c	Ø	
Hydrangea paniculata	hydrangea, panicled		Ø	Ø	F		Ø			F		f/c	Ø*	Ø		Ø	
Hydrangea quercifolia	hydrangea, oak-leaf		Ø	Ø	F		Ø			F		f/c		Ø	f/c	Ø	

Key:
F = registered for use in the field (or landscape)
f/c= registered for both field and container use
Ø = label prohibits use on this species
F*, C*, or f/c* = registered for some species or cultivars; consult label for details.
Ø* = prohibited for some cultivars within a species; consult label for details.

References

Abou Dahab, T.A.M. 2007. *In vitro* propagation of *Hydrangea macrophylla* Thunb. Arab J. Biotech. 10:161-178.

Agro, E. and Y. Zheng. 2014. Controlled-release fertilizer application rates for container nursery crop production in southwestern Ontario, Canada. HortScience. 49:1414–1423.

Allen, T. C., J. P. McMorran, and R. H. Lawson. 1985. Detection and identification of viruses in hydrangeas. Acta Hort. 164: 85-89.

Altland, J. and M. Lanthier. 2007. Influence of container mulches on irrigation and nutrient management. J. Environ. Hort. 25:234–238.

Anonymous. 2010. How to apply PGRs to the Invincibelle® Spirit hydrangea. Spring Meadow Nursery. Grand Haven, MI. Accessed November 23, 2015. <http://springmeadownursery.com/content-media/pdf-articles/How_to_Apply_PGR_to_Invincibelle_Spirit.pdf>.

Artetxe, A., V. Terés, and A.I. Beunza. 1997. Effects of container size and substrates on *Hydrangea macrophylla* growth. Acta Hort. 450:419-424.

Bailey, D.A. 1989. Hydrangea Production. Timber Press. Portland, Oregon. 91 pp.

Bailey, D.A., Clark, B. (1992) Summer applications of plant growth retardants affect spring forcing hydrangeas. HortTechnology 2:213–216.

Bailey, D.A., and P.A. Hammer. 1990. Possible nonpathogenic origin of hydrangea distortion. HortScience 25:1808.

Bailey, K.L., J. Derby, S.M. Boyetchko, K. Sawchyn, E. Becker, G. Sumampong, S. Shamoun, D. James, S. Masri, and A. Varga. 2012. *In vivo* studies evaluating commercial biofungicide suppression of blight caused by *Phytophthora ramorum* in selected ornamentals. Biocontrol Sci. and Tech.. 22: 1268-1283.

Bir, D. 2000. Pruning hydrangeas. Accessed November 23, 2015. <http://www2.ca.uky.edu/HLA/Dunwell/hydprun.html>.

Blom, T.J. and B.D. Piott. 1992. Florists' hydrangea blueing with aluminum sulfate applications during forcing. HortScience. 27:1084-1087.

Bounfour, M. and L.K. Tanigoshi. 2001. Effect of temperature on development and demographic parameters of *Tetranychus urticae* and *Eotetranychus carpini borealis* (Acari: Tetranychidae). Annu. Entomol. Soc. Amer. 94:400-404.

Boyer, C., E. Blythe, J. Griffin, and B. Morales. 2013. Use of root -promoting products for vegetative propagation of nursery crops. Kansas State University Research and Extension Publication MF-3105. Accessed November 23, 2015. <https://www.bookstore.ksre.ksu.edu/pubs/MF3105.pdf>.

Byrne, J. 2010. Bacterial leaf spot on hydrangea. Michigan State University Extension. Accessed January 13, 2016. <http://msue.anr.msu.edu/news/bacterial_leaf_spot_on_hydrangea>.

Capinera, J.L. 2014. Green peach aphid, *Myzus persicae* (Sulzer) (Insecta: Hemiptera: Aphididae). University of Florida IFAS Extension EENY222. Accessed January 13, 2016. <http://edis.ifas.ufl.edu/in379>.

Cayanan, D.F., M. Dixon, Y. Zheng, and J. Llewellyn. 2009. Response of container-grown nursery plants to chlorine used to disinfest irrigation water. HortScience 44:164-167.

Chappell, M.R., M. van Iersel, A. Bayer, L. O'Meara, S. Dove, P. Thomas, P. Alem and R. Ferrarezi. 2011. Monitoring environmental conditions and substrate water content to increase efficiency of irrigation in nurseries. Proceedings of the 2011 Irrigation Association Conference. November 3-8, 2011, San Diego, CA.

Chappell, M.R., S.K. Braman, J. Williams-Woodward, and G. Knox. 2012. Optimizing plant health and pest management of *Lagerstroemia* spp. in commercial production and landscape situations in the Southeastern United States: A review. J. Environ. Hort. 30:161-172.

Chong, J.-H., L. Reid, and M. Williamson. 2009. Distribution, host plants, and damage of the black twig borer, *Xylosandrus compactus* (Eichhoff), in South Carolina. J. Agric. Urban Entomol. 29: 199-208.

Clark, M.J. and Y. Zheng. 2015. Use of species specific controlled release fertilizer rates to manage growth and quality of container nursery crops. HortTechnology. 25:370-379.

Cleveland, T. 2012. Bacterial leaf spot on oakleaf hydrangea. University of Illinois. Home, Yard, and Garden Pest Newsletter 5. Accessed January 13, 2016. <http://hyg.ipm.illinois.edu/article.php?id=367>.

Cochran, D., M. Benitez-Ramirez, and A. Fulcher. 2014. Effect of branch-inducing treatments on growth of tissue culture and cutting-propagated *Hydrangea quercifolia* 'Alice'. J. Environ. Hort. 32:182–188.

Cochran, D. and A. Fulcher. 2013. Type and rate of plant growth regulator influence vegetative, floral growth, and quality of Little Lime™ hydrangea. HortTechnology. 23:306-311.

Cochran, D., A. Fulcher, and G. Bi. 2013. Efficacy of dikegulac sodium applied to pruned and unpruned 'Limelight' hardy hydrangea grown at two locations in the southeastern U.S. HortTechnology, 23:836-842.

Cole, J.C., R.O. Brown, and M.E. Payton. 2013. Two cultivars of oakleaf hydrangea respond to ancymidol, uniconazole, or pinching. HortTechnology. 23:339-346.

Conwell, T., K. Tilt, D. Findley, H. Ponder, and K. Bowman. 2002. Pruning decisions for containerized hydrangeas. Proc. So. Nur. Res. Assoc. 47:495-498.

Daughtrey, M.L., R.L. Wick, and J.L. Peterson. 2000. Botrytis blight of flowering potted plants. Plant Health Progress. DOI:10.1094/PHP-2000-0605-01-HM. Accessed January 13, 2016. <https://www.plantmanagementnetwork.org/pub/php/management/botrytis/>.

Day, E. 2009. Scale insects. Accessed July 23, 2015. <http://pubs.ext.vt.edu/2808/2808-1012/2808-1012.html>.

Dirr, M. 2004. Hydrangeas for American gardens. Timber Press, Portland, OR.

Dirr, M. 2009. Manual of woody landscape plants: their identification, ornamental characteristics, culture, propagation and uses. Stipes Pub., Champaign, IL.

Dirr, M.A. and C.W. Heuser, Jr. 2006. The reference manual of woody plant propagation: From seed to tissue culture. 2nd Ed. Varsity Press, Inc. Cary, NC. 410 pp.

Drees, B.M. 2004. Water wands: High pressure water spray devices for insect and mite control. Texas AgriLife Ext. EEE-00006. Accessed November 23, 2015. <http://extentopubs.tamu.edu/eee_00006.html>.

Dudek, T. 2011. Red headed flea beetle: a new pest of nursery crops in Michigan. Accessed November 23, 2015. <http://msue.anr.msu.edu/news/red_headed_flea_beetle_a_new_pest_of_nursery_crops_in_michigan>.

Dunwell, W., D. Wolfe, and J. Johnson. 2001a. Hydrangeas for cut flowers: Two years of bloom data observations. University of Kentucky Nursery and Landscape Res. Rep. PR 450. Lexington, KY. Accessed November 23, 2015. <http://www.ca.uky.edu/agc/pubs/pr/pr450/PR450.PDF>.

Frank, S.D. and C.S. Sadof. 2011. Reducing insecticide volume and non-target effects of ambrosia beetle management in nurseries. J. Econ. Entomol. 104:1960-1968.

Fulcher, A.F., N. Ward-Gauthier, W.E. Klingeman, and F. Hale. 2015. Blueberry culture and pests, disease, and abiotic disorder management during nursery production in the southeastern U.S.: A review. J. Environ. Hort. 33:33-47.

Gibson, J. and J. Groninger. 2007. Enhancing branching of oakleaf hydrangea. USDA IR-4 Woody Branching Protocol Report. Accessed November 23, 2015. <http://ir4.rutgers.edu/Ornamental/OrnData/20080116p.pdf>.

Gilman, E. 2012. An illustrated guide to pruning. Delmar, Clifton Park, NY.

Gotoh, T. 1997. Annual life cycles of populations of the two-spotted spider mite, *Tetranychus urticae* Koch (Acari: Tetranychidae) in four Japanese pear orchards. Appl. Entomol. Zoology 32: 207-216.

Hagan, A. 2005. Nematode pests of annual and perennial flowers, herbs, woody shrubs, and trees. Alabama Cooperative Extension System ANR-689. Accessed January 13, 2016. <http://www.aces.edu/pubs/docs/A/ANR-0689/ANR-0689.pdf>.

Hagan, A.K. and J.M. Mullen. 2000. Phytophthora rot on woody ornamentals. Alabama Cooperative Extension System ANR-571. Accessed January 13, 2016. <http://www.aces.edu/pubs/docs/A/ANR-0571/ANR-0571.pdf>.

Hagan, A.K. and J.M. Mullen. 2001. Diseases of hydrangea. Alabama Cooperative Extension System ANR-1212. Accessed January 13, 2016. Accessed January 19, 2016. <http://www.aces.edu/pubs/docs/A/ANR-1212/ANR-1212.pdf>.

Hagan, A.K., J.W. Olive, J. Stephenson, and M.E. Rivas-Davila. 2004. Impact of application rate and interval on the control of powdery mildew and cercospora leaf spot on bigleaf hydrangea with azoxystrobin. J. Env. Hort. 22:58-62.

Hagan, A.K., J.W. Olive, J. Stephenson, and M.E. Rivas-Davila. 2005. Control of powdery mildew and cercospora leaf spot on bigleaf hydrangea with Heritage and Milstop fungicides. Alabama Agricultural Experiment Station Bulletin 658.

Hagen, E., S. Nambuthiri, A. Fulcher, and R. Geneve. 2014. Growth and water consumption of *Hydrangea quercifolia* irrigated by on demand and daily water use irrigation regimes. Scientia Hort. 179:132-139.

Halcomb, M. and A. Windham. 2011. Southern blight. The University of Tennessee Extension. Accessed January 13, 2016. <https://extension.tennessee.edu/mtnpi/Documents/handouts/Insect%20and%20Disease%20Control/Southern_Blight.pdf>.

Halcomb, M., S. Reed, and A. Fulcher. 2013. Hydrangeas. UT-UK IPM for Shrub Production Manual. University of Tennessee. Accessed November 22, 2015. <https://ag.tennessee.edu/plantsciences/Pages/AFulcher/SustainableNurseryCrop.aspx>.

Handrick, K.A. 1997. Production of blue hydrangea flowers without aluminum drenches. Commun. Soil Sci. Plant Analysis. 28:1191-1198.

Herms, D.A. 2004. Using degree-days and plant phenology to predict pest activity. In: IPM of Midwest Landscapes, pp. 49-59. V. Krischik and J. Davidson, eds. Minnesota Agricultural Experiment Station Publication SB-07645.

Hester, K.A., G. Bi, M.A. Czarnota, A. Fulcher, G.J. Keever, J.H. Leith, J.D. Orsi, B.E. Whipker, K. Sullivan, and C.L. Palmer. 2013. Impact of Augeo, Configure, and Florel on hydrangea branching. J. Environ. Hort. 31:27-29.

Holland, A.S., G.J. Keever, J.R. Kessler, and F. Dane. 2007. Cyclanilide applications promote branching of woody ornamentals. J. Environ. Hort. 25:139-144.

Jacques, R.L. and D.C. Peters. 1971. Biology of *Systena frontalis*, (Coleoptera: Chrysomelidae) with special reference to corn. J. Econ. Entomol. 61: 135-138.

Johnson, W.T. and H.H. Lyon. 1991. Insects that feed on trees and shrubs, 2nd ed. Cornell Univ. Press, Ithaca, NY. 560 pp.

Knox, G.W. 2013. French hydrangea for gardens in north and central Florida. University of Florida IFAS Extension ENH1069. Accessed January 13, 2016. <https://edis.ifas.ufl.edu/ep330>.

Krueger, R. and R. McSorley. 2015. Solarization for pest management in Florida. University of Florida IFAS Extension ENY902. Accessed January 19, 2016. <https://edis.ifas.ufl.edu/in824>.

Kunkel, B.A. 2012. Sustainable management tools for the redheaded flea beetle in nurseries. SARE, NE Region Final ONE12-163 Project Report. Accessed November 23, 2015. <https://mysare.sare.org/sare_project/one12-163/>.

Leonard, M.D. 1964. Additional records of New Jersey aphids. J. New York Entomol. Soc. 72: 79-101.

Li, Y., R. Trigiano, S. Reed, T. Rinehart, and J. Spiers. 2009. Assessment of resistance components of bigleaf hydrangeas (*Hydrangea macrophylla*) to *Erysiphe polygoni* in vitro. Canadian J. of Plant Path. 31(3):348-355. Accessed January 13, 2016. <http://www.tandfonline.com/doi/pdf/10.1080/07060660909507609>.

Lockhart, B., D. Mollov, and M. Daughtry. 2013. First report of *alfalfa mosaic virus* occurrence in Hydrangea in the United States. Plant Dis. 97:1258.

Machado Caballero, J.E., B.E. Lockhart, S.L. Mason, and M. Daughtrey. 2009. Identification and properties of a carlavirus causing chlorotic mottle of florists' hydrangea (*H. macrophylla*) in the United States. Plant Dis. 93: 891-895.

Maltais, P.M. and M.C. Ouellette. 2000. A note on *Systena frontalis* (Coleoptera:Chrysomelidae) adults on lowbush blueberry, *Vaccinium angustifolium*. Phytoprotection 81:129-131.

Mangold, J.R., R.C. Wilkinson and D.E. Short. 1977. Chlorpyrifos sprays for control of *Xylosandrus compactus* in flowering dogwood. J. Econ. Entomol. 70:789-790.

McNiel, R., B. Vaneva, J. Snyder, and S. Bale. 2007. Hydrangea production in containers as a system to generate floral cut stems. University of Kentucky Nursery and Landscape Res. Rep. Lexington, KY. Accessed November 23, 2015. <http://www.ca.uky.edu/agc/pubs/pr/pr554/pr554.pdf>.

Midcap, J. 2004. Low phosphorus and slow release iron effects on hydrangea production. Center for Applied Nursery Res. Rep. Dearing, GA. Accessed November 23, 2015. <http://www.canr.org/pastprojects/2004008.pdf>.

Midcap, J. and T. Bilderback. 2002. Evaluating hydrangea production with improved substrates. Center for Applied Nursery Res. Rep. Dearing, GA. Accessed November 23, 2015 <http://www.canr.org/pastprojects/2002014.pdf>.

Miralles, J., R. Valdéz, J.A. Franco, S. Bañón, and M.J. Sánchez-Blanco. 2013. Irrigation of *Hydrangea* with saline reclaimed wastewater: Effects of fresh water flushing. Acta Hort. 1000:229-236.

Mizell, R.F. and T.C. Riddle. 2004. Evaluation of insecticides to control the Asian ambrosia beetle, *Xylosandrus crassiusculus*. Proc. Southern Nursery Assoc. Res. Conf. 49:152-155.

Mmbaga, M.T. and J.B. Oliver. 2007. Effects of biopesticides on foliar diseases and japanese beetle (*Popillia japonica*) adults in roses (*Rosa* spp.), oakleaf hydrangea (*Hydrangea quercifolia*), and crapemyrtle (*Lagerstroemia indica*). Arbor. Urban For. 33:210-219.

Mmbaga, M.T., M.S. Kim, L. Mackasmiel and Y. Li. 2012. Evaluation of *Hydrangea macrophylla* for resistance to leaf-spot diseases. J. Phytopath. 160(2): 88-97. Accessed January 13, 2016. <http://onlinelibrary.wiley.com/doi/10.1111/j.1439-0434.2011.01862.x/full>.

Morel, P. 2001. Growth control of *Hydrangea macrophylla* through water restriction. Acta Hort. 548:51-58.

Mullen, J. 2001. Southern blight, southern stem blight, white mold. The Plant Health Instructor. DOI: 10.1094/PHI-I-2001-0104-01. Accessed January 13, 2016. <http://www.apsnet.org/edcenter/intropp/lessons/fungi/Basidiomycetes/Pages/SouthernBlight.aspx>.

Mussey, G.J. and D.A. Potter. 1987. Phenological correlations between flowering plants and activity of urban landscape pests in Kentucky. J. Econ. Entomol. 90: 1615-1627.

NPB (National Plant Board). 2013. U.S. D*omestic Japanese Beetle Harmonization Plan*. Accessed November 23, 2015. <http://nationalplantboard.org/japanese-beetle-harmonization-plan/>.

Ngoan, N.D., R.C. Wilkinson, D.E. Short, C.S. Moses, and J.R. Mangold. 1976. Biology of an introduced ambrosia beetle, *Xylosandrus compactus*, in Florida. Ann. Entomol. Soc. Am. 69:872-876.

O'Meara, L., M.W. van Iersel, and M.R. Chappell. 2013. Modeling daily water use of *Hydrangea macrophylla* and *Gardenia jasminoides* as affected by environmental conditions. HortScience 48:1040-1046.

O'Meara, L., M.R. Chappell, and M.W. van Iersel. 2014. Water use of *Hydrangea macrophylla* and *Gardenia jasminoides* in response to a gradually drying substrate. HortScience 49:493-498.

Opena, G.B. and K.A. Williams. 2002. Use of precharged zeolite to provide aluminum during blue hydrangea production. J. Plant Nutrition. 26:1825-1840.

Paul, T.M. and A.Q. Jhon. 1992. Influence of node number and IBA treatments on rooting of *Hydrangea macrophylla* (Thunb.) cuttings. Adv. Plant Sci. 5:619-622.

Pershey, N.A. 2014. Reducing water use, runoff volume, and nutrient movement for container nursery production by schedule irrigation based on plant daily water use. East Lansing, Mich. State Univ., East Lansing, M.S. Thesis.

Plant and Supply Locator. 2015. Accessed November 22, 2015. <http://www.plantlocator.net/>.

Potter, M.F. 2008. Spider mites on landscape plants. Univ. Ky. ENTFACT-438. Accessed November 23, 2015. <http://www2.ca.uky.edu/entomology/entfacts/ef438.asp>.

Pratt, P.D., P. Schausberger, and B.A. Croft. 1999. Prey-food types of *Neoseiulus fallacis* (Acari: Phytoseiidae) and literature versus experimentally derived prey-food estimates for five phytoseiid species. Exp. Applied Acarology 23:551-565.

Pscheidt, J.W. and C.M. Ocamb (Eds.). 2015. Pacific Northwest plant disease management handbook. Oregon State University, Corvalis, OR. Accessed January 13, 2016. <https://pnwhandbooks.org/plantdisease>.

Raulston, J.C. 1995. Propagation guide for woody plants in the NCSU Arboretum. NCSU Arboretum, Raleigh, NC. 73 pp.

Raupp, M.J., A.B. Cumming, and E.C. Raupp. 2006. Street tree diversity in Eastern North America and its potential for tree loss to exotic borers. Arboric. Urban For. 32:297-304.

Rebek, E.J., and C.S. Sadof. 2003. Effects of pesticide applications on the euonymus scale (Homoptera: Diaspididae) and its parasitoid, *Encarsia citrina* (Hymenoptera: Aphelinidae). J. Econ. Entomol. 96:446-452.

Reding, M., O.P. Schultz, and C. Ranger. 2010. Monitoring flight activity of ambrosia beetles in ornamental nurseries with ethanol-baited traps: Influence of trap height on captures. J. Envir. Hort. 28: 85-90.

Regan, R. 1994. Variation in water use of container-grown plants. Proc. Intl. Plant Prop. Soc. Annu. Conf. 44:310-312.

Riley, T.J. 1983. Damage to soybean seeds and seedlings by larvae of the flea beetle, *Systena frontalis* (Coleoptera: Chrysomelidae). J. Georgia Entomol. Soc. 18: 287-291.

Roeber, R. and H. Hass. 1997. Plant quality and growth of *Hydrangea* x*hybrida* as influenced by temperature and water quantity. Acta Hort. 450:425-432.

Ruffoni, B., S. Ermanno, and M. Savona. 2013. In vitro propagation of *Hydrangea* spp. Methods Mol Biol. 994:231-244. DOI: 10.1007/978-1-62703-074-8_18.

ScaleNet. 2013. Accessed November 23, 2015. < http://scalenet.info/flatcat/>.

Schweigkofler, W., K. Kosta, V. Huffman, S. Sharma, K. Suslow and S. Ghosh. 2014. Steaming inactivates *Phytophthora ramorum*, causal agent of sudden oak death and ramorum blight, from infested nursery soils in California. Plant Health Progress 15:43.

Smith, E.H. 1970. Taxonomic revision of the genus *Systena* Chevrolat (Coleoptera: Chrysomelidae: Alticinae) North of Mexico, M.S. Thesis, Purdue Univ. 179 p.

Sinclair, W.A. and H.H. Lyon. 2005. Diseases of trees and shrubs (2nd ed.). Comstock Publishing, Cornell University Press. Ithaca, NY.

Stanyard, M.J., R.E. Foster, and T.J. Gibb. 1997. Effects of orchard ground cover and mite management options on the population dynamics of European red mite (Acari: Tetranychidae) and *Amblyseius fallacis* (Acari: Phytoseiidae) in apple. J. Econ. Entomol. 90:595-603.

Stoven, H. and J. Owen. 2008. Comparison of substrate amendments for the adjustment of hydrangea [*Hydrangea macrophylla* (Thunb.) SER 'Bailmer', Endless Summer®] flower color. Proc. So. Nur. Res. Conf. 53:32-35.

Sun, Y., G. Bi, G. Niu, and C. Perez. 2015. Foliar application of dikegulac sodium increases branching of ‘Merritt’s Supreme’ bigleaf hydrangea. HortTechnology 25:306-312.

Uddin, W., S.M. McCarter, and R.D. Gitaitis. 1996. First report of oakleaf hydrangea bacterial leaf spot caused by a pathovar of *Xanthomonas campestris*. Plant Dis. 80(5):599. Accessed January 13, 2016. <https://www.apsnet.org/publications/plantdisease/backissues/Documents/1996Abstracts/PD_80_0599B.htm>.

USDA. 2009. 2007 Census of agriculture. United States Department of Agriculture, Washington, DC.

van Iersel, M., K. Seader, and S. Dove. 2009a. Exogenous abscisic acid application effects on stomatal closure, water use, and shelf life of hydrangea (*Hydrangea macrophylla*). J. Environ. Hort. 27:234-238.

van Iersel, M., R.M. Seymour, M. Chappell, and F. Watson. 2009b. Soil moisture sensor-based irrigation reduces water use and nutrient leaching in a commercial nursery. Proc. So. Nursery Res. Conf. 54:17-21.

Vann, S. 2009. Cercospora leaf spot of hydrangea. University of Arkansas Division of Agriculture. FSA-7570-PD-11-09N. Accessed January 13, 2016. < https://uaex.edu/publications/PDF/FSA-7570.pdf>.

Veerakone, S., L.W. Liefting, B.S.M. Lebas, and L. Ward. 2012. First report of *Cherry leaf roll virus* in *Hydrangea macrophylla*. Plant Dis. 96(3):463. Accessed January 13, 2016. <http://apsjournals.apsnet.org/doi/abs/10.1094/PDIS-08-11-0708>.

Warsaw, A.L., R.T. Fernandez, B.M. Cregg, and J.A. Andresen. 2009. Water conservation, growth, and water use efficiency of container-grown woody ornamentals irrigated based on daily water use. HortScience. 44:1308-1318.

Weiler, T.C. and L.C. Lopes. 1974. Hydrangea distortion. Focus on floriculture. 2:9.

Windham, M.T., S.M. Reed, M.T. Mmbaga, A.S. Windham, Y. Li, and T.A. Rinehart. 2011. Evaluation of powdery mildew resistance in *Hydrangea macrophylla*. J. Env. Hort. 29(2):60-64. Accessed January 13, 2016. <http://pubag.nal.usda.gov/pubag/downloadPDF.xhtml?id=49710&content=PDF>.

Wood, S.L. 1982. The bark and ambrosia beetles of North and Central America (Coleoptera: Scolytidae), a taxonomic monograph. Great Basin Nat. Mem. 6:1-1359.

Yeh, D.M. and H.H. Chiang. 2001. Growth and flower initiation in hydrangea as affected by root restriction and defoliation. Sci. Hort. 91:123-132.

Notes:
Discussion of Japanese beetles (*Popillia japonica)* is largely reprinted from text prepared for Optimizing Plant Health and Pest Management of *Lagerstroemia* spp. in Commercial Production and Landscape Situations in the Southeastern U.S.: A Review (Chappell et al. 2012. J. Environ. Hort. 30:161-172). Reprint of this content herein is made possible by permission of the original content authors.

Discussion of twospotted spidermite (*Tetranychus urticae)* is largely reprinted from text prepared for Chong et al. 2012. Cherry – *Prunus* spp., p. 79-108. In: A.F. Fulcher and S.A. White (eds.). IPM for select deciduous trees in southeastern US nursery production. Southern Nursery IPM Working Group, Knoxville, TN. Reprint of this content herein is made possible by permission of the original content authors.

Discussion of Oystershell scale (*Lepidosaphes ulmi*) management is largely based on text prepared for A Review of Arthropod Pests, Plant Diseases and Abiotic Disortders and their Management on Viburnum species in the Southeastern U.S. (Klingeman et al. 2014; J. Environ. Hort. 32:84-104). Reprint of this content herein is made possible by permission of the original content authors.

Discussion of Cottony camellia scale (*Pulvinaria floccifera*) management is largely adapted from text prepared for Knox et al. 2014. Camellia - *Camellia* spp., p. 31-62. In: IPM for Shrubs in Southeastern U.S. Nursery Production: Vol. 1. S.A. White and W.E. Klingeman, Eds. Clemson, SC: Southern Nursery IPM Working Group. Print ISBN: 978-0-9854998-2-2. Reprint of this content herein is made possible by permission of the original content authors.

Loropetalum - *Loropetalum chinense*

Matthew Chappell, University of Georgia
Juang-Horng (JC) Chong, Clemson University
Geoff Weaver, University of Georgia
Jean Williams-Woodward, University of Georgia

History, Culture and Management

Figure 2.1 Petals of loropetalum flowers, shown here as the pink flowers of *Loropetalum chinense* var. *rubrum* 'Burgundy', look like straps - as do petals of many species in the witchhazel family.

INTRODUCTION

1. Historical Perspective & Ornamental Value
2. Cultivars
3. Propagation
4. Container Size, Timing and Pruning
5. Fertility & Irrigation
6. Plant Growth Regulators to Control Size and Enhance Branching

Introduction

Loropetalum (*Loropetalum chinense* Oliv.) is in the witchhazel family, with its name derived from the Greek text 'strap' and 'petal' (Figure 2.1). There are many ornamental taxa in this family, with some of the more popular genera being *Fothergilla*, *Hamamelis*, and *Liquidambar* (Figure 2.2) (Gapinski, 2015). Many genera in the witchhazel family are concurrently found in both North America and central Asia, at similar latitudes with similar climate, and therefore many Asian species have been successfully introduced into the North American ornamental marketplace. Loropetalum's native range is quite vast, and generally falls along and to the south of the Yangtze River in China as well as the Japanese mountainous areas on Kumanu and Miyazaki on Kyushu Island and Chubu and Kansai on Honshu Island (Gong et al., 2016).

Figure 2.2 Flowers of (a) *Fothergilla*, (b) *Hamamelis*, and (c) *Liquidambar* all share similar floral structure, with strap-like petals borne on flowers originating from the axils of nodes.

Historical Perspective

The history of *L. chinense* from a landscape plant perspective has two distinct periods. The first loropetalum taxa were introduced into the U.S. via the Arnold Arboretum in 1880. These green leaved (white flowering) forms (Figure 2.3) could be traced back to the original taxa collected in the vicinity of Jiujiang (Kiukiang) in south central China at an elevation of approximately 1,000ft. (330m). Plants were initially introduced from these natural populations into the European market by Charles Maries of Veitch and Son Nursery in the mid 1800's and later to the U.S. It should be noted that initial loropetalum taxa had cold hardiness of approximately USDA Zone 8-9 (Figure 2.4; Table 2.1), due to the origin of the taxa. Therefore, the species was only grown in southern Europe and once introduced to the U.S.; southern states and California (ironic considering Arnold Arboretum is in USDA Zone 6). Later, in the 1960's, additional green foliage taxa were introduced into the U.S. market from collections made from more northern latitudes, likely Japan. These taxa were vastly more cold hardy, and records exist of plantings in the Washington D.C. and Baltimore area that were cold hardy (Weaver Jr., 1976).

Figure 2.3 Upon its introduction into the U.S. in 1880 until red-foliaged forms were introduced in 1989, all loropetalum in the U.S. market were green foliage forms.

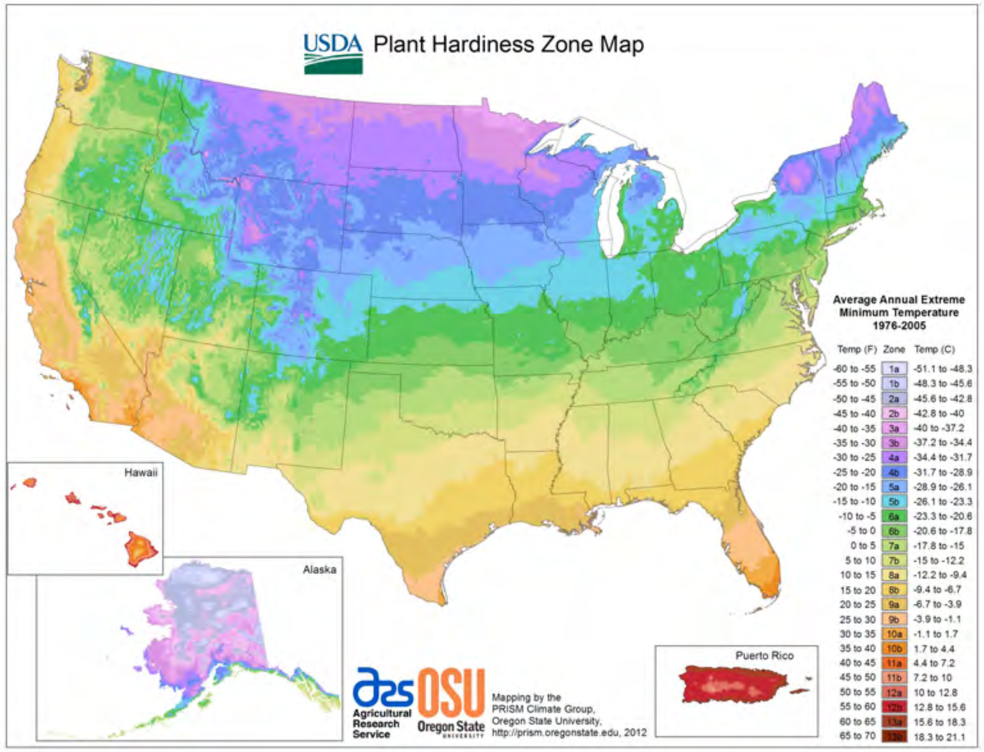


Figure 2.4 USDA Plant Hardiness Zone Map. USDA-ARS and OSU

Ornamental Value

The ornamental popularity of loropetalum increased dramatically once red-foliaged cultivars (Figure 2.5) were introduced into the U.S. in 1989. Red foliage (pink flowering) forms were actually found in the wild, purportedly by Nihonkaki Nursery in Japan, and are a naturally occurring variety. Hence, the taxonomic classification of these forms is *Loropetalum chinense* (Oliv.) var. *rubrum* Yieh. Dr. John Oliver, former director of the U.S. National Arboretum, introduced the first two cultivars of *Loropetalum chinense* var. *rubrum* into the trade, 'Blush' and 'Burgundy' (Figure 2.6). These two cultivars had a cold hardiness of USDA Zone 7, with cold injury observed between 0 – 5°F (U.S. National Arboretum, 1999). This level of cold hardiness is common among all red-foliage cultivars on the market today (Table 2.1).

Figure 2.5 The red-foliage *Loropetalum chinense* var. *rubrum* cultivars 'Blush' and 'Burgundy' were introduced in 1989. Pictured here is a mature 'Blush' plant on the campus of The University of Georgia.

Cultivars

Currently (as of March 2017), there are 56 cultivars of loropetalum on the market (Table 2.1; footnotes depict cross-listed names). The majority of these cultivars were introduced in the last decade, with the average number of cultivars released annually over the last 5-years at six. Some of these cultivars are simply 'Blush', 'Burgundy' or other unpatented cultivars that have been renamed in local or regional markets. Also of interest is that the majority of cultivars fall into several closely-related groups, likely based on shared parentage due to limited initial genotypic variation (Gawel et al., 1996). This, in the long-term, may prove to negatively affect the genera should a species-specific pest or disease complex emerge. While national statistics do not rank loropetalum sales within the top ten shrubs, in the Southeast it is a staple nursery crop, with virtually every nursery producing at least one cultivar of the species (Plant Locator, 2016).

Figure 2.6 The red-foliage *Loropetalum chinense* var. *rubrum* cultivars 'Blush' and 'Burgundy' were introduced in 1989. Pictured here is 'Burgundy'.

Table 2.1. Cultivars of *Loropatalum chinense* available on the market as of spring 2017.

Trade or Cultivar Name	Foliage Color	Flower Color	Plant Size (W x H)	Habit
Bicolor [1]	deep maroon	white	3-4 ft X 3-4 ft	small shrub
Bill Wallace [2]	purple-green	hot pink	4-5 ft X 1-2 ft	small spreading shrub
Blush [3]	olive green	fuchsia pink	4-6 ft X 6-8 ft	medium shrub
Burgundy [4]	purple-green	hot pink	6-10 ft X 6-10 ft	large shrub to small tree
Carolina Moonlight	green	white	4-5 ft X 3-4 ft	small compact shrub
Carolina Ruby Red	burgundy	fuschia pink	5-10 ft X 5-10 ft	large shrub
Cerise Charm [5]	burgundy	hot pink	2-3 ft X 2-3 ft	small shrub
Chang's Ruby	burgundy	hot pink	3-5 ft X 3-5 ft	small shrub
Cherry Blast	dark burgundy	dark pink-red	5-6 ft X 5-6 ft	medium compact shrub
Crimson Fire	deep plum	bright pink	3-4 ft X 2-3 ft	dwarf mounding shrub
Dark Fire [6]	dark purple	pink	5-6 ft X 5-6 ft	small compact shrub
Daruma [7]	deep plum	bright pink	3-5 ft X 2-5 ft	dwarf mounding shrub
Daybreak's Flame [8]	bronze purple	hot pink	5-10 ft X 5-8 ft	large spreading shrub
Emerald Snow [9]	green	white	4-5 ft X 4-5 ft	small shrub
Ever Red [10]	dark burgundy	dark pink-red	5-6 ft X 5-6 ft	medium compact shrub
Firedance [11]	red-purple	fuchsia pink	3-6 ft X 3-6 ft	medium rounded shrub
Garnet Fire [12]	burgundy	dark pink-red	3-5 ft X 3-5 ft	small mounding shrub
Green Elf	green	white	2-3 ft X 1-3 ft	dwarf compact shrub
Hillier Compacta [13]	green	white	2-3 ft X 1-3 ft	dwarf spreading shrub

[1] Vigorous growth. Older leaves turn olive green. Flowers may be streaked with pink.
[2] Derived from a branch mutation of 'Burgundy'. May suffer from cold damage in USDA zone 7.
[3] New growth is bronze-red. One of two original introductions of Loropetalum chinense var. Rubrum. Occassionally sold as 'Razzleberry' or 'Raspberry Fringe'.
[4] New growth is reddish purple and leaves may turn bright red in Autumn. One of 2 original introductions of Loropetalum chinense var. Rubrum.
[5] Also known as 'Kurobijin'.
[6] Foliage color persists through Summer heat.
[7] Superior dwarf cultivar.
[8] New foliage is bronze-red. Older leaves turn green and may turn orange in Autumn. Tips of flowers pale pink to white. Arching, spherical growth habit.
[9] Also known as 'Shang-White'. New foliage is lime green.
[10] Also known as 'Chang Nian Hong'. Foliage color persists through Summer.
[11] New foliage is ruby red. Older leaves turn green.
[12] Leaves hold color well.
[13] Groundcover-like growth habit. Parent plant for some other white-flowering forms. May also be known as "Hillier form" or simply "Hillier's".

Table 2.1 (*continued*) Cultivars of *Loropatalum chinense* available on the market as of spring 2017.

Trade or Cultivar Name	Foliage Color	Flower Color	Plant Size (W x H)	Habit
Hindwarf [14]	burgundy	dark pink-red	3-5 ft X 1-2 ft	dwarf spreading shrub
Jade Confetti [15]	green	white	4-6 ft X 6-8 feet	medium shrub
Jazz Hands Bold [16]	dark purple	hot pink	5-6 ft X 5-6 ft	medium shrub
Jazz Hands Dwarf Pink	deep purple	hot pink	2-3 ft X 1-3 ft	dwarf mounding shrub
Jazz Hands Dwarf White [17]	green	white	2-3 ft X 1-3 ft	dwarf mounding shrub
Jazz Hands Mini [18]	black-purple	hot pink	3 ft X 1 ft	dwarf shrub
Jazz Hands Variegated [19]	variegated deep purple	hot pink	4 ft X 4-6 ft	medium mounded shrub
Little Rose Dawn [20]	burgundy	rose pink	12-15 ft X 7-8 ft	large spreading shrub
Longwood Hardy	green	white	3-6 ft X 3-6 ft	medium shrub
Melting Pot	burgundy	white	2 ft X 2 ft	dwarf shrub
Merlot Lace	burgundy-purple	hot pink	5 ft X 4 ft	small shrub
Mosaic [21]	variegated burgundy	pink	6-8 ft X 5 ft	medium shrub
Pipa's Red	burgundy	hot pink	5-6 ft X 5 ft	medium shrub
Pizazz [22]	dark purple	dark pink	6-8 ft X 6-8 ft	medium shrub
Plaze [23]	dark purple	dark pink	4 ft X 4 ft	small shrub
Plum Delight [24]	bronze-purple	dark pink	6-8 ft X 6-8 ft	medium shrub
Plum Gorgeous	deep purple	hot pink	5-6 ft X 4-5 ft	medium shrub
Purple Daydream	dark purple	hot pink	3-4 ft X 2-3 ft	dwarf shrub
Purple Diamond [25]	deep purple	hot pink	4-5 ft X 4-5 ft	small shrub
Purple Majesty	burgundy-purple	fuchsia pink	4-6 ft X 6-8 ft	large shrub

[14] Mature leaves turn green. Similar to 'Sato's Red Dwarf'.
[15] Hardy to USDA zone 6.
[16] Leaves and flowers are larger than most loropetalum cultivars.
[17] Blooms repeatedly throughout Summer.
[18] Very small cultivar.
[19] New foliage is variegated with white and pink splotches.
[20] Compact growth and profuse blooms, mature foliage turns green. Originated from a sport of 'Ruby'.
[21] Sectoral yellow variegation.
[22] New foliage is reddish-purple.
[23] Derived from a 'Pizzaz' seedling.
[24] Also known as 'Hines Purpleleaf'. New growth is rose-purple.
[25] Also known as 'Shang-hi'. Foliage color persists through Summer.

Table 2.1 (*continued*) Cultivars of *Loropatalum chinense* available on the market as of spring 2017.

Trade or Cultivar Name	Foliage Color	Flower Color	Plant Size (W x H)	Habit
Purple Pixie [26]	deep purple	hot pink	4-5 ft X 1-2 ft	small spreading shrub
Razzleberri [27]	burgundy	pale pink	4-6 ft X 4-5 ft	medium shrub
Red Diamond [28]	dark burgundy	dark pink-red	6 ft X 6 ft	medium compact shrub
Rosy Baby	dark plum	pink	2-3 ft X 4-5 ft	low spreading shrub
Ruby [29]	burgundy	rose pink	3-5 ft X 3-5 ft	small shrub
Ruby Parfait [30]	variegated red-purple	hot pink	3-4 ft X 3-4 ft	small shrub
Ruby Snow	burgundy	white	6 ft X 6 ft	medium shrub
Sato's Dwarf Red [31]	burgundy	dark pink-red	3-5 ft X 1-2 ft	small spreading shrub
Shidare [32]	green	white	5-8 ft X 5-8 ft	large weeping shrub
Sizzling Pink [33]	burgundy-purple	dark pink	4-5 ft X 4-6 ft	medium shrub
Snow Muffin [34]	green	white	2-3 ft X 1-3 ft	dwarf mounding shrub
Snow Panda [35]	green	white	6-8 ft X 8-10 ft	large columnar shrub
Sparkling Sangria	deep maroon	electric pink	6-10 ft X 6-10 ft	large shrub
Suzanne [36]	red-maroon	hot pink	3-4 ft X 3-4 ft	small shrub
Tokyo Weeping [37]	green	white	10 ft X 10 ft	large weeping shrub
Zhuzhou Fuschia [38]	black-maroon	deep pink	4-6 ft X 8-12 ft	large shrub to small tree

[26] Also known as 'Peack'. Foliage color persists through Summer.
[27] Also known as 'Monraz'. Flowers have a reddish fringe.
[28] Also known as 'Shang-red'.
[29] Mature foliage turns green.
[30] New foliage is red to purple with marbled white variegation. Leaves mature to a dark red-purple.
[31] Similar to 'Hindwarf' but mature leaves do not turn green.
[32] Weeping growth habit. Specimen at J.C. Raulston arboretum.
[33] Flowers may be tinted slightly purple. Reported to have improved cold hardiness although this is not reflected in the UDA zone recommendation.
[34] Also known as 'Snowmound'. Very dense growth.
[35] Columnar growth habit.
[36] Mature plants have peeling bark.
[37] Weeping growth habit. Specimen at J.C. Raulston arboretum.
[38] Can reach heights up to 20 ft with age. Said to be the most cold-hardy of the pink-flowering loropetalums although this is not reflected in the USDA zone recommendations.

Note: A number of cultivars available in Europe were excluded from this list (e.g. ‘Black Pearl’. ‘Chang Nian-Hong’, ‘Ming Dynasty’, and ‘Tang Dynasty’) as there is limited to no commercial availability of these cultivars in North America. .

Propagation

Propagation of loropetalum is typically carried out using vegetative methods to maintain genetic purity of a cultivar (Figure 2.7). However, seed propagation is possible and rather easily accomplished, as seed are prevented from germinating by a simple physiological dormancy mechanism. Seed is produced as an aggregate of samaras (Figure 2.8) and should be collected when it starts to brown in early to mid-summer. Seed should be cold/wet stratified for 90 days at 36-40°F (2.2-4.4°C) and planted immediately after removing from cold storage (Chappell, 2016). Dirr and Heuser Jr. (2006) and Blazich & LeBude (2015) reported successful vegetative propagation (80%) in late July, when stems are at a semi-hardwood stage, using 3,000 ppm IBA quick dip in a peat:perlite mix under mist. Care should be taken to use a light mix, at least 50% perlite. Authors of this chapter have rooted in 100% perlite. If a heavy mix is used, root-rot will become problematic. Cuttings should be overwintered in the propagation trays and not shifted up until the year after propagated. Shifting up in the fall of the propagation year results in massive losses over the initial winter.

Figure 2.7 Propagation of loropetalum is typically via vegetative (“cutting”) methods, as seen here.

Figure 2.8 Pictured here is a seed capsule collected from the cultivar ‘Blush’.

Weed control in propagation (Figure 2.9) has been an increasingly important subject, as maintaining clean propagation material will result in cleaner nursery stock downstream. This is a central theme in any IPM strategy. Cochran et al. (2008) examined three herbicides in the propagation of loropetalum, to determine efficacy and damage to propagules: Gallery (isoxaben), Ronstar 2G (oxadiazon), and Regal O-O (oxyfluorfen + oxadiazon). Herbicides were applied at three separate times during the propagation process: before sticking, lightly rooted, or fully rooted. One year after sticking, growth indices of ‘Ruby’ loropetalum were similar regardless of when Gallery was applied. Shoot growth was similar about one year later when Ronstar and Regal O-O were applied; however, root coverage was suppressed with Ronstar applied before sticking and at lightly rooted, while Regal O-O suppressed root coverage on all dates of application.

Figure 2.9 Weed control in propagation can be difficult due to label restrictions coupled with phytotoxic effects of preemergence herbicides on newly rooted cuttings.

Container Size, Timing and Pruning

In nursery production, the majority of loropetalum are grown in #1 or #3 containers; with in-ground production only producing specimen plants for high-end landscapes. In #1 containers, the production cycle is generally 9-12 months, with single liners shifted up into #1 containers in early summer and grown out one growing season, overwintered, and shipped after a spring growth flush. Plants are generally sheared while in propagation trays, prior to shifting up, and again in late winter prior to the spring growth flush and sale date. In #3 containers, a single or up to 3 liners are placed in the containers. ‘Double-sticking’ (placing two liners; Figure 2.10) or ‘triple-sticking’ (placing three liners) in a container is done to speed up the production cycle. Most growers double-stick into #3 containers, which results in a 12 to 18-month production cycle (Figure 2.11). Liners are shifted up into #3 containers in early summer (May-July depending on location) and grown out one growing season, overwintered, and grown out a second growing season, with estimated sale date in fall of the second growing season or spring of the third season immediately after new growth is initiated. Plants are generally sheared while in propagation trays, prior to shifting up, again in late winter prior to the spring growth flush of the second season and if not sold that fall again in winter prior to the third season spring growth flush.

Figure 2.10 Many growers ‘double stick’ liners into #3 containers to reduce the time needed to finish a container.

Planting depth is an important factor in container production of shrubs. Planting too deep can lead to stem rot and eventual failure of the

Figure 2.11 'Double sticking' liners can significantly reduce finishing time of larger containers, by as much as a year. Pictured here is a double-stuck loropetalum 'Crimson Fire', six (top photo) and twelve (bottom photo) months after shifting up from liners into a #3 container.

liner/plant once put into general production. Pecot (2004) examined the effect of planting loropetalum at grade (properly), 1.5-inches below-grade (too deep) and 3-inches below-grade in #1 containers. The effects of planting too deep were significant in loropetalum, more so than other common shrubs (including gardenia and Indian hawthorn). Both growth index and plant quality were reduced when planted even slightly below-grade (1.5-inches).

Fertility

A major obstacle to the production of loropetalum, particularly *L. chinense* var. *rubrum* cultivars, prior to 2006, was a disorder termed 'little-leaf disorder' (Ruter, 2006) or loropetalum decline (Figure 2.12). Symptoms included darkening of older foliage, shortening of internodes, upward cupping of foliage, crinkling of new growth and decrease

Figure 2.12 Pictured here is a comparison between loropetalum that are unaffected and affected by little leaf disorder (a.k.a. loropetalum decline). The cause of this disorder is copper deficiency.

in leaf size. Initially, growers suspected mites to be responsible for the damage, but miticides did not solve the problem. Foliar analysis conducted by Ruter (2006) determined that symptomatic plants were deficient in copper, zinc and nickel. Of these, plants treated with copper sulfate foliar spray returned to a normal growth pattern. This discovery resulted in further work with a number of foliar sprays, to determine if foliar application of additional nutrients could enhance loropetalum growth. Shober at al. (2008) foliar applied copper hydroxide and sulfate of boron, manganese and zinc. They also applied Peter's S.T.E.M, which contained sulfur, boron, copper, iron manganese, molybdenum, and zinc. Only those products with copper resulted in increased growth rate. This solidified copper as a limiting nutrient in loropetalum production. Today, many growers fertilize with a standard pelletized controlled-release fertilizer product and apply foliar copper one to three times throughout the growing season to maintain growth. The goal of a fertility program is to maintain optimal levels of foliar nutrition, determined by Simonne et al. (2008) to be (in percent) 1.1-2.3 N, 0.1-0.3 P, 0.5-1.0 K, 0.7-1.5 Ca and 0.1-0.2 Mg (and in ppm) 5-20 B, 5-10 Cu, 20-80 Fe, 10-40 Mn, and 10-40 Zn.

Irrigation

Interestingly, very little formal research has been conducted on the water requirements of loropetalum in containers, with anecdotal information suggesting the species falls into a medium water use grouping (SNA, 2014). However, a survey of growers indicated that most growers consider loropetalum to be a low water user between shifting up and

Figure 2.13 Many commercial nurseries are investigating the use of reclaimed wastewater as an irrigation source, with some nurseries (e.g. Hackney Nursery; Quincy, FL) installing systems to pump water from nearby wastewater treatment facilities to the nursery.

establishment in the container and a medium water user once established in a container. Yeager at al. (2009) did conduct a trial to determine the effect of reclaimed irrigation water on nursery stock (Figure 2.13). Reclaimed water typically has an electrical conductivity higher than groundwater or surface water in the southeast (approximately 1.0 dS/m). Reclaimed water in this study had an EC of 0.96 dS/m and chloride, sodium and pH of the water exceeded desired levels based on established BMPs. Also present in reclaimed water were low levels of nitrogen and phosphorous, although not at levels high enough to enhance growth. Overall, growth of loropetalum when irrigated with reclaimed water was slightly lower than plants irrigated with municipal water, yet the reduction was not great enough to offset the significant reduction in cost of using reclaimed water, which is half the cost of municipal water.

Plant Growth Regulators to Control Size and Enhance Branching

Some cultivars of loropetalum are known for their fast growth rate and/or lack of branching unless pruned. While no work has been conducted on application of PGRs to containerized plants, Smith et al. (2014) and Chen et al. (2012) have reported that flurprimidol, applied as a granular or soil drench, significantly reduced loropetalum growth in landscape situations. In the case of Chen et al. (2012), the researchers made note that the ‘effects were immediate and long-lasting, with reductions in growth at label rate of two years’. In the case of Smith et al. (2014), reductions in growth were approximately 40% when applied at label rate. Both studies indicate the effectiveness of this PGR, but caution should be employed if applying to container-grown crops as not to significantly reduce growth rate and therefore delay sale date. However, if a crop is ready for sale, PGRs may be an option to hold a crop for an additional season. Also, no data has indicated that flurprimidol (or any other PGR) enhances branching of loropetalum in production environments.

Section 2

Arthropod Pest Management

Common Arthropod Pests

1. Ambrosia Beetle
2. Aphids

Figure 2.14 Granulate ambrosia beetle (*Xylosandrus crassiusculus*) is the predominant borer of nursery grown trees in the southeastern U.S.

Loropetalum is a relatively pest-free ornamental shrub. The following arthropod pests are considered occasional pests.

Ambrosia Beetle

Ambrosia beetles (Coleoptera: Curculionidae) are known to attack loropetalum in nurseries occasionally, particularly when the plants are stressed by other environmental factors, such as drought and flood. The granulate ambrosia beetle (*Xylosandrus crassiusculus*) (Figure 2.14) is the predominant borer of nursery grown trees in the southeastern U.S. However, the black twig borer (*Xylosandrus compactus*) is the most common ambrosia beetle pest of loropetalum. Adult ambrosia beetles become active as early as early March in South Carolina (Chong, unpublished data), mid-March in Virginia and Tennessee, and late-April in Ohio (Reding et al., 2010) (Table 2.2). Adult females bore into the branches and trunk of susceptible plants, and construct a gallery

Table 2.2 Seasonal activity of the major arthropod pests of Loropetalum in the mid-southern U.S., and unless otherwise noted, represent occurrence in USDA Plant Hardiness Zone 7[Z].

Arthropod Pest	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Aphids												
Granualte ambrosia beetle												

[Z] Depicted activity may be early or later than shown depending on location. Activities represented in the table are of the most common insect pests.

where eggs are deposited. The adult beetles also deposit symbiotic ambrosia fungi, which serve as the sole food source for the larvae. Adults push out the saw dust through the entry hole as they excavate the gallery. When the moisture content of the saw dust is high enough, the saw dust stick together and form a 'frass tube' (Figure 2.14). The frass tube can be used as a monitoring and diagnostic characteristic, but it can be fragile and often lost on windy and/or rainy days. The beetles complete development in about 55 days (Oliver and Mannion, 2001). Males are small, flightless, and remain in the gallery. Only the females disperse from the original gallery.

Management
Once the beetles bore into the wood, they become protected from insecticide sprays and their host plants become infected with the ambrosia fungi. Therefore, the best management approach is to prevent attacks by the ambrosia beetles. Attacks by the ambrosia beetles are associated with the attraction of the ambrosia beetles to ethanol released by the stressed trees or shrubs (Ranger et al., 2016). The first step in preventing attacks is to reduce stresses on the trees and shrubs by employing proper irrigation practices to avoid flooding or over-saturation of substrate.

Pyrethroids, particularly bifenthrin and permethrin (Table 2.3; send of section), are effective in repelling ambrosia beetle attacks for up to 10 days (depending on ambient temperature) (Mizell and Riddle, 2004). The short residual longevity of the pyrethroid insecticides necessitates repeated applications at 7- to 10-day intervals from the initiation of adult flight to complete flush of the canopy. The initiation of flight activity can be monitored with ethanol-baited traps (Figure 2.15) beginning in late-February in the southeastern U.S. Home-made soda bottle trap, Lindgren funnel trap, or modified Japanese beetle traps functioned well when paired with an ethanol lure (Oliver et al., 2004). Ethanol bait can be prepared in a container and released from a wick, or through purchases of slow-release ethanol baits. Traps should be hung 1.5 to 5.5 feet (0.5 to 1.7 m) above ground (Reding et al., 2010) and checked regularly. Preventive sprays should be initiated as soon as the first ambrosia beetle is captured in the trap, and stopped when the canopy completely flushed. The insecticide solution should be uniformly sprayed to cover all trunk and branch surfaces. Because of the high cost of chemical management, growers are often advised to protect high value, susceptible trees instead of spraying all plant species in the nursery. Systemic neonicotinoids are not effective in preventing attacks by the ambrosia beetles or killing ambrosia beetles that are already in the gallery (Chong, unpublished data).

Figure 2.15 There are many types of traps, using ethanol, that are used to trap boring insects. Pictured here is a Lindgren funnel trap baited with 95% ethanol and fresh redbay stems.

Attacked trees, which are supposedly stressed and more attractive to the ambrosia beetles, could be left in the nursery for 3-4 weeks after the initial attacks to allow more infestations before removing and discarding in order to spare other healthy trees in the nursery from attacks. The attacked trees are best burnt. No effective biological control option is currently available.

Aphids
Aphids (Hemiptera: Aphididae) attack new growth of loropetalum, resulting in the deposition of a large amount of honeydew and the growth of black sooty mold on the foliage. Extensive feeding by aphids can also cause distortion or stunting of the new leaves and buds. Such distortion of new growth can become an issue for producing high quality cuttings or young plants in greenhouses or nurseries.

Figure 2.16 Aphids attack new growth of *Loropetalum*, resulting in the deposition of a large amount of honeydew and the growth of black sooty mold on the foliage. Pictured here is the pea aphid.

Aphids are easily recognized. These are small (often less than 1/10" in length), pear-shaped, soft-bodied, with color ranging from green, yellow to red and black (depending on the species and season) (Figure 2.16). The most characteristic feature of an aphid is the pair of cornicles (or tailpipes) on the rear end of the abdomen. The majority of a population is consisted of wingless nymphs and adults. However, when the feeding site is crowded or during the dispersal phase in the spring and fall, winged adults are produced. These winged adults catch the wind and disperse to new plant parts or new plants.

There is no information on the species of aphids that can infest loropetalum. Generally, aphids are active from spring to fall (Table 2.2), and can infest loropetalum at any time during these seasons. Many aphid species produce live nymphs without mating (parthenogenesis). An enormous population can be built up within a short period of time. There are multiple generations per year.

Management
Aphid populations do not typically remain on loropetalum for long, nor do they cause significant long-term damage. Therefore, infestation by aphids does not always require management. The first step in developing an aphid management program is to develop good weed management and sanitation programs where weeds, debris and infested plants are removed regularly. When the aphid population is small, spraying with a forceful jet of water can also dislodge many from the infested plant. This method, however, may not be practical when a large number of densely packed loropetalum shrubs are infested.

No study has investigated resistance of various loropetalum cultivars to aphids.

Aphid populations are constantly attacked by natural enemies, such as lady beetles, lacewings, syrphid flies and parasitoid wasps. In warm and wet weather, aphid populations are often decimated by outbreak of naturally existing entomopathogenic fungi. The use of broad-spectrum insecticides (such as organophosphates and pyrethroids) (Table 2.3) should be limited to conserve natural enemies. Ants are known to collect honeydew of aphids and interfere with the actions of natural enemies. Therefore, it is advisable to also develop an ant management program to reduce their population in order to improve biological control.

Aphid population is seldom large enough to warrant chemical management. When the need arises, insecticides registered for the management of aphids in the production nursery include carbaryl, acephate, pyrethroids, neonicotinoids (acetamiprid, dinotefuran, imidacloprid and thiamethoxam), pymetrozine, azadiractin, abamectin, spirotetramat, neem oil, horticultural oil and insecticidal soap (Table 2.3). The entomopathogenic fungus *Beauveria bassiana* is also very effective. Systemic insecticides, such as the neonicotinoids, could be applied as topical sprays, soil drench, broadcast granules, or trunk sprays. The neonicotinoids applied through indirect method may require days or weeks to move into the plant tissues. Therefore, these chemicals should be applied before the population increases. They can provide long residual protection against aphid infestation. For an existing population, topical applications of listed insecticides to the infested terminals on a weekly or biweekly basis can be very effective in eliminating the infestation and preventing reinfestation.

Mites
An unidentified species of eriophyid mite was initially identified as the cause of loropetalum 'little leaf disorder' (a.k.a 'loropetalum decline') in central and southern Florida landscapes. However, further investigation indicated that the decline was most likely the result of nutrient deficiency (Stewards and Shober, 2014)

Table 2.3 [1] Pest directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Loropetalum chinense*.

IRAC Code [2,3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Aphids	Black twig borer
1A	Acetylcholinesterase inhibitors	Carbamates	carbaryl[10]	Sevin SL	L, N, G	12	x	
			methiocarb	Mesurol 75W	N, G	24	x	
1B [2]	Acetylcholinesterase inhibitors	Organophosphates	acephate[10]	Orthene T&O	L, N, G	24	x	
				Lepitect	L, N, G	24	x	
				Precise GN	G, N	12	x	
			chlorpyrifos	Dursban 50W	N	24	x	x
				DuraGuard ME	N, G	24	x	
			dicrotophos	Inject-A-Cide B	L	N/A	x	
			dimethoate	Dimethoate 4E, 4EC	N	10-14 days	x	
			malathion[10]	Malathion 5EC	L	12	x	
			oxydemeton methyl	Harpoon [11]	L	0	x	
				MSR Spray Concentrate	N	240	x	
3A	Sodium channel modulators	Pyrethroids / Pyrethrins	bifenthrin[10]	Attain TR	G	12	x	
				Menace GC	L, N, G	12	x	x
				Onyx	L	N/A	x	x
				OnyxPro	L, N, I	12	x	x
				Talstar S Select	N, G	12	x	
			cyfluthrin	Decathlon	L, N, G, I	12	x	
			beta-cyfluthrin	Tempo Ultra WP	L, I	N/A	x	
				Tempo SC Ultra	L, I	N/A	x	
			lambda-cyhalothrin	Scimitar CS; Scimitar GC	L ; L, N, G	N/A; 24	x	
				Demand	L	N/A	x	
			cypermethrin	Demon WP	L, I	N/A	x	
			fenpropathrin	Tame 2.4 EC	N, G, I	24	x	
			tau-fluvalinate	Mavrik Aquaflow	L, N, G, I	12	x	
			permethrin	Astro	L, G, I	12	x	
				Permethrin Pro	L, I	N/A	x	
				Perm-Up 3.2 EC	L, N, G, I	12	x	

Table 2.3 (*continued*) [1] Pest directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Loropetalum chinense*.

IRAC Code [2,3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Aphids	Black twig borer
3A	Sodium channel modulators	Pyrethroids / Pyrethrins	pyrethrins	Tersus	N, G	12	x	x
				Pyganic	N, G	12	x	
			pyrethrum	Pyrethrum TR	N, G	12	x	
3A + 4A[2]	Sodium channel modulators	Pyrethroids + Neonico-tinoids	bifenthrin + clothianidin	Aloft LC G, LC SC	L	N/A	x	
			bifenthrin + imidacloprid	Allectus SC	L, I	N/A	x	
			cyfluthrin + imidacloprid	Discus N/G	N, G, I	12	x	
			lambda-cyhalothrin + thiamethoxam	Tandem	L	N/A	x	
			zeta-cypermethrin + bifenthrin + imidacloprid	Triple Crown T&O	L, I	N/A	x	x
3A + 27	Sodium channel modulators	Pyrethroids + Piperonyl-butoxide (PBO)	pyrethrins + piperonyl butoxide	Pyreth-It	N, G	12	x	
4A [2]	Nicotinic acetylcholine receptor agonists	Neonicotinoids	acetamiprid	TriStar 8.5 SL	L, N, G	12	x	
			clothianidin	Arena 0.25 G	L, I	12	x	
				Arena 50 WDG	L, I	12	x	
			dinotefuran	Safari 20 SG	L, N, G, I	12	x	
				Zylam Liquid	L	N/A	x	
				Transtect 70 WSP	L	N/A	x	
			imidacloprid[10]	Xytect 75WSP; 2F	L, N, G, I	12	x	
				Marathon II	N, G, I	12	x	
				Marathon 60WP	N, G, I	12	x	
			thiamethoxam	Merit	L, I	N/A	x	
				CoreTect	L, I	N/A	x	
				Discus Tablets	N, G, I	12	x	
				Flagship 25WG	N, G, I	12	x	
				Meridian 0.33G	L, I	N/A	x	
				Meridian 25WG	L, I	N/A	x	

Table 2.3 (*continued*) [1] Pest directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Loropetalum chinense*.

IRAC Code [2,3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Aphids	Black twig borer
4C + 5	Nicotinic acetylcholine receptor agonists	Sulfoxaflor + Spinosyns	sulfoxaflor + spinetoram	XXpire	L, N, G	12	x	
6	Chloride channel activators	Avermectins, Milbemycins	abamectin	Lucid, Avid				
				Aracinate TM[11]	L, N, G, I	N/A	x	
			emamectin benzoate	Arbormectin[11]	L	N/A	x	x
6 + 20D	Chloride channel activators	Avermectins, Milbemycins	abamectin + bifenazate	Sirocco	L, N, G, I	12	x	
7A	Juvenile hormone mimics	Juvenile hormone analogues	s-kinoprene	Enstar AQ	G, I	4	x	
7B	Juvenile hormone mimics	Fenoxycarb	fenoxycarb	Preclude TR	G	12	x	
7C	Juvenile hormone mimics	Pyriproxyfen	pyriproxifen	Distance IGR	L, N, G, I	12	x	
				Fulcrum	L, N, G, I	12	x	
8D	Misc. non-specific (multi-site) inhibitors	Borates	sodium tetraborohydrate decahydrate	Prev-AM Ultra	N, G	12	x	
9B [2]	Selective homopteran feeding blockers	Pymetrozine	pymetrozine	Endeavor	L, N, G, I	12	x	
		Pyrifluquinazon	pyrifluquinazon	Rycar	G	12	x	
21A	Mitochondrial complex I electron transport inhibitors	METI acaricides and insecticides	tolfenpyrad	Hachi-Hachi SC	G	12	x	
23 [2]	Inhibitors of acetyl CoA carboxylase	Tetronic and Tetramic acid derivatives	spiromesifen	Kontos	N, G, I	24	x	
28	Ryanodine receptor modulators	Diamides	chlorantraniliprole	Acelepryn	L, I	N/A	x	
				Acelepryn G	L, I	N/A	x	
			cyantraniliprole	Mainspring	L, G, I	4	x	
29	Chordotonal organ modulators – undefined target site	Flonicamid	flonicamid	Aria	L, N, G	12	x	
Unknown	Unknown	Azadirachtin	azadirachtin[10]	Azatin O	L, N, G, I	4	x	
				Azatin XL	N, G, I	4	x	
				Azatrol EC	L, N, G, I	4	x	
				Ornazin EC	L, N, G, I	12	x	

Table 2.3 (*continued*) [1] Pest-directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Loropetalum chinense*.

IRAC Code [3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Aphids	Black twig borer
Not classified	Various		*Beauveria bassiana*	BotaniGard ES; Mycotrol ESO; Mycotrol WPO	L, N, G, I	4	x	
				BotaniGard 22 WP	L, N, G, I	4	x	
				Naturalis-L	L, N, G	4	x	
			Chromobacterium subtsugae	Grandevo PTO	L, N, G	4	x	
			Isaria formosorosea	NoFly	G	12	x	
				Preferal	L, N, G	4	x	
			horticultural oil[10]	Ultra-Pure Oil, TriTek	L, N, G, I	4	x	
			insecticidal soap[10]	M-Pede	L, N, G, I	12	x	
			neem oil[10]	Trilogy	L, N	4	x	
				Triact 70	L, N, G, I	4	x	

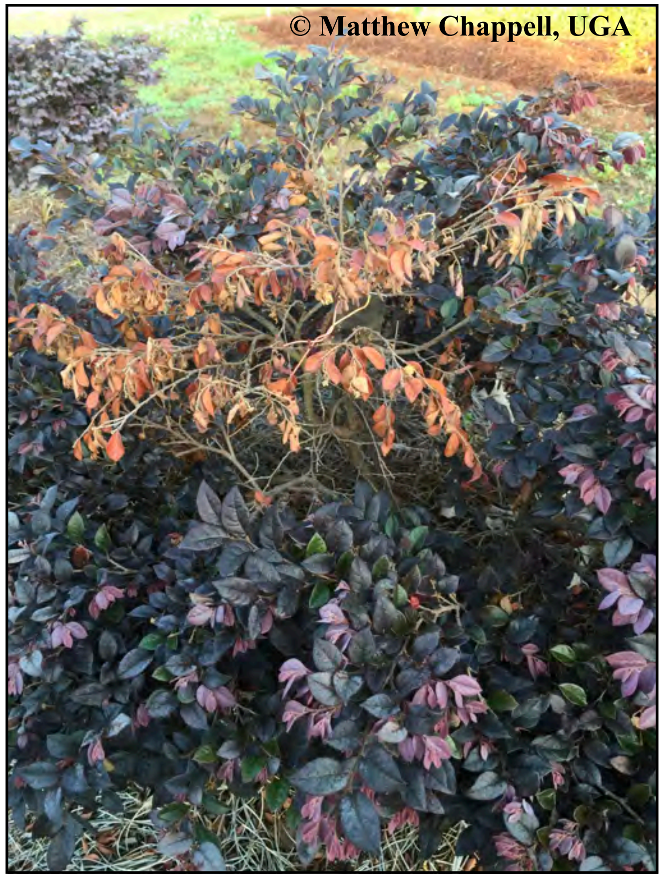
Table 2.3 (*continued*) [1] Pest-directed insecticidal activity and Insecticide Resistance Action Committee (IRAC) Codes for use in developing a pesticide rotation plan to manage key pests of *Loropetalum chinense*.

Footnotes and Supplemental Information:

1. Pests listed on pesticide labels are subject to change, as are re-entry intervals, trade names, and formulations. Within states and counties, products may have additional permitted uses by 2(ee) and Special Local Need allowances. Consult your County Extension Agent or State Agency to determine if other uses are allowable. The label should always be consulted to confirm that chart-listed pests still appear on the label. Check the product labels for specific site restriction information, notes on application, sensitive plant species and specific target pest species.
2. Applications of products from these footnoted IRAC groups are optimized when not preceded by/or followed by products in subsequently noted IRAC Group(s), particularly when managing the indicated pest. Suggested rotation combinations to avoid include serial treatments with a.i. products from: IRAC 1B, 4A, 9B, 23 (aphids).
3. IRAC Code designations and Related Modes of Action are explained at the Insecticide Resistance Action Committee Database 2017 (IRAC 2017; http://www.irac-online.org/modes-of-action/).
4. See product label for information about potential crop phytotoxicity, known plant sensitivity, and how to test for phytotoxicity.
5. Check label for additional restrictions (e.g., on the number of times the product can be applied in a growing season or year, or additional application restrictions within permitted use sites; and crop types or flowering status that may not be legally treated.)
6. Trade names of products are provided as examples only. No endorsement of mentioned product, nor criticism of unmentioned products, is intended.
7. Products may not be registered or renewed for use in all southeastern U.S. states. Consult your state's Department of Agriculture to confirm legal use of products in your state.
8. Use site information is provided for reference only: L = landscape; N = nursery; G = greenhouse; I = interiorscape. t/s*Products listed for use in L, N, G or I sites may include active ingredients in differently labeled products that are designated for use in turfgrass or sod production use sites. Mention of those products is beyond the scope of this resource guide, and products listed herein may not be legal or appropriate for site uses in turfgrass or sod production
9. Re-Entry Interval (REI) designations apply to agricultural (nursery) uses. Within REI column, 'N/A' is used to indicate products with Landscape and/or Interiorscape site uses and that present Non-Agricultural Use Requirements. Consult product labels for specific details required for compliance within those application conditions and use sites, including those for which REI listings do not apply. Additionally, some product labels may list site uses that are beyond the scope of this publication (e.g., sod farms, silvicultural & Christmas tree nurseries, pastures, rights-of-way, etc.). These site uses may involve Agricultural Use Requirements that list an REI not presented here.
10. Multiple formulations and trade names of the same active ingredients are available. Labels among these products often differ in legally allowable uses (regarding sites, pest, REI, etc.); representative example(s) presented.
11. Product formulated for tree or shrub injection; specialized equipment may be required.

SECTION 3

Disease Management



COMMON DISEASE PESTS

1. Phytophthora Root Rot
2. Bacterial Gall
3. Leaf spot
4. Ramorum blight

Few plant diseases affect *Loropetalum chinense*. Root rot, particularly Phytophthora root rot, is the most common root disease. Relatively new diseases on loropetalum include a fungal leaf spot and stem gall/canker disease.

Phytophthora Root Rot

The most common root disease affecting liners and containerized loropetalum is Phytophthora root rot caused by several *Phytophthora* species including *P. cinnamomi* Rands and *P. nicotianae* Breda de Haanof. Infected roots are brown, softened and necrotic. The lower stem at the soil line can develop a reddish-brown discoloration as infection progresses into the crown (Figure 2.19). Infected plants may wilt; however, scorching along the leaf margins, foliage browning (Figure 2.20) and leaf dropping are the most common foliar symptoms of root disease.

Figure 2.19 In severe Phytophthora infections, lower stems at the soil line can develop a reddish-brown discoloration as infection progresses into the crown.

In production, plants can be affected by root rot if they are not planted in a well-drained soil (Williamson and Scott, 2015). Phytophthora is a "water mold" pathogen that requires a wet environment to infect and spread. *Phytophthora* spp. produce motile spores (zoospores) that move in water from irrigation water splash, within puddles under containers and low areas, or wherever there is standing water. *Phytophthora* spp. also produce chlamydospores and oospores that provide long-term survival within infected roots, plant residue and in soil. Roots growing through infested rooting substrate or coming in contact with infested soil can become infected (Hagan, 2011; Hagan and Mullen, 2000).

The most critical factor in preventing Phytophthora diseases is to avoid over-watering. Substrates and containers that allow sufficient drainage should be used, and irrigation should be scheduled such that over-watering is avoided by taking into account rainfall and weather conditions before irrigating. For field production, loropetalum should be planted in well-drained soil, and areas prone to flooding or with a fragipan or hardpan should be avoided.

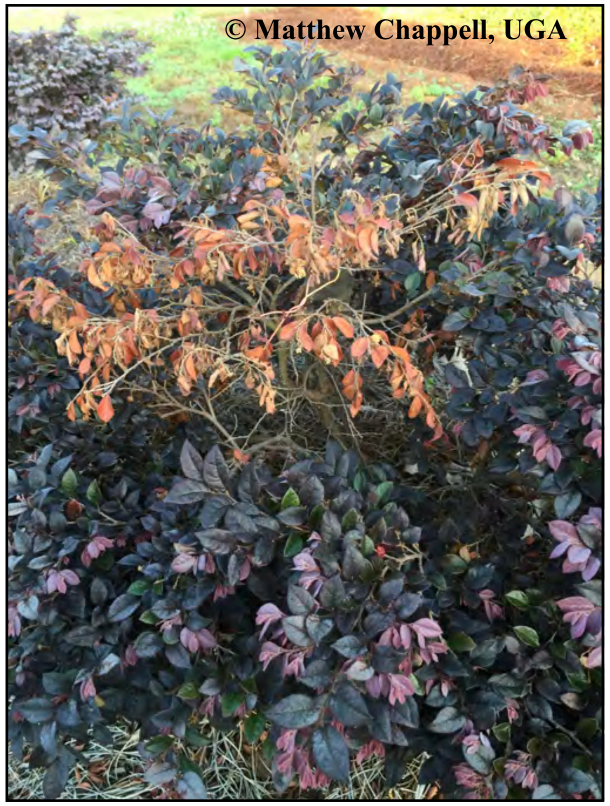


Figure 2.20 Typically, the initial symptom of Phytophthora infection is branch dieback.

Root infection can occur year-round in southeastern plant nurseries; however, plant decline symptoms are most commonly seen during the summer months (June-August) when high temperatures and heat injury to container-grown roots further stress the diseased root system. *Phytophthora cinnamomi* has optimal growth at moderate temperatures of 20-28°C (68-82°F); whereas, temperatures higher than 32°C (90°F) tend to limit or suppress growth (Eggers et al., 2012). Optimal temperatures for growth of *P. nicotianae* are similar to *P. cinnamomi*, except that isolates of *P. nicotianae* can continue to grow at warmer temperatures (Erwin and Ribeiro, 1996). What this means is that targeting chemical control when foliage symptoms are observed during the summer months is too late and most likely misses the infection period of the pathogen.

Management

Management of Phytophthora root rot requires prevention, cultural and chemical approaches. Since *Phytophthora* spp. can survive in all infected plant parts and in substrate, diseased plants must be removed and destroyed to prevent future infections. Containers and substrate from infected plants should also be disinfected (containers) and/or discarded (substrate). All debris should be removed, and work surfaces and tools must be sanitized between crops. Container-grown plants should be kept on crowned gravel pads to ensure proper drainage and avoid transmission of propagules between containers. Store substrates and containers on concrete pads or asphalt, and avoid re-using containers or sanitize them between uses. Used substrate should be discarded, but can be sanitized by steaming or composting if necessary (Schweigkofler et al., 2014).

Fungicides are an important component in reducing Phytophthora infection; however, fungicides are not labeled for use on loropetalum. Test all products for possible phytotoxicity before using on the entire crop. Fungicides cannot cure Phytophthora root rot but can be used as a preventative measure against infections (Table 2.4). They are most effectively used as drenches or when incorporated into the potting substrate during propagation (Bailey et al., 2012).

The most commonly used fungicides (Table 2.4) contain mefenoxam, fosetyl-Al, and formulations of phosphorus acid derivatives (e.g. phosphonate and phosphite fungicides). Other effective fungicides include cyazofamid, dimethomorph, fenamindone, fluopicolide and mandipropamid. Fungicides drenches should be applied beginning in the spring when container substrate temperatures reach 15-20°C (59-68°F) and *Phytophthora* infection occurs. Waiting until symptoms develop in the warmer, summer months is too late to effectively manage this root disease.

Bacterial Gall

Loropetalums are also at risk of infection by the bacteria *Pseudomonas savastanoi* (syn. *Pseudomonas syringae* subsp. *savastanoi)* (Sinclair and Lyon, 2005). This is the most damaging plant disease on loropetalum, commonly referred to as bacterial stem gall and canker. It manifests as knotted, swollen sections on stems (Figure 2.21) and can eventually lead to shoot or plant death. It was first discovered in May 2012 in Aiken, SC and has since been seen from NY to FL and west to AL (Hagan and Conner, 2013). This bacterium was also thought to cause olive gall on *Olea europaea* and oleander knot in *Nerium oleander* (Sinclair and Lyon, 2005). Ornamental species in the Oleaceae attacked by *P. savastanoi* include ash, privet, and forsythia. It is not recognized as attacking any other member of the Hamamelidaceae family. This is interesting as recent additional pathogen identification and cross-inoculation testing indicates that the causal bacterium only infects loropetalum and does not infect oleander nor olive, and may be a yet unnamed bacterium closely related to *P. savastanoi* (Sun et al., 2013).

Figure 2.21 Bacterial stem gall and canker is a relatively new, yet devastating bacterium. It manifests as knotted, swollen sections on stems.

Management

In nurseries, infected plants should be disposed of and cuttings should be taken from disease-free plants (Figure 2.22). Tools must be sanitized between uses using a (10% v/v) bleach or isopropyl alcohol solution between cuts or at a minimum between small blocks of plants. If plants become infected, at a minimum remove the infected branch below the gall (Hagan and Conner, 2013; Williamson and Scott, 2015); a better action would be to discard the plant. There is no treatment once plants have been infected. Copper-containing compounds can be applied during periods when disease transmission is maximized (Table 2.4). Applications should be applied weekly. This generally coincides with wet periods. Bacteria ooze from the galls during extended periods of wet, cloudy weather (Fichtner, 2011). The bacterium is then dispersed to adjoining healthy shoots via water splash from rain showers or overhead irrigation.

Figure 2.22 Cuttings should never be taken from infected plants. Pictured here are loropetalum liners infected with bacterial stem gall and canker.

Fungal Leaf Spot

A leaf spot disease (Figure 2.23) caused by the fungus, *Pseudocercospora liquidambaricola* (J.M. Yen) U. Braun, can affect loropetalum. Not much is known about this disease. The leaf spot fungus is described to also infect sweetgum (*Liquidambar* spp.); however, cross inoculation studies have not been conducted. Infection causes purplish, angular to irregularly-shaped leaf spots that are mostly seen within the lower plant canopy. The entire plant can become infected under wet, humid conditions. Generally, the leaf spot is more commonly seen during the late summer months in the southeastern U.S. Infection is usually of minor importance; however, it can cause significant leaf drop if infection is severe.

Management

Prolonged periods of leaf wetness favors fungal leaf spot diseases. Keeping the foliage as dry as possible can help reduce disease development. Irrigate only when plants will dry quickly. Avoid irrigating plants late in the day so the foliage has time to dry before nightfall. Discarding severely infected plants can help reduce disease spread. Propagate only from healthy stock plants. Fungicides labeled to manage other leaf spot diseases caused by similar *Cercospora*, *Passalora*, or *Pseudocercospora* spp. (Table 2.4) may help reduce leaf spot development such as azoxystrobin, pyraclostrobin, myclobutanil, mancozeb, thiophanate methyl, and copper hydroxide. Applications should begin at bud break to protect new growth in the spring and late summer. No fungicide is labeled for use on loropetalum; therefore, test fungicides for possible phytotoxicity prior to use on the entire crop.

Figure 2.23 Another new pathogen of loropetalum is a leaf spot disease caused by the fungus, *Pseudocercospora liquidambaricola.*

Ramorum Blight

Loropetalum is a known host to *Phytophthora ramorum* Werres, De Cock & Manin 't Veld, cause of Sudden Oak Death and Ramorum blight (Blomquist et al., 2012). Infection causes very small, dark-colored leaf spots and defoliation. Leaf spotting is inconspicuous and may be overlooked during routine scouting. Of greatest concern is that *P. ramorum* infection may be missed on loropetalum and thereby allow the pathogen to spread to other nearby susceptible host plants. Loropetalum plants should be inspected routinely for leaf spotting symptoms due to the ramifications of *P. ramorum* infection, which is a federally-regulated pathogen.

Management

Management depends upon exclusion. Vigilant inspection of incoming plants from areas where *P. ramorum* is known to occur is a must. Leaf spotting could be confused with Pseudocercospora leaf spot. If suspect symptoms are seen, a sample should be submitted to a state or University plant disease clinic for identification. Keep plant foliage as dry as possible to reduce disease development and spread for all foliar diseases affecting loropetalum. Infected plants are required to be destroyed.

Table 2.4. Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Loropetalum chinense*.

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)[Y]	Fungal leaf spot	Bacterial gall	Phytophthora root rot
1: MBC Benzimidazoles: Upwardly systemic. Broad spectrum fungicide for various fungi. Fungicide resistance risk high. Tank mix with fungicides from a different fungicide group (FRAC) to prevent or delay resistance development. Do not mix with copper or with highly alkaline pesticides such as lime or sulfur.			
thiophanate methyl: Cleary's 3336 F, EG; Allban Flo, 50 WSB; OHP 6672 50 WP, 4.5F; Transom; T-bird; T-methyl; Fungo Flo	x		
1 + 2: MBC Benzimidazoles + Dicarboximides: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.			
thiophanate methyl + iprodione: Cleary 26/36 [W]	x		
1 + M5: MBC Benzimidazoles + Multi-site inhibitor: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.			
thiophanate methyl + chlorothalonil: Spectro 90 WDG [W]	x		
1 + 14: MBC Benzimidazoles + Aromatic Hydrocarbons: Systemic. Effective against water molds, or oomycetes. Long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.			
thiophanate methyl + etridiazole: Banrot 40WP[W]			x
2: Dicarboximides: Locally systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.			
iprodione: Chipco 26019 FLO, N/G; 26GT; Iprodione [V]	x		
3: DMI or SI Triazoles: This group was formerly known as De-Methylation Inhibitors (DMI) and are now classified as Sterol Biosynthesis Inhibitors (SBI or SI). Upwardly systemic. Rain-fast in 2 hours. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.			
myclobutanil: Eagle 20W; Siskin	x		
metconazole: Tourney[W]	x		
propiconazole: Banner Maxx; Banner Maxx II; Strider; Fathom	x		
tebuconazole: Torque	x		
triticonazole: Trinity; Trinity TR	x		
3 + 11: DMI or SI Triazoles + Quinone outside Inhibitors (QoI): Upwardly and locally downward systemic. Rain-fast in 2 hours. Some curative activity. Medium risk for resistance.			
triadimefon + trifloxystrobin: Strike Plus 50 WDG; Trigo	x		
4: Phenylamides: Systemic. Effective against diseases caused by oomycetes, or water molds, including damping-off, root and stem rots, and foliar diseases. Use as soil drench or foliar application.			
mefenoxam: Ridomil Gold; Subdue GR; Subdue Maxx[W]			x
4 + 12: Phenylamides + Phenylpyrroles/Osmotic signal transducers: Systemic. Broad spectrum folair-applied fungicide.			
mefenoxam + fludioxonil: Hurricane; Hurricane WDG [U,V,W]			x

Table 2.4 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Loropetalum chinense*.

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s) [Y]	Fungal leaf spot	Bacterial gall	Phytophthora root rot
7 + 11: Carboxamides/Succinate Dehydrogenase Inhibitors (SDHI) + QoI Q: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.			
benzovindiflupyr + azoxystrobin: Mural [W]	x		
fluxapyroxad + pyraclostrobin: Orkestra Intrinsic [W]	x		
pyraclostrobin + boscalid: Pageant; Pageant Intrinsic [W]	x		x
9 + 12: Anilino-Pyrimidines/Methionine biosynthesis inhibitors + Phenylpyrroles: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.			
cyprodinil + fludioxonil: Palladium [W]	x		
11: Quinone outside Inhibitors (QoI): These fungicides are also known as strobilurins. Locally systemic. Effective on mildews, foliar pathogens, and most fungi. Some control of oomycetes, or water molds. Fungicide resistance risk high.			
azoxystrobin: Heritage; Strobe 50WG	x		x
fenamidone: FenStop	x		x
fluoxastrobin: Disarm O	x		x
kresoxium methyl: Cygnus	x		
pyraclostrobin: Empress Intrinsic; Insignia SC			x
trifloxystrobin: Compass; Compass O 50 EDG [V]	x		x
12: Phenylpyrroles/Osmotic signal transducers: Non-systemic but good residual protection. Broad spectrum fungicide, not effective against oomycetes, or water molds.			
fludioxonil: Medallion; Medallion WDG; Mozart TR	x		
14: Aromatic Hydrocarbons: Locally systemic. Effective against water molds, or oomycetes.			
etridiazole: Terrazole 35% WP, CA, L; Truban 25EC, 30WP [V]			x
19: Polyoxins: Locally systemic chitin synthase inhibitor. For use in outdoor and greenhouse nursery crops. Resistance risk high.			
polyoxin D zinc salt: Affirm; Veranda O; Endorse WP [W]	x		
21: Quinone inside Inhibitors: Locally systemic. Effective against water molds, or oomycetes. Resistance risk unknown but presumed to be medium to high.			
cyazofamid: Segway, Segway O, Segway SC			x
25: Glucopyranosyl Antibiotics: Bacterial protectant. Broad spectrum bacterial control.			
streptomycin sulfate: Agri-Mycin 17		x	

Table 2.4 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Loropetalum chinense*.

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s) [Y]	Fungal leaf spot	Bacterial gall	Phytophthora root rot
28: Carbamates: Cell membrane permeability, fatty acid interruption (proposed). Low to medium resistance risk.			
propamocarb hydrochloride: Banol			x
33: Phosphonates: Fully systemic; when applied to leaves, product can translocate to lower parts. Effective against water molds, or oomycetes, such as Phytophthora, Pythium, and downy mildew pathogens. Low risk for fungicide resistance development.			
phosphorous acid: Alude; Fosphite; K-Phite T/O, 7LP; Magellan		x	x
potassium phosphite: Vital			x
fosetyl-Al: Aliette WDG		x	x
40: Carboxylic Acid Amides: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes.			
mandipropamid: Micora			x
dimethomorph: Stature DM, SC			x
45 + 40: Quinone outside Stigmatellin-binders (QoS) + Carboxylic Acid Amides: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes.			
ametoctradin + dimethomorph: Orvego [W]			x
43: Pyridinemethyl-benamides Delocalisation of spectrin-like proteins: Locally systemic, translaminar. Control of oomycetes, or water molds. Medium to high resistance risk.			
fluppicolide: Adorn			x
44: Bacillus spp.: Microbial disruptor of cellular membranes.			
Bacillus subtilis: Cease, Triathlon		x	x
49: Piperidinyl-thiazole- isoxazolines: Locally systemic. Control of oomycetes, or water molds. Rain fast. Medium to high resistance risk.			
oxathiapiprolin: Segovis			x
M: Multi-site inhibitors: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.			
(M1) copper hydroxide: Champ WG, DP, Formula 2; CuPro 2005 T/N/O, 5000; Nu-Cop 50DF, HB, 3L	x	x	
(M1) copper salts of fatty and rosin acids: Camelot O	x	x	
(M1) copper sulphate pentahydrate: Phyton 27; Phyton 35	x	x	
(M1) tribasic copper sulfate: Basicop; Cuproxat	x	x	
(M1 + M3) copper hydroxide + mancozeb: Junction [X]	x	x	
(M3) mancozeb: Protect T/O, DF; Fore 80WP; Dithane 75DF; Pentathlon DF, LF; Mancozeb 4F, Flowable with Zinc	x	x	

Table 2.4 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Loropetalum chinense*.

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s) [Y]	Fungal leaf spot	Bacterial gall	Phytophthora root rot
M: Multi-site inhibitors: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.			
(M5) chlorothalonil: Daconil Ultrex, Zn Flowable, Weather Stik [U]; Manicure 6FL; Manicure Ultra; Thalonil 90 DF; Thalonil 6L; Mainsail	x		
M3 + 1: Multi site inhibitor + MBC Benzimidazoles: Upwardly systemic. Broad spectrum fungicide for various fungi. Fungicide resistance risk medium. Do not mix with copper or with highly alkaline pesticides such as lime or sulfur.			
thiophanate methyl + mancozeb: Zyban [X]	x		
M3 + 3: Multi site inhibitor + Sterol Biosynthesis Inhibitor: Effective as protectants (and upwardly systemic) on broad spectrum including most fungi and mildews. Fungicide resistance risk low.			
mancozeb: + myclobutanil: Clevis[W]	x		
M5 + 3: Multi site inhibitor + SI Triazoles: Upwardly systemic. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.			
propiconazole + chlorothalonil: Concert II [X]	x		
NC: Not a Classified substance: Contact fungicide for greenhouse and nursery use. Low risk for resistance.			
potassium bicarbonate: MilStop	x		

[Z] (FRAC 2017).
[Y] Check current products for labeled pesticides, sites for control and plant safety and efficacy on fungal species. This table reports information on fungicide labels and does not necessarily reflect product efficacy. Refer to fungicide labels for rates and usage, specific host information, possible phytotoxicity, re-entry intervals and resistance management. Within columns, products indicated by "x" are labeled for use against the listed pathogen type. Always test product on a sample (small number) of plants before treating an entire crop, as some fungicides listed are for control of the disorder but not specifically labeled for use in *Hudrangea* spp.
[X] Including the causal agent of sudden oak death, *Phytophthora ramorum*.
[W] Chemical contains more than one active ingredient, thus product may be listed within more than one FRAC code designation (FRAC 2017).
[V] Not for use in residential landscapes; commercial use only.
[U] Do not apply with mist blowers or high pressure spray equipment in greenhouses.

References

Bailey, K.L., J. Derby, S.M. Boyetchko, K. Sawchyn, E. Becker, G. Sumampong, S. Shamoun, D. James, S. Masri, and A. Varga. 2012. In vivo studies evaluating commercial biofungicide suppression of blight caused by *Phytophthora ramorum* in selected ornamentals. Biocontrol Sci. Tech. 22:1268-1283.

Blazich, F. and A. LeBude. 2015. The North Carolina Extension Gardener Handbook: Chapter 13, Propagation. Accessed August 29, 2016 <http://content.ces.ncsu.edu/extension-gardener-handbook/13-propagation>.

Blomquist, C.L., S. Rooney-Latham, M.C. Soriano, and J.C. McCarty. 2012. First report of *Phytophthora ramorum* causing a leafspot on *Loropetalum chinense*, chinese fringe flower in California. Plant Disease 96(12):1829-1829. Accessed May 13, 2016. <http://apsjournals.apsnet.org.proxy-remote.galib.uga.edu/doi/abs/10.1094/PDIS-01-12-0062-PDN>.

Chappell, M. 2016. Seed propagation of *Loropetalum chinense*; Personal communication.

Chen, Y., R.P. Bracy and A.D. Owings. 2012. Using plant growth regulators in the landscape. LA Ag. Mag. Winter 2012 Ed.

Cochran, D.R., C.H. Gilliam, D.J. Eakes, G.R. Wehtje and P.R. Knight. 2008. Herbicide use in propagation of *Loropetalum chinense* 'Ruby'. J. Environ. Hort. 26:139-143.

Dirr, M.A. and C.W. Heuser Jr. 2006. The Reference Manual of Woody Plant Propagation: From Seed to Tissue Culture (p. 232). Varsity Press, Inc. Cary, N.C.

Eggers, J. E., Balci, Y., and MacDonald, W. L. 2012. Variation among *Phytophthora cinnamomi* isolates from oak forest soils in the eastern United States. Plant Dis. 96:1608-1614.

Erwin, D.C., and Ribeiro, O.K. 1996. *Phytophthora* Diseases Worldwide. St. Paul, MN: The American Phytopathological Society.

Fichtner, E. J. 2011. Olive knot. UC ANR Publication 74156. UC Statewide IPM Program, University of California, Davis, CA. Accessed August 20, 2016 <http://www.ipm.ucdavis.edu/PMG/PESTNOTES/pn74156.html>.

Gapinski, A. 2015. Hamamelidaceae, Part 2: Exploring the witch-hazel relatives of the Arnold Arboretum. Arnoldia 72:20-35.

Gawel, N.J., G.R. Johnson and R. Sauve. 1996. Identification of genetic diversity among *Loropetalum chinense* var. *ruburm* introductions. J.Environ. Hort. 14:38-41.

Gong, W., W. Liu, L. Gu, S. Kaneko, M. Koch and D. Zhang. 2016. From glacial refugia to wide distribution range: demographic expansion of *Loropetalum chinense* (Hamamelidaceae) in Chinese subtropical evergreen broadleaved forest. Organisma Div. Evol. 16:23-28.

Hagan, A.K. 2011. Control of diseases on trees and shrubs. Alabama Cooperative Extension. Accessed May 13, 2016 <http://www.aces.edu/timelyinfo/PlantPathology/2011/January/pp359.pdf>.

Hagan, A.K. and K. Conner. 2013. Bacterial gall on loropetalum. Alabama Cooperative Extension System PP-726.

Hagan, A.K. and J.M. Mullen. 2000. Phytophthora rot on woody ornamentals. Alabama Cooperative Extension System ANR-571. Accessed May 13, 2016 <http://www.aces.edu/pubs/docs/A/ANR-0571/ANR-0571.pdf>.

Mizell, R. F., and T. C. Riddle. 2004. Evaluation of insecticides to control the Asian ambrosia beetle, *Xylosandrus crassiusculus*. Proc. Southern Nurs. Assoc. Res. Conf. 49:152-155.

Oliver, J. B., and C. M. Mannion. 2001. Ambrosia beetle (Coleoptera: Scolytidae) species attacking chestnut and captured in ethanol-baited traps in middle Tennessee. Env. Entomol. 30:909-918.

Oliver, J. B., N. N. Youssef, and M. A. Halcomb. 2004. Comparison of different trap types for collection of Asian ambrosia beetle, pp. 158-163. *In* J.B. Oliver [ed.], Proc. Southern Nurs. Assoc. Res. Conf.

Pecot, H.C. 2004. Influence of planting depth and mulch on the growth of nine species of ornamental plants in landscape and container settings: A thesis. LSU Electronic Thesis & Dissertation Database. Accessed July 14, 2016 <http://sites01.lsu.edu/wp/graduateschool/thesis-and-dissertation-library/>.

Plant Locator. 2016. Accessed July 13, 2016 <http://www.plantlocator.net>.

Ranger, C.M., M.E. Reding, P.B. Schultz, J.B. Oliver, S.D. Frank, K.M. Adesso, J.-H. Chong, B. Simpson, C. Werle, S. Gill, and C. Krause. 2016. Biology, ecology, and management of nonnative ambrosia beetles (Coleoptera: Curculionidae: Scolytinae) in ornamental plant nurseries. J. Integrated Pest Mgmt. 7:1-23.

Reding, M., J. Oliver, P. Schultz, and C. Ranger. 2010. Monitoring flight activity of ambrosia beetles in ornamental nurseries with ethanol-baited traps: Influence of trap height on captures. J. Environ. Hort. 28:85-90.

Ruter, J. 2006. Controlling little-leaf disorder on *Loropetalum chinense* var. *rubrum* with foliar applications of copper. HortScience 40:501.

Schweigkofler, W., K. Kosta, V. Huffman, S. Sharma, K. Suslow and S. Ghosh. 2014. Steaming inactivates *Phytophthora ramorum*, causal agent of sudden oak death and ramorum blight, from infested nursery soils in California. Plant Health Res. 15:43-47.

Shober, A.L., G. Leibee, and M.L. Ko-Yokomi. 2008. Response of *Loropetalum chinensis* var. *rubrum* 'Ruby' to foliar applications of micronutrient fertilizers and miticide. J. Environ. Hort. 26:235-238.

Simonne, E.H., C.E. Harris, J.O. Sichivitsa, J.E. Altland, C.H. Gilliam and D.J. Eakes. 2008. A simple format for reporting methods used in nutritional studies: Application with *Loropetalum* response to fertilizer rates. J. Plant Nutrition 22:1797-1806.

Sinclair, W.A. and H.H. Lyon. 2005. Diseases of Trees and Shrubs: Second Edition. Cornell University Press, Ithica N.Y.

Smith, H.C., J.A. Ferrell and T.J. Koschnick. 2014. Flurprimidol performance on ornamental species in relation to trimming time and method of application. HortScience 49:1305-1308.

Southern Nursery Association. 2014. Irrigation Management Practices. IN: SNA Best Management Practices Guide: Guide for Producing Nursery Crops. Accessed July 14, 2014 <http://contents.sna.org/bmpv30.html>.

Stewards, J., and A.L. Shober. 2014. Improving the health of declining *Loropetalum* in the home landscape. University of Florida, IFAS Extension, Publication #SL354. Accessed July 14, 2016 <http://edis.ifas.ufl.edu/ss556>.

Sun, X., A. Jeyaprakash, D. Davison, D. Jones, T. Schubert and B. Sutton. 2013. *Pseudomonas* sp. Found on Loropetalum stem canker in Florida. Phytopathology 103:S2.141

U.S. National Arboretum. 1999. *Loropetalum chinense* var. *rubrum* 'Blush' and 'Burgundy'. Accessed July 14, 2016 <http://www.usna.usda.gov/Newintro/loro.pdf>.

Weaver Jr., R.E. 1976. The witch hazel family (Hamamelidacese). Arnoldia 36:69-109.

Williamson, J. and J.M. Scott. 2015. Loropetalum. Clemson Cooperative Extension HGIC 1085. Accessed May 13, 2016 <http://www.clemson.edu/extension/hgic/plants/landscape/shrubs/hgic1085.html>.

Yeager, T., C. Larsen, J. von Merveldt and T. Irani. 2009. Use of reclaimed water for irrigation in container nurseries. University of Florida – IFAS Extension ENH-1119.

CHAPTER 3

Holly - *Ilex* spp.

Francesca Peduto Hand, The Ohio State University

Winston Dunwell, University of Kentucky

Frank A. Hale, University of Tennessee

Juang-Horng (JC) Chong, Clemson University

Sarah A. White, Clemson University

Gary Knox, University of Florida

Section 1

History, Culture and Management

Ilex verticillata 'Winter Red'

Introduction

History and Landscape Value

Ilex is a large group of evergreen and deciduous plants that belong to the *Aquifoliaceae* family. Linnaeus named the genus after the holm oak, *Quercus ilex*, a spiny leaved oak native to the Mediterranean region. Holm is *Ilex* in Latin (Galle, 1997; Beales, 2006). Commonly referred to as holly, they are found native and planted throughout the southern and eastern United States (Table 3.1). Considered versatile and of significant ornamental value, they are also prevalent throughout other temperate regions of world. Long a garden favorite, there are a staggering number of species and numerous cultivars registered with the Holly Society of America (Galle, 1997; Holly Society of America, 2015). Many outstanding native hollies are found in southern states such as *Ilex vomitoria* (yaupon) in the lower south to *I. verticillata* (winterberry) of the upper south with species such as *I. opaca* (American holly) occurring in all southern states. They rank in size from ground covers to large trees and are used as specimen plants, foundation plants (Figure 3.1) and hedges (Figure 3.2). Many of the cultivars currently in catalogs were available as early as 1994 (Meyer et. al., 1994). Broadleaf evergreen forms (Figure 3.3) were grown by 1,654 operations across the U.S. in 2014, with a wholesale value of $10.3 million (USDA-NASS, 2014).

Figure 3.1 In this photo, three distinct sized hollies are visible. In the foreground, *Ilex cornuta* 'Rotunda' is visible (although typically larger, this plant is regularly pruned to maintain lower growth habit). The larger rounded shrub behind *I. cornuta* 'Rotunda' and against the building is *I. cornuta* 'Burfordii'. Immediately to the right is a large specimen of *I. ×attenuata* 'Forsterii'.

Figure 3.2 *Ilex ×attenuata* 'Savannah' is routinely used as a hedge plant, as seen here.

Figure 3.3 *Ilex* 'Robin™ Red' is a newer broadleaf evergreen holly.

Table 3.1 Number of production nurseries growing various *Ilex* species reported [plant & supply locator] for southern states.

Species/Taxon	Number of nurseries	States where produced
Ilex 'Nellie R. Stevens'	154	AL, AR, CA, FL, GA, LA, MD, MS, NC, SC, TN, TX, VA
Ilex vomitoria 'Stokes Dwarf'	73	FL, SC, GA, NC, TN
Ilex cornuta 'Dwarf Burford'	69	AL, FL, GA, LA, MD, MS, NC, SC, TN, VA
Ilex cassine - Dahoon Holly	66	AL, FL
Ilex cornuta 'Needlepoint' ('Anicet Delcambre', 'Delcambre', 'Willowleaf')	56	AL, FL, GA, LA, MS, NC, SC, TN
Ilex crenata 'Sky Pencil'	49	AL, FL, GA, LA, MS, NC, SC, TN
Ilex vomitoria - Yaupon Holly	47	FL, GA, LA, NC, SC
Ilex ×attenuata 'Eagleston'	46	FL, LA, SC
Ilex 'Emily Bruner'	44	AL, FL, GA, LA, NC, SC, VA
Ilex cornuta 'Burfordii'	42	AL, FL, GA, LA, MS, NC, SC
Ilex ×attenuata 'Savannah'	38	AL, FL, GA, LA, NC, SC, TX
Ilex cornuta 'Carissa'	36	AL, FL, GA, LA, MS, NC, SC, TN
Ilex 'Conaf' pp9487 Oak Leaf™	32	AL, GA, FL, LA, MD, NC, SC, VA
Ilex 'Mary Nell'	31	AL, FL, GA, LA, NC, SC, VA
Ilex vomitoria 'Nana' - Dwarf Yaupon	28	AL FL, GA LA, MS, NC, SC, TN
Ilex crenata 'Soft Touch'	26	AL, FL, GA, LA, MD, MS, NC, SC, TN
Ilex crenata 'Compacta'	25	AL, FL, GA, MD, NC, SC,TN
Ilex opaca 'Greenleaf'	25	AL, GA, NC, SC, TN, VA
Ilex vomitoria var. *pendula* - Weeping Yaupon	25	AL, FL, GA, NC, SC
Ilex 'Conin' pp9486 Robin™ (Ilex ×Robin™ pp9486 - Red Robin Holly)	22	AL, FL, GA, MD, NC, SC, VA
Ilex crenata 'Steeds'	16	AL, GA, FL, MD, MS, NC, SC, TN
Ilex verticillata 'Winter Red'	16	AL, GA, NC, MD, NC, TN, VA
Ilex glabra - Gallberry, Inkberry	15	FL, MD, NC, SC
Ilex glabra 'Shamrock'	15	AL, FL, MD, NC, TN,VA
Ilex ×meserveae 'China Girl'	12	AL, FL, GA, MD, MS, NC, SC, TN, VA

Table 3.1 (***continued***) Number of production nurseries growing various *Ilex* species reported [plant & supply locator] for southern states.

Species/Taxon	Number of nurseries	States where produced
Ilex cornuta - Chinese/Needlepoint Holly	12	FL, LA, SC
Ilex crenata 'Helleri'	12	AL, GA, FL, LA, MD, MS, NC, SC, TN
Ilex crenata 'Hoogendoorn'	12	FL, GA, MD, MS, NC, SC, TN
Ilex opaca 'Oakleaf'	12	FL, GA, MS, SC, TN
Ilex 'Wirt L. Winn'	12	AL, NC, MD, PA, TN, VA
Ilex opaca 'Satyr Hill'	11	AL, MD, NC, PA, TN, VA
Ilex vomitoria 'Pride Of Houston' - Yaupon	10	FL, GA, SC
Ilex cornuta 'Compacta'	9	AL, GA, LA, NC, SC, VA
Ilex cornuta 'Sizzler'	9	AL, GA, SC, NC
Ilex cassine 'Tensaw'	9	AL, FL, GA
Ilex crenata 'Green Luster'	8	FL, GA, SC, MS, NC, TN
Ilex opaca - American Holly	8	AL, FL, MD NC, VA
Ilex 'Dr. Kassab'	8	NC, MD, VA
Ilex 'STBB' Aspire™	8	FL, GA, SC, VA
Ilex verticillata 'Red Sprite'	7	AL, MD, NC, VA
Ilex vomitoria 'Shadow's Female'	7	AL, FL, GA, SC
Ilex ×meserveae 'Dragon Lady'	7	AL, MD, NC
Ilex cornuta 'Fine Line'	6	AL, GA, NC, SC
Ilex latifolia - Lusterleaf Holly	6	GA, SC
Ilex ×meserveae 'Blue Princess'	6	FL, MD, NC, SC
Ilex glabra 'Nigra'	5	AL, GA, NC, TN, VA
Ilex opaca 'Carolina #2'	5	AL, GA, SC
Ilex 'Conty' pp12009 Liberty™	5	AL, GA, NC, SC
Ilex 'Magland' pp14417 Oakland™	5	AL, FL, GA
Ilex verticillata - Common Winterberry	5	MD, NC
Ilex 'Conive' pp9498 Festive™	4	GA, NC, SC

Table 3.1 (***continued***) Number of production nurseries growing various *Ilex* species reported [plant & supply locator] for southern states.

Species/Taxon	Number of nurseries	States where produced
Ilex ×'HL 10 90' pp14,477 ‘Christmas Jewel’®	4	GA, NC, SC, VA
Ilex decidua	3	FL, LA, TN
Ilex decidua 'Warren's Red’	3	MS,AL
Ilex 'Conal' pp9485 ‘Cardinal’™	3	NC, SC
Ilex 'Conot' pp12010 ‘Patriot’™	3	FL, MD, NC
Ilex cornuta 'Rotunda'	2	FL
Ilex cornuta 'Thunderhead'	2	SC
Ilex crenata - Japanese Holly	2	FL, GA
Ilex decidua southern selection	2	AL, MS
Ilex opaca 'Croonenburg'	2	SC
Ilex opaca 'Dan Fenton'	2	MD, NC
Ilex opaca 'Miss Helen' (female)	2	MD
Ilex verticillata 'Jim Dandy'	2	NC
Ilex verticillata 'Southern Gentleman'	2	GA, NC
Ilex vomitoria 'Scarlet's Peak' pp20581	2	FL
Ilex vomitoria 'Shadow's Big Leaf'	2	FL
Ilex vomitoria 'Will Flemings'	2	FL
Ilex crenata 'Bennett's Compacta'	1	NC
Ilex crenata 'Bullata'	1	FL
Ilex crenata 'Chandler'	1	AL
Ilex crenata 'Cherokee'	1	NC
Ilex crenata 'Golden Tip Helleri'	1	NC
Ilex crenata 'Howardi'	1	NC
Ilex crenata 'Kingsville Green Cushion'	1	GA
Ilex crenata 'Northern Beauty'	1	AL
Ilex crenata 'Rotundifolia'	1	NC

Table 3.1 (***continued***) Number of production nurseries growing various *Ilex* species reported [plant & supply locator] for southern states.

Species/Taxon	Number of nurseries	States where produced
Ilex crenata 'Sky Pointer'™	1	MD
Ilex crenata 'Stropkey'	1	AL
Ilex glabra 'Compacta'	1	FL
Ilex glabra 'Georgia Wine'	1	GA
Ilex opaca 'Delia Bradley'	1	NC
Ilex opaca 'Jersey Princess'	1	NC
Ilex opaca (female) - American Holly	1	VA
Ilex 'East Bay'	1	GA
Ilex cassine var. *myrtifolia* - Myrtle-leaved Holly	1	FL
Ilex cassine 'Perdido'	1	AL
Ilex cornuta 'O'Spring'	1	SC
Ilex latifolia 'Auburn'	1	SC
Ilex serrata 'Xanthocarpa'	1	SC
Ilex verticillata 'Compacta'	1	LA
Ilex verticillata 'FarrowBP' Berry Poppins™	1	NC
Ilex verticillata 'Winterberry'	1	TN
Ilex verticillata 'Wintergold'	1	AL
Ilex vomitoria 'Dodd's Cranberry'	1	SC
Ilex vomitoria 'Folsum's Weeping'	1	FL
Ilex vomitoria 'Gray's Green Leaf'	1	AL
Ilex vomitoria 'Hightower'	1	GA
Ilex vomitoria 'Kathy Ann'	1	GA
Ilex vomitoria 'Roundleaf'	1	GA
Ilex vomitoria 'Taylor's Rudolph'	1	FL
Ilex ×'Acadiana'™	1	AL
Ilex ×'Carolina Sentinel'	1	GA

Table 3.1 (***continued***) Number of production nurseries growing various *Ilex* species reported [plant & supply locator] for southern states.

Species/Taxon	Number of nurseries	States where produced
Ilex ×'Emerald Colonnade'®	1	FL
Ilex ×meserveae 'Blue Boy'	1	NC
Ilex ×meserveae 'Blue Girl'	1	GA
Ilex ×meserveae 'Blue Maid'	1	VA, MD
Ilex ×meserveae 'Heckfee' Castle Spire™	1	VA
Ilex ×meserveae - Blue Holly	1	GA
Ilex ×meserveae 'China Boy'	1	FL

In the landscape, hollies are used as specimen plants, foundation plantings, mass plantings, hedges, and topiary. One of the principle ornamental characteristics of hollies are the berries (Figure 3.4), which are typically red but can also be purple to black, white, or yellow; depending on species or cultivar. Berries follow flowering by weeks to several months and typically persist 90 days or more before falling or being consumed by birds or mammals (Galle, 1997). In the 1900s-1950s, cultivars with heavy berry set drove a significant cut-stem industry in the U.S., when holly orchards and nursery stock blocks provided cut stems with fruit for grave blankets, garlands, fall floral arrangements and Christmas wreaths (James and Larry Sanders, personal communication).

Figure 3.4 *Ilex cornuta* 'Burfordii' is a typical example of berry color on hollies in the landscape, although berry color can also be purple to black, white or yellow depending on species and cultivar.

Species and Cultivar Characteristics

Ilex can be shrubs or trees; most are branched to the container top or the ground in nursery production unless specialty market pruned to expose the trunk. They have entire, alternate leaves that are evergreen or deciduous depending on the species (Table 3.2). The leaves may have spines, as does *I. opaca* and many of the hybrids; *I. ×attenuata, I. ×merserve, I.* 'Nelly R. Stevens' and *I.* 'Mary Nell', or not have them as is observed on the deciduous *I. glabra*, *I. decidua,* and *I. verticillata*.

Figure 3.5 Male flowers of *Ilex ×merserve* 'BluePrince®'.

Ilex are dioecious; the male and female flowers occur on separate plants. Male flowers have 4-6 stamens and from 4-6 petals alternating with the stamens (Figure 3.5). The female flowers have a globose round pistil with petals in multiples of 4 (Figure 3.6). Flowers occur on last year's growth and are not considered showy on most species.

Virtually all female *Ilex* require a male plant be nearby to provide (mostly insect transferred) pollen to ensure abundant fruiting. One exception is *I. cornuta* 'Burfordii',

Figure 3.6 Female flowers of *Ilex cornuta* 'Burfordii'.

which is able to set fruit parthenocarpically (without a male present). Male plants that flower at the same time as the female plants need to be selected and planted to ensure proper pollination. The late Bob Simpson, (Simpson's Nursery, Vincennes, IN) created a listing that has the flowering time for male and female cultivars of *I. verticillata* (Figure 3.7; Simpson, 2015).

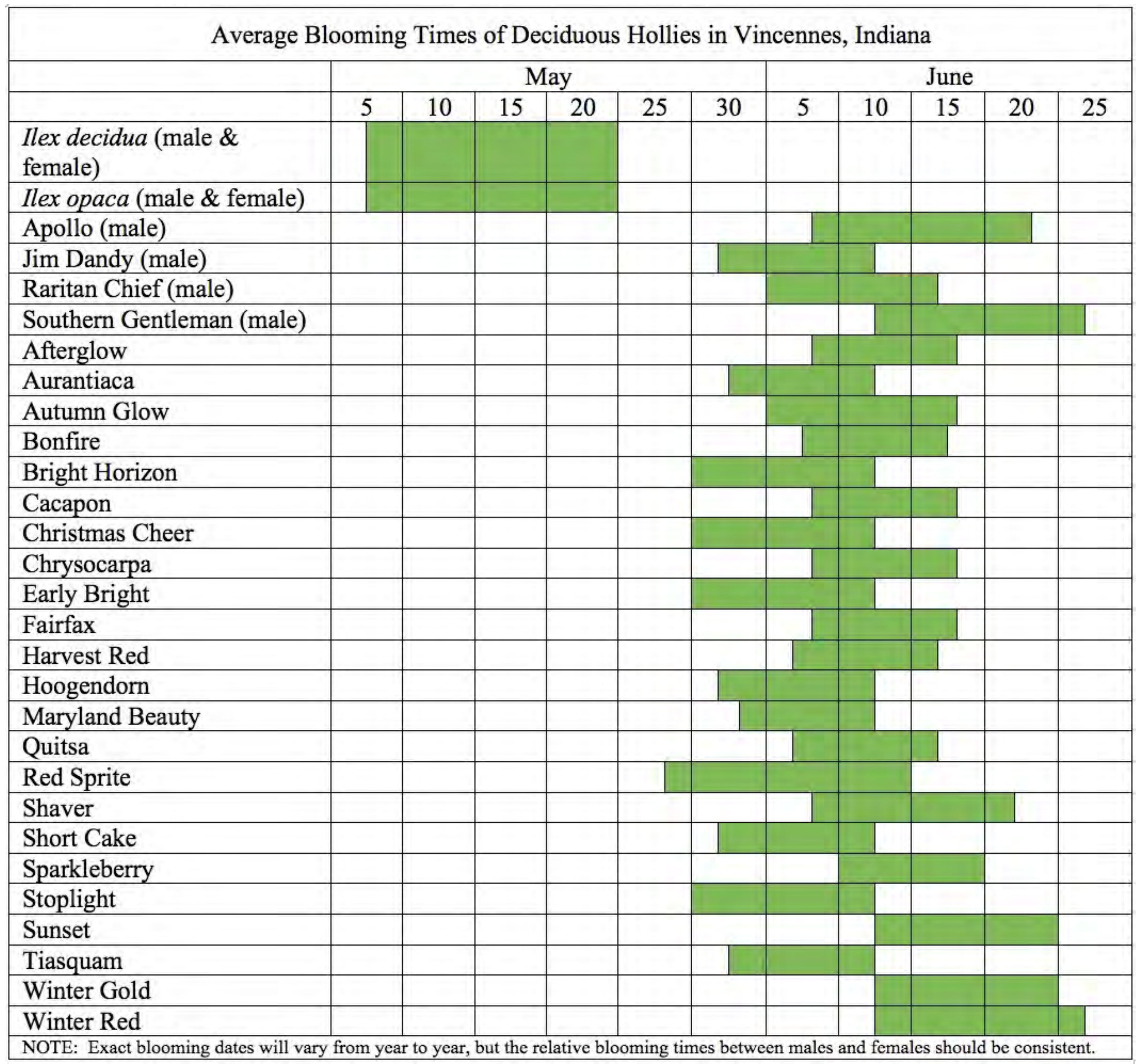


Figure 3.7 The late Bob Simpson, (Simpson's Nursery, Vincennes, IN) created a listing that has the flowering time for male and female cultivars of *Ilex verticillata.* Bob Simpson, Simpson Nurseries

Table 3.2 Characteristics of selected *Ilex* spp. and cultivars currently in production in southeastern United States nurseries

Ilex Species	Cultivar	Common name	Hardiness zone	Size/habit	Foliage	Fruit color	Notes
Ilex ×attenuata	*Ilex ×attenuata*	Fosteri holly	6-9	25-30 ft, narrow pyramid	Narrow with spines	Red	#2 & #3 most common
	Fosters #2	Foster holly	6-9	25-30 ft, narrow pyramid	Glossy with spines	Red	The standard
	'Savannah'	Savannah holly	6-9	25-30 ft	*I. opaca*-like	Red	Large abundant fruit
	'Eagleston'	Eagleston holly	6-9	more rounded than Foster	Glossy with spines	Red	Considered fast growing
	'Greenleaf'	Greenleaf holly	6-9	pyramidal	Spiny green	Red	Precocious fruiting. Frequently listed as *I. opaca*
I. aquifolium	*I. aquifolium*	English holly	7-9	30-50 ft tree	Spiny dark green	Red	A parent of hybrids and sheared hedges
	'Monvilla'	Gold Coast® English holly	6-9	4-10 ft tall rounded shrub	Dark green edged yellow	Male (no fruit)	Pollinator for *I. aquifolium*
I. cassine L.	*I. cassine L.*	Dahoon holly	7-9	20-30 ft height × 8-15 ft wide	Evergreen leaves	Red, rarely yellow	Parent of *I. ×anattenuata* hybrids
	'Tensaw'	Tensaw dahoon holly	7-9	12-15 ft height	Wide leaves	Red	
I. cornuta	*I. cornuta*	Chinese holly	7-9	8-10 ft (to 20 ft) wider than tall rounded shrub	Evergreen with spines	Red on short stalk	Have been grown in zone 6 in protected sites.
	'Burfordii'	Burford holly	7-9	10-20 ft	Green with 1-3 spines	Red	Parthenocarpic, doesn't require a male to set showy fruit
	'Dwarf Burford'	Dwarf burford holly	6-9	5-6 ft height, wider than tall	Like Burfordii	Red	Common to Southeastern landscapes
	'Needlepoint'	Needlepoint holly	7-9	15 ft height × 10 ft wide	Green	Vivid red	Showy fruit
	'Carissa'	Carissa holly	7-9	3-4 ft height × 4-6 ft wide	Simple, glossy green	Red	Smaller form than species
I. crenata	*I. crenata*	Japanese holly	6-10 (Galle), 5-8 (Dirr)	16 ft height × 10 ft wide	Evergreen	Black	Dense habit
	'Sky Pencil'	Sky pencil holly	7-8	20 ft height and tightly columnar	Evergreen - dark green	Black	Marketed as Sky Sentry™, the best of the tight upright hollies.
	'Soft Touch'	Soft touch holly	6-9	2 ft × 3 ft dwarf	Evergreen - dark green	Black	Syn. 'Soft Helleri'
	'Compacta'	Compact Japanese holly				Male	"Bennett Hybrid Group" sold under several names

Table 3.2 (*continued*) Characteristics of selected *Ilex* spp. and cultivars currently in production in southeastern United States nurseries

Ilex Species	Cultivar	Common name	Hardiness zone	Size/habit	Foliage	Fruit color	Notes
I. crenata continued	'Helleri'	Helleri holly	5-9	3 ft tall, wider than tall	Small leaved evergreen	Black	Popular, 2015 HSA Plant of Year.
	'Steeds'	Steeds holly	5-9	pyramidal	Evergreen	Black	Syn. 'Steeds Upright'
	'Hoogendoorn'	Hoogendoorn holly	5-9	2 ft × 2 ft, tight round	Dark green	Male (no fruit)	
I. decidua	*I. decidua*	Possumhaw	5-9	20-30 ft but usually smaller in the landscape	Deciduous tapering to petiole	Orange to dark red	White, 4-merous
	'Warren's Red'	Warren's Red holly	5-8	25 ft-20 ft, more upright than species	Glossy leaved	Abundant bright red	Silver gray branches and trunk
I. 'Emily Bruner'		Emily Bruner holly	7-?	Broadly pyramidal, 20-30 ft	Dark green; evergreen	Vivid red	Popular
I. glabra	*I. glabra*	Inkberry holly	5-9	10 ft; wider than tall	Evergreen	Black	Spreading native
	'Shamrock'	Shamrock holly	5-9	compact	Shiny dark green leaves	Black	Known for leaf glossiness and color.
I. ×*koehneana*	*I.* ×*koehneana*	Koehne holly	6-9	60 ft when mature	Evergreen	Red	*I. aquifolium* × *I. latifolia* hybrids
	'Wirt L. Winn'	Wirt L. Winn holly	6-9	20-25 ft broadly pyramidal	Dark green	Dark red	Female, very good plant (Galle)
I. latifolia	*I. latifolia*	Lusterleaf holly	7-10	60 ft, tree	Evergreen, thick, finely serrate, glossy green leaves.	Red	Used as parent for shrub hybrids
	'Mary Nell'	Mary Nell holly	7-10	10 ft. × 20 ft.	Glossy olive green	Red	
I. 'Nellie R. Stevens'	*I.* 'Nellie R. Stevens'	Nellie R, Stevens holly	6-9	15-25 ft height	Evergreen, 2-3 spines per margin	Vivid Red	Twice as many nurseries grow 'Nellie R. Stevens' than any other holly cultivar.
I. Red Hybrid Hollies	'Magland'	Oakland™ holly	6-9	20 ft height × 12 ft width	Shiny dark green leaves	Male (no fruit)	
	'Conin'	Robin™ holly	6-10	20 ft height × 15 ft width	Nellie R. Stevens-like.	Red	Southern Living Selection. Some leaf damage at minus 14°F in KY
I. opaca	*I. opaca*	American holly	5-9	40 ft height × 30 ft width	Medium green with heavy spines	Red	Slow growth limits nursery interest.

Table 3.2 (*continued*) Characteristics of selected *Ilex* spp. and cultivars currently in production in southeastern United States nurseries

Ilex Species	Cultivar	Common name	Hardiness zone	Size/habit	Foliage	Fruit color	Notes
I. opaca continued	'Carolina #2'	Carolina #2 holly	5-9	30 ft height × 20 ft width	Evergreen	Red	Popular in south.
	'Satyr Hill'	Satyr Hill holly	5-9	30 ft height × 20 ft width	Evergreen	Red	HSA 2003 Plant of the year. Popular as fast growing *I. opaca* cultivar
I. ×meserveae	*I. ×meserveae*	Merserve holly	4-8	15 ft rounded shrub	Evergreen	Red	Susceptible to phytophora in wet, clay soils.
	Blue Princess®	Blue Princess® blue holly	5-7	15 ft height × 10 ft wide	Bluish green	Dark red	Cold hardy
	Blue Prince®	Blue Prince® blue holly	5-7	8-12 ft height rounded shrub	Dark green	Male (no fruit)	Cold hardy
	China Girl®	China Girl® blue holly	5-8	10 ft height × 8 ft wide	Green, leaves cup downward	Vivid red	Heat tolerant, not a blue holly cross
	China Boy®	China Boy® blue holly	5-8	10 ft height × 8 ft wide	Green	Male (no fruit)	Heat tolerant, not a blue holly cross
	Berri-Magic™	Berri-Magic™ blue holly	5-8	10 ft height × 8 ft wide	Evergreen	Red	Combinations of 'Blue Girl' and 'Blue Boy' or China Girl® and China Boy®
I. pedunculosa	*I. pedunculosa*	Long-stalked holly	5-9	15-30 ft height	Evergreen	Bright red on 1-2 inch pedicels, unique, showny	Outstanding for leaf and fruit. Moist woodlands preferred. (Fredericks, 1992)
I. verticillata	*I. verticillata*	Winterberry holly	4-9	6-10 ft height	Deciduous, no fall color	Vivid red	Greatest range of any holly. "Southern form" larger leaves and faster growing.
	'Winter Red'	Winter red winterberry holly	4-9	10 ft rounded	Leaves "Southern Form"	Vivid red, abundant and persistent	The standard for female *I. verticillata.*
	'Red Sprite'	Red sprite winterberry holly	4-9	3-5 ft height	Deciduous	Large vivid red, not as persistent as other cultivars	Dwarf; HSA 2010 Holly of the Year
I. vomitoria	*I. vomitoria*	Yaupon holly	7-10	20 ft × 20 ft	Lustrous green	Red	Soil tolerant, adaptable to moist and dry environs. "Caffeine Hollies"
	'Nana'	Dwarf yaupon holly	7-10	3-5 ft × 4-6 ft	Shiny green	Red not showy	Less than half the size of the species
	'Pendula'	Weeping yaupon holly	7-10	20-25 ft height	Medium green	Vivid red, showy	'Pendula' cultivar is *I. vomitoria* var. *pendula* (Galle)
	'Pride Of Houston'	Pride of Houston yaupon holly	7-10	Upright spreader	Dark green	Vivid red	Heavy flowering and fruit set
	'Stokes Dwarf'	Stokes dwarf yaupon holly	7-10	3-4 ft × 3-4 ft	Purplish new growth to shiny green	Male (no fruit)	Syn. 'Schilling's Dwarf'

Holly fruit are drupes and contain 4-6 seeds (pyrenes). Red is the most common fruit color but yellow, white, and purple to black also occur (Table 3.2). While not uncommon for female hollies to have male names, as is the case with *I. opaca* ‘Chief Paduke’, yellow-fruited ‘Cecil’ or ‘Henry Hicks’, it does reduce their market appeal when consumers mistakenly consider male names represent non-fruiting males.

Management and Production Practices

Holly is a high value, staple crop partly because of its esteem as a garden plant but primarily because many *Ilex* species and cultivars grow more slowly than other nursery crops and consequently have a longer production cycle; additionally, this popular plant requires more intensive management. With the sheer number of species and cultivars on the market (and being grown at individual nurseries), managing the genus as a single crop is impossible (Table 3.1). For example, growth rate of *I.* ‘Nellie R. Stevens’ is nearly double that of *I. vomitoria* in many nurseries, which has a downstream effect on management of irrigation, fertility, pruning, etc.; regardless of if the crop is grown in a container or in-ground.

Figure 3.8 *Ilex aquifolium* ‘Argenta Marginata’ in a large container.

Current U.S. nursery production of *Ilex* is predominantly in containers, because most consumers prefer purchasing containerized plants (Figure 3.8). Container sizes for the homeowner market are typically #1 to #3, with smaller foundation type species/cultivars (e.g. *I. crenata*) being grown in smaller containers and larger tree-type cultivars grown in #3 containers (e.g. *I.* ‘Nellie R. Stevens’). Plants destined for a commercial market are typically grown in larger containers, with smaller foundation type species/cultivars (e.g. *I. crenata*) being grown in smaller #3 containers and larger tree-type cultivars like *I.* ‘Nellie R. Stevens’ grown in #15 - #100 containers. Generally, it takes (depending on location) 6-8 months to finish a #1 container, 12-24 months to finish a #3 container and 24+ months to finish a #7 or larger container.

Most growers producing species of container-grown holly in the southeast use pine bark as the principal substrate component. Depending on the grower or the species of *Ilex*, various organic amendments can be added to the substrate to adjust the physical and chemical properties that contribute to growing quality plants. These amendments may include peat moss, other types of bark from species other than loblolly pine (*Pinus taeda*), or various composted materials, for example, composted cotton gin trash and cotton stalks. The nature of systems, bark suppliers, compost sources and feed stocks, nutrient source, climate, as well as irrigation water quality and the irrigation delivery method and volume applied all preclude the idea of a single or even several recipes of specified growing media for producing high quality plants. There is no holy grail in terms of creating the perfect growing substrate. However, most hollies prefer a slightly acid soil or substrate, pH range 5.5 – 7.0 with 6.0 being ideal (Galle, 1997). ‘Foster’s #2’ prefers a more acidic pH of 5.5 -6.0 (Halcomb and Fulcher, 2010). Pine bark fits that range and this is one of the reasons it is the substrate of choice in container production of *Ilex*. *I. cornuta* ‘Burfordii Compacta’, *I. crenata*, *I. vomitoria* ‘Nana’, *I.* ×*attenuata* ‘Forsterii’, and *I.* ‘Nellie R. Stevens’ all have a high nutrient requirement while *I. glabra*, *I.* ×*attenuata* ‘East Palatka’ and *I.* ×*merserveae* have medium nutrient requirements (Chappell et al*.*, 2013; Table 3.3).

Field production of holly (Figure 3.9) is generally limited to those species and cultivars that are employed as trees in landscapes, with the vast majority being broadleaf evergreen cultivars. Broadleaf evergreen cultivars are generally more receptive to summer digging and transplanting compared to deciduous species and cultivars. In the southeast, examples of cultivars commonly grown in-ground and sold as B&B include ‘Emily Bruner’, ‘Nellie R. Stevens’, ‘Burfordii’, ‘Savannah’, and ‘Wirt L. Winn’. A typical size B&B tree will be 6-8 feet (1.8-2.4 m) tall at sale, and require 24-36 months to attain this size in the field under optimal growing conditions.

Figure 3.9 *Ilex* ×*attenuata* ‘Fosterii’ being grown in a field situation.

To provide the customer with a healthy vigorous plant, it is best to use the American Standard for Nursery Stock, ANSI Z60.1-2014 (AmericanHort, 2014), as the basis for container size and root ball size for a given plant size.

Propagation

Ilex propagation can be accomplished using seed, cuttings, grafting, budding, layering, division, and micropropagation. Seed propagation can be difficult and time consuming (12 months or 2-3 years to germinate) and is mostly used for rootstock production and by breeders. The seed has a morphophysiological dormancy that requires a warm period for the embryo to develop; then a cold stratification. *I. crenata* seems to germinate the easiest, yet like most *Ilex*, seeds needs to be removed from the flesh to germinate. Gently crushing fruit followed by soaking reduces the effort required to remove the seed from the fruit. In his 1953 book, Harold Hume states; “freshly gathered seeds of *I. myrtifolia*, *cassine*, *glabra*, and *crenata* sown in flats in a mixture of sand, soil and peat on January 2, 1950,

Table 3.3 Average range of foliar concentrations reported for macronutrients (pages 92-93) and micro-nutrients (pages 94-95) measured in recently mature leaves collected at mid-season from that current season's growth of select *Ilex* species.

Macronutrient (% dry weight)	Nitrogen (N)	Phosphorus (P)	Potassium (K)	Calcium (Ca)	Magnesium (Mg)	Sulfur (S)
Ilex 'Emily Bruner'	0.80 – 1.69	0.05 – 0.13	0.80 – 1.59	0.72 – 1.62	0.40 – 0.44	0.12 – 0.17
Ilex 'Herbert Kahrs'	1.12 – 1.87	0.07 – 0.19	0.46 – 1.65	1.33 – 2.04	0.33 – 0.43	0.15 – 0.19
Ilex 'Nellie R. Stevens'	1.80 – 2.00	0.13 – 0.14	1.32 – 2.02	0.93 – 1.51	0.33 – 0.36	0.16 – 0.23
Ilex 'Sparkleberry'	1.89 – 2.54	0.12 – 0.24	0.95 – 2.08	0.39 – 0.82	0.35 – 0.67	0.19 – 0.29
Ilex (deciduous cultivars)	1.91 – 2.22	0.11 – 0.17	0.52 – 1.98	0.77 – 1.36	0.40 – 0.97	0.18 – 0.27
Ilex ambigua	2.01 – 3.11	0.12 – 0.21	2.22 – 3.20	0.86 – 0.94	0.4 – 0.65	0.16 – 0.21
Ilex aquifolium cultivars	1.39 – 1.57	0.11 – 0.14	1.33 – 1.72	0.73 – 0.82	0.32 – 0.35	0.19 – 0.26
Ilex aquipernyi 'Dragon Lady'	1.65 – 2.22	0.08 – 0.19	0.69 – 1.44	0.55 – 0.66	0.16 – 0.33	0.12 – 0.19
Ilex ×attenuata 'East Palatka'	1.27 – 2.14	0.07 – 0.14	0.94 – 1.76	0.93 – 1.33	0.31 – 0.41	0.11 – 0.16
Ilex ×attenuata 'Foster #2'	1.46 – 2.34	0.07 – 0.19	0.7 – 1.77	0.59 – 1.25	0.22 – 0.33	0.17 – 0.24
Ilex ×attenuata 'Savannah'	1.55 – 2.29	0.06 – 0.13	0.58 – 1.87	0.94 – 1.21	0.33 – 0.43	0.15 – 0.20
Ilex ×attenuata cultivars	1.46 – 1.51	0.07 – 0.10	0.69 – 0.90	0.63 – 0.86	0.28 – 0.39	0.14 – 0.24
Ilex cassine	0.90 – 1.53	0.05 – 0.11	0.55 – 1.15	0.71 – 1.29	0.41 – 0.56	0.13 – 0.17
Ilex cornuta 'Burfordii'	1.22 – 2.18	0.1 – 0.17	0.87 – 2.18	1.28 – 1.54	0.25 – 0.3	0.18 – 0.23
Ilex cornuta 'Carissa'	1.48 – 2.9	0.15 – 0.22	1.45 – 1.84	0.97 – 1.16	0.33 – 0.41	0.15 – 0.22
Ilex cornuta 'Dwarf Burford'	1.86 – 1.90	0.12 – 0.15	1.33 – 1.42	1.77 – 2.13	0.30 – 0.33	0.15 – 0.20
Ilex cornuta 'Rotunda'	1.35 – 2.40	0.12 – 0.30	0.80 – 2.20	0.70 – 1.50	0.30 – 1.00	0.14 – 0.25
Ilex cornuta cultivars	1.43 – 1.92	0.12 – 0.13	0.80 – 1.87	0.72 – 1.57	0.19 – 0.45	0.12 – 0.17
Ilex crenata 'Compacta'	2.18 – 2.36	0.12 – 0.13	0.72 – 1.00	1.21 – 1.59	0.39 – 0.43	0.23 – 0.29
Ilex crenata 'Convexa'	1.85 – 2.26	0.17 – 0.18	0.77 – 1.56	0.84 – 1.46	0.33 – 0.47	0.17 – 0.23
Ilex crenata 'Helleri'	1.30 – 2.25	0.08 – 0.11	0.52 – 0.93	0.76 – 1.34	0.29 – 0.44	0.17 – 0.21
Ilex crenata 'Hetzii'	2.29 – 2.94	0.13 – 0.19	1.11 – 1.45	1.04 – 1.45	0.24 – 0.32	0.13 – 0.20

Table 3.3 (*continued*) Average range of foliar concentrations reported for macronutrients (pages 92-93) and micro-nutrients (pages 94-95) measured in recently mature leaves collected at mid-season from that current season's growth of select *Ilex* species.

Macronutrient (% dry weight)	Nitrogen (N)	Phosphorus (P)	Potassium (K)	Calcium (Ca)	Magnesium (Mg)	Sulfur (S)
Ilex crenata cultivars	1.88 – 2.74	0.12 – 0.27	0.71 – 1.68	0.67 – 2.12	0.33 – 0.64	0.21 – 0.32
Ilex decidua	2.06 – 2.30	0.13 – 0.20	0.94 – 1.40	1.08 – 1.45	0.53 – 0.89	0.50 – 0.55
Ilex glabra cultivars	1.41 – 1.63	0.06 – 0.10	0.51 – 0.55	0.45 – 0.60	0.09 – 0.19	0.14 – 0.21
Ilex integra	1.53 – 2.31	0.11 – 0.21	1.22 – 1.96	0.99 – 1.28	0.26 – 0.39	0.15 – 0.23
Ilex ×koehneana 'Wirt L. Winn'	1.41 – 2.29	0.08 – 0.11	1.09 – 1.69	1.11 – 1.32	0.29 – 0.38	0.13 – 0.18
Ilex laevigata	2.08 – 2.97	0.11 – 0.18	2.25 – 2.77	0.69 – 1.18	0.33 – 0.75	0.15 – 0.22
Ilex latifolia	1.10 – 1.42	0.08 – 0.12	0.99 – 1.38	1.64 – 1.91	0.25 – 0.47	0.09 – 0.11
Ilex myrtifolia	1.89 – 2.05	0.09 – 0.12	0.6 – 1.57	0.71 – 1.27	0.28 – 0.35	0.13 – 0.17
Ilex opaca	1.59 – 2.94	0.08 – 0.23	0.65 – 1.23	0.33 – 0.80	0.20 – 0.36	0.13 – 0.22
Ilex opaca (dwarf) cultivars	1.68 – 1.81	0.09 – 0.13	0.61 – 0.81	0.65 – 0.82	0.37 – 0.49	0.17 – 0.20
Ilex pernyi	1.54 – 1.91	0.13 – 0.15	0.86 – 1.35	1.15 – 1.48	0.30 – 0.65	0.15 – 0.17
Ilex verticillata 'Winter Red'	2.14 – 2.33	0.12 – 0.20	0.56 – 1.48	0.28 – 0.67	0.24 – 0.49	0.21 – 0.24
Ilex verticillata cultivars	1.90 – 2.58	0.11 – 0.19	0.50 – 2.59	0.42 – 1.21	0.12 – 0.82	0.20 – 0.26
Ilex vomitoria	1.74 – 2.56	0.10 – 0.15	0.67 – 1.04	0.23 – 0.60	0.32 – 0.56	0.12 – 0.18
Ilex vomitoria 'Pendula'	1.89 – 2.08	0.15 – 0.28	1.17 – 1.63	0.52 – 0.87	0.33 – 0.47	0.15 – 0.20
Ilex vomitoria cultivars	2.02 – 2.55	0.10 – 0.13	0.78 – 1.11	0.43 – 0.48	0.45 – 0.53	0.11 – 0.15
Ilex Meserve-type hybrids	1.97 – 2.16	0.14 – 0.18	1.05 – 1.42	0.88 – 1.15	0.27 – 0.32	0.14 – 0.18
Ilex ×meserveae cultivars	1.91 – 3.80	0.11 – 0.14	1.01 – 1.64	0.66 – 1.36	0.21 – 0.51	0.18 – 0.27

Table 3.3 (*continued*) Average range of foliar concentrations reported for macronutrients (pages 92-93) and micro-nutrients (pages 94-95) measured in recently mature leaves collected at mid-season from that current season's growth of select *Ilex* species.

Micronutrient [ppm (µg/g)]	Iron (Fe)	Manganese (Mn)	Boron (B)	Copper (Cu)	Zinc (Zn)	Molybdenum (Mo)
Ilex 'Emily Bruner'	17 – 40	262 – 1763	13 – 66	2 – 8	10 – 91	0.12 – 0.30
Ilex 'Herbert Kahrs'	33 – 64	288 – 3812	33 – 79	3 – 11	34 – 168	0.11 – 0.34
Ilex 'Nellie R. Stevens'	79 – 102	1131 – 1442	59 – 74	4 – 13	257 – 384	0.12 – 0.30
Ilex 'Sparkleberry'	30 – 65	425 – 2133	19 – 33	3 – 9	55 – 421	0.12 – 1.74
Ilex (deciduous cultivars)	55 – 90	1241 – 3175	32 – 58	5 – 8	106 – 263	0.02 – 0.08
Ilex ambigua	56 – 132	267 – 1749	24 – 58	5 – 10	21 – 40	0.08 – 0.22
Ilex aquifolium cultivars	20 – 70	387 – 889	25 – 29	11 – 83	183 – 363	0.12 – 2.10
Ilex aquipernyi 'Dragon Lady'	25 – 40	778 – 1789	13 – 32	3 – 6	17 – 49	0.12 – 0.30
Ilex ×attenuata 'East Palatka'	35 – 89	248 – 1092	19 – 33	1 – 11	33 – 239	0.11 – 0.2
Ilex ×attenuata 'Foster #2'	34 – 89	32 – 834	18 – 29	3 – 11	34 – 183	0.11 – 0.23
Ilex ×attenuata 'Savannah'	32 – 69	266 – 1067	22 – 37	1 – 8	45 – 301	0.1 – 0.32
Ilex ×attenuata cultivars	40 – 66	223 – 647	26 – 30	2 – 6	71 – 227	0.09 – 0.12
Ilex cassine	37 – 50	536 – 2122	34 – 46	2 – 8	117 – 157	0.12 – 0.30
Ilex cornuta 'Burfordii'	45 – 86	276 – 944	29 – 99	3 – 14	34 – 510	0.09 – 0.15
Ilex cornuta 'Carissa'	44 – 75	121 – 236	21 – 42	5 – 11	44 – 197	0.08 – 0.17
Ilex cornuta 'Dwarf Burford'	53 – 59	478 – 1031	87 – 93	5 – 7	96 – 200	0.12 – 0.30
Ilex cornuta 'Rotunda'	35 – 200	75 – 1150	30 – 50	10 – 20	34 – 235	0.19 – 0.61
Ilex cornuta cultivars	37 – 45	1550 – 2081	34 – 72	2 – 7	26 – 139	0.11 – 0.30
Ilex crenata 'Compacta'	52 – 173	289 – 1566	49 – 53	5 – 9	568 – 588	0.12 – 0.34
Ilex crenata 'Convexa'	56 – 102	235 – 2430	29 – 65	6 – 9	33 – 252	0.11 – 0.24
Ilex crenata 'Helleri'	39 – 112	72 – 1962	21 – 39	4 – 11	54 – 357	0.03 – 0.56
Ilex crenata 'Hetzii'	38 – 94	344 – 2524	30 – 74	5 – 9	45 – 393	0.09 – 0.24
Ilex crenata cultivars	43 – 170	248 – 2103	14 – 137	1 – 13	63 – 949	0.12 – 0.26

Table 3.3 (*continued*) Average range of foliar concentrations reported for macronutrients (pages 92-93) and micro-nutrients (pages 94-95) measured in recently mature leaves collected at mid-season from that current season's growth of select *Ilex* species.

Micronutrient [ppm (µg/g)]	Iron (Fe)	Manganese (Mn)	Boron (B)	Copper (Cu)	Zinc (Zn)	Molybdenum (Mo)
Ilex decidua	59 – 103	982 – 1855	37 – 48	4 – 7	38 – 264	0.12 – 0.30
Ilex glabra cultivars	26 – 41	50 – 770	29 – 53	2 – 5	80 – 209	0.12 – 1.16
Ilex integra	44 – 85	154 – 1868	28 – 55	5 – 11	55 – 256	0.12 – 0.12
Ilex ×koehneana 'Wirt L. Winn'	33 – 67	324 – 1384	26 – 60	5 – 11	27 – 98	0.11 – 0.29
Ilex laevigata	34 – 88	126 – 542	24 – 67	5 – 13	31 – 117	0.1 – 0.26
Ilex latifolia	25 – 50	613 – 918	19 – 50	3 – 8	6 – 21	0.09 – 0.30
Ilex myrtifolia	42 – 75	348 – 1559	26 – 40	5 – 11	40 – 286	0.02 – 0.15
Ilex opaca	19 – 199	97 – 1281	13 – 76	3 – 10	21 – 634	0.11 – 0.30
Ilex opaca (dwarf) cultivars	30 – 39	1077 – 1590	38 – 58	4 – 6	142 – 151	0.12 – 0.30
Ilex pernyi	59 – 162	1397 – 1804	62 – 148	1 – 4	20 – 285	0.12 – 0.30
Ilex verticillata 'Winter Red'	30 – 75	589 – 3067	10 – 33	4 – 10	44 – 326	0.12 – 0.30
Ilex verticillata cultivars	28 – 132	1652 – 5545	18 – 73	4 – 17	133 – 563	0.05 – 0.12
Ilex vomitoria	25 – 63	701 – 3190	15 – 98	4 – 8	19 – 80	0.12 – 0.30
Ilex vomitoria 'Pendula'	41 – 60	56 – 192	29 – 90	5 – 10	47 – 100	0.11 – 0.32
Ilex vomitoria cultivars	42 – 57	215 – 1415	57 – 82	2 – 11	48 – 209	0.01 – 0.12
Ilex Meserve-type hybrids	43 – 104	891 – 2466	55 – 70	4 – 10	155 – 203	0.12 – 2.55
Ilex ×meserveae cultivars	103 – 194	538 – 1633	40 – 72	2 – 21	123 – 389	0.12 – 0.56

Figure 3.10 A rooted cutting of *Ilex decidua* 'Council Fire'. The cutting was taken in May 20, 2015 and ready for transplanting July 17, 2015.

and handled in a greenhouse began to germinate February 2, 1950." Patience is required for *I. aquifolium, I. cornuta,* and *I. opaca*. Plants raised from seed typically are a ratio of one male to three or four female with female to male sometimes as high as ten to one (Hume, 1953).

Semi-hardwood tip cuttings from well-matured, current season's growth produce the best plants (Hartmann et al*.*, 2011) when employing vegetative propagation. Recipes for rooting cultivars and species can be fairly routine but frequently require fine tuning to get high percentage rooting at a given propagation facility. To root one of the more difficult species, *I. decidua* and its cultivars, Bon Hartline (2009) recommends taking a cutting 6-8 inches long after a couple of late frosts (Galle, 1997). Strip off all leaves, wound each side, use 3,000 ppm IBA talc or liquid; stick in 50% peat:50% perlite substrate under mist with bottom heat of 70-75°F (21-24°C) (Hartline, 2009). Hartmann and Kester recommend July cuttings for *I. serrata*, *I decidua* and *I. verticillata* with the bottom leaf removed, and quick-dipped in 5,000 ppm IBA (Hartmann et al*.*, 2011; Figure 3.10).

Ilex crenata is plagiotropic and requires terminal cuttings for upright growth. *I. vomitoria* can suffer burn damage and defoliation if treated with NAA (Hartmann et al., 2011). Red shade cloth has been used in Kentucky to increase root development of *Ilex* cuttings (Hartmann et al., 2011; Hopkins, 2009), which is important in liner development (Figure 3.11). Red coverings can continue to increase roots and branching throughout holly container production (Stephens, 2007).

Figure 3.11 Hollies growing under red shade.

Pruning

The majority of hollies have shrub growth habits. Several *Ilex* spp. have an excurrent growth habit (one dominant leader), but in production still require pruning to maintain a single central leader with dense branching and foliage. Examples would be *I. opaca* and *I. ×attenuata*. Halcomb and Fulcher recommend a 3:2 height to width ratio for *I. ×attenuata* (Halcomb and Fulcher, 2010). Excurrent growth hollies are sometimes grown to a shrub-like habit by planting more than one liner in a container. The shrub hollies, *I. cornuta* and *I. crenata,* are trimmed to maintain balanced growth. All hollies will have random vigorous branches grow that can negatively alter their appearance and will need to be removed. Hollies are not pruned during months that would result in stimulating soft growth that could be cold or freeze damaged (mid-July, August, September and October or later in southern states).

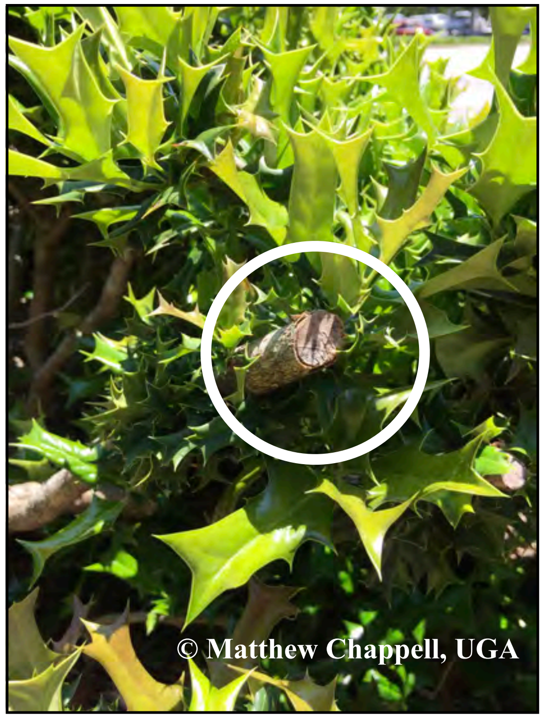


Figure 3.12 Larger pruning cuts, particularly when using shears to prune as seen here on *Ilex cornuta* 'Rounnda', are common.

Pruning by machine and shears leads to a plant with cut ends (Figure 3.12). Time should be given for a flush of new growth to cover the cuts before being sold (Figure 3.13). In the event of plants being left in the container or field for long periods, roots and tops may be severely pruned to restore a holly into a saleable plant, but it is generally not recommended because of the cost related to these efforts.

Irrigation

Most hollies are listed as having a medium irrigation requirement except for *I. dimorphophylla* and *I. vomitoria*, which have low irrigation requirements. In the case of *I. vomitoria*, average water use over a 4-month production cycle in summer averaged 0.46 L (0.26 gallons) per day for a #3 container (Niu et al., 2006). However, many growers are irrigating with 2-4 times that volume, which can lead to increased root pathogen pressure. Newly developed soil moisture monitoring and control systems have been employed by growers and results have been promising. Growth has been almost doubled in containers and the field when compared to timed or manually controlled irrigation systems saving water and time in production at Waverly Farm in Adamstown, MD (Faulring, 2014).

Figure 3.13 Growth over a previous pruning on field grown 'Oakland™' Holly.

SECTION 2

Abiotic Stressors & Mammalian Pests

Ilex opaca with snow resting on the foliage.

ILEX

1. Abiotic Stressors
2. Mammalian Pests

Abiotic Disorders

Hollies, as a genus, are generally known as resilient shrubs and trees that are capable of tolerating many abiotic stress factors. Among the most critical abiotic factors to consider when managing *Ilex* is tolerance to cold. Most of the information related to minimum temperatures that species and cultivars can tolerate is based on visual observations, principally based on extreme winters such as the winter of 1983-84 (Dirr et al., 1984). Still, there has been some research conducted to determine maximum (terminal) cold hardiness of selected species and cultivars. Cold tolerance of *I. aquifolium* was determined to be -11° F (-24°C), as measured in mid-winter after plants had been allowed to acclimate to typical winter conditions (Rutten and Santarius, 1988). The cold hardiness of other species and cultivars, as determined in Georgia after plants were allowed to acclimate to winter conditions (Dirr and Lindstrom Jr., 1990), included *I. ×attenuata* 'East Palatka' (-17°F; -27°C), *I. ×attenuata* 'Foster's #2' (-22°F; -30°C), *I. ×attenuata* 'Savannah' (-17°F; -27°C), *I. ×koehneana* 'Wirt L. Winn' (-6°F; -21°C), *I. latifolia* (0°F; -18°C), *I.* 'Lydia Morris' (0°F; -18°C), *I.* 'Nellie R. Stevens' (-6°F; -21°C), *I. opaca* (-22°F; -30°C), and *I. opaca* 'Greenleaf' (-22°F; -30°C).

Cold damage (not mortality) in holly can be manifest in three ways. First, many of the broadleaf evergreen species and cultivars can experience mid-winter cold damage (Figure 3.14). Mid-winter cold damage typically occurs on days with temperatures well below freezing, ample sunshine and windy conditions. The mechanism of damage is actually more closely related to drought damage. This is because when the circumstances mentioned above are present, the stem/trunk of the plant will be shaded and frozen, while ample sunlight warms the foliage above freezing and initiates transpiration. With the stem/trunk frozen, no water can replace the moisture lost from foliage due to transpiration and desiccation of foliage occurs. Symptoms may take days or weeks to manifest, due to the slowed physiology of the plant (Chappell, 2015). The second mechanism of damage occurs when temperatures drop quickly, as in the case of a strong early fall cold front or late spring cold front, from well above freezing to well below freezing. In these circumstances, sap in the cambium can freeze and cause the cambium to separate from the stem. Growers term this "bark blast" or "bark separation". The third mechanism typically occurs in spring, if spring growth is initiated and temperatures drop below freezing. In this case, new growth will rapidly turn black

Figure 3.14 Mid-winter cold stress/ damage is often first noticed as a discoloration, often reddish to purple, beginning at the leaf margin and progressing inward with time.

Figure 3.15 When freezing temperatures occur after new growth has initiated, the tender new growth often turns black/purple immediately after the freezing event. New growth will have to be initiated, which can cause significant production delays.

(Figure 3.15). This can cause substantial delays in production, from weeks to a year in severe cases. In all three cases, winter protection is available to plants grown in containers (Figure 3.16) (LeBude et. al, 2012). Winter protection for field-grown plants is much more difficult due to the inability of growers to cover and/or shelter plants (unlike container growers who can provide cover to plants). As a result, winter damage is more likely in field-grown nurseries.

Mammalian Pests

Ilex are not a favorite food of deer, but they have been reported and observed by the authors to damage *I.* ×*meserveae*, *I. crenata*, *I.* 'Nellie R. Stevens', *I. pernyi* (Drake et al., 2003) and *I.* 'Burfordii' (Chappell, 2015). Nurseries have become havens, refuges even, for deer in areas of high deer populations. High value crops such as hollies will need protection in the form of a high fence up to 8-10 feet (2.4-3.0 m) (Figure 3.17). Square metal fence is preferred over plastic because rabbits and other rodents can chew through plastic fence and create holes allowing deer to get their heads under the fencing and tear an opening in the fence. Monitoring for deer is still required as under severe stress (being chased, threatened by a car or truck on a nearby roadway) they are able to jump even an 8-foot (2.4 m) fence and will have to be herded to an open gate and out of the fenced nursery.

Figure 3.16 To protect plants from winds and extreme cold, many southern U.S. growers place container plants in covered houses over the winter months. Opaque (white) plastic is preferred because temperatures are moderated, typically lower on sunny days, compared to clear plastic.

Figure 3.17 Wire fence with border plants to keep out deer and to protect from herbicide sprays in neighboring fields.

Arthropod Pest Management

COMMON ARTHROPOD PESTS

1. Leafminers
2. Armored scales
3. Soft scales
4. Other scale insects
5. Twolined spittlebug
6. Aphids
7. Weevils
8. Thrips
9. Spider mites
10. Ambrosia beetles
11. Clearwing borers

Leafminers

Holly species are all susceptible to dipterous leafminers in the genus *Phytomyza* (Shetlar, 2002). These leafminers are the most damaging pests of hollies in the eastern U.S. (Hale, 2015). The native holly leafminer (*Phytomyza ilicicola*) predominantly attacks American holly (Figure 3.18) although it is also a pest of Chinese and English hollies. The holly leafminer (*P. ilicus*) is only a pest of English holly while inkberry leafminer (*P. glabricola*) attacks inkberry and winterberry leafminer (*P. verticillatae*) attacks winterberry (Shetlar, 2002).

Figure 3.18 Holly leafminer (*Phytomyza ilicicola*) damage on *Ilex opaca*.

There are two types of damage caused by these dipterous leafminers. The female leafminer flies use their ovipositor (egg laying structure) to make pinholes in the leaves in the spring. Native holly leafminer flies emerge in early spring when leaves are about 1/2-inch long (Shetlar, 2002). The black 1/8-inch long flies (Hale, 2015) feed on the sap that exudes from the pinholes although very few holes are produced by the inkberry leafminer. This feeding allows the leafminer flies to mature as they prepare to lay their eggs in the pinholes. The numerous pinholes distort the leaves by making them rough, twisted and stunted (Shetlar, 2002).

The second type of damage is caused by the larvae that hatch from the eggs in one or two weeks. The first instar larva of the native holly leafminer forms a thin ¼-inch long trail and then goes dormant until October or November. It molts into a second instar larva, which widens the mine as it feeds. The larva is inactive during freezing periods and resumes activity and molts into a third instar with warmer spring temperatures (Shetlar, 2002). The 1/8 inch long third instar larva is yellow to white (Hale, 2015). Each larva forms a blotch leaf mine as it finishes its larval development. It cuts a small exit hole in the upper leaf surface prior to pupating. Adults emerge and exit the leaf within a couple weeks. There is only one generation per year for the native holly leafminer and the other leafminer species are thought to have a similar life cycles (Shetlar, 2002).

One exception is inkberry leafminer that has three or four generations per year in Ohio. Most of the eggs are laid in last year's leaves in the spring. The first instar larva makes a short straight mine and does not go dormant but continues to develop into a second and third instar larva which produce a blotch mine. Mining from subsequent generations can cause the tip of the leaf to turn black (Shetlar, 2002).

Management

Resistant hybrids such as the Meserve types ('Blue Princess' and 'Blue Prince') should be used where leafminers have been problematic (Shetlar, 2002). Parasitoid wasps can attack the larvae and pupae of inkberry leafminer and possibly other leafminers. These parasitoids are very sensitive to insecticide, so sprays should not be used when parasitoids are active, which is usually once the mines are about half formed (Shetlar, 2002).

Contact insecticides targeting the adults such as carbaryl, chlorpyrifos, spinosad, permethrin and others (Table 3.4) need to be timed to when the adults are active but before they have laid many eggs. For native holly leafminer control, sprays should be applied when the leaves of American holly are only ½ inch long (Shetlar, 2002). For holly leafminer, the black adult flies begin to emerge when the English holly twigs have three to four new leaves (Hale, 2015).

Systemic insecticides (Table 3.4) are used to control young larvae. Applications to the soil should be made early before the leaves expand. Foliar systemic sprays should be applied to English and American hollies when the leaves are nearly half expanded and to inkberry when the short straight mines of the first instar larvae are formed (Shetlar, 2002).

Armored scales

Fifty-nine armored scale insect species (Hemiptera: Diaspididae) have been recorded on *Ilex* spp. (ScaleNet 2015). However, only a handful of species are observed frequently and even fewer are economically important. Tea scale (*Fiorinia theae* Green) (Figure 3.19) and Florida red scale (*Chrysomphalus aonidum* L.) (Figure 3.20) are the most frequently encountered and problematic in southern nurseries and landscapes. Other armored scale species, such as the California red scale (*Aonidiella aurantii* Maskell), oleander scale (*Aspidiotus nerii* Bouche), false Florida red scale (*Chrysomphalus bifasciculatus* Ferris), lantana scale (*Hemiberlesia lataniae* Signoret), greedy scale (*Hemiberlesia rapax* Comstock), camellia scale (*Lepidosaphes camelliae* Hoke), purple scale (*Lepidosaphes beckii* Newman), oystershell scale (*Lepidosaphes ulmi* L.), Japanese maple scale (*Lopholeucaspis japonica* Cockerell), peony scale (*Pseudaonidia paeoninae* Cockerell), false oleander scale (*Pseudaulacaspis cockerelli* Cooley) and euonymus scale (*Unaspis euonymi* Comstock), also are found occasionally on various species of hollies. These occasional armored scale species can be damaging at some locations.

Figure 3.19 Tea scale (*Fiorinia theae* Green) can be an extremely devastating pest on a wide range of holly species and cultivars.

Figure 3.20 A severe infestation of Florida red scale (*Chrysomphalus aonidum*).

Field identification of armored scales typically begins with the overall shape of the scale covers. Those with an oval or round cover (subfamily Aspidiotinae) include California red scale, false Florida red scale, Florida red scale, greedy scale, lantana scale, oleander scale and peony scale. Those with an elongated or oyster-shaped cover (subfamily Diaspidinae) include camellia scale, euonymus scale, false oleander scale, Japanese maple scale, oystershell scale, purple scale and tea scale. Each species has characteristics useful for identification, which will be briefly described in the following sections. A field identification guide to some of the armored scale species on hollies is available from Gyeltshen and Hodges (2013).

Armored scales feed by sucking plant cell content from leaves and woody tissues through a piercing-sucking mouthpart. After the cell content is removed, the plant cells collapse or are left empty. On leaves, the resulting damage often appears as yellowing or chlorotic blotches in the immediate areas where the scale insects are feeding; some blotches may appear on the upper surface even though the insects are feeding on the underside of the leaves (such as the tea scale) (Figure 3.21). Heavily infested plants may abort their leaves, flowers or fruits prematurely. Feeding damage by armored scales on woody tissues may include localized necrosis of woody tissues, which may progress to twig, branch or trunk diebacks during severe infestations. The destruction of vascular tissues also leads to declined plant vigor, stunted plant growth, and premature senescence. The presence of the scale insects, particularly the tests of males, which are often white, on the leaves and trunks can also be aesthetically distracting. For example, the waxy deposits and tests of male tea scales cause a white fuzzy appearance on the underside of leaves. Unlike the soft scales, armored scales do not produce honeydew and sooty mold.

Figure 3.21 Damage from scale, if severe enough, can result in yellowing of foliage on the top-side of foliage, as seen here.

Among the aspidiotines, Florida red scale (Figure 3.20) is one of the most common armored scale species on hollies. This species is widely distributed from Florida to Texas and north to South Carolina and Missouri (Miller and Davidson, 2005, ScaleNet, 2015; Chong, 2015). Historically a pest of citrus, the Florida red scale is now commonly found on the underside of leaves of hollies, camellias, ivy and palms. The test or cover of an adult female Florida red scale is round (2-2.5 mm in diameter), dome-shaped, dark reddish brown, with three concentric rings and a light brown center (appearance similar to a nipple). The female bodies and the eggs are yellow. The covers of mature males are elongated and reddish brown; adult males look like gnats and are short-lived. In the southern U.S., development of the Florida red scale appears to be continuous throughout the year. There may be 3-6 generations per year, depending on the location (Miller and Davidson, 2005). The development from eggs to adults can be completed in 40-55 days in the summer and up to 180 days in the winter.

False Florida red scale (also known as the bifasciculate scale) (Figure 3.22) can be easily confused with the Florida red scale. The two species are almost identical in every aspect of their appearance, except that the covers of the false Florida red scale are of a lighter color. Identification of the two species can only be confirmed through examination of slide-mounted specimens. This species is more polyphagous, and is distributed much farther north than the Florida red scale, with field populations established as far north as Virginia (Miller and Davidson, 2005). The biology of this species is poorly known. In Japan, two generations have been reported, with egg hatch occurring in late May to early June and in late July and August (Murakami, 1970).

Figure 3.22 False Florida red scale (syn. False armored scale) can be easily confused with the Florida red scale. The two species are almost identical in every aspect of their appearance, except that the covers of the false Florida red scale are of a lighter color.

California red scale (Figure 3.23) is one of the most important pests of citrus in the world. In the U.S., this species is distributed in California, and from Florida west to Texas and north to Georgia. This is a polyphagous species, with 11 plant genera reported as hosts in Florida (Dekle, 1977). On hollies, however, the California red scale is generally considered an occasional and minor pest. Adult female covers are flat, circular and translucent (about 1.8 mm in diameter), showing the yellow to red or reddish brown bodies under the cover. Male covers are elongated and similar in color to the female covers. The California red scale primarily feeds on holly foliage, but they will attack all plant parts in other plant species. There are two generations per year along the coast of California,

Figure 3.23 On hollies, the California red scale is generally considered an occasional and minor pest.

whereas three generations occur in the inland areas (Rosen and DeBach, 1978). All life stages are present throughout the year.

Greedy scale (Figure 3.24) is widely distributed in the U.S., and considered one of the most important pest species in the world (Beardsley and Gonzalez, 1975; Miller and Davidson, 1990). This is a polyphagous species with 38 plant genera reported as hosts in Florida (Dekle, 1977). Adult female covers are circular, dome-shaped, whitish to gray, and 2-3 mm in diameter. The "nipple" of the cover is located closer to the side of the cover. Female body and eggs are yellow. The greedy scale is most often found on the bark, encrusting branches and trunks of hollies during heavy infestations. The biology of this species in the U.S. is poorly known. Two overlapping generations per year have been reported in New Zealand (Blank et al., 1995) and Italy (Bianchi et al., 1994), with crawlers likely present throughout the spring and summer.

Lantana scale is polyphagous (reported on 276 plant genera by Dekle, 1977), and similar in appearance to the greedy scale. Distinguishing the two species requires examination of slide-mounted specimens. This species is widely distributed from Florida to California, and north to Indiana and New York. Miller and Davidson (2005) reported that outdoor populations of the lantana scale are established as far north as Maryland. There are two generations per year on Foster hollies in Maryland, with second-instar nymphs overwintering on twigs and leaves (Stoetzel and Davidson, 1974). Crawlers are present from mid-June to mid-July (first generation) and mid-August (second generation).

Figure 3.24 Although an extremely important pest worldwide, greedy scale is only an occasional pest of *Ilex*.

Oleander scale is another widely reported species, but populations are established outdoors only in the southern states (Miller and Davidson, 2005). This species feeds on 61 host genera in Florida (Dekle, 1977), with avocado, bay, citrus, ivy, oleander, olive and palms being the common hosts (Miller and Davidson, 2005). All life stages are found on holly foliage. Adult female covers are flat, circular, white to tan, transparent, about 2.5 mm in diameter, and with a "nipple" in the middle of each cover. Female body is orange yellow and the egg is pale yellow. Male covers are similar in color, oval and smaller. There are huge variations in the biology of this species, which suggest that this may be a species complex instead of a single species. There are three overlapping generations per year, with crawlers beginning to appear in May but continue to be present throughout the spring and summer. The unreproductive adult female is likely the overwintering stage.

Figure 3.25 The peony scale feeds on the twigs and branches of hollies.

Peony scale (sometimes called the Japanese camellia scale) (Figure 3.25) feeds on the twigs and branches of hollies. The cover is about 2.5 mm in diameter, circular to oval, grayish brown, with the “nipple” closer to the side of the cover. Often the covers are dusted or covered with plant materials. Female body is purple with a light yellowish rear end. This species is known to occur in an area covering New Jersey to Pennsylvania, Arkansas, Texas and Florida, as well as California (ScaleNet, 2015), but it is only a pest of hollies in the southern U.S. (Nash, 1973; Baker, 1980). There is one generation per year in the U.S. (English and Turnipseed, 1940; McComb and Davidson, 1969; Tippins et al., 1977). Crawler emergence occurs in early April to late May in Alabama (English and Turnipseed, 1940), April to May in Georgia (Tippins et al., 1977), late May in Virginia (Johnson and Lyon, 1991), late May to mid-June in Maryland (McComb and Davidson, 1969).

Figure 3.26 Tea scale infections can quickly become severe, as seen here. Chazz Hesselein, Alabama Cooperative Extension System, Bugwood.org

Tea scale (Figure 3.26) is more commonly encountered and damaging on camellias, but it can also be found on many hollies grown in nurseries and landscapes. This species is distributed from Florida to Massachusetts and Texas to Indiana, and California (ScaleNet, 2015). All life stages of the tea scale are found on the underside of the leaves and infestations can be severe (Figure 3.27). Crawlers are tiny, yellow and mobile. Adult females have yellow bodies covered by elongated, brown and single-ridged tests (or covers, about 1.5 mm in length) (Figure 3.27). Male nymphs produce large amount of white waxy

Figure 3.27 Tea scale crawlers are tiny, yellow and mobile. Adult females have yellow bodies covered by elongated, brown and single-ridged tests.

filaments (Figure 3.26). Each female produces 17-43 eggs in a month (Chiu and Kouskolekas, 1980; Munir and Sailer, 1985). There may be as many as three overlapping generations per year. Each generation completes development in 60-70 days (English and Turnipseed, 1940). All life stages are present throughout the year, and crawlers likely emerge from spring to fall. The emergence of crawlers of the first generation begins in February in Georgia (Hodges and Braman, 2004), March in Alabama (English and Turnipseed, 1940), and April in Maryland (Gill, 2011).

Camellia scale is a serious pest of camellias but a minor pest of hollies. This species can be found from Florida to Massachusetts, Pennsylvania, Oklahoma and Texas (ScaleNet, 2015), but it is only a major pest in the southern U.S. Covers are slightly flattened, oyster-shaped, grayish brown, smooth and about 2.5 mm long. Female body and eggs are purple in color. Male tests are smaller and lack the white wax deposits that are found on tea scale male tests. Despite the high frequency of its encounter, biology of the camellia scale is poorly known. In the field, all life stages are present throughout the year (Miller and Davidson, 2005). Development of females is completed in 45 days at 20°C (68°F), 23 days at 25°C (77°F) and 18 days at 30°C (86°F) , while the developmental duration is about 4-13 days longer for the males (Cooper and Oetting, 1989). Each female is capable of producing 30-169 eggs (Cooper and Oetting, 1989). There may be 4-5 overlapping generations per year in central Georgia (Cooper and Oetting, 1989). Crawler emergence first occurs in May (Chong, 2015) and additional emergence peaks in July, September and November in central Georgia (Cooper and Oetting, 1989).

The preferred host of the euonymus scale (Figure 3.28) is euonymus; this species attacks hollies (on leaves) only occasionally. This is a species native to Asia, and currently distributed in Florida to California and north to New York, Massachusetts, Indiana, Illinois, Missouri and Oklahoma (ScaleNet, 2015). Female covers are broadly oyster- or pear-shaped, brownish yellow, and about 1.6 mm long. Male covers are elongated, white, felted, and with three ridges on the back. Female body is orange or yellow; egg and crawler are yellow. There are two overlapping generations per year in the northern, and three overlapping generations in the southern, U.S. (Gill et al., 1982). The population overwinters as adult females. In Maryland, crawlers emerge in early May to early June (first generation) and mid-July (second generation).

Figure 3.28 Euonymous scale, although a minor pest of hollies, has a very unique appearance. Gyorgy Csoka, Hungary Forest Research Institute, Bugwood.org

False oleander scale, also known as the magnolia white scale, is the most common armored scale species infesting southern magnolias. It is a minor pest of hollies. Its host range includes 263 plant species from 79 families (ScaleNet, 2015). This species feeds on leaves. Outdoor populations are established from Florida to South Carolina, Texas and Arkansas. Female covers are broadly oyster- or pear-shaped, 2-3 mm long, flat, and yellowish to brown. Female body, egg and crawler are yellow. Males are rarely seen. False oleander scale originated from East Asia, and was first detected in Florida in 1942. Tippins (1968) reports that false oleander scale reproduces year-round in southern Georgia, and each generation is completed within two months. Each female reproduces for about 60 days. All life stages are present throughout the year, and there may be several generations per year. Crawlers are present at anytime during the year and that there is no distinctive crawler emergence time.

Japanese maple scale (Figure 3.29) has recently become a serious issue for maples in urban landscapes. This is a minor pest of hollies, infesting branches and trunks. This species is distributed in the eastern U.S. from Georgia to New York and Connecticut, and west to Louisiana and Indiana. Adult female covers are elongated, with a whitish outer waxy cover and the under layer reddish brown. Male covers are similar to those of the females, but smaller. Female body, egg and crawler are purple. There is one generation per year in Pennsylvania (Miller and Davidson, 2005), and two generations in Maryland, Tennessee and Virginia (Fulcher et al., 2011; Gill et al., 2012).

Figure 3.29 Japanese maple scale has recently become a serious issue for maples in urban landscapes. Although this is currently a minor pest of hollies, only occasionally infesting branches and trunks, it should be monitored as little is currently known about this pest. Pictured here you see the underside of one scale insect and the topside of several others.

Oystershell scale (Figure 3.30) feeds on twigs and branches. Host plants include more than 100 species. Female covers are about 2.5 mm long, oyster-shaped, and gray or brown and closely resemble bark in coloration. Female body is white with a brown end; eggs and crawlers are also white. The oystershell scale is distributed throughout the U.S., but more commonly found in the northern states. There is generally one generation per year. Crawlers emerge in April in Kentucky (Mussey and Potter, 1997), May in Michigan and Ohio (Herms, 2004), and May and July (two generations) in Virginia (Day, 2009).

Figure 3.30 Oystershell scale is an uncommon pest of holly in southern states but is more frequestly seen north of the Mason-Dixon line.

Purple scale feeds on 45 plant genera in Florida (Dekle, 1977). This species is distributed in California, and from Florida west to Louisiana and north to Missouri and North Carolina. Female covers are oyster-shaped, purplish brown to pale yellow brown, and about 2 mm long. Male covers are similar to those of females, but shorter and smaller. Female body and egg are white; crawler is whitish yellow. There are three generations per year; each generation takes 2-3 months to complete in Florida and Alabama (English and Turnipseed, 1940, Thompson and Griffiths, 1949). In Florida, crawler emergence peaks in March-April, June-July and September-October (Thompson and Griffiths, 1949).

Soft scales

Thirty-eight soft scales (Hemiptera: Coccidae) are known to feed on *Ilex* spp. (ScaleNet, 2015). Among these, Florida wax scale (*Ceroplastes floridensis* Comstock), Indian wax scale [*Ceroplastes ceriferus* (Fabricius)] and cottony camellia scale [*Pulvinaria floccifera* (Westwood)] are the most frequently encountered species in southern nurseries and landscapes. Other regionally or occasionally important species include barnacle scale (*Ceroplastes cirripediformis* Comstock), brown soft scale (*Coccus hesperidum* L.), tessellated scale [*Eucalymnatus tessellatus* (Signoret)], acuminate scale [*Kilifia acuminata* (Signoret)], glassy scale [*Inglisia vitrea* or *Pseudokermes vitreus* (Cockerell)], hemispherical scale [*Saissetia coffeae* (Walker)] and black scale [*Saissetia oleae* (Olivier)]. A field identification guide to scale insect species on hollies is available from Gyeltshen and Hodges (2013).

Similar to the armored scales, soft scales also use piercing-sucking mouthparts to feed on the plants. The difference is armored scales feed on contents of individual cells while soft scales usually remove sap from the phloem tissues. Plant sap is low in proteins and amino acids, which is needed by the soft scales. Therefore, the soft scales have to ingest a large quantity of plant sap, absorb the nutrients, and excrete the sugary waste called honeydew. The shiny, sticky honeydew droplets on the leaves can be aesthetically distracting. Honeydew is a superb growing medium for black sooty mold, which forms a film on the leaf surface and interferes with photosynthesis. Removal of sap and nutrients from the plants also retards plant growth and eventually leads to dieback.

Figure 3.31 Florida wax scale is commonly seen on a vast number of hosts across the southeast, including *Ilex*.

Florida wax scale (Figure 3.31) is the most commonly encountered scale insect species on hollies, particularly in the southern states. Its hosts include more than 250 plant species, and it is distributed from Florida to New York and westward to Texas and Pennsylvania. This species is not considered a pest in the northeastern U.S. because the population cannot survive winter (Kosztarab, 1996). Females are oval, about 3 mm in diameter, and covered with a thick, gummy wax layer. Nymphs are oval, and the wax deposits do not cover the entire body but appear as blooms of wax on the fringe and top of body, giving it almost a star-like appearance. The bodies of adults and nymphs are pink or reddish. There are two generations per year in the southern range (spanning Florida, North Carolina and Texas) and one generation in its northern range (Virginia to Pennsylvania). In South Carolina, crawler emergence occurs in May and July (Chong, 2015). Nymphs and adults feed on twigs, branches and along leaf veins. Adult female is the overwintering stage. In spring, each female can produce more than 150 eggs.

Figure 3.32 Indian wax scale purplish females (immature) and white wax covered females (mature) on plant stem.

Indian wax scale is a common pest of hollies, and its pest status appears more important north of the range dominated by the Florida wax scale. This polyphagous species feeds primarily on the twigs and branches of hollies, and can be found from Florida to Texas and Oklahoma and northward to Illinois and New York (ScaleNet, 2015). Similar to the Florida wax scale, this species may not establish permanent populations in the northeastern U.S. because of its poor tolerance for low winter temperatures (Kosztarab, 1996). Eggs, crawlers, and immature nymphs are purple or reddish while similarly colored adult females are circular, about 6 mm in diameter, and covered by a thick layer of white wax (Figure 3.32). There is one generation per year, and crawler emergence occurs from late May to mid-June in Georgia (Hodges and Braman, 2004) and Virginia (Smith et al., 1971) and late June to July in Maryland (Smith et al., 1971). Each female is capable of producing 1000-2000 eggs.

Figure 3.33 Barnacle scale is primarily a pest in the deep south, but is occasionally found as far north as Pennsylvania and New York.

Barnacle scale (Figure 3.33) is another species of wax scale. Although this species has a wide distribution that spans Florida to California, Missouri, Ohio and Pennsylvania (ScaleNet, 2015), the barnacle scale is a pest of hollies mainly in Florida and the southern U.S. It overwinters as adult females in Florida, but may not survive the winter in the northeastern U.S. (Kosztarab, 1996). The barnacle scale can be found feeding on all parts of a plant. There is one generation per year in California (Gill, 1988), and several overlapping generations in the southern U.S. (Kosztarab, 1996).

Cottony camellia scale (a.k.a. cottony taxus scale) is distributed in the U.S. stretching from Florida to Texas, Massachusetts and Illinois, as well as California and the Pacific Northwest (ScaleNet, 2015). This is a polyphagous species and the host range includes camellia, holly, yew and rhododendron. Adult females are about 3 mm in length, elongated oval, yellowish brown in the back, with dark brown margin (Figure 3.34). There is only one generation per year. Adults produce hundreds of reddish or purplish eggs in white, waxy, fluffy ovisacs (about twice the length of female) on the underside of leaves in May (Figure 3.34). Crawlers emerge in May in Tennessee. They emerge in June in Virginia (Day, 2009) and Connecticut (Johnson and Lyon, 1991). Crawlers and nymphs feed on the foliage. Just before leaf senescence in the fall, the second-instar nymphs migrate to the twigs to overwinter. Adult females migrate back to the leaves to reproduce.

Figure 3.34 Cottony camellia scale adult female overwintering (Left) and laying the white ovisac (Right).

Brown soft scale is common pest of many ornamental plants, particularly in greenhouses, but it is an occasional pest of hollies in nurseries and landscapes. This is one of the most widely distributed soft scale species in the world. In the U.S., permanent populations are established in areas south of Maryland and eastern Virginia (Kosztarab, 1996). Adult females are 2.5 to 4 mm in diameter, oval, yellowish brown, and mottled with dark brown markings (Figure 3.35). There are 3 to 5 generations per year in California (Gill, 1988). Crawlers are present throughout the year. All life stages feed mainly on the leaves, but will

Figure 3.35 Brown soft scale is common pest of many ornamental plants. Adult females are 2.5 to 4 mm in diameter, oval, yellowish brown, and mottled with dark brown markings.

also feed on twigs and branches when the population is large.

Tessellated scale is distributed from Florida to Texas, north to Massachusetts, New York, Indiana, Illinois and Michigan (ScaleNet, 2015). This is a polyphagous species. Adult females are pear-shaped, 2-5 mm in diameter, flat, reddish to dark brown, and with a raised ridge in the middle of the back. The main distinguishing feature of this species, i.e. the polygonal areas (tessellations) on the back, can only be observed in slide-mounted specimens. No male is known for this species. Most individuals feed on leaves, and some on stems. This is mainly a greenhouse pest, but may have one or two generations per year outdoors in Florida.

Acuminate scale is a minor pest of hollies and a poorly known species. Specimens of this species have been reported from Alabama, Georgia, Missouri, New York and Texas (ScaleNet, 2015). Female body is broadly oval with head narrowly rounded (thus appearring triangular), flat, and pale green or yellowish green. This species feeds on leaves. This species appear to have three generations per year when feeding on mango in Egypt, with crawlers emerging in March, July and November (Hassan et al., 2012).

Glassy scale receives its common name due to its glassy, transparent cover. The cover also has a conspicuous longitudinal ridge. The body under the cover is oval, yellowish to reddish brown, about 3 mm in diameter. This species feeds on leaves, and is only found in Florida. Biology of this species is not known.

Figure 3.36 Life stages of hemispherical scale.

Similar to the brown soft scale, hemispherical scale is a common pest of ornamental plants grown in greenhouses, shade houses and nurseries. This is an extremely polyphagous species and widely distributed in the tropical region and greenhouses of the world. Because it is a tropical species, established outdoor populations are only found in southern Florida and California (Johnson and Lyon, 1991). Adult females are 3-5 mm in diameter, hemispherical from side view and oval from top view and dark brown to black (Figure 3.36). There are 1 or 2 generations per year in Florida (Hamon and Williams, 1984) and two or more overlapping generations in California (Gill, 1988).

Figure 3.37 Black scale is yet another pest that is more prevalent in the southern states compared to northern states, but can be found nation-wide.

Black scale (Figure 3.37) is a relative of the hemispherical scale. Similar to the hemispherical scale, the black scale is also extremely polyphagous. Differently, however, the black scale is widely distributed in the U.S., from Florida to California, north to Washington, Colorado, Kansas, Indiana, New York and Massachusetts (ScaleNet, 2015). It is a more important pest species in the southern U.S. Females are 3-5 mm in diameter, brown to black, hemispherical in profile, and have a distinctive “H” marking on the back. There is one generation per year in the inland region of California, but two generations along the coast (Johnson and Lyon, 1991). Each female may deposit 2,000 eggs. The black scale and the hemispherical scale reproduce parthenogenetically, therefore, no males are known in these species. Older nymphs and adults prefer to feed on twigs and branches, while younger nymphs also feed on the foliage.

Other scale insects

Holly pit scale, *Asterolecanium puteanum* (Hemiptera: Asterolecaniidae) (Figure 3.38), is a native species of North America, and is commonly found on American holly, Burford holly and Japanese holly. This species is distributed in the eastern U.S. stretching from Florida to Alabama, Pennsylvania and New Jersey. This is a small scale insect (about 1 mm). Females are oval, convex, and yellowish green or golden. No cover can be found over the insects. They are most often found feeding on stems. Their feeding causes sunken pits on the stems; severe infestations often distort the stem, retard growth, cause premature leaf drop, and lead to stem dieback. Holly pit scales overwinter as adult females. There is likely one generation per year, with crawlers emerging in June and July.

Figure 3.38 Holly pit scale is a native species of North America, and is commonly found on American holly, burford holly and Japanese holly. This species is distributed in the eastern U.S. stretching from Florida to Alabama, Pennsylvania and New Jersey.

Management – Scale insects

Scale insects are cryptic (which makes them difficult to detect), have enormous fecundity (which allows populations to build up quickly), are covered with shells or wax (which makes them difficult to control with insecticide sprays), and some species have long crawler emergence time (which makes accurate timing of application quite difficult and may require repeated applications). Successful management of scale insects (including armored scales, soft scales and pit making scales) depends on good understanding of scale insect biology, feeding habits, natural enemies, and management approaches that are available.

Figure 3.39 Often scale infestations begin on single branches, so scouting and removal early in the pest cycle can significantly reduce spread.

Prevention is the first step of any management program. Pruning off lightly or locally infested branches (Figure 3.39) or foliage removes the scale insect population and prevents dispersal. Heavily infested plants should be removed from the production area and discarded properly. Cleaning up debris, leaves and pruned tissues ensure no scale insect crawlers return to the holly shrubs.

No resistant holly species or cultivar has been developed against scale insects.

A large number of insecticides are registered for use against scale insects in nurseries and landscapes. These insecticides are generally categorized as contact (carbaryl, chlorpyrifos, pyrethroids, buprofezin, azadirachtin, pyriproxyfen, insecticidal soap and horticultural oil), systemic (spirotetramat, acetamiprid, acephate, dimethoate, dinotefuran, imidacloprid, thiamethoxam and chlorantraniliprole), and biological (the entomopathogenic fungi *Beauvaria bassiana*) (Table 3.4).

When applying contact, systemic and biological insecticides as sprays, the most effective timing is during or shortly after crawler emergence. Armored scale covers are made of wax, cast skin and fecal materials, and function as protection against the environment and natural enemies. Insecticide solution cannot penetrate the covers and make contact with the bodies of scale insects. Crawlers have little or no waxy shells and so they are more susceptible.

Crawler emergence can be monitored with:

1. Visual inspection. Inspect infested plants or leaves weekly near the reported time of crawler emergence. Hand lens and magnifying glass are great aids. Look for yellowish, reddish or purplish crawlers that are dispersing or emerging from the female shells.
2. Sticky band. A sticky band can be easily constructed by wrapping double-sided tape around an infested branch a few weeks before the reported emergence time of the crawlers of each species. A sticky trap can also be made by wrapping electrical tape around the twig and coating it with a thin layer of petroleum jelly. Check the sticky bands weekly and look for yellowish, reddish or purplish crawlers that are stuck to the sticky bands.
3. Degree-day models. Degree-day models are available for tea scale (526 centigrade degree-day, at base temperature of 5°C) (Hodges and Braman, 2004), Indian wax scale (846 centigrade degree-day, at base temperature of 12.78°C) (Hodges and Braman, 2004), Japanese maple scale (806 Farenheit degree-day for the first generation and 2220 Farenheit degree-day for the second generation; base temperature of 50°F and start date of January 1) (Gill et al., 2012), and oystershell scale (761 Farenheit degree-day, at base temperature 40°F) (Mussey and Potter, 1997). It is important to understand that degree-day models are tools that help forecast crawler emergence; visual inspection or sticky bands should still be used to confirm the presence of crawlers before applying insecticides.
4. Plant phenological indicators. The phenology of plants and insects are intimately linked to ambient temperature, which make plant phenological indicators useful tools in forecasting the emergence of crawlers or other life stages. Crawler emergence of Indian wax scale occurs when *Yucca filamentosa* L. is blooming, whereas those of tea scale occur when Chinese wisteria is in 50% bloom, in Georgia (Hodges and Braman, 2004). Crawlers of the oystershell scale emerge around the time when Japanese flowering crabapple (*Malus floribunda*), eastern redbud (*Cercis canadensis*), flowering dogwood (*Cornus florida*), Sargent's crabapple (*Malus sargentii*) and lilac (*Syringa vulgaris*) are at 95% bloom, and snowball viburnum (*Viburnum plicatum* var. *tomentosum*) are in first bloom in Kentucky (Mussey and Potter, 2004). In Maryland, the first-generation crawlers of Japanese maple scale emerge when *Cotinus coggygrias* (smokebush) and *Syringa chinensis* (Chinese lilac) are in full bloom, and the second-generation crawlers emerge when *Aralia spinosa* (devil's walkingstick) is in bud (Gill et al., 2012). Again, confirmation with visual inspection or sticky trap still is needed. When using degree-day models and plant phenological indicators, it is advisable to keep detailed records of the variation between predicted and actual dates of emergence, as well as differences among locations and cultivars, and adjust accordingly for future predictions.

Feeding site of scale insects has significant influence on the efficacy of systemic insecticides (Table 3.4). Systemic insecticides (particularly the neonicotinoids), when applied through indirect application methods such as soil drench, soil injection, trunk injection, trunk spray and granules, are more effective against scale insects (nymphs or adults) that are feeding on leaves (such as tea scale, camellia scale or cottony camellia scale). Systemic insecticides applied using indirect methods are not nearly as effective against scale insect species (such as oystershell scale, Japanese maple scale and holly pit scale) or life stages (such as ovipositing adults) that settled on the woody tissues. Repeated sprays of horticultural oil, insect growth regulators (buprofezin and pyriproxyfen), spirotetramat and the neonicotinoids timed for, or shortly after, the peak emergence of crawlers perform better against these scale insect species and life stages feeding on woody tissues. Dormant oil (applied after leaf drop and before bud swell) is also effective in reducing scale insect populations.

Figure 3.40 Black citrus (tea) aphid all life stages with a ladybird beetle larva (arrow) hunting aphids.

A large number of pathogens, predators and parasitoids attack scale insects in nurseries and landscapes (Figure 3.40). Cooper and Oetting (1987) reported three parasitoids of tea and camellia scales, while Hodges and Braman (2004) reported five and six predator and parasitoid species of Indian wax scale and tea scale, respectively, in Georgia. In Florida, about 13.5% of the tea scale population is lost to parasitoids and predators annually (Munir and Sailer, 1985). However, the populations and activities of the arthropod predators and parasitoids can be disrupted by the use of broad-spectrum insecticides (such as acephate, carbaryl, chlorpyrifos and pyrethroids). For example, the use of carbaryl had been shown to increase the population of the Florida red scale through the elimination of the parasitoid population (Rehman et al., 2000). Whenever possible, limit or eliminate the use of broad-spectrum insecticides to reduce the negative impact and conserve the population of natural enemies.

Twolined Spittlebug

Twolined spittlebug [*Prosapia bicincta* (Say)] (Figure 3.41) nymphs are pests of grasses (preferred hosts are centipedegrass and St. Augustinegrass) while the adults feed on grasses or the foliage of several ornamentals, especially hollies (Gorsuch, 2003; Brandenburg et al*.,* 2002). The nymphs produce a bubbly spittle mass while feeding on the grass stems with their piercing-sucking mouthparts. Their feeding can stress grass and cause it to yellow or even turn brown. The wingless nymphs are white, yellow or orange with a brown head and red eyes (Brandenburg et al*.,* 2002).

Figure 3.41 Twolined spittlebug nymphs are pests of grasses (preferred hosts are centipedegrass and St. Augustinegrass) while the adults feed on grasses or the foliage of several ornamentals, especially hollies.

The adults are 1/3 inch long, dark brown to black with red eyes and legs, wings held rooflike over the back, two red or orange lines across the wings and a narrower band across the top of the thorax (Gorsuch, 2003; Brandenburg et al*.,* 2002). Later in the summer or early fall, the adults fly to ornamentals such as holly to feed which causes splotchy yellow leaves which may prematurely drop (Brandenburg et al*.,* 2002).

If spittlebug nymphs are damaging turfgrass, control of the nymphs with insecticide (Table 3.4) will reduce the number of subsequent adults and thus limit potential damage to holly by the adults. Insecticides can also be applied in late summer or whenever adults are seen in the holly canopy.

Aphids

Black citrus aphid [*Toxoptera aurantii* (Boyer de Fonscolombe)] is relatively large (1-2 mm long) when compared to other aphid species. The black citrus aphid appears pear-shaped, and with a pair of black tailpipes (or cornicles) at the rear end. Nymphs are reddish brown, and adults are usually shiny black (but can range from dull black to mahogany) (Johnson and Lyon, 1991) (Figure 3.42). The antennae and legs of both nymphs and adults are striped. The black citrus aphid is a pest of citrus, but it will attack the growing terminals of hollies (particularly English holly) in large numbers. A population often includes both winged and wingless forms. This species is distributed from Florida westward to Texas and northward to New York, as well as from California to Washington. The adult female aphids reproduce without mating (parthenogenesis) by producing unfertilized eggs that hatch within the body so that nymphs are born. An average of 50 offspring per adult female over 7-10 days plus a generation time of as little as 6 days allow for rapid population growth. Multiple overlapping generations occur from spring to fall.

Similar to soft scales, aphids feed on the phloem of hollies. Aphids produce honeydew and promote the growth of sooty mold. Distortion may occur on heavily infested terminals. The presence of honeydew, sooty mold, the aphids and their cast skins can reduce the aesthetic value of the hollies.

Management

Aphid populations begin to appear as early as March and will remain until the winter. Because of the explosive growth potential of an aphid population, management should begin before or immediately after the detection of aphid infestation.

Figure 3.42 Black citrus aphid is relatively large when compared to other aphid species. The black citrus aphid appears pear-shaped, and with a pair of black tailpipes (or cornicles) at the rear end. Nymphs are reddish brown, and adults are usually shiny black.

Many aphid species can use common weeds and other plant materials in the production areas as alternative hosts. The first step in developing an aphid management program is to develop good weed management and sanitation programs where weeds, debris and infested plants are removed regularly. Growing plant species that are attractive to black citrus aphids, such as citrus, camellias, *Pittosporum, Ixora* and *Elaeagnus*, downwind from the hollies can help to reduce the dispersal of aphids onto the hollies by wind.

When the aphid population is small, spraying with a forceful jet of water can also dislodge many from the infested plant. This method, however, may not be practical when a large number of densely packed holly shrubs are infested.

No study has investigated resistance in various holly species and cultivars against aphids.

Typically, aphid infestations do not cause long-term deterioration in plant health. In a typical landscape setting, management may not be needed because aphid populations suffer very high mortality due to detrimental environmental factors and natural enemies. Lady beetles, lacewings, syrphid flies and parasitoid wasps are frequently found to attack aphids. In warm and wet weather, aphid populations are often decimated by outbreaks of naturally existing entomopathogenic fungi. The use of broad-spectrum insecticides (Table 3.4) should be limited to conserve the population of natural enemies (see discussion in the section on *Management – Scale insects*). Ant treatment may also be necessary when biological control is practiced because ants are known to collect honeydew of aphids and interfere with the actions of natural enemies.

When aphid infestations are large and existing biological control failed to prevent injuries to hollies, chemical management options may be considered. Insecticides registered for the management of aphids in production nursery include carbaryl, acephate, pyrethroids, neonicotinoids (acetamiprid, dinotefuran, imidacloprid and thiamethoxam), pymetrozine, azadiractin, abamectin, spirotetramat, neem oil, horticultural oil and insecticidal soap (Table 3.4). The entomopathogenic fungus *Beauveria bassiana* is also very effective in reducing aphid populations when used repeatedly. Systemic insecticides, such as the neonicotinoids, could be applied by topical sprays, soil drench, broadcast granules, or trunk sprays. The neonicotinoids may require days or weeks to move into the plant tissues but can provide long residual protection against aphid infestation; therefore, these chemicals should be applied before the population increases. For existing populations, topical applications of listed insecticides to the infested terminals on a weekly or biweekly basis can be very effective in eliminating infestations and preventing reinfestation of the holly terminals.

Weevils

Black vine weevil [*Otiorhynchus sulcatus* (F.)] (Coleoptera: Curculionidae) is a common pest of ornamental shrubs in the northern half of the U.S. (Johnson and Lyon, 1991). The black vine weevil was introduced from northern Europe in 1835 (Smith, 1932). This species attacks 150 plant species including yews, rhododendrons, euonymus, camellia, and many other evergreen species (Reding, 2008). Adult beetle is 3/8 inch long, dull gray to black, with short broad snout, and sporting numerous small golden spots (actually pits with short golden hairs) on the elytra (wing covers) (Figure 3.43). Adult females become active at night, climb onto the host plant (they do not fly) and cause mainly esthetic damage to nursery crops by creating elongated notches on the edges of the leaves (Figure 3.44). Additional damage is done by the larvae (legless, white, C-shaped grubs with brown heads), which feed on the roots, eventually girdling the roots and stem (Figure 3.45). The population consists of females only and reproduces through parthenogenesis. Adult females drop the eggs on the soil surface or insert them into plant crevices (Smith, 1932). There is only one generation per year, with older larvae overwintering in the soil. Adults emerge in mid-June in the Northeast, and in March in California (Johnson and Lyon, 1991). Reproduction occurs about one month after adult emergence and may last until the winter, with each female depositing up to an average of 880 eggs depending on the host plant species (Fisher, 2006).

Figure 3.43 Black vine weevil adult.

Figure 3.44 Damage from weevil feeding on camellia leaf. Damage is similar on *Ilex* foliage.

Twobanded Japanese weevil (*Pseudocneorhinus bifasciatus* Roelofs) is another common weevil pest of hollies. This species is distributed in New England, the Midwest and the mid-Atlantic states (Johnson and Lyon, 1991). Adults create notches on the margin of leaves similar to those created by the black vine weevil. The twobanded Japanese weevil is widely distributed throughout the eastern U.S. and feeds on a wide variety of ornamental plants (Baker, 1994; Boyd and Wheeler, 2005). Adult is about ¼ inch long, light brown, with indistinct white lines on the elytra (Figure 3.46). Adult feeds on new leaves or shoots during the day (but will drop to the ground when disturbed), and the grub feeds on the roots. There is one generation per year, with adult emerging from the soil in April on Long Island, NY (Johnson and Lyon, 1991).

Management

The black vine weevil and the twobanded Japanese weevil could feed and develop on a wide variety of ornamental plants grown in the same nursery. Infested plants should be removed from the production area or treated with chemical or biological products so that they do not become a source of infestation for other plants in the production area. In addition, by laying down a nursery mat or by removing leaf litter or other debris, growers can deny hiding, pupating and egg deposition sites for the weevils.

Figure 3.45 Black vine weevil grub.

In landscape settings where only a few plants are impacted, a sticky material (such as Tanglefoot) can be applied to the trunk to trap adult weevils climbing up the trunk at night. Leaves and branches touching the ground could be removed to eliminate a route for the weevils to climb up the plants. Also, hand removal of adults can also be effective (Masiuk, 2003). However, these cultural and physical methods may be too labor intensive for commercial nurseries.

The presence of weevils in nurseries is determined by monitoring for damage (notches on the leaves). For the black vine weevil, monitoring efforts should focus on the lower branches and leaves because the flightless adults often do not climb too high from their hiding sites in the soil and litter.

Visual monitoring of damage, however, does not provide growers with information on the spring emergence of adults so that timely application of control methods can begin before feeding and reproduction. A recent study showed that a 1:1 mixture of (Z)-2-pentenol and methyl eugenol was effective in attracting the black vine weevils in Oregon (van Tol et al., 2012). However, the effectiveness of such an attractant for monitoring black vine weevils in commercial tree and shrub nurseries remains to be tested. Therefore, the monitoring of foliage feeding weevils is limited to the time- and labor-intensive observations for damage. In Ohio, adult emergence of the black vine weevil was found to begin when black locust and yellowwood are in full bloom and mountain laurel and multiflora rose are in first bloom (Herms, 2004).

Figure 3.46 Twobanded Japanese weevil adult.

Chemical management of black vine weevils should begin after adult emergence in May. For adult control, topical applications of acephate, chlorpyrifos, pyrethroids and azadirachtin should be applied at least twice (2-3 weeks between the applications) after adult emergence (Table 3.4). Systemic neonicotinoid insecticides applied as a soil drench to containerized or field-grown plants are effective in reducing the numbers of larvae as well as feeding damage by the adults (Reding and Persad, 2009; Reding and Ranger, 2011). Bifenthrin can be used as a replacement for the more hazardous chlorpyrifos for root ball dipping (Cowles, 2001).

An additional and effective tool in management of larvae in the soil or potting substrate is a soil drench with entomophathogenic namatodes (Grewal, 2012). Species currently available commercially include *Steinernema carpocapsae, Steinernema feltiae, Steinernema kraussei* and *Heterorhabditis bacteriophora*. There are indications that *Hetetorhabditis* species are more effective in reducing the abundance of black vine weevil larvae due to its higher dispersal within the potting substrate (Ansari and Butt, 2011). Soil application of the entomopathogenic fungi, *Beauveria bassiana*, is also suggested for adult and larvae control (Anonymous, 2004). The soil or substrate should be thoroughly wet before application of nematodes or fungi to increase their efficacy.

Thrips

Chilli thrips, *Scirtothrips dorsalis* Hood (Figure 3.47), is also known as the yellow tea thrips. The chili thrips is widely distributed in the world, inflicting serious damage to vegetables, ornamentals and fruit crops. Widespread damage by the chili thrips began in Florida in 2005. Currently, the chili thrips are found in Florida, Georgia and Texas. A study by Holtz (2006) suggested that this species would be a pest primarily in the southern states and the Pacific states. The chili thrips feed primarily on growing plant tissues by puncturing the cell wall and removing cell content. As a result, the feeding damage on hollies and other susceptible plant species typically presents as distortion and blotches on foliage (Figure 3.48) and shoots on hollies. In serious infestations, the plants and fruits are so severely stunted or disfigured that sale is not possible. No study has investigated the potential resistance in various holly species and cultivars.

Figure 3.47 Chili thrips are widely distributed in the world, inflicting serious damage to vegetables, ornamentals and fruit crops. However, in the U.S. they are predominantly located along the gulf coast (as of 2016).

Adult chili thrips is a small insect (less than 1 mm; smaller than typical flower thrips), with pale yellow or almost white body banded with small blotches on the abdominal segments (Hodges et al.*,* 2009).

Figure 3.48 Seen here is chili thrip damage to camellia. However, damage to hollies and other broadleaf evergreens should not differ greatly and present as distorted growth, with bronzing of midveins and secondary veins on new foliage in the spring and early summer with contorted new growth. Damage eventually becomes darkened as the summer progresses.

Females insert 60 to 200 tiny eggs into the plant tissues over her lifetime. Nymphs feed on the foliage immediately after hatching. Duration of development was 18-21 days (Seal et al*.,* 2010). There may be as many as 14-18 generations per year in southern Florida (Holtz, 2006), where the chili thrips is expected to remain active for most of the year.

Management

Similar to other insect and mite pests that have a large number of potential hosts, the management of chili thrips should begin with sanitation. Remove weeds, debris and old or infested plants within and around the production areas and propagation greenhouses.

Thrips can be monitored by using yellow or blue sticky cards that are installed vertically just above the canopy. Check the cards regularly for thrips that are stuck to the sticky cards. The presence of thrips on the sticky cards does not warrant immediate management action because many thrips species can be carried on the wind. Check the surrounding plants for feeding damage to new leaves and growing buds (necrotic scars and distortion). Alternatively, tap foliage or flowers on a white background (a piece of paper or board) and look for pale yellow or white cigar-shaped thrips. Generally thrips do not require management on hollies; however, if the presence of chili thrips is suspected, samples should be collected and sent to state extension specialists or diagnostic services for identification.

Several biological control agents are available for the management of thrips. Predatory mites (*Neosieuslus cucumeris*, *Amblyseius swirskii* and *Hypoaspis* miles) and bugs (*Orius* species) are commercially available. These biological control agents have been shown to be effective in laboratory and greenhouse tests (Arthurs et al*.,* 2009; Dogramaci et al., 2011) but their efficacy and practical deployment in nurseries and landscapes still requires more research. Entomopathogenic nematodes (*Steinernema feltiae*) and fungi (*Beauveria bassiana*) have also been shown to be effective alternatives to insecticides. When a biological control program is being implemented, it is important to understand the potential negative interactions selected insecticides, miticides and fungicides can have with the biological control agents. This information can usually be found on the suppliers' websites.

Insecticides should be used before the thrips population becomes too large and damage becomes obvious. Because of the continuous dispersal of adults from the surrounding areas, reapplication on at least a biweekly basis may be necessary. Rotate among insecticides of different modes of action or IRAC numbers with each treatment (or each thrips generation of 14-21 days) to prevent or delay the development of insecticide resistance in the thrips population. Chemicals registered for treatment against thrips in the nurseries include acephate, abamectin, acetamiprid, azadirachtin, dinotefuran, flonicamid, novaluron, pyrethroids, spinosad, thiamethoxam, insecticidal soap and horticultural oil (Table 3.4). Most of these chemicals are evaluated against the western flower thrips but efficacy data against chilli thrips are also available (Bethke et al*.,* 2011). It is important to recognize that soil drench or indirect treatment with systemic chemicals (dinotefuran and thiamethoxam) is more effective against thrips feeding on the leaves than those feeding on the flowers.

Spider mites

Southern red mite [*Oligonychus ilicis* (McGregor)] (Figure 3.49) is the most common spider mite species to attack hollies in the nursery and landscape. This species is widely distributed in the eastern U.S., affecting many broadleaved ornamentals including holly, azalea, camellia and boxwood. Adult female is about 1/50 inch long, rounded and reddish brown body, no clear division of body segments and four pairs of legs. This is a "cool-season" mite, meaning that the population grows in the cooler months in the spring and fall while quiescent in the summer months. There are multiple generations per year, with the first peak activity observed in April in New Jersey (Mague and Streu, 1980). These spider mite species feed on the foliage using a piercing mouthpart to remove cell content. This mode of feeding causes a characteristic silvery stippling or chlorosis on the foliage. Young leaves that are severely damaged may be permanently distorted.

Figure 3.49 Southern red mite is the most common spider mite species to attack hollies in the nursery and landscape.

The twospotted spider mite [*Tetranychus urticae* (Koch)] is a common mite pest of hundreds of ornamental and horticultural crops. The small female (1/50 inch) is pale yellowish or translucent with two greenish spots on the sides of the body. The twospotted spider mite is a 'warm-season' mite that becomes active in spring and progressively more damaging in the summer. It feeds in the same way and causes similar damage as the southern red mite.

Figure 3.50 Twospotted spider mite damage can be spotted with a hand lens, but adults require higher magnification (as is the case with most spider mites).

Management

Scout in the spring and fall for the southern red mite and late spring and summer for the twospotted spider mite. Inspect the underside of leaves carefully for damage using a hand lens. If damage is suspected, use a dissecting microsope (40x or greater magnification) for the presence of adults (Figure 3.50), immature adults and eggs. Southern red mites have reddish-orange eggs while twospotted spider mite eggs are milky white. Another monitoring method is to strike the leaves over a piece of white paper, then look for tiny red or yellowish-green specks on the paper with a hand lens. The plant feeding mites usually move much slower than the predatory mites. Plant feeding mites are easily crushed by swiping your hand across the paper, which leaves streaks on the paper. Also, inspect plants that show damage (stippling, bronzing, curling) and flag them for treatment.

Keeping the production area free of weeds and infested plants is the first step in preventing infestation by spider mites. Most mite species do not survive well in a humid environment or they can be easily knocked off the plants with a forceful jet of water. This method may not work well for plantings with many holly shrubs. Growing hollies in a less dusty area can help to reduce the incidence of spider mite infestations.

Most commercial suppliers of biological control agents sell predators of spider mites; the most commonly used is the predatory mite *Phytoseiulus persimilis.* The predatory mites come in bottles (filled with bran) or individual sachets. The bran (together with the mites) can be sprinkled on the plants, and the sachets hang on the plants. The use of the predatory mites in the winter may be limited because the predatory mites perform the best at 65% relative humidity and over 20°C (68°F). In nurseries, *Neosiulus fallacis* had been shown to be more effective.

Many miticides are available for control of mites in the nursery: abamectin, acequinocyl, bifenazate, clofentezine, etoxazole, fenazaquin, fenbutatin-oxide, fenpyroximate, hexythiazox, pyridaben and spiromesifen (Table 3.4). Horticultural oil, insecticidal soaps and neem oil can also be used with limited efficacy. Chlorpyrifos, carbaryl and pyrethroids also have low activity against mites and their uses are typically discouraged because of potential negative impacts on the existing natural enemies. Regardless of the chemical used, the applications have to be repeated every 7-14 days and complete coverage of the canopy is needed for successful management. For eriophyid mite control, surfactant or penetrant may be added to the solution to increase penetration of the solution into hard to reach crevices or between bud scales. It is also important to rotate among different modes of action to prevent or delay the development of miticide resistance.

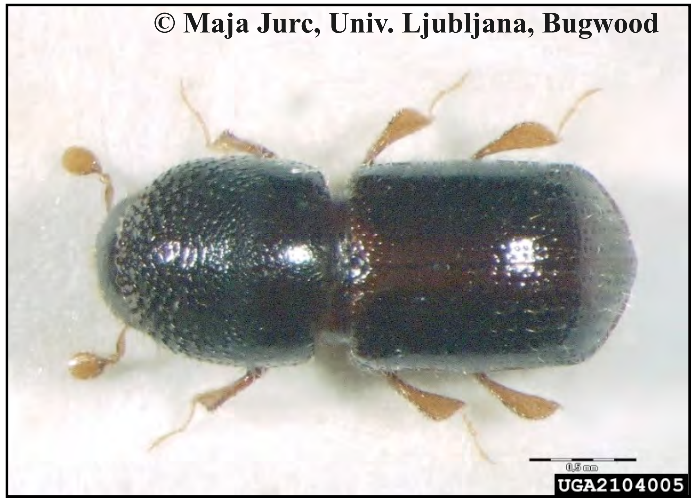


Figure 3.51 The black stem borer has been reported as an occasional pest of hollies in the southeastern U.S.

Ambrosia beetles

Ambrosia beetles (Coleoptera: Curculionidae) are major pests of ornamental trees and shrubs in nurseries and landscapes. Hollies are typically not preferred by the ambrosia beetles, unless they become stressed by other environmental factors, such as drought and flood. The black stem borer, *Xylosandrus germanus* (Blandford) (Figure 3.51) has been reported as an occasional pest of hollies. The black stem borer is a native of Asia; it was introduced to the U.S. in the 1930s. The species is currently distributed in the eastern and midwestern U.S., and Oregon (Rabaglia et al*.,* 2006). The black stem borer appears to be more aggressive in its northern range, whereas the granulate ambrosia beetle, *Xylosandrus crassiuslus* (Motschulsky) (Figure 3.52) is the predominant borer of nursery grown trees in the southeastern U.S. Adult female black stem borers become active as early as early March in South Carolina (Chong, 2015), mid-March in Virginia and Tennessee, and late-April in Ohio (Reding et al*.,* 2010). Adult females bore into the branches and trunk of susceptible plants. As the beetles are excavating the gallery, they push out the wash dust, which form a ‘frass tube’ (Figure 3.53). The frass tube can be used as a monitoring and diagnostic characteristic, but it can be fragile and often lost on windy or rainy days. The ambrosia beetles introduce the ambrosia fungi, on which the larvae of the beetles feed. Males are small, flightless, and remain in the gallery. A new generation of females emerges from the gallery about 55 days after the initial attack (Oliver and Mannion, 2001).

Figure 3.52 Granulate ambrosia beetles are major pests of ornamental trees and shrubs in nurseries and landscapes, including most *Ilex* spp. and cultivars.

Management

Flight activity of ambrosia beetles can be monitored with ethanol-baited traps beginning in late-February in the southeastern U.S. A homemade soda bottle trap, Lindgren funnel trap, or modified Japanese beetle trap are effective (Oliver et al., 2004). Ethanol bait can be prepared in a container and released from a wick, or by purchases of slow-release ethanol baits. Traps should be hung 0.5 to 1.7 m above ground (Reding et al., 2010) and checked regularly.

Figure 3.53 Granulate ambrosia beetles, like many ambrosia beetles, are only seen once the damage has been done. These ambrosia beetles tend to attack smaller hardwood trees and can be devastating to nursery stock.

Attacked trees, which are supposedly stressed and more attractive to the ambrosia beetles, could be left in the nursery for 3-4 weeks after the initial attacks to allow more infestations before removing and discarding in order to spare other healthy trees in the nursery from attacks. The attacked trees are best burnt. No effective biological control option is currently available.

Chemical management of ambrosia beetles remains preventive in nature. Pyrethroids, particularly bifenthrin and permethrin, are effective in repelling ambrosia beetle attacks for up to 10 days (depending on ambient temperature) (Mizell and Riddle, 2004) (Table 3.4). The insecticide solution should be uniformly sprayed to cover all trunk and branch surfaces. Reapplications are necessary until the trees fully leaf out. Because of the high cost of chemical management, growers are often advised to protect high value, susceptible trees instead of spraying all plant species in the nursery. Systemic neonicotinoids are not effective in preventing attacks by ambrosia beetles or killing ambrosia beetles that are already in the gallery (Chong, 2015).

Clearwing borers

Holly clearwing borer, *Synanthedon kathyae* Duckworth and Eichlin (Lepidoptera: Sesiidae) (Figure 3.54), has been reported as a pest of nursery-grown hollies in New Jersey (Ghidiu et al., 1987). This is a native clearwing borer species that is distributed in Maryland, Massachusetts, New Jersey, New York, North Carolina, Nova Scotia, South Carolina and Virginia (Solomon, 1995). This species attacks American holly, as well as hybrids. Adult moth is small, with a wingspan of 18 to 25 mm; head blue black with yellow fringe; thorax blue black with yellow stripes along the sides; abdomen blue black with two yellow stripes. This species appears to mimic small wasps. Larvae inside the tunnel are white, with brown heads, and about 15 to 21 mm long when mature. There is one generation per year. Adult moths are active from late May until late July. Adults lay eggs on bark, and the larvae bore into the wood after hatching. The borer appears to prefer stems 8 to 40 mm in diameter. The gallery created by the larvae is usually 3-7 cm above the soil line. When the adults emerge, they often leave pupal skins protruding from the exit holes. The tunneling activity of the larvae destroys the vascular tissues of plants, causing gradual wilting, chlorotic foliage, premature leaf senescence, and dieback. Often, oozy brown frass can be found around the entrance hole.

Figure 3.54 The holly clearwing borer has been reported as a pest of nursery-grown hollies in New Jersey, but is also found in mid-Atlantic states including Maryland, New Jersey, North Carolina and Virginia. This species attacks American holly, as well as *Ilex opaca* hybrids.

Management

Pheromone lure ("general" clearwing lure) and the sticky wing or delta traps are commonly used as monitoring tools for the initiation of flight activity of clearwing borers. These lures may also be used for monitoring the flight of the holly clearwing borer (Neal and Eichlin, 1983). However, the "general" clearwing lure also attracts other similar non-target clearwing moth species, such as the greater and lesser peachtree borers (*Synanthedon exitiosa* and *Synanthedon pictipes*, respectively), and confirmation of the targeted clearwing moth species may be needed (Braxton and Raupp, 1995). Check all susceptible trees at the base of the trunk and on branches to detect light brown frass messes that are indicative of larvae activity.

Attacks by clearwing borers do not kill the infested trees immediately. Removal of the larvae (by physical or chemical means) followed by appropriate fertilization and irrigation practices could help the infested trees maintain vigor and even survive and recover from the attacks.

Management approaches to the holly clearwing borer are poorly known. Preventive and curative applications of the nematode *Steinernema carpocapsae* (Weiser) and *Heterorhabditis bacteriophora* are effective in suppressing greater peachtree borer activity in peach orchards (Cottrell and Shapiro-Ilan, 2006; Shapiro-Ilan et al., 2009). It is not clear if entomopathogenic nematodes have similar efficacy against the holly clearwing borer.

Preventive bark sprays of pyrethroid, carbaryl or chlorpyrifos in the spring should coincide with the initiation of flight activity as indicated by captures in pheromone traps (Table 3.4). Sprays should achieve complete coverage of the bark on trunk and branches; foliage does not need to be sprayed. Reapplications may be needed every 7-14 days. Soil or trunk applications of systemic neonicotinoids do not provide reliable clearwing borer control.

Table 3.4 [1] Insecticides available to control common insect pests of *Ilex* spp. in commercial production environments.

IRAC Code [3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Armored scales	Soft scales	Thrips	Weevils	Clear-wing borers	Ambrosia beetles	Spider mites	Leafminers	Aphids	Twolined spittlebug
1A	Acetylcholinesterase inhibitors	Carbamates	carbaryl[10]	Sevin SL	L, N, G	12	x	x	x	x				x	x	x
			methiocarb	Mesurol 75W	N, G	24			x				x		x	
1B [2]	Acetylcholinesterase inhibitors	Organophosphates	acephate[4, 10]	Orthene T&O	L, N, G	24	x	x	x	x				x	x	x
				Lepitect	L, N, G	24	x	x	x	x			x	x	x	
				Precise GN	G, N	12				x					x	
			chlorpyrifos	Dursban 50W	N	24	x	x	x	x	x	x	x	x	x	x
				DuraGuard ME	N, G	24	x	x	x	x			x		x	
			dicrotophos	Inject-A-Cide B[11]	L	N/A	x	x			x		x		x	
			dimethoate	Dimethoate 4E, 4EC	N	10-14 days	x	x	x				x	x	x	
			malathion[10]	Malathion 5EC	L	12	x	x	x				x	x	x	
			methidathion	Supracide 2E	N	30 days	x	x								
			oxydemeton methyl	Harpoon[11]	L	0	x	x			x		x		x	
				MSR Spray Concentrate	N	10 days			x	x			x	x	x	
3A	Sodium channel modulators	Pyrethroids / Pyrethrins	bifenthrin[10]	Attain TR	G	12	x	x	x				x		x	x
				Menace GC	L, N, G	12	x	x	x	x		x	x		x	x
				Onyx	L	N/A	x	x	x		x	x	x	x	x	x
				OnyxPro	L, N, I	12	x	x	x		x	x	x	x	x	x
				Talstar S Select	N, G	12	x	x		x			x		x	x
				Talstar Nursery G	N	12				x						x
			cyfluthrin	Decathlon	L, N, G, I	12	x	x	x	x					x	x
			beta-cyfluthrin	Tempo Ultra WP	L, I	N/A	x	x	x	x					x	x
				Tempo SC Ultra	L, I	N/A	x	x	x	x					x	x
			lambda-cyhalothrin	Demand	L	N/A	x	x	x	x			x	x	x	
				Scimitar CS; Scimitar GC	L ; L, N, G	24; 24	x	x	x	x			x	x	x	x

Table 3.4 (***continued***) Insecticides available to control common insect pests of *Ilex* spp. in commercial production environments.

IRAC Code [3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Armored scales	Soft scales	Thrips	Weevils	Clear-wing borers	Ambrosia beetles	Spider mites	Leafminers	Aphids	Twolined spittlebug
3A	Sodium channel modulators	Pyrethroids / Pyrethrins	cypermethrin	Demon WP	L, I	N/A									x	x
			deltamethrin	DeltaGard G	L, I	N/A				x						x
			fenpropathrin	Tame 2.4 EC	N, G, I	24	x		x	x			x	x	x	x
			tau-fluvalinate	Mavrik Aquaflow	L, N, G, I	12			x	x			x		x	x
			permethrin	Astro	L, G, I	12			x	x	x			x	x	x
				Permethrin Pro	L, I	N/A			x	x	x			x	x	x
				Perm-Up 3.2 EC	L, N, G, I	12			x	x	x			x	x	x
			pyrethrins	Tersus	N, G	12	x	x	x	x	x	x	x	x	x	x
				Pyganic	N, G	12	x	x	x				x		x	x
			pyrethrum	Pyrethrum TR	N, G	12	x	x	x				x		x	x
3A + 4A [2]	Sodium channel modulators	Pyrethroids + Neonico-tinoids	bifenthrin + clothianidin	Aloft LC G, LC SC	L	N/A	x	x	x	x				x	x	x
			bifenthrin + imidacloprid	Allectus SC	L, I	N/A	x	x	x	x			x	x	x	x
			cyfluthrin + imidacloprid	Discus N/G	N, G, I	12	x	x	x	x				x	x	x
			lambda-cyhalothrin + thiamethoxam	Tandem	L	N/A	x	x	x	x				x	x	x
			zeta-cypermrthrin + bifenthrin + imidacloprid	Triple Crown T&O	L, I	N/A	x	x	x	x		x	x		x	x
3A + 27A	Sodium channel modulators	Pyrethroids + Piperonyl butoxide (PBO)	pyrethrins + piperonyl butoxide[8]	Pyreth-It	N, G	12			x						x	x
4A [2]	Nicotinic acetylcholine receptor agonists	Neonicotinoids	acetamiprid	TriStar 8.5 SL	L, N, G	12	x	x	x	x				x	x	
			clothianidin	Arena 0.25 G	L, I	12									x	
				Arena 50 WDG	L, I	12									x	

Table 3.4 (***continued***) Insecticides available to control common insect pests of *Ilex* spp. in commercial production environments.

IRAC Code [3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Armored scales	Soft scales	Thrips	Weevils	Clear-wing borers	Ambrosia beetles	Spider mites	Leafminers	Aphids	Twolined spittlebug
4A [2]	Nicotinic acetylcholine receptor agonists	Neonicotinoids	dinotefuran	Safari 2G	L, N, G, I	12	x	x	x	x				x		
				Safari 20 SG	L, N, G, I	12	x	x	x	x					x	
				Zylam Liquid	L	N/A	x	x	x	x				x	x	
				Transtect 70 WSP	L	N/A	x	x	x	x	x			x	x	
			imidacloprid[10]	Xytect 75WSP; 2F	L, N, G, I	12	x	x	x	x					x	x
				Marathon II	N, G, I	12	x	x	x	x				x	x	x
				Marathon 60WP	N, G, I	12	x	x	x	x				x	x	x
			thiamethoxam	Merit	L, I	N/A	x	x	x	x				x	x	
				CoreTect	L, I	N/A	x	x	x	x				x	x	
				Discus Tablets	N, G, I	12	x	x	x	x				x	x	
				Flagship 25WG	N, G, I	12		x	x	x				x	x	
				Meridian 0.33G	L, I	N/A		x	x	x				x	x	
			thiamethoxam	Meridian 25WG	L, I	N/A		x	x	x				x	x	
4C + 5	Nicotinic acetylcholine receptor agonists	Sulfoxaflor + Spinosyns	sulfoxaflor + spinetoram	XXpire	L, N, G	12	x		x				x		x	
5	Nicotinic acetylcholine receptor allosteric activators	Spinosyns	spinosad	Conserve SC	L, N, G	4			x				x			
				Entrust	L, N, G	4			x				x			

Table 3.4 (*continued*) Insecticides available to control common insect pests of *Ilex* spp. in commercial production environments.

IRAC Code [3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Armored scales	Soft scales	Thrips	Weevils	Clear-wing borers	Ambrosia beetles	Spider mites	Leafminers	Aphids	Twolined spittlebug
6	Chloride channel activators	Avermectins, Milbemycins	abamectin	Lucid, , Minx, Avid	L, N, G	12			x				x		x	
				Aracinate TM[11]	L, N, G, I	N/A	x	x	x	x	x		x	x	x	
			emamectin benzoate	Arbormectin[11]	L	N/A	x					x	x	x	x	
				Tree-age [11]	L	N/A	x				x			x		
				Enfold	N	12							x	x		
			milbemectin	Ultiflora	N	12							x			
6 + 20D	Chloride channel activators	Avermectins, Milbemycins	abamectin + bifenazate	Sirocco	L, N, G, I	12			x				x		x	
7A	Juvenile hormone mimics	Juvenile hormone analogues	s-kinoprene	Enstar AQ	G, I	4	x	x	x					x	x	
7B	Juvenile hormone mimics	Fenoxycarb	fenoxycarb	Preclude TR	G	12	x	x		x			x		x	
7C	Juvenile hormone mimics	Pyriproxyfen	pyriproxifen	Distance IGR	L, N, G, I	12			x							
				Fulcrum	L, N, G, I	12	x	x							x	
8C	Misc. non-specific (multi-site) inhibitors	Fluorides	cryolite (sodium alumino-fluoride)	Kryocide	L	N/A				x						
8D	Misc. non-specific (multi-site) inhibitors	Borates	sodium tetrabor-ohydrate decahydrate	Prev-AM Ultra	N, G	12	x	x					x		x	
9B [2]	Selective homopteran feeding blockers	Pymetrozine	pymetrozine	Endeavor	L, N, G, I	12									x	
		Pyrifluquinazon	pyriflquinazon	Rycar	G	12									x	
10A	Mite growth inhibitors	Clofentezine	clofentazine	Ovation SC	N, G	12							x			
		Hexythiazox	hexythiazox	Hexygon DF	L, N, G, I	12							x			
10B	Mite growth inhibitors	Etoxazole	etoxazole	Beethoven TR	G	24							x			
				TetraSan 5 WDG	L, N, G, I	12							x			

Table 3.4 (*continued*) Insecticides available to control common insect pests of *Ilex* spp. in commercial production environments.

IRAC Code [3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Armored scales	Soft scales	Thrips	Weevils	Clear-wing borers	Ambrosia beetles	Spider mites	Leafminers	Aphids	Twolined spittlebug
12B	Inhibitors of mitochondrial ATP synthase	Organotin miticides	fenbutatin-oxide	Meraz, ProMite 50WP	L, N, G	48							x			
13	Uncouplers of oxidative phospho-rylation via disruption of the proton gradient	Chlorfenapyr	chlorfenapyr	Pylon	G	12			x				x	x		
15	Inhibitors of chitin biosynthesis, type 0	Benzoylureas	diflubenzuron	Adept	G	12								x		
				Dimilin 25W	L, N	12				x				x		
				Dimilin 4L	L, N	12				x				x		
			novaluron	Pedestal	N, G	12			x					x		
16	Inhibitors of chitin biosynthesis, type 1	Buprofezin	buprofezin	Talus 70DF	L, N, G	12	x	x								
20B	Mitochondrial complex III electron transport inhibitors	Acequinocyl	acequinocyl	Shuttle 15 SC	L, I	12							x			
				Shuttle-O	N, G	12							x			
20D	Mitochondrial complex III electron transport inhibitors	Bifenazate[18]	bifenazate	Floramite SC	L, N, G, I	12							x			
				Floramite SC/ LS	L	N/A							x			
21A	Mitochondrial complex I electron transport inhibitors	METI acaricides and insecticides	fenazaquin	Magus	L, N, G, I	12							x			
			fenpyroximate	Akari 5SC	N, G, I	12							x			
			pyridaben	Sanmite	N, G	12							x			
			tolfenpyrad	Hachi-Hachi SC	G	12	x	x	x						x	
23 [2]	Inhibitors of acetyl CoA carboxylase	Tetronic and Tetramic acid derivatives	spiromesifen	Forbid 4F	L	N/A							x			
			spirotetramat	Kontos	N, G, I	24	x	x	x				x		x	x
25	Mitochondrial complex II electron transport inhibitors	Betaketonitrile derivatives	cyflumetofen	Sultan	L, N, G, I	12							x			
28	Ryanodine receptor modulators	Diamides	chlorantraniliprole	Acelepryn	L, I	N/A		x			x				x	
				Acelepryn G	L, I	N/A									x	
			cyantraniliprole	Mainspring	G, I, L	4		x	x						x	

Table 3.4 (***continued***) Insecticides available to control common insect pests of *Ilex* spp. in commercial production environments.

IRAC Code [3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Armored scales	Soft scales	Thrips	Weevils	Clear-wing borers	Ambrosia beetles	Spider mites	Leafminers	Aphids	Twolined spittlebug
29	Chordotonal organ Modulators - undefined target site	Flonicamid	flonicamid	Aria	L, N, G	12	x	x	x						x	
Unknown	Unknown	Azadirachtin	azadirachtin[10]	Azatin O	L, N, G, I	4	x	x	x	x	x			x	x	
				Azatin XL	N, G, I	4			x	x				x	x	
				Azatrol EC	L, N, G, I	4	x	x	x	x			x	x	x	
				Ornazin 3% EC	L, N, G, I	12	x	x	x	x	x			x	x	x
				TreeAzin	L, N, G	until dry								x		
		Pyridalyl	pyridalyl	Overture 35WP	G	12			x							
Not classified	Various		*Beauveria bassiana*	BotaniGard ES	L, N, G, I	4			x	x			x		x	
				BotaniGard 22 WP	L, N, G, I	4			x	x					x	
				Naturalis-L26	L, N, G	4			x	x			x		x	
			Chromobacterium subtsugae	Grandevo PTO	L, N, G	4			x				x		x	
			Heterorhabtditis bacteriophera	LarvaNem	L	N/A				x						
				ExhibitlineH	N	N/A				x						
				NemaShield	L, N, G, I	N/A				x						
			Isaria formosorosea (=*Paecilomyces fumosoroseus*)	NoFly, Preferal	L, N, G	12			x	x			x	x	x	
			Metarhizium anisopliae	Met52, Tick-EX	L, N, G	4			x	x						
			Steinernema carpocapsae	Millenium, EntoNem	L, N, G, I	N/A			x	x	x					

Table 3.4 (***continued***) Insecticides available to control common insect pests of *Ilex* spp. in commercial production environments.

IRAC Code [3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names [4, 5, 6, 7]	Use Site [8]	REI (hrs) [9]	Armored scales	Soft scales	Thrips	Weevils	Clear-wing borers	Ambrosia beetles	Spider mites	Leafminers	Aphids	Twolined spittlebug
Not classified	Various		*Steinernema feltiae*	NemaSys, Steinernema-System	L, N, G, I	N/A			x							
				NemaShield	L, N, G, I	N/A			x							
				EntoNem	L, N, G, I	N/A			x	x						
			Steinernema kraussei	Nemasys L	L, N, G, I	N/A				x						
				Kraussei-System	N	N/A				x						
			horticultural oil[10]	Ultra-Pure Oil, TriTek	L, N, G, I	4	x	x	x				x	x	x	
			insecticidal soap[10]	M-Pede	L, N, G, I	12	x	x	x				x		x	
			neem oil[10]	Trilogy	L, N	4		x	x				x		x	
				Triact 70	L, N, G, I	4	x	x					x		x	
			capsicum extract, garlic oil, soybean oil	Captivia	L, N	4			x				x			

Table 3.4 (***continued***) Insecticides available to control common insect pests of *Ilex* spp. in commercial production environments.

Footnotes and Supplemental Information:

1. Pests listed on pesticide labels are subject to change, as are re-entry intervals, trade names, and formulations. Within states and counties, products may have additional permitted uses by 2(ee) and Special Local Need allowances. Consult your County Extension Agent or State Agency to determine if other uses are allowable. The label should always be consulted to confirm that chart-listed pests still appear on the label. Check the product labels for specific site restriction information, notes on application, sensitive plant species and specific target pest species.
2. Applications of products from these footnoted IRAC groups are optimized when not preceded by/or followed by products in subsequently noted IRAC Group(s), particularly when managing the indicated pest. Suggested rotation combinations to avoid include serial treatments with a.i. products from: IRAC 1B, 4A, 9B, 23 (aphids).
3. IRAC Code designations and Related Modes of Action are explained at the Insecticide Resistance Action Committee Database 2017 (IRAC 2017; http://www.irac-online.org/modes-of-action/).
4. See product label for information about potential crop phytotoxicity, known plant sensitivity, and how to test for phytotoxicity.
5. Check label for additional restrictions (e.g., on the number of times the product can be applied in a growing season or year, or additional application restrictions within permitted use sites; and crop types or flowering status that may not be legally treated.)
6. Trade names of products are provided as examples only. No endorsement of mentioned product, nor criticism of unmentioned products, is intended.
7. Products may not be registered or renewed for use in all southeastern U.S. states. Consult your state's Department of Agriculture to confirm legal use of products in your state.
8. Use site information is provided for reference only: L = landscape; N = nursery; G = greenhouse; I = interiorscape. t/s*Products listed for use in L, N, G or I sites may include active ingredients in differently labeled products that are designated for use in turfgrass or sod production use sites. Mention of those products is beyond the scope of this resource guide, and products listed herein may not be legal or appropriate for site uses in turfgrass or sod production
9. Re-Entry Interval (REI) designations apply to agricultural (nursery) uses. Within REI column, 'N/A' is used to indicate products with Landscape and/or Interiorscape site uses and that present Non-Agricultural Use Requirements. Consult product labels for specific details required for compliance within those application conditions and use sites, including those for which REI listings do not apply. Additionally, some product labels may list site uses that are beyond the scope of this publication (e.g., sod farms, silvicultural & Christmas tree nurseries, pastures, rights-of-way, etc.). These site uses may involve Agricultural Use Requirements that list an REI not presented here.
10. Multiple formulations and trade names of the same active ingredients are available. Labels among these products often differ in legally allowable uses (regarding sites, pest, REI, etc.); representative example(s) presented.
11. Product formulated for tree or shrub injection; specialized equipment may be required.

Disease Management

COMMON DISEASE PESTS

1. Black root rot
2. Phytopthora root rot
3. Rhizoctonia web blight
4. Botryosphaeria canker
5. Powdery mildew
6. Anthracnose

Black Root Rot

Black root rot is caused by the ascomycete fungus *Thielaviopsis basicola* (Berk & Br.) Ferraris. This pathogen has a very wide host range including more than 130 genera of plants, mainly herbaceous, with *Ilex* species being among the few woody plants affected. Particularly, Japanese holly (*I. crenata*), inkberry (*I. glabra*), and blue holly (*I. ×meservae*) are the major species affected in both nurseries and landscapes (Sinclair and Lyon, 2005d).

Figure 3.55 Severe black root rot symptoms on root ball of potted holly.

Thielaviopsis basicola affects the plant vigor by colonizing and killing the root system (Figure 3.55). Initially, small dark lesions can be observed on the root tips of affected plants. Over time, these lesions may coalesce and form extensive blackened areas on the feeder roots (Figure 3.56). This dark-brown to black appearance is the result of the formation of dark-colored spores (chlamydospores) on and in the root lesions. Depending on the extent of the root damage and the age of the plant, aboveground symptoms may range from stunting of terminal growth to interveinal foliar chlorosis to plant defoliation and dieback (Figure 3.57).

Thielaviopsis basicola is a soilborne pathogen. Chlamydospores represent the survival structures of the pathogen in the soil where it can persist for many years in absence of a susceptible host. Environmental conditions that favor disease development include cool soil temperature 17.2 – 22.8°C (63-73°F), high soil moisture and alkaline pH (6-7).

Figure 3.56 Extensive blackened areas on black root rot infected roots.

Figure 3.57 Defoliation and dieback symptoms on holly plants with extensive black root rot damage.

Management
Infected nursery plants are usually the major source of pathogen dissemination (Pscheidt, et al., 2001). Because this is a soilborne pathogen, using soilless mix for propagation will help limit the chances of introducing the pathogen into the nursery. Liners purchased from outside nurseries should be inspected for symptoms before introducing them into the production (Pscheidt, et al., 2001). Regardless of their origin, symptomatic plants should be discarded together with any plant debris and contaminated soil. Containers should not be recycled or should be properly disinfected before re-use. Keeping the pH below 6 will help control symptom development. Fungicides that have proved to provide good control of this disease include drench applications of thiophanate methyl, triflumizole, and polyoxin D (Williams-Woodward et al., 2014; Table 3.5).

Phytopthora Root Rot
The water mold *Phytopthora cinnamomi* Rands is a well-known root rot pathogen of numerous ornamental plants in nursery production, including *Ilex* species. As other Phytopthora species, *P. cinnamomi* thrives in conditions of poorly drained soils and can become problematic in container production. Because they are very intolerant of poorly drained soils, Japanese hollies are especially susceptible to this disease (Hagan, 2005).

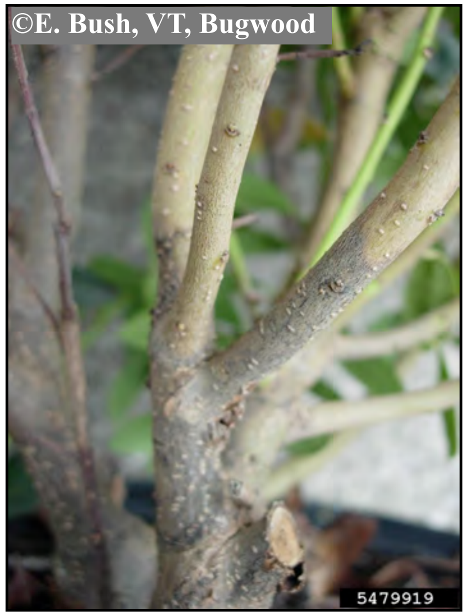


Figure 3.58 Blackening of stem near base of potted *Ilex glabra* plant caused by Phytopthora root rot.

Infection normally starts on the small feeder roots and subsequently extends to the main roots. The roots of infected plants are darkly discolored and mushy. Over time, the dark brown discoloration may extend upward into the crown and the lower stem (Figure 3.58). Because the functionality of the root system is compromised, aboveground symptoms may be confused with those induced by other root rot causing organisms (e.g. *Thielaviopsis basicola*). An overall lack of plant vigor, yellowing of the foliage, early leaf drop, wilting, and eventually stem dieback (Figure 3.59) and plant death (Figure 3.60) can be observed on infected plants (Hagan, 2005).

Figure 3.59 Symptoms of Phytopthora dieback on a potted *Ilex glabra* plant.

Phytophthora cinnamomi produces two types of spores. The first ones, called zoospores, have motile appendages, called flagella that allow them to swim. Wet soils and saturated substrates provide a medium for the pathogen to move into the root zones, thus infections are favored by prolonged periods of saturated soil. Because water is the means by which zoospores move, they can be washed out of containers and into recirculated sources of irrigation water or splashed on tools, equipment, containers, shoes, and healthy plant material. The second types of spores, called chlamydospores, are thick-walled spores that represent the overwintering stage of the pathogen. These spores can survive in native soil, contaminated rooting substrate, and nursery beds for years in a variety of climate extremes (Agrios, 1997).

Figure 3.60 Holly plant killed by Phytopthora root rot in a landscape bed.

Root infection by zoospores most likely happens in spring and fall, when environmental conditions are favorable. However, aboveground symptoms are normally not seen until the summer when warmer temperatures increase plant water demand. Holly grown under a combination of heat and/or moisture stress are much more sensitive to the disease than are well-maintained, vigorous plants (Hagan, 2005).

Management
Management of Phytopthora root rot requires a combination of good sanitation practices, water management and preventative fungicide applications. In commercial nurseries, root rot outbreaks can be often traced back to the use of contaminated potting media or diseased liners (Hagan, 2005). Thus, it is important to take cuttings from healthy plants and root them in well-drained rooting substrates such as aged bark in clean containers on raised benches rather than in ground beds (Hagan, 2005). Avoid over-irrigating plants to reduce saturated rooting conditions. Potting media or containers should not be re-used or at least should be thoroughly washed and disinfested. Diseased cuttings and container plants should be timely discarded. Fungicides should be applied from the time cuttings are rooted until finished container-grown plants are shipped. Effective fungicides to be used as preventative drench applications include etridiazole, cyazofamid, fluopicolide, fosetyl-Al, mono- and di- potassium salts of phosphorous acid, and potassium phosphite (Williams-Woodward et al., 2014; Table 3.5). Between 6% to 66% of the Phytophthora isolates recovered from nurseries and greenhouses in the southeastern U.S. have been reported to have developed resistance to mefenoxam, which is the most commonly used fungicide for

Phytopthora control (Hwang and Benson, 2005; Olson and Benson, 2011; Olson et al., 2013).

Figure 3.61 Death of lower leaves of meserve holly (*Ilex ×meserveae*) affected by Rhizoctonia web blight.

Figure 3.62 White to tan mycelium on branches of meserve holly (*Ilex ×meserveae*) affected by Rhizoctonia web blight.

Rhizoctonia Web Blight

Rhizoctonia solani Kühn is the basidiomycete fungus responsible for the disease known as web blight. The disease is common on rooted liners and container-grown cultivars of Japanese and Yaupon holly, and in general all those cultivars with dense canopies (Pscheidt, et al., 2001).

Initial symptoms of the disease include small, tan to black spots on the leaves that may enlarge to cover the entire surface and eventually cause the leaves to drop (Figure 3.61). Leaf spots may go unnoticed and plant defoliation may be the first symptom observed (Pscheidt et al., 2001). In conditions of warm temperatures and high humidity, the fungal mycelium will grow from the infected leaves fallen on the container surface developing a web (Figure 3.62). Sometimes mycelium strings can be observed holding the abscised leaves to the plant stem (Pscheidt, et al., 2001).

Management

Maintaining good air circulation around and in the plant canopy is the most important practice to manage this disease (Pscheidt, et al., 2001). Fungicides can be used after proper cultural practices have been undertaken and include drench applications of thiophanate methyl, propiconazole, tebuconazole, azoxystrobin, pyraclostrobin, or fludioxonil among others (Williams-Woodward et al*.,* 2014; Table 3.5).

Botryosphaeria Canker

Different species of the fungal genus *Botryosphaeria* can cause canker and dieback on holly, most commonly *B. obtusa* (Schwein.) Shoemaker and *B. rhodina* (Berk. & M.A. Curtis) Arx (Sinclair and Lyon, 2005c). Nearly all cultivated species of holly including Foster, dahoon, Japanese, Chinese, inkberry, American, and yaupon holly (Hagan, 2005).

Figure 3.63 Limb dieback caused by Botryosphaeria canker.

Bot canker is a stress-related dieback disease. In fact, Botryosphaeria fungi are non-specialized opportunistic pathogens that attack plants that have been exposed to environmental stresses and wounds. Holly plants in good physical condition are rarely damaged by this disease (Hagan, 2005).

Cankers form at wounds, around the base of dead twigs or on limbs that have been exposed to freezing injuries or sunburn and enlarge during the growing season, most rapidly when the plant is subjected to periods of water stress (Sinclair and Lyon, 2005c). The phloem and xylem tissues below the canker, as well as several inches above and below the canker margin, turn brown (Hagan, 2005). Cankers may continue to expand until the diseased limb is girdled, killing all parts of the plant above the canker (Hagan, 2005) (Figure 3.63). Cankers that kill limbs during the growing season can cause wilting, yellowing and premature leaf drop (Sinclair and Lyon, 2005c).

Management

Because Botryosphaeria canker is a stress-related disease, management should focus on proper plant establishment and maintenance practices rather than the use of fungicides.

New plantings should be installed in the fall. Prior to planting, a soil test should be conducted to identify the need for mineral or pH amendments (Hagan, 2005). On poorly drained soils or sites prone to flood, plants should be planted on raised beds and not too deep in the soil. Mulching should be done to help hold moisture in the soil and avoid mechanical injury to the trunk by mowers and weed trimmers (Hagan, 2005). Watering is recommended during periods of extended hot, dry weather. Cankered branches or twigs should be pruned back several inches below the discolored tissues.

Powdery Mildew

Powdery mildew of holly is caused by the fungal pathogen *Erysiphe nemopanthi* (Peck) U. Braun & S. Takam. White patches of powdery growth can be observed on the upper leaf surface of infected plants during summer and fall, which may eventually coalesce to cover the entire leaf surface. Young succulent tissues are more susceptible to infection than mature leaves (Sinclair and Lyon, 2005a). Even though leaf tissues are not killed by the disease, infection can interfere with the normal photosynthetic activity of the plant and thus reduce plant growth.

Figure 3.64 Powdery mildew dulls foliage and causes a white cast to the upper leaf surface.

The white powdery growth (Figure 3.64) represents the fungal mycelium bearing thousands of airborne spores. These spores perpetuate infections throughout the season. In late summer, scattered black fruiting structures, called cleistothecia, form on the underside of the leaves. These serve as the overwintering stage of the fungus and will initiate new infections in the following spring (Sinclair and Lyon, 2005a).

Powdery mildew fungi are obligate parasites, which means that they require living tissues to grow and reproduce. These pathogens are also host specific, so for example, infected holly plants do not represent a threat to nearby roses or viburnum, and vice versa. Infections are favored by mild temperatures, between 70 and 81°F (21 and 27°C), and high relative humidity, between 90 to 100% so disease may be prevalent in situations of poor air circulation. Too high of a temperature (above 90°F; 33°C), as well as prolonged periods of leaf wetness, can inhibit disease development.

Management

Cultural practices for the control of powdery mildew include improving air movement through pruning and plant spacing, and reducing excessive nitrogen and irrigation, which promote succulent growth (Sinclair and Lyon, 2005a). Infected plant tissues should be removed and destroyed to reduce inoculum load. Several fungicides are available and should be applied at the first sign of disease. These include azoxystrobin, fenarimol, propiconazole, thiophanate-methyl, or triadimefon among others (Williams-Woodward et al., 2014; Table 3.5).

Anthracnose

The fungal pathogens *Colletotrichum acutatum* and *C. gleosporoides* are responsible for the disease known as anthracnose on holly and on many other annual and perennial plants. The disease is most common on English, Chinese, American, inkberry, and winterberry holly (Hagan, 2005).

Figure 3.65 Irregular tan to brown spots on anthracnose infected leaves of *Ilex decidua*.

Symptoms of anthracnose are various and include leaf spots, leaf blights, cankers and diebacks (Sinclair and Lyon, 2005b). Circular to irregular tan to brown blotches can be observed on infected leaves (Figure 3.65). Pink-orange spore masses appear during humid weather inside the blotches, and can be easily seen with a hand lens (Hagan, 2005). A shoot dieback may also be seen on anthracnose-damaged ‘Burford’ holly (Hagan, 2005).

Colletotrichum pathogens can overwinter on and in leaves and stems, including plant debris fallen on the ground. In spring, infections are initiated by different types of spores (conidia or ascospores) released from the overwintering structures (acervuli) and dispersed by either rain or air currents (Sinclair and Lyon, 2005b). Once they land on the plant surface, these spores penetrate the host tissues directly or through wounds and natural openings creating lesions, which in conditions of warm and wet weather may enlarge quickly (Sinclair and Lyon, 2005b).

Management

Avoid overhead irrigation and splashing water to limit infection and disease spread. Fungicides should be applied preventatively starting in late spring and repeated at 7-14 days interval as needed (Hagan, 2005). Effective active ingredients include mancozeb, chlorothalonil, or thiophanate methyl (Table 3.5).

Table 3.5 Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Ilex*[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Black root rot	Powdery mildew	Rhizoctonia	Botryosphaeria	Phytophthora root rot, water molds[X]	Anthracnose
1: MBC Benzimidazoles: Upwardly systemic. Broad spectrum fungicide for various fungi. Fungicide resistance risk high. Tank mix with fungicides from a different fungicide group (FRAC) to prevent or delay resistance development. Do not mix with copper.						
thiophanate methyl: Cleary's 3336 F, EG; Allban 50 WSB; OHP 6672 50WP, 4.5F	x	x	x			x
1 + 2: MBC Benzimidazoles + Dicarboximides: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.						
thiphanate methyl + iprodione: 26/36[W]	x					x
1 + 3: MBC Benzimidazoles + DMI or SI Triazoles: Systemic. Long protection during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.						
thiophanate methyl + propiconazole: Protocol[W]		x				x
1 + 14: MBC Benzimidazoles + Aromatic Hydrocarbons: Systemic. Effective against water molds, or oomycetes. Long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.						
thiophanate methyl + etridiazole: Banrot 8G, 40WP[W]	x		x		x	
1 + M5: MBC Benzimidazoles + Multi-site inhibitor: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.						
thiophanate methyl + chlorothalonil: Spectro 90 WDG, Zyban[W]	x	x	x			x
2: Dicarboximides: Locally systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.						
iprodione: Chipco 26019 FLO, N/G; 26GT; Iprodione Pro 2SE; Raven[V]			x			x
3: DMI or SI Triazoles: This group was formerly known as De-Methylation Inhibitors (DMI) and are now classified as Sterol Biosynthesis Inhibitors (SBI or SI). Upwardly systemic. Rain-fast in 2 hours. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.						
fenarimol: Rubigan A.S.		x				
imazalil: Fungaflor TR		x				
metconazole: Tourney[W]		x				x
myclobutanil: Eagle, 20W; Siskin		x				x
myclobutanil: Rally 40WSP		x				

Table 3.5 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Ilex*[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Black root rot	Powdery mildew	Rhizoctonia	Botryosphaeria	Phytophthora root rot, water molds[X]	Anthracnose
3: DMI or SI Triazoles: This group was formerly known as De-Methylation Inhibitors (DMI) and are now classified as Sterol Biosynthesis Inhibitors (SBI or SI). Upwardly systemic. Rain-fast in 2 hours. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.						
propiconazole: Banner Maxx, Banner Maxx II, Strider		x	x			x
tebuconazole: Torque		x	x			x
triadimefon: Bayleton 50, Flo; Strike 50 WDG[V]		x				x
triflumizole: Terraguard SC, SC/LS	x	x				
triticonazole: Trinity , Trinity TR		x				x
3 + 11: DMI or SI Triazoles + Quinone outside Inhibitors (QoI): Rain-fast in 2 hours and locally systemic. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.						
trifloxystrobin + triadimefon: Trigo		x				x
4: Phenylamides: Systemic. Effective against diseases caused by oomycetes, or water molds, including damping-off, root and stem rots, and foliar diseases. Use as soil drench or foliar application.						
mefenoxam: Subdue Maxx, Subdue GR			x		x	
4 + 12: Phenylamides + Phenylpyrroles/Osmotic signal transducers: Systemic. Broad spectrum drench-applied fungicide.						
mefenoxam + fludioxonil: Hurricane, Hurricane WDG[U,V,W]			x		x	
5: Morpholine: Inhibits sterol biosynthesis in membranes. For use only in greenhouse and similar enclosed structures. Do not use after flower buds visible.						
piperalin: Pipron		x				
7: Carboxamides/Succinate Dehydrogenase Inhibitors (SDHI): Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Spray apply for leaf disorders.						
utolanil:Prostar 70WP			x			
7 + 11: Carboxamides/Succinate Dehydrogenase Inhibitors (SDHI) + QoI Q: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Spray apply for leaf disorders, soil drench for Phytophthora.						
pyraclostrobin + boscalid: Pageant, Pageant Intrinsic[W]		x	x		x	x
fluxapyroxad + pyraclostrobin: Orkestra Intrinsic[W]		x				x
benzovindiflupyr + azoxystrobin: Mural						x

Table 3.5 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Ilex*[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Black root rot	Powdery mildew	Rhizoctonia	Botryosphaeria	Phytophthora root rot, water molds[X]	Anthracnose
9 + 12: Anilino-Pyrimidines/Methionine biosynthesis inhibitors + Phenylpyrroles: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.						
cyprodinil + fludioxonil: Palladium[W]		x				x
11: Quinone outside Inhibitors (QoI): These fungicides are also known as strobilurins. Locally systemic. Effective on mildews, foliar pathogens, and most fungi. Some control of oomycetes, or water molds. Fungicide resistance risk high. Apply as a soil drench for Phytophthora root rot.						
azoxystrobin: Heritage		x	x		x	x
azoxystrobin: Strobe 50WG		x	x			x
fenamidone: FenStop		x			x	x
fluoxastrobin: Disarm O		x	x		x	x
kresoxim methyl: Cygnus		x				x
pyraclostrobin: Insignia; Insignia SC		x	x		x	
pyraclostrobin: Empress Intrinsic			x		x	
trifloxystrobin: Compass; Compass O 50 EDG[V]		x	x		x	x
12: Phenylpyrroles/Osmotic signal transducers: Non-systemic but good residual protection. Broad spectrum fungicide, not effective against oomycetes, or water molds.						
fludioxonil: Emblem, Medallion; Medallion WDG, Mozart TR	x		x			x
14: Aromatic Hydrocarbons: Locally systemic. Effective against water molds, or oomycetes.						
etridiazole: Terrazole 35% WP, CA, L; Truban 25EC, 30WP[V]					x	
19: Octopamine Receptor Agonists: Locally systemic. For use in outdoor and greenhouse nursery crops.						
polyoxin D: Affirm, Veranda O, Endorse	x	x	x			x
21: Quinone inside Inhibitors: Locally systemic. Effective against water molds, or oomycetes. Resistance risk unknown but presumed to be medium to high. Apply as a soil drench for Phytophthora root rot.						
cyazofamid: Segway, Segway O, Segway SC					x	
28: Carbamates: Cell membrane permeability, fatty acid interruption (proposed). Low to medium resistance risk.						
propamocarb: Banol[V]			x		x	

Table 3.5 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Ilex*[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Black root rot	Powdery mildew	Rhizoctonia	Botryosphaeria	Phytophthora root rot, water molds[X]	Anthracnose
33: Phosphonates: Fully systemic; when applied to leaves, product can translocate to lower parts. Effective against water molds, or oomycetes, such as Phytophthora, Pythium, and downy mildew pathogens. Low risk for fungicide resistance development. Apply as a soil drench for Phytophthora root rot.						
phosphorous acid: Alude		x			x	
phosphorous acid: K-Phite T/O, 7LP		x			x	x
potassium phosphite: Vital		x			x	
fosetyl-Al: Aliette WDG		x			x	
40: Carboxylic Acid Amides: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes. Apply as a soil drench for Phytophthora root rot.						
mandipropamid: Micora					x	
dimethomorph: Stature DM, SC					x	
43: Pyridinemethyl-benamides Delocalisation of spectrin-like proteins: Locally systemic, translaminar. Control of oomycetes, or water molds. Medium to high resistance risk. Apply as a soil drench for Phytophthora root rot.						
fluopicolide: Adorn					x	
44: *Bacillus subtilis*: Microbial disruptor of cellular membranes. Spray apply for leaf disorders, soil drench for Phytophthora.						
Bacillus subtilis: Cease		x	x		x	x
45 + 40: Quinone outside Stigmatellin-binders (QoS) + Carboxylic Acid Amides: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes. Apply as a soil drench for Phytophthora root rot.						
ametoctradin + dimethomorph: Orvego[W]					x	
49: Oxysterol binding protien homologue inhibition (OSBPI): Resistance risk assumed to be medium to high (single site inhibitor). Previously U15 FRAC code.						
oxathiapiprolin: Segovis					x	
M: Chemicals with multi-site activity: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.						
(M1) copper hydroxide: Champ DP, Champ Formula 2		x				x
(M1) copper hydroxide: CuPro 5000, T/N/O; Nu-Cop 50DF, HB, 3L		x				x

Table 3.5 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Ilex*[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Black root rot	Powdery mildew	Rhizoctonia	Botryosphaeria	Phytophthora root rot, water molds[X]	Anthracnose
M: Chemicals with multi-site activity: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.						
(M1) copper octanoate: Camelot O		x				x
(M1) copper sulphate pentahydrate: Phyton 27, Phyton 35		x				x
(M1) tribasic copper sulfate: Cuproxat		x				x
(M1 + M3) copper hydroxide + mancozeb: Junction		x				x
(M3) mancozeb: Mancozeb 4 F, Flowable with Zinc	x	x	x	x		x
(M3) mancozeb: Pentathlon LF	x	x	x	x		x
(M3) mancozeb: Protect DF	x	x	x	x		x
(M5) chlorothalonil: Daconil Ultrex, Zn Flowable, Weather Stik [T]		x				x
M3 + 3: Multi site inhibitor + Sterol Biosynthesis Inhibitor: Effective as protectants (and upwardly systemic) on broad spectrum including most fungi and mildews. Fungicide resistance risk low.						
mancozeb: + myclobutanil: Clevis[W]		x				x
M5 + 3: Multi site inhibitor + Sterol Biosynthesis Inhibitor: Effective as protectants (and upwardly systemic) on broad spectrum including most fungi and mildews. Fungicide resistance risk low.						
propiconazole + chlorothalonil: Concert II		x				x
NC: Not a Classified substance: Contact fungicide for greenhouse and nursery use. Low risk for resistance.						
potassium bicarbonate: MilStop		x				x

[Z] (FRAC, 2017).
[Y] Check current products for labeled pesticides, sites for control and plant safety and efficacy on fungal species. This table reports information on fungicide labels and does not necessarily reflect product efficacy. Refer to fungicide labels for rates and usage, specific host information, possible phytotoxicity, re-entry intervals and resistance management. Within columns, products indicated by "x" are labeled for use against the listed pathogen type. Always test product on a sample (small number) of plants before treating an entire crop, as some fungicides listed are for control of the disorder but not specifically labeled for use in *Hudrangea* spp.
[X] Including the causal agent of sudden oak death, *Phytophthora ramorum*.
[W] Chemical contains more than one active ingredient, thus product may be listed within more than one FRAC code designation (FRAC, 2017).
[V] Not for use in residential landscapes; commercial use only.
[U] Only use as a suppressant, does not grant preventative control.
[T] Do not apply with mist blowers or high pressure spray equipment in greenhouses.

References

Agrios, G.N. 1997. Plant Pathology. 4th ed. Academic Press, New York.

AmericanHort. 2014. American Standard for Nursery Stock, ANSI Z60.1-2014. Accessed October 6, 2015 <http://americanhort.org/documents/ANSI_Nursery_Stock_Standards_AmericanHort_2014.pdf>.

Anonymous. 2004. Black vine weevil. In: IPM of Midwest Landscapes, pp. 77-78. V. Krischik and J. Davidson, eds. MN Ag. Exp. St.Publication SB-07645.

Ansari, M.A. and T.M. Butt. 2011. Effect of potting media on the efficacy and dispersal of entomopathogenic nematodes for control of black vine weevil, *Otiorhynchus sulcatus* (Coleoptera: Curculionidae). Biological Control 58:310-318.

Arthurs, S.P., C.L. McKenzie, J.J. Chen, M. Dogramaci, M. Brennan, K. Houben and L. Osborne. 2009. Evaluation of *Neoseiuslus cucumeris* and *Amblyseius swirskii* (Acari: Phytoseiidae) as biological control agents of chilli thrips, *Scirtothrips dorsalis* (Thysanoptera: Thripidae), on pepper. Biological Control 49:91-96.

Baker, J.R. (ed.) 1980. Insect and related pests of shrubs. NC State Univ. Cooperative Ext. Services, Bulletin AG-189. Raleigh, NC. 199 pp.

Baker, J. R. 1994. Twobanded Japanese weevil. Accessed September 17, 2015. <http://www.ces.ncsu.edu/depts/ent/notes/O&T/shrubs/note34/note34.html>.

Beales, P.A., C.R. Lane, V.C. Barton, and P.M. Giltrap. 2006. *Phytophthora kernoviae* on ornamentals in the UK. EPPO Bulletin, 36:377–379.

Beardsley, J.W., and R.H. Gonzalez. 1975. The biology and ecology of armored scales. Ann. Rev. Entomo. 20:47-73.

Bethke, J., J. Chamberlin, R. Cloyd, J, Dobbs, M. Faver, D. Gilrein, K. Heinz, R. Lindquist, S. Ludwig, C. McKenzie, G. Murphy, R. Oetting, L. Osborne, C. Palmer, M. Parrella, N. Rechcigl and R. Yates. 2011. Thrips management program for ornamental horticulture. Accessed September 17, 2015. <http://mrec.ifas.ufl.edu/lso/DOCUMENTS/ThripsManagementProgram-February%202011-FINAL.pdf>.

Bianchi, A., A. Pacchiacucchi, L. Guarino, and E. Maffeo. 1994. Segnalata una nuova cocciniglia su actinidia nel Lazio. Informatore Agrario 50:46, 73-75.

Blank, R.H., C. Gill, M.H. Olson, and M.P. Upsdell. 1995. Greedy scale phenology on Taraire based on Julian days and degree-day accumulations. Env. Entomo. 24:1569-1575.

Boyd, D.W. and A.G. Wheeler. 2005. The twobanded Japanese weevil, *Pseudocneorhinus bifasciatus* (Roelofs) (Coleoptera: Curculionidae): southeastern U.S. distribution of an adventive pest of ornamental shrubs. Proc. Southern Nursery Assoc. Res. Conf. 49:192-194.

Brandenburg, R.L., S. Bambara and J.R. Baker. 2002. Managing the twolined spittlebug in the home landscape. Ornamentals and Turf, Department of Entomology Insect Note, NC State Univ. Coop. Ext. Accessed October 18, 2015 <https://www.ces.ncsu.edu/depts/ent/notes/O&T/lawn/note97/note97.html>.

Braxton, S.M., and M.J. Raupp. 1995. An annotated checklist of clearwing borer pests of ornamental plants trapped using commercially available pheromone lures. J. Arboriculture 21:177-180.

Chappell, M., J. Owen, S. White and J. Lea-Cox. 2013. Container Nutrition Management Practices IN T. Yeager, T. Bilderback, D. Fare, C. Gilliam, J. Lea-Cox, A. Niemiera, J. Ruter, K. Tilt, S. Warren, T. Whitwell and R. Wright (eds.) Best Management Practices: Guide for Producing Nursery Crops 3rd edition. Southern Nursery Assoc., Atlanta, GA.

Chappell, M. 2015. Preliminary studies indicate the mechanism of winter injury in broadleaf species and deer herbivory in landscape plantings in the Atlanta metro. Personal Communication.

Chiu, C.-H. and C.A. Kouskolekas. 1980. Observations on reproductive biology of tea scale, *Fiorinia theae* Green. J. GA Entomo. Soc. 15:317-327.

Chong, J-H. 2015. Distribution of scale insects across the Southeast and preliminary results on granualate ambrosia research. Personal Communication.

Cooper, R.M. and R.D. Oetting. 1987. Hymenopterous parasitoids of tea scale and camellia scale in Georgia. J. Entomo. Sci. 22:297-301.

Cooper, R.M. and R.D. Oetting. 1989. Life history and field development of the camellia scale (Homoptera: Diaspididae). Annals Entomo. Soc. America 82:730-736.

Cottrell, T.E., and D.I. Shapiro-Ilan. 2006. Susceptibility of the peachtree borer, *Synanthedon exitiosa*, to *Steinernema carpocapsae* and *Steinernema riobrave* in laboratory and field trials. J. Invertebrate Path. 92:85-88.

Cowles, R.S. 2001. Protecting container-grown crops from black vine weevil larvae with bifenthrin. J. Env. Hort. 19:184-189.

Day, E. 2009. Scale insects. Accessed September 17, 2015. <http://pubs.ext.vt.edu/2808/2808-1012/2808-1012.html>.

Dekle, G.W. 1977. Florida armored scale insects. FL Dept. Ag., Division of Plant Industry, Gainesville, FL. 345 pp.

Dirr, M.A., G.E. Smith, D. Ehrlinger, T. Smith and L. Thomas. 1984. Casualties and survivors of the 1983-84's freeze. American Nurseryman 160:33-55.

Dirr, M.A. and O. Lindstrom Jr. 1990. Leaf and stem cold hardiness of 17 broadleaf evergreen taxa. J. Env. Hort. 8:71-73.

Dogramaci, M., S.P. Arthurs, J.J. Chen, C. McKenzie, F. Irrizary and L. Osborne. 2011. Management of chilli thrips, *Scirtothrip dorsalis* (Thysanoptera: Thripidae), on peppers by *Amblysieus swirskii* (Acari: Phytoseiidae) and *Orius insidiosus* (Hemiptera: Anthocoridae). Biological Control 59:340-347.

Drake, D., P. Nitzsche, and P. Perdomo. 2003. Landscape plants rated by deer resistance. Rutgers NJAES Cooperative Ext. Bulletin 271.

English, L.L. and G.F. Turnipseed. 1940. Insect pests of azaleas and camellias and their control. Alabama Polytechnic Institute Ag. Exp. Station Circular 84.

Faulring, J. 2014. Soil moisture sensors in *Ilex* production. Personal Communication.

Fisher, J.R. 2006. Fecundity, longevity and establishment of *Otiorhynchus sulcatus* (F.) and *Otiorhynchus ovatus* (L.) (ColeopteraL Curculionidae) from the Pacific North-west of the United States of America on selected host plants. Ag. and Forest Entomo. 8:281-287.

Fredericks, W.H., Jr. 1992. The Exuberant Garden and the Controlling Hand. Little, Brown and Company, Boston, MA.

Fulcher, A., F. Hale, and M. Halcomb. Japanese maple scale: An important new insect pest in the nursery and landscape. Univ. TN Ext. Pub. W277. Accessed September 15, 2015 <https://extension.tennessee.edu/publications/Documents/W277.pdf>.

Galle, F. 1997. Hollies: The genus *Ilex*. Timber Press, Portland, OR.

Ghidiu, G.M., L. Vasvary, T.D. Eichlin, and J.D. Solomon. 1987. Injury and biology of the clearwing borer *Synanthedon kathyae*. J. Lepidopterists' Soc. 41:154-158.

Gill, R.J. 1988. The scale insects of California: Part 1. The soft scales (Homoptera : Coccoidea : Coccidae). CA Dept. Food & Ag., Sacramento, CA. 132 pp.

Gill, S.A., D.R. Miller, and J.A. Davidson. 1982. Bionomics and taxonomy of the euonymus scale, *Unaspis euonymi* (Comstock), and detailed biological information on the scale in Maryland (Homoptera: Diaspididae). Univ. MD Ag. Exp. Station Misc. Pub. 969.

Gill, S. 2011. TPM/IPM weekly report for arborists, landscape managers & nursery managers, April 8, 2011. Accessed September 17, 2015 <https://www.extension.umd.edu/sites/default/files/_docs/programs/ipmnet/11Apr08L.pdf>.

Gill, S., P. Shrewsbury, and J. Davidson. 2012. Japanese maple scale (*Lopholeucaspis japonica*): A pest of nursery and landscape trees and shrubs. Univ. MD Ext. Publication. Accessed 15 September, 2015 <https://extension.umd.edu/sites/default/files/_docs/programs/ipmnet/JapaneseMapleScale-UMD.pdf>.

Gorsuch, C.S. 2003. Two-lined spittlebug. Dept. Entomo. Soils, and Plant Sci., Clemson Univ. EIIS/TO-16. Accessed September 17, 2015. <http://www.clemson.edu/cafls/departments/esps/factsheets/turforn/two_lined_spittlebug_to16.html>.

Grewal, P.S. 2012. Entomopathogenic nematodes as tools in integrated pest management. *In* Integrated Pest Management: Principles and Practice. D. P. Abrol and U. Shankar, eds. CABI Publishing, Wallingford, England.

Gyeltshen, J., and A.C. Hodges. 2013. Field key to identification of scale insects on holly (*Ilex* spp.). Publication #IPM-141. IFAS Ext. Accessed September 17, 2015. <http://edis.ifas.ufl.edu/in649>.

Hagan, A. Common diseases of holly and their control. 2005. AL Cooperative Ext. ANR-1087. Accessed September 15, 2015 <http://www.aces.edu/pubs/docs/A/ANR-1087/ANR-1087.pdf>.

Halcomb, M. and A. Fulcher. 2010. IPM for Shrub Production: Chapter 2. Accessed September 29, 2015. <http://plantsciences.utk.edu/pdf/fulcher_IPM_shrub_online_version_foster_hollies.pdf>

Hale, F.A., originally developed by J. Yanes, Jr. and H. Williams. Revised 2015. Holly leafminers, Univ. TN Ext. Pub. SP 290. Accessed September 15, 2015. <https://extension.tennessee.edu/publications/Documents/SP290-T.pdf>.

Hamon, A.B., and M.L. Williams. 1984. The soft scale insects of Florida. Arthropods of Florida and Neighboring Land Areas. Vol. II. FL Dept. Ag. Cons. Serv., Div. Plant Ind., Gainesville, FL. 194 pp.

Hartline, B.J. 2009. Propagating *Ilex decidua*. Personal Communication.

Hartmann, H.T., D.E. Kester, F.T. Davies, Jr. and R.L. Geneve. 2011. Hartmann and Kester's Plant Propagation, 8th Ed. Prentice Hall, New York.

Hassan, A.S., H.A. Nabil, A.A. Shahein, and K.A.A. Hammad. 2012. Some ecological aspects of *Kilifia acuminata* (Hemiptera: Coccidae) and its parasitoids on mango trees at Sharkia Governorate, Egypt. Egyptian Acad. J. Biological Sci. 5:33-41.

Herms, D.A. 2004. Using degree-days and plant phenology to predict pest activity. *In* IPM of Midwest Landscapes, pp. 49-59. V. Krischik and J. Davidson, eds. MN Ag. Exp. Station Pub. SB-07645.

Hodges, A., S. Ludwig, L. Osborne and G.B. Edwards. 2009. Pest Thrips of the United States: Field Identification Guide. Accessed September 14, 2015. <https://firstdetector.org/pdf/chili_thrips_deck.pdf>.

Hodges, G.S. and S.K. Braman. 2004. Seasonal occurrence, phenological indicators and mortality factors affecting five scale insect species (Hemiptera: Diaspididae, Coccidae) in the urban landscape setting. J. Entomol. Sci. 39:611-622.

Holly Society Of America, Inc. Holly Cultivar Registrations:1994-Present. Accessed October 4, 2015. <http://www.hollysocam.org/registrations.htm>.

Holtz, T. 2006. NPAG Report: *Scirtothrips dorsalis* Hood. New Pest Advisory group, center for Plant Health Sci. and Technology, USDA-APHIS, Raleigh, NC. 7 pp.

Hopkins, Robert. 2009. Personal Communication.

Hume, H. Harold. 1953. Hollies. The MacMillan Company, New York

Hwang, J. and D.M. Benson. 2005. Identification, mefenoxam sensitivity, and compatibility type of *Phytophthora* spp. attacking floriculture crops in North Carolina. Plant Dis. 89:185-190.

Johnson, W.T., and H.H. Lyon. 1991. Insects that Feed on Trees and Shrubs. 2nd Ed. Comstock Publishing Associates.

Kosztarab, M. 1996. Scale insects of Northeastern North America: Identification, Biology, and Distribution. VA Museum Nat. History, Special Publication Number 3, Martinsville, VA.

LeBude, A.V., T.E. Bilderback, H.T. Krauss, S.A. White, M. Chappell and J. Owen. 2012. Preparing nursery crops for winter in the Southeastern United States. NC State Cooperative Ext. Bulletin AG-454.

Mague, D., and H.T. Streu. 1980. Life history and seasonal population growth of *Oligonychus ilicis* infesting Japanese holly in New Jersey. Env. Entomo. 9:420-424.

Masiuk, M. 2003. Black vine weevil fact sheet. Penn State Cooperative Ext. Accessed September 17, 2015. <http://ento.psu.edu/extension/factsheets/black-vine-weevil>.

McComb, C.W. and J.A. Davidson. 1969. Armored scale insects. A checklist of the Diaspididae of Maryland and the District of Columbia. MD Univ., Entomology Leaflet 50.

Meyer, F.G., P.M. Mazzeo, and D.H. Voss. 1994. A catalog of Cultivated Woody Plants of The Southeastern United States. U.S. National Arb. Contribution Number 7.

Miller, D.R., and J.A. Davidson. 1990. A list of armored scale pests, pp. 299-306. *In* D. Rosen (ed.), Armored scale insects. Vol. 4B. Elsevier, Amsterdam, The Netherlands.

Miller, D.R. and J.A. Davidson. 2005. Armored Scale Insect Pests of Trees and Shrubs. Cornell Univ. Press, Ithaca, NY.

Mizell, R.F., and T.C. Riddle. 2004. Evaluation of insecticides to control the Asian ambrosia beetle, *Xylosandrus crassiusculus*. Proc. Southern Nurs. Assoc. Res. Conf. 49:152-155.

Munir, B. and R.I. Sailer. 1985. Population dynamics of the tea scale, *Fiorinia theae* (Homoptera: Diaspididae), with biology and life tables. Env. Entomol. 14:742-748.

Murakami, Y. 1970. A review of biology and ecology of diaspine scale in Japan (Homoptera: Coccoidea). MUshi 43:65-114.

Mussey, G.J. and D.A. Potter. 1997. Phenological correlations between flowering plants and activity of urban landscape pests in Kentucky. J. Economic Entomo. 90:1615-1627.

Nash, R.F. 1973. Control of the peony scale, *Pseudaonidia paeoniae*, and a wax scale, *Ceroplastes ceriferus*, with granular systemic insecticides on *Camellia japonica*. J. GA Entomol. Soc. 8:149-151.

Neal, J.W., and T.D. Eichlin. 1983. Seasonal responseof six male Sesiidae of woody ornamentals to clearwing borer (Lepidoptera: Sesiidae) lure. Env. Entomol. 12:206-209.

Niu, G., D.S. Rodriguez, R. Cabrera, C. McKenney, and W. Mackay. 2006. Determining water use and crop coefficients of five woody landscape plants. J. Env. Hort. 24:160-165.

Oliver, J.B., and C.M. Mannion. 2001. Ambrosia beetle (Coleoptera: Scolytidae) species attacking chestnut and captured in ethanol-baited traps in middle Tennessee. Env. Entomol. 30:909-918.

Oliver, J.B., N.N. Youssef, and M.A. Halcomb. 2004. Comparison of different trap types for collection of Asian ambrosia beetle, *In* J.B. Oliver [ed.], Proc. Southern Nursery Assoc. Res. Conf. pp. 158-163.

Olson, H.A. and D.M. Benson. 2011. Characterization of *Phytophthora* spp. on floriculture crops in North Carolina. Plant Disease 95:1013-1020.

Olson, H.A., S.N. Jeffers, K.L. Ivors, K.C. Steddom, J.L. Williams-Woodward, M.T. Mmbaga, D.M. Benson, and C.X. Hong. 2013. Diversity and mefenoxam sensitivity of *Phytophthora* spp. associated with the ornamental horticulture industry in the southeastern United States. Plant Disease 97:86-92.

Pscheidt, J.W., Wick, R.L., and D.M. Benson. 2001. Holly diseases. *In: Diseases of woody ornamentals and trees in nurseries*. R.K. Jones and D.M. Benson (Eds.). APS Press, St. Paul, MN.

Rabaglia, R.J., S.A. Dole, and A.I. Cognato. 2006. Review of American Xyleborina (Coleoptera: Curculionidae: Scolytinae) occurring north of Mexico, with an Illustrated key. Annals Entomo. Soc. America 99:1034-1056.

Reding, M.E. 2008. Black vine weevil (Coleoptera: Curculionidae) performance in container- and field-grown hosts. J. Entomol. Sci. 43:300-310.

Reding, M.E. and A.B. Persad. 2009. Systemic insecticides for control of black vine weevil (Coleoptera: Curculionidae) in container- and field grown- nursery crops. J. Economic Enotmo. 102:927-933.

Reding, M.,J. Oliver, P. Schultz, and C. Ranger. 2010. Monitoring flight activity of ambrosia beetles in ornamental nurseries with ethanol-baited traps: Influence of trap height on captures. J. Env. Horticulture 28:85-90.

Reding, M.E. and C.M. Ranger. 2011. Systemic insecticides reduce feeding, survival, and fecundity of adult black vine weevils (Coleoptera: Curculionidae) on a variety of ornamental nursery crops. J. Economic Entomol. 104:405-413.

Rehman, S.U., H.W. Browning, H.N. Nigg and J.M. Harrison. 2000. Increases in Florida red scale populations through elimination of *Aphytis holoxanthus* Debach in Florida citrus. Biological Control 18:87-93.

Rosen, D., and P. DeBach. 1978. Diaspididae, *In* C.P. Clausen (ed.), Introduced parasites and predators of arthropod pests and weeds: A world review. USDA Handbook No. 480. Washington, DC.

Rutten, D. and K. Santarius. 1988. Cold acclimation of *Ilex aquifolium* under natural conditions with special regard to the photosynthetic apparatus. Physiologia Plantarum 72:807-815.

Sanders, L. and J. Sanders. Hollies for cut stems in Paducah, KY. Personal communication.

ScaleNet. 2015. Accessed September 16, 2015. <http://www.sel.barc.usda.gov/scalenet/scalenet.htm>.

Seal, D.R., W. Klassen and V. Kumar. 2010. Biological parameters of *Scirtothrips dorsalis* (Thysanoptera: Thripidae) on selected hosts. Env. Entomol. 39:1389-1398.

Shapiro-Ilan, D.I., T.E. Cottrell, R.F. Mizell, D.L. Horton, and J. Davis. 2009. A novel approach to biological control with entomopathogenic nematodes: Prophylactic control of the peachtree borer, *Synanthedon exitiosa*. Biol. Control 48:259-263.

Shetlar, D. 2002. Holly & inkberry leafminers. The Ohio State Univ. Ornamental Fact Sheet. Accessed September 21, 2015 <http://entomology.osu.edu.edu/bugdoc/Shetlar/factsheet/ornaMENTAL/fsHOLLY-INKBERRYlm.HTM>.

Simpson, R. Average bloom times of deciduous hollies in Vincennes, Indiana. Accessed September 29, 2015 <http://www.simpsonnursery.com/what-we-grow/bloom-chart-2>.

Sinclair W.A. and H.H. Lyon. 2005a. Powdery mildews. *In:* Diseases of trees and shrubs, 2nd Ed. Cornell University Press, Ithaca, NY.

Sinclair W.A. and H.H. Lyon. 2005b. Anthracnose and diebacks caused by Glomerella and Colletotrihum. *In:* Diseases of trees and shrubs, 2nd Ed. Cornell University Press, Ithaca, NY.

Sinclair W.A. and H.H. Lyon. 2005c. Botryosphaeria cankers and diebacks. *In:* Diseases of trees and shrubs, 2nd Ed. Cornell University Press, Ithaca, NY.

Sinclair W.A. and H.H. Lyon. 2005d. Phymatotrichum and Thielaviopsis Root Rots. *In:* Diseases of trees and shrubs, 2nd Ed. Cornell University Press, Ithaca, NY.

Smith, F.F. 1932. Biology and control of the black vine weevil. USDA Technical Bulletin 325.

Smith, F.F., A.K. Ota, C.W. McComb and J.A. Weidhaas. 1971. Development and control of a wax scale, *Ceroplastes ceriferus*. J. Economic Entomol. 64:889-893.

Solomon, J.D. 1995. Guide to insect borers of North American broadleaf trees and shrubs. Agricultural Handbook 706. U.S. Department of Agriculture & Forest Service, Washington, DC.

Stephens, W. 2007. Colored shade cloth and plant growth. Comb. Proc. Intl. Plant Prop. Soc. 57:212–17.

Stoetzel, M.B., and J.A. Davidson. 1974. Biology, morphology and taxonomy of immature stages of 9 species in the Aspidiotini. Annals Entomol. Soc. America 67:475-509.

Thompson, W.L., and J.T. Griffiths. 1949. Purple scale and Florida red scale as insect pests of citrus in Florida. Univ. FL, Ag. Exp. Station Bulletin 462. Gainesville, FL.

Tippins, H.H. 1968. Observations on *Phenacaspis cockerelli* (Cooley), a pest of ornamental plants in Georgia. J. GA Entomol. Soc. 3:13-15.

Tippins, H.H., H. Clay and R.M. Barry. 1977. Peony scale: a new host and biological information. J. GA Entomol. Soc. 12:68-71.

U.S. Department of Agriculture - NASS. 2014. 2014 Census of Horticultural Specialties: Table 19: Broadleaf Evergreens. Washington, D.C.

van Tol, R., D.J. Bruck, F.C. Griepink and W.J. de Kogel. 2012. Field attraction of the vine weevil *Otiorhynchus sulcatus* to kairomones. J. Economic Entomol. 105:169-175.

Williams-Woodward, J., K. Ivors, A. Windham, N. Ward-Gauthier. 2014. Relative effectiveness of various chemicals for disease control of ornamental plants. Accessed September 16, 2015. <http://wiki.bugwood.org/uploads/Orn_efficacy_table2014.pdf>.

Rhododendron and Azalea – *Rhododendron* spp.

William E. Klingeman, University of Tennessee

Anthony V. LeBude, North Carolina State University

Angelia Rateike, University of Tennessee

Geoff M. Weaver, University of Georgia

Jean Williams-Woodward, University of Georgia

Discussion of cranberry rootworm (*Rhadopterus picipes*) is largely reprinted from text prepared by Knox et al. (2012); information about weevils and black stem borer (*Xylosandrus germanus*) is adapted from Chong et al. (2012); details regarding caterpillar defoliators and twospotted spider mite (*Tetranychus urticae*) management is largely reprinted from text prepared by Fulcher et al. (2015); cottony camellia scale (*Pulvinaria floccifera*) and southern red mite (*Oligonichus ilicis*) management are largely based on text prepared by Knox et al. (2014); and *Oberea* longhorned borer management is largely based on text prepared by Klingeman et al. (2014). Discussion of Phytophthora leaf blight and dieback (*Phytophthora ramorum* and *Phytophthora* spp.) management is based in part on text prepared by Klingeman et al. (2014). Reprint of this content herein is made possible by permission of the original content authors.

Section 1

History, Culture and Management

Introduction

1. Introduction
2. Horticultural Management and Production Practices
3. Container Production
4. Pruning and Landscape Plant Form

Introduction

Rhododendron evokes imagery of exotic Far East adventures, steep mountain terrain and deep river valleys, intoxicating fragrances, overwhelming beauty, and bold textures in the garden (Figure 4.1). Plant specimens were collected from rugged areas in the orient and the southern Appalachian Mountains by some of the earliest plant explorers. Indeed, rhododendrons have been captivating nursery producers and gardeners for literally centuries. Rhododendrons grown in nurseries in the southeastern U.S. consist mainly of the large leaved evergreen species and their hybrids with large colorful trusses, the deciduous azaleas with beautifully vibrant colored flowers on long thin stems, and the small-leaved evergreen azaleas, which provide a kaleidoscope of color every spring and early summer so common to many southern landscapes.

Figure 4.1 The bold colors and textures of many Rhododendron taxa, such as *Rhododendron austrinum*, offer focal points in landscapes across the southeast.

History

The genus *Rhododendron* contains over 1,000 species and was thought to have originated along the south slope of the Himalaya Mountains extending from Nepal into northern Burma through to the Yunnan and Szechuan provinces of southwestern China. These south Asian mountainous origins, in addition to mountainous regions in the western and eastern U.S., are home to many of the large leaved evergreen species and most deciduous azaleas found in the U.S. The genus extends southward to Malaysia, the Philippine Islands and as far south as Australia. The small-leaved evergreen azaleas that are common in many southeastern U.S. landscapes are indigenous to a maritime distribution along Thailand, Viet Nam, and eastern China, Korea, and Japan (Leach, 1961). Coincidentally, in the southeastern U.S., the planting plans of many landscapes, as well as the inventory of many nurseries (Figure 4.2), contain some form of small leaved evergreen azalea. They are ubiquitous.

Figure 4.2 Nursery production of azalea is widespread across the southeast. Pictured here is a large block of azaleas growing at van der Giessen nursery in Semmes, AL.

Classification of Species and Cultivars

The phylogeny and classification of *Rhododendron* spp. will continue to be rearranged as more powerful tools discern relationships among species more confidently. According to Goetsch (2005), *Rhododendron* spp. can be placed into two subgenera and three sections that represent the deciduous azaleas (section *Pentanthera*), large leaved evergreen species (section *Ponticum*) and small leaved evergreen species (section *Tsutsusi*) (Table 4.1). Various subsections within each section categorize further the species. The number of cultivars of all three subgenera and sections of *Rhododendron* are numerous and legendary, and many volumes, as well as careers, have been dedicated to their description and lineage (Leach, 1961; Galle, 1987; Salley and Greer, 1986; Towe, 2004). Salley and Greer (1986) cover over 4,800 cultivars introduced and registered since the early 1800's. The Royal Horticultural Society (RHS) is the official international registrar for rhododendron and the American Rhododendron Society (ARS) serves as the North American representative to the RHS for genus *Rhododendron*. The approximate number of registered rhododendron hybrids at one point was 14,298 hardy rhododendron hybrids suitable for temperate regions; 12,989 azalea hybrids including both evergreen and deciduous azaleas; 108 azaleodendron hybrids which are crosses between azaleas and other rhododendrons; and 680 vireya hybrids which are tropical members of the *Rhododendron* genus (Anon., 2015).

Table 4.1 Proposed changes in classification of the genus *Rhododendron* (Goetsch, 2005)[z].

Subgenus	Section	Common Name	Common species and hybrid groups within this section (Galle 1985)
Hymenanthes K. Koch	Pentanthera G. Don	Deciduous azaleas	Species[y], Aromi deciduous, Ghents, Mollis, Occidentale, Knap Hill, Northern Lights, North American hybrids
Hymenanthes K. Koch	Ponticum G. Don	Large leaved evergreen azaleas	Species[x] (groups not identified)
Azaleastrum Planch.	Tsutsusi Sweet	Small leaved evergreen azaleas	Species[w], Aromi evergreen, Belgian Indian, Encore, Southern Indian, Kurume, Kaempferi, Satsuki, interhybrid groups.

[z] Table not meant to be reliable classification compared to published data nor is it comprehensive of each section, species list or hybrid group.

[y] Includes *Rhododendron alabamense, R. arborescens, R. atlanticum, R. austrinum, R. calendulaceum, R. canescens, R. cumberlandense, R. flammeum, R. japonicum, R. luteum, R. molle, R. periclymenoides, R. prinophyllum, R. prunifolium* and *R. viscosum* (Anon., 2015).

[x] Includes *Rhododendron aureum, R. brachycarpum, R. catawbiense, R. caucasicum, R. degronianum, R. hyperythrum, R. macrophyllum, R. makinoi, R. maximum, R. minus, R. ponticum, R. smirnowii, R. ungernii* (Anon., 2015).

[w] Includes *Rhododendron amagianum, R. atrovirens, R. breviperulatum, R. decandrum, R. dilatatum, R. eriocarpum, R. farrerae, R. hidakanum, R. hongkongense, R. indicum, R. kaempferi, R. kanehirai, R. kiusianum, mucronatum, R. nipponicum, R. oldhamii, R. ovatum, R. yedoense* var. *poukhanense* (Anon., 2015).

Figure 4.3 *Rhododendron catawbiense* on Roan Mountain in North Carolina.

Economic Value

The real number of nurseries in the southeastern U.S. that grow these individual groups of plants is probably unknowable, but group distribution is related to the climate of both the nursery and its final customer and relative sales figures (Table 4.2). More small-leaved evergreen azaleas are grown in the southeastern U.S., and perhaps the United States, than the large leaved evergreen cultivars. Deciduous azaleas are the least likely to be grown in large quantities compared to the other two sections of rhododendron, however, there are nurseries that specialize in growing deciduous azaleas exclusively. For example, nurseries in the deep southern states like Florida and Louisiana might grow more coastal deciduous azalea species and small leaved evergreen azaleas than the large leaved rhododendron species due to heat tolerance of the former groups. In contrast, growers in the upper piedmont and mountainous areas of North Carolina might grow larger leaved rhododendron species or their hybrids, as well as the native azaleas found locally throughout the steep terrain (Figure 4.3).

Horticultural Management and Production Practices

Adaptability and Culture

New, novel, and desirable rhododendron and azalea cultivars continue to be introduced by combining various species, introgressing new traits or creating new breeding pathways to otherwise reluctant crosses. Remontancy in hardy, little leaved evergreen azaleas have produced 30+ Encore® azalea cultivars that rebloom from spring through fall in wide ranging colors. Encore® azalea cultivars are hardy to USDA Plant Hardiness Zone (6)7 and thrive in the deep south. The 'Confederate Series' of deciduous azaleas married the heat tolerance of *R. austrinum*, Florida flame azalea, with the elegance of *R.* x'Hotspur Yellow' Exbury azalea to create a vigorous group of plants that withstand southern gardening conditions, particularly powdery mildew. A revolution in breeding has occurred with polyploid rhododendron. Polyploids have three or more sets of chromosomes. Seedlings of diploid (2x) parents can be treated with the herbicide oryzalin, a mitotic inhibitor, to produce polyploid (3x or greater) populations. Polyploid plants can have

Table 4.2 Cumulative rhododendron and azalea crop values reported by respondents from 36 state(s) for wholesale and/or retail sales (USDA, 2009).

Region [z]	States [y]	Total Crop Value (in $1000s)	
		Rhododendron [x]	Azalea [x]
1	CT, MA, NH(R[y]), RI, VT	11,318	1,820
2	NJ, NY	4,744	9,230
3	MD, PA, VA, WV	1,956	4,826
4	AL, FL, GA, KY, MS, NC, SC, TN	4,072[W]	40,210
5	IL, IN, MI, MN (R[y]), OH, WI	2,723	3,609
6	AR, LA, TX	19[V]	13,653
7	IA(RA[y]), KS(RA[y]), MO	37	19[U]
8	CO	*NR*	5
9	CA, HI	1,573[T]	6,320
10	OR, WA	19,263	7,982
Total		45,705	87,674

[Z] Standard federal regional boundaries.

[Y] State's respondents reported values for retail (R) sales of both rhododendrons and azaleas, or retail azalea sales (RA) only. Where no value was reported for a given state, then that state was excluded from listing within its region.

[X] Rhododendron includes large-leaf evergreen forms; Azaleas includes only evergreen little-leaf azalea forms.

[W] Rhododendron crop value excludes FL and MS, from which no data were reported.

[V] Rhododendron crop value excludes AR and LA, from which no data were reported.

[U] Rhododendron crop value excludes KS and azalea crop values exclude MO, from which no data were reported.

thicker leaves and larger flowers that persist longer (Jones et al., 2008). Furthermore, wide hybrids between distantly related species can be sterile and the ability to create polyploids of that cultivar can restore fertility, opening another pathway to breeding that otherwise was unavailable.

Figure 4.4 Well-rooted plug liners of various deciduous azaleas.

Propagation

Almost all *Rhododendron* species are propagated vegetatively from stem cuttings (Figure 4.4) or to a lesser extent, sexually from seeds (Figure 4.5). Individual nurseries in the south can collect millions of stem cuttings per year from cultivars on site. Successful vegetative propagation methods include rooting stem cuttings collected from hedged stock plants, two-leaf stem cuttings with a wounded base dipped in various rooting hormone treatments, stem layering both above ground and within mounds, grafting, or tissue culture (Harrington, 1990; Preece et al., 1993; Sommerville, 1998; Towe, 2004; Dirr and Heuser, 2006; Jones et al., 2010). Seeds of rhododendron require light to germinate but have no pretreatment requirements, therefore simply dusting them onto sandy peat or sphagnum peat moss under a constant temperature of 25°C (77°F) produces seedlings within 14-21 days (Blazich and Rowe, 2008).

Figure 4.5 Seeds being cleaned from capsules and sorted from trash prior to sowing.

Sanitation during Propagation

Preventing pathogen spread is important for establishing new evergreen azalea crops without disease or insects (Figure 4.6). In a series of experiments to completely control *Rhizoctonia* anastomosis group-P, the pathogen responsible for azalea web blight on stem cuttings of various little leaved evergreen rhododendron cultivars, Copes and Blythe

Figure 4.6 Sanitation during propagation is essential to the future health of crops. Reducing inoculum by maintaining a clean propagation environment is an obvious method of improving crop health.

(2009, 2011, 2012) found that dipping stem cuttings in 50°C (120°F) hot water baths for 20 minutes was the only sanitation method that completely controlled the pathogen. Moreover, they determined that stem cuttings could bathe from 20-40 minutes in 50°C (120°F) water and still develop adequate roots, which provided a wide margin of error in operations. Bathing cuttings for shorter durations in warmer water was less successful and caused more leaf damage. Even with 25% leaf damage caused by the hot water bath (50°C), stem cuttings still rooted adequately (Copes and Blythe, 2011). The method was successful for stem cuttings collected from 12 different hybrid groups of azaleas (Copes and Blythe, 2014), indicating that this method might be used successfully among other hybrid groups as yet untested.

Container Production

Containers and Container Type (Figure 4.7; 4.8)

Many container types exist for growing azaleas and rhododendrons. Largely, blow molded plastic containers in normal trade sizes are used for production of many species of rhododendron. In some cases, the type of container does not affect overall root growth or root architecture because rhododendrons, particularly small leaved evergreen azaleas, have small fibrous root systems (Figure 4.8), which are not prone to circling like larger tree root systems, but are more prone to matting on the container sides (Appleton, 1989). Nevertheless, *R.* 'Hershey Red' Kurume azaleas had fewer matted roots but similar

Figure 4.7 Evergreen rhododendron in container production.

Figure 4.8 Evergreen rhododendron allowed to dry down in late summer to encourage flower bud production for following spring. Note the extremely fibrous root system that is common in rhododendron and azalea taxa.

biomass when grown in containers treated with copper sulfate [Spinout® (Griffin Corporation, Valdosta, GA)] compared to the same azaleas grown in untreated containers. One year after transplanting to the landscape, azaleas that had been grown in Spinout® treated containers had more roots grown into the surrounding native soil (Appleton and French, 1999).

Small leaved evergreen azaleas grown in trade gallon sized containers are a staple in the south and they can consume more water than other small leaved species of plants during summer (Figure 4.9; 4.10). To alleviate this problem, growers use "squat" pots, which are generally wider and shorter than containers with similar growing volumes. Since the perched water table at the bottom of each container has a relatively similar height, the wider base and shorter stature of the squat pot increases the surface area for the perched water volume and lowers the height of the roots above this area making water more available to plants. Irrigation management is important because squat pots drain poorly and over-irrigation can cause nutrient uptake problems, root drowning, root rots, or all of these conditions. Another alternative for optimizing irrigation efficiency is to space plants more closely. Plants of *R.* 'Girard's Crimson' grown in trade #3 containers, as well as four other woody plant species, used less water when spaced 10

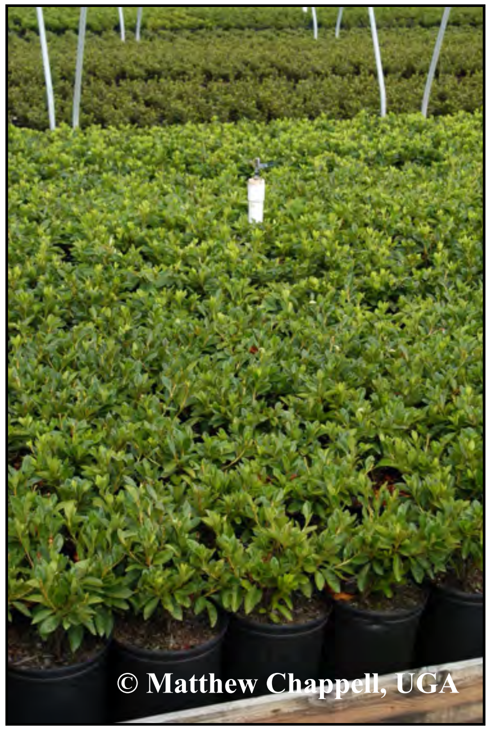


Figure 4.9 A staple crop for southeastern nursery growers is trade gallon sized azaleas, as seen here at Martin's Nursery in Semmes, AL.

Figure 4.10 Azaleas being grown under natural shade from pine trees.

inches apart within rows and 9.5 inches apart between rows, hexagonally, compared to further spacing (16 inches, 18 inches). This indicates that plant species may not be as important an indicator of water use as plant spacing (Yeager and Million, 2014).

Substrates and Amendments
Most growers producing species of container-grown rhododendron in the southeast use pine bark as the principal substrate component (Figure 4.11). Depending on the grower or the species of *Rhododendron*, various organic amendments can be added to the substrate to adjust the physical and chemical properties that contribute to growing quality plants. These amendments may include peat moss, other types of bark from species other than loblolly pine (*Pinus taeda* L.), or various composted materials, for example, composted cotton gin trash and cotton stalks. The nature of systems, bark suppliers, compost sources and feed stocks, nutrient source, climate, as well as irrigation water quality and the irrigation delivery method and volume applied all preclude the idea of a single or even several recipes of specified growing media for producing high quality plants. There is no holy grail in terms of creating the perfect growing substrate.

For example, Cole et al. (2005) amended pine bark with either nothing, peat moss or cotton gin compost in various ratios then grew *Rhododendron indicum* 'Formosa' azalea in that substrate irrigated under three different volumes of water. All azalea growth was similar in all treatments after one growing season. Root growth ratings, however, were highest when substrates were amended with 25% peat moss and lowest when amended with 50% cotton gin compost (CGC) because the water holding capacity was highest and air space lowest in CGC. Substrates amended with CGC tended to have a higher pH and more nutrients leached from the substrate than pine bark alone or pine bark amended with peat moss. Alternatively, Riley et al. (2012) found that cotton gin trash and composted cotton stocks used as an amendment to pine bark substrates increased growth of container-grown *R. obtusum* 'Sunglow' azalea as long as overhead irrigation was used in production. Plant growth was reduced when these amendments were added to whole pine tree substrates (substrates created using mostly wood-based particles) instead of pine bark only, or when substrates were watered with low volume drip irrigation. In another study, addition of 40% or more of bark of *Juniperus virginiana* as an amendment to pine bark substrates reduced growth of *Rhododendron* x'Formosa' azalea. Contributors to poor growth may have been increased airspace and total porosity, as well as lower bulk density, which might make the substrates hold less water and dry more frequently between watering (Edwards et al., 2012). Additionally, pH of the substrate was over 6.2 during growth, which might reduce availability of some nutrients.

Figure 4.11 Pine bark is the predominant substrate used in the production of *Rhododendron* spp. in the southeast.

Comparing findings between studies examining substrates is difficult because discreet physical properties are often not established among studies, nor is there a continuous spectrum of physical properties, (e.g., air space) by which to compare. Most typically, comparisons are made between the substrates themselves, due in part to the nature and properties of substrates and their amendments and the system in which they are tested. Thus, particle size, water holding capacity, and cation exchange capacity of the amendments affects the same physical properties of the final substrate. With commonly grown azaleas, like 'Formosa' or 'Sunglow' azaleas, these subtle changes might not be important in terms of producing a quality plant, but for other more difficult-to-grow species of rhododendron (e.g., *R. calendulaceum* flame azalea), management of all inputs can play a critical role.

As a rule of thumb, do not add more than 25% of any organic amendment to the substrate regardless of cost or perceived benefit without checking the resulting changes in physical or chemical properties. Additions closer to 5-10% are more typical. Additionally, if large-scale changes are made to routine production practices, run small-scale tests first using intended ratios of the product, and more importantly, determine if future supplies of organic amendments will be available consistently with similar and reliable quality from batch to batch. Otherwise, nutrient content and physical properties can change with each new shipment and cause unplanned problems without a clue to which part of the production system is contributing to the problem. Unfortunately, problems with physical properties of substrates might not begin presenting themselves visually as foliar symptoms until well into production.

Nutrient Use and Fertilization
Rhododendrons around the globe grow in diverse climates from the tropics to boreal forests and variable soil types from moist, steep river valleys, to dry, rocky outcroppings, high mountain balds, and peat-laden bogs. Mineral type, form, and availability, as well as relationship to other nutrients is important to provide the necessary nutrients for optimal growth and flower production. *Rhododendron* spp. grown in the field generally prefer

Table 4.3 Typical foliar concentrations reported for macro- and micro-nutrients measured in recently mature leaves collected at mid-season from that current season's growth of large leaved evergreen rhododendron species (ER) and various small leaved evergreen and deciduous azaleas (AZ). Sources: Mills and Jones, 1996; Cresswell and Weir, 1997; Scagel et al., 2014.

Macronutrient	ER	AZ
	Dry weight (%)	
Nitrogen	1.6-2.0	1.5-2.5
Phosphorus	0.21-0.29	0.20-0.50
Potassium	0.40-0.80	0.50-1.50
Calcium	0.80-1.40	0.50-1.50
Magnesium	0.19-0.27	0.25-1.0
Sulfur		0.20-0.50
	Parts per million	
Boron	33-47	25-75
Copper	1-3	6-25
Iron	93-245	50-250
Manganese	602-1511	40-200
Zinc	21-37	20-200

acidic soils of pH 4.8-5.5 (Bir, 1992). As pH climbs, however, micronutrients, especially iron (Fe), are essential so nutrients should be added in a chelated form if possible (McAleese and Rankin, 2000). Iron chlorosis can occur in new growth of small-leaved evergreen species deficient in the nutrient, and magnesium deficiency can occur in large leaved species in field soils of mountainous regions in the southeast. Refer to Table 4.3 for "normal' levels of nutrient concentrations in foliage of both large leaved rhododendrons and azaleas.

Nitrogen (N) is the most important macronutrient for plant growth in rhododendron because it affects growth, flowering, and uptake of other nutrients. Scagel et al. (2008) grew *R.* 'P.J.M.' evergreen rhododendron, and *R.* 'Cannon's Double' deciduous azalea with and without N while all other nutrients were made available. Increased N was associated with demand for and uptake of other nutrients. This indicates that if N is increased in nutrient applications, and all other nutrients are at their *optimum* levels, there does not have to be an associated increase in other nutrients: the plant will take nutrients up in relation to available N. However, a decrease in N through reduced fertilizer applications might affect uptake of other nutrients even if they are present at optimum levels (Scagel et al., 2008). If container plants are more than 12 months from potting date and just N is reapplied for a quick green-up, however, there could be a deficiency of other nutrients within the plant because need is increased, but supply is not.

The ammonium nitrate form of N is preferred by many azaleas (little-leaved evergreen forms) as those receiving 75% of N in the ammonium form were higher quality than those receiving more nitrate-N (Ryan, 1990). However, *R. catawbiense* (large-leaved evergreen) preferred nitrate N to either ammonium-N or urea, as long as iron was in the chelated form when pH was above 4 in field soils. High rates of nitrate N can also lead to iron deficiency in rhododendrons. Regardless of N form, however, foliar N rates of 2% caused leaf damage symptoms. Ammonium N leaches less frequently than nitrate N in soils and rhododendrons can assimilate it preferentially, if used excessively, leading to high tissue levels and subsequent damage. Sensitivity to high rates of N or different forms of N might also be sensitivity to high soluble salts in the soil. Therefore, use a fertilizer with a balance between nitrate and ammonium forms of N, or slightly more N in the ammonium form.

The notion that increased phosphorous (P) increases flower production in rhododendron has resulted in growers making late summer additions of triple super phosphate (0-46-0) to container plants to improve sale the following spring. Original reports stating that P increased flower bud formation in plants may have been misunderstood or misreported. Many other reports show no increase in flower bud formation with excess P, but do show an increase when P is added compared to soils deficient in P (Ryan, 1990, and references therein). Therefore, adding P at the lowest recommended levels for rhododendron growth is sufficient. Excess P does not increase flower bud formation; indeed it might lead to iron (Fe) deficiency chlorosis because P may bind Fe especially if substrate pH is high (Ryan, 1990).

Nitrogen is better correlated with increased bud initiation though it is difficult to separate the effect of N and P on flower bud formation. Increasing soluble N rates applied through irrigation increased flower bud initiation of container-grown *R.* 'English Roseum,' but not *R.* 'Catawbiense Boursault' (Wright, 1992). The number of flower buds was loosely correlated with the number of shoots emerging from the last flush of growth for the year. Increased flower bud initiation coincided with fewer emerging terminal shoots, which were larger in diameter and had larger leaves. Removing trusses after flowering and pruning out stems lead to increased flower bud initiation and fewer, but larger stems and leaves (Wright, 1992). Ryan (1990 and references therein) suggested that foliar tissue N levels between 1.6-2.0% were optimal for flower bud initiation across both small-leaved and large-leaved common evergreen rhododendron (deciduous azaleas were not mentioned) whether plants were grown in containers or field soil. There is some caution in raising tissue N levels because Casey and Raupp (1999a) found that leaves of *R.* 'Delaware Valley White' azalea with N levels 1.1% and 1.5% experienced increased colonization of azalea lace bug. The severity of injury, however, did not increase with increasing N. Although more lace bugs colonized this azalea cultivar, individual insects did not cause more damage than if they had colonized a plant with decreased tissue levels of N. Therefore, azaleas grown at optimum N levels for growth (Table 4.3) and reproduction can be utilized as key indicator plants when scouting for azalea lace bug: a key insect pest.

Irrigation Interactions with Nutrient Use

The combination of N and irrigation (Figure 4.12) can affect growth and flowering. Scagel et al. (2011) irrigated several rhododendron species with the same volume of water applied either all at once or in two increments (cyclic) during the day. Increasing N rates increased allocation to new leaves in *R.* 'Gibraltar' deciduous azalea and *R.* 'English Roseum,' (large leaved evergreen) and to old leaves in *R.* 'P.J.M.,' (large-leaved evergreen) which aids quality of plant material when N is sufficient. When those plants experienced drought stress, however, biomass was allocated to stems in 'Gibraltar' azalea, and to the roots in 'English Roseum' and 'P.J.M.'. Total biomass of these drought stressed plants was not affected by water volume (only allocation of biomass) while grown in containers, but flowering was affected the following year in the landscape (Scagel et al., 2014). In the deciduous azalea cultivar, water stress in containers increased vegetative growth after planting in the landscape, but decreased flower production, whereas in the evergreen cultivars, it decreased vegetative growth and generally increased flower production. When growing rhododendron at optimum nutrient levels, especially N, small amounts of drought stress experienced the summer prior to spring sales might increase flowering in large leaved evergreen species and landscape survival in deciduous species. Excessive late-season drought may induce leaf scorch or surface bleaching on foliage (Figure 4.13).

Figure 4.12 Irrigation interacts with fertilizers and can be managed to optimize growth and flowering.

Roots of small leaved evergreen azaleas can be susceptible to high salt injury and water logging from over-irrigation, therefore managing irrigation and nutrient applications are important techniques to reduce stress and ultimately pest or plant disease damage. In addition, when irrigation is managed well for container-grown plants, lower N levels than prescribed on fertilizer labels can be used to grow small-leaved evergreen azaleas. When grown with 1.0 lb N per cubic yard of substrate, plants of *R.* 'George L. Tabor' had similar root and shoot dry weights when leaching fractions were close to zero (Million et al., 2007). A leaching faction is the ratio between the water leached from the bottom of the container and the total volume that enters the container. Recording a lower leaching fraction, for example 0.1, means that approximately 10 percent of the irrigation volume that was applied consequently left the containers (or conversely, that 90% of the water remained and did not run through to leach nutrients from the container). A low leaching fraction results in more nutrients that remain available for plants, thereby requiring lower than prescribed N levels to yield adequate crop growth.

Figure 4.13 Leaf scorch or foliar bleaching can occur when plants are permitted to dry down excessively in summer.

Pruning and Landscape Plant Form

Most large-leaved evergreen rhododendrons are unlikely to readily regenerate new bud initials on old wood after pruning, so it is best to prune early in production to create a branched architecture. Shoots on young plants without flower buds grow from terminal buds and can become leggy over time if left unpruned. Leggy and heavily shaded plants can be pruned to remove the terminal leaf bud and then letting the nearby axillary buds break; a process that helps to increase branching. This is done early in production to create well-branched plants. In some years, these plants may break bud more than once, usually in the fall, and terminal vegetative buds may be removed before bud break after flowering the following year. If terminal flower buds appear on branch tips as plants mature, then terminal vegetative buds are absent and several axillary buds present will break after flowering, thus maintaining the branched effect without pruning. If plants become too leggy and mature, renewal pruning or coppicing is required to allow plants to regrow from buds at or just below ground level. Plants may not flower for a few years after coppicing.

Small leaved evergreen azaleas can be sheared just after flowering to remove deadheads and increase branching. These common plants break freely from older wood and can be maintained in almost any shape by shearing after blooms expire, but before new growth occurs. Plants break bud more than once per year, especially in the southeastern U.S., so several pruning efforts might be required to maintain desired shape (Figure 4.14)

Figure 4.14 While an extreme example, bonsai of azalea demonstrates the concept of lateral bud break that can enhance flowering.

Deciduous azaleas can be pruned in much the same way as small-leaved evergreen azaleas as they also break freely from buds on old wood. Simply shear or prune after flowering to desired shape. The natural habit of most native deciduous azaleas is more upright, open, with several long-stemmed branches originating from the base. To create more branches from seed grown or cutting grown plants, nursery workers can pinch young plants even before they are potted into containers. Repeated pinching early in production creates a well-branched architecture that appears fuller in containers (Irving, 2000).

Increased branching can also be improved by the application of plant growth regulators (PGRs). Use of PGRs depends heavily on plant development at the time of application as well as climate. Therefore, very specific instructions are usually given for PGR rate, time of year, and plant phenology during application. For example, increased branching occurred on container-grown *R.* 'PJM Compact,' *R. occidentale* deciduous azalea, and *R.* 'Girard's Variegated Hot Shot' evergreen azalea when sprayed to runoff once in late June near Portland, Oregon, with a combination of Atrinal (dikegulac-sodium) at 2000 ppm in a 1:10 dilution of Off-Shoot-O (methyl esters of fatty acids) (Ticknor and Skinner, 1991). Flower bud initiation as well as plant quality (more branching and reduced growth) may be increased in some taxa by spraying plant growth regulators (PGRs). Some PGRs have a long-term effect on flowering and growth in woody plants due to their slow growth compared to herbaceous plants. Plants may be stunted or flowers deformed as a result. Additionally, timing and dose of spray application is important and optimum levels for both can vary among the three types of rhododendrons certainly and even within those groups. The number of flower buds on five cultivars of hybrid large-leaved evergreen rhododendrons (*R.* 'Purpureum Elegans,' 'Roseum Elegans,' 'Chinoides,' 'English Roseum,' and 'Nova Zembla') were significantly increased when sprayed with as low as 50 ppm Sumagic® (uniconazole-P) in mid-July as the first, post flower flush of foliage began to harden (Bir and Conner, 1998). Application of four PGRs to two cultivars of little leaved evergreen rhododendrons, *R.* 'Johanna' and 'Blauuw's Pink', effected plant growth and flowering differently. Paclobutrazol (190 ppm) and daminozide (6375 ppm) increased the number of flowers in both cultivars as well as maintained a height to diameter ratio without pinching that is necessary for marketing. Unfortunately, daminozide caused flowers of 'Blauuw's Pink' to be deformed for three years afterward, but not flowers of 'Johanna,' a more robust cultivar (Meijona et al., 2009). Due to feasibility, the authors suggested spraying paclobutrazol only on both cultivars. The remaining PGRs, chlormequat chloride in drench and fatty acid sprays were phytotoxic, caused deformed foliage, and reduced flowering to different degrees. These results indicate that testing each PGR with various cultivars is important to both produce branching that controls growth and increase floral bud initiation.

Section 2

Abiotic Stressors

Figure 4.15 Azaleas being overwintered in a polyhouse may also be covered with cloth.

Abiotic Stressors

1. Winter injury and cold tolerance
2. Deer damage

Winter Injury and Cold Tolerance

Cold temperatures can damage more mature rhododendrons grown in containers, for example, in response to early frosts in autumn and late frosts in spring, as well as extended freezing temperatures in winter. Symptoms include root death and subsequent stem blight or wilting when growth reoccurs in large leaved evergreens, bark cracking at the base or complete stem breakage, foliage desiccation along leaf margins, flower bud abortion or loss of blooms while flowering. After intense record keeping of bud count on every rhododendron in his landscape for 17 years, Gilkey (1996) suggested anecdotally that an increase in flower initiation was correlated with bud blast or bud set the previous year due to cold temperatures. Therefore, loss of flower buds one year due to low temperature might increase flower formation the following year.

Overwintering survival of deciduous azaleas in containers has also been a perennial problem (Figure 4.15). Plants might root well from stem cuttings, but less than half of rooted plants survive overwintering (personal observations). Some reports claim that increasing growth after rooting, but before dormancy, can increase overwintering survival. Banko and Stefani (1996) tested this hypothesis by spraying 1000 ppm GA_3 (gibberellin), 10 ppm BA (benzyladenine, cytokinin), and a combination of 500 ppm GA_3 +10 ppm BA on rooted cuttings of two deciduous azaleas. The treatments did increase shoot break before dormancy in one cultivar. None of the PGR treatments, however, increased overwintering survival significantly even though plants were placed in a covered structure kept just above freezing. In a similar study with *R. flammeum* and *R. austrinum*, rooted stem cuttings were potted in August and new growth was marked prior to overwintering. Plants were divided evenly among two groups with or without new growth, and then placed into either (1) a dark, cold room maintained at 41°F (5°C) continuously; (2) a double layer, white polyethylene covered structure with an inflator fan, heating, and ventilation maintained between 41-65°F (5-18.3°C), and (3) a single layer, white polyethylene-covered structure with no supplemental heating or ventilation. The variation in day/night temperatures increased from cold room to the unventilated overwintering structure. These three environments are the most feasible to adopt for use in nursery production. Nevertheless, only 50% of plants survived overwintering regardless of species, whether or not new growth occurred prior to overwintering, or environment (LeBude, data unpublished). Many deciduous azaleas rooted in June and potted in August or September may not have enough time to become completely dormant before the first frost or freezing temperatures occur, therefore survival decreases. Other reports suggest use of high concentrations of rooting hormones necessary to stimulate rooting can decrease overwintering survival, but those theories need further scientific testing.

More cold hardy cultivars can be grown and planted to prevent damage during production and increase landscape survival in more northerly climates. A gene isolated from *R. catawbiense* plays a critical role in cryoprotection of cells during the cold hardening process. Thus, cultivars of and hybrids with *R. catawbiense* in their parentage can be more cold hardy than other species (Arora et al., 2008). Cold resistance of hybrid rhododendrons is designated by a code that indicates the lowest temperature the flower

Table 4.4 Cold hardiness code designations for large leaved evergreen hybrid rhododendrons.

Code	Temperature
H-1	-25°F (°C)
H-2	-15°F (°C)
H-3	-5°F (°C)
H-4	5°F (°C)
H-5	15°F (°C)
H-6	25°F (°C)
H-7	32°F (°C)

buds can tolerate during the winter and still open perfectly in spring (Table 4.4). Most rhododendrons grown in the southeast U.S. are in the category H-1 to H-4. Anecdotally, more cold hardy cultivars will have their leaf angles decrease (hang down from stem) earlier in winter and more acutely than those that are considered less hardy (Morsink, 2005). The USDA plant hardiness zone map designates cold hardiness of azaleas, both deciduous and evergreen. Many small–leaved evergreen azaleas are hardy to zones (6)7 to 9, while deciduous azaleas can be hardy from zones (4)5 to 9 depending on species and hybrid. Table 4.5 lists many azalea and rhododendron taxa with the lowest midwinter temperature experienced by flower buds that sustained <50% injury. These are temperatures experienced at the peak of dormancy and may not indicate the ability to survive similar temperatures experienced during early or late winter freezing events during the hardening, rehardening, or dehardening process that can occur throughout winter. Nevertheless, they can be used to generalize about relative hardiness among various taxa.

In the south, Harden (1990) suggests that heat tolerance is not needed as a virtue for survival; more important traits are disease resistance and cold hardiness. Plants need excellent drainage and high volumes of water. Therefore, a spray program dominated by Benlate and to a lesser extent, Subdue and Daconil might aid survival. Species or hybrids with *R. arboreum, R. auriculatum, R. barbatum, R. fortunei, R. triflorum, or R. taliense* have grown well in his trials. Additionally, protect plants in winter from winds by shading, barriers, or mounds of leaves or mulch.

Figure 4.16 Deer damage can be a significant problem both in commercial and landscape situations.

Deer Damage

Rhodendron indicum ‘Judge Soloman’ experienced high deer browse activity (Figure 4.16) in eastern Alabama and requires protection if these animals are nearby. Various deer repellents (Buck Off!, Deer Out™, Deer Stopper®, Plantskydd™, and PredaScent™) prevented deer browse, yet untreated control plants were also not browsed. Therefore, testing at nursery combined with prevention of deer reaching this plant is advised (Hoffman et al., 2012).

Table 4.5 Midwinter cold hardiness (°F) of flower buds among tested Azalea taxa (Pellet et al., 1986).

Azalea species & cultivars	Lowest Temp (°F) tested with < 50% injury	Lowest Temp (°F) tested with < 50% injury, and some survival	Azalea species & cultivars with some survival	Lowest Temp (°F) tested with < 50% injury	Lowest Temp (°F) tested with some survival
'Amoena Coccinea'	-8	-13	'Marathon'	-13	-13
'Annamarie'	-13	-13	'Orange Essence'	-24	-29
R. arborescens	-24	-29	'Parade'	-24	-29
'Buzzard'	-24	-29	'Pink and Sweet'	-29	-29
'Bonfire'	-29	-35	'Pink Array'	-8	-13
'Cherry Bomb'	-24	-29	'Pink Clusters'	-13	-18
'Deep Rose'	-24	-29	'Pink Rocket'	-29	-29
'Delaware Valley White'	-8	-8	'Polar Bear'	-13	-13
'Everglow'	-24	-29	'Popsicle'	-35	-35
'Frank Abbott'	-24	-29	*R. prunifolium*	-24	-29
'Gibraltar'	-24	-24	'Jane Abbott' ('Miss Jane')	-29	-35
'Golden Showers'	-24	-29	'Rainbow'	-29	-29
'Herbert'	-13	-18	'Sparkler'	-24	-29
'Hinode Giri'	-8	-8	'Stewartstonian'	-8	-8
'Hino Scarlet'	-2	-8	'Summer Time'	-29	-29
'Iridescent'	-29	-29	'Viking'	-13	-18
'Jane Abbott' ('Miss Jane')	-29	-35	'Viscocephalum'	-29	-35
'Jane Abbott' ('Peach')	-29	-40	*R. yedoense* var. *poukhanense*	-18	-24
R. kiusianum 'Album'	-18	-24	*R. yedoense* var. *alba*	-13	-13
'Lemon Drop'	-35	-35	'Weston's Innocence'	-29	-29
Light Orange-Pink'	-29	-29			

Table 4.5 (*continued*) Midwinter cold hardiness (°F) of flower buds among tested Rhododendron taxa (Pellet et al., 1986).

Rhododendron species & cultivars	Lowest Temp (°F) tested with < 50% injury	Lowest Temp (°F) tested with some survival	Rhododendron species & cultivars	Lowest Temp (°F) tested with < 50% injury	Lowest Temp (°F) tested with some survival
'Aglow'	-24	-24	'Lucky Rock'	-8	-13
'April Snow'	-18	-18	'Lunar'	-18	-24
'Arctic Cold'	-13	-18	'Mary Kittel'	-18	-24
'Balta'	-18	-24	'Molly Fordham'	-18	-24
'Big Deal'	-13	-18	'Nantucket'	-13	-18
'Big Red'	-13	-18	'Noreaster'	-18	-24
'Boule de Neige'	-13	-18	'Nova Zembla'	-18	-18
'Bristol Cream'	-13	-24	'Olga Mezitt'	-24	-24
'Burgundy Eye'	-13	-18	'Pauline Bralit'	-2	-13
'Carolina Rose'	-24	-24	'Purple Gem'	-18	-24
'Caronella'	-18	-18	'Scintillation'	-13	-18
R. catawbiense 'Album'	-18	-24	'Summer Rose'	-18	-18
R. fortunei 'Waterway'	-13	-13	'Tapestry'	-24	-29
'Francesca'	-18	-24	'Thunder'	-18	-18
'Goldsworth Yellow'	0	-2	'Tottenham' (azaleodendron)	-13	-18
'Henry's Red'	-24	-29	'Wally'	-18	-24
'Ignatius Sargent'	-13	-18	'Waltham'	-18	-24
'Laetevirens'	-2	-18	'Weston'	-13	-18
'Laurie'	-24	-24	'Weston's Pink Diamond'	-18	-18
'Llenroc'	-18	-18	'Years of Peace'	-13	-18
'Low Red Frilled'	-18	-24			

Arthropod Pest Management

Figure 4.17 Adult rhododendron stem borer, *Oberea myops*.

Azalea and broadleaf rhododendron species and cultivars grown in the southeastern U.S. have an extensive cohort of insect and mite pests that can cause significant economic crop losses and lasting aesthetic injury in nursery and landscape sites.

COMMON ARTHROPOD PESTS

1. Stem-feeding Arthropod Pests
2. Root-, Bud- and Leaf-chewing Pests
3. Root- and Foliar-feeding Weevils
4. Caterpillar Defoliators
5. Piercing-sucking Arthropod Pests

Stem-feeding Arthropod Pests

Oberea *Longhorned Beetle Borers*

Rhododendron stem borer (*Oberea myops* Haldeman), which is a pest of azalea and rhododendron (*Rhododendron* spp.), blueberry (*Vaccinium* spp.), and mountain laurel (*Kalmia* spp.) (Baker, 1994), is a 12.5 to 15.6 mm (0.5 to 0.625 in) long, slender longhorned woodboring beetle (Figure 4.17) (Coleoptera: Cerambycidae). *Oberea myops* has a yellowish brown head and thorax with two black spots on the thorax, and yellowish gray wing covers with dark outer margins (Baker, 1994). Adult azalea stem borer emerges from mid-May through mid-July and chews two rows of girdling channels about 12.5 mm (0.5 in) apart on a new-growth twig (Figure 4.18) and then inserts an egg under the bark (Figure 4.19) (Culin et al., 1993; Baker, 1994). Larvae hatch, and then bore down the twig into the main stem, tunneling into the crown and eventually into the roots where they spend the winter (Baker, 1994). Once infested, twigs and stems of deciduous azaleas rapidly wilt as larvae move downward; stems of broadleaf rhododendron take longer to wilt (Culin et al., 1993). As larvae tunnel and mature, a series of holes about 1 mm in diameter are also cut in the bark at intervals averaging about 6.2 cm apart along stems. These holes serve as points from which sawdust-like frass is pushed out (Culin et al., 1993; Baker, 1994). Larvae (Figure 4.20) feeding in broadleaf rhododendron (e.g., 'English Roseum') may complete development in either one or two years. By contrast, a few *O. myops* larvae feeding in deciduous azaleas can complete develop within a single year, a majority take two years to mature, and some individuals may require up to three years to reach adulthood (Culin et al., 1993). Tunneling within stems and holes cut for frass expulsion can lead to breakage of portions of the crown due to wind and water interactions in the plant canopy (Culin et al., 1993).

Figure 4.18 *Oberea myops* frass and girdled stems with exit holes on rhododendron stems.

Figure 4.19 Rhododendron stem borer egg laid beneath the bark.

Larvae of the dogwood twig borer (*Oberea tripunctata* Swederus) also attack azaleas and rhododendrons, blueberry, elm (*Ulmus* sp.), mountain laurel, apple and crabapple (*Malus* sp.), *Prunus sp.,* mulberry (*Morus* sp.), poplar (*Populus* sp.), sourwood (*Oxydendrum* spp.), viburnum (*Viburnum* spp.), and willow (*Salix* sp.) (Solomon, 1995).

Figure 4.20 Rhododendron stem borer larvae feed beneath the bark on rhododendron stems.

Dogwood twig borers are also cylindrical in shape and chew round exit holes through the wood and bark just prior to pupation and adult emergence. Dogwood twig borer adults are about 3 mm wide (0.12 in) and 10 to 15 mm (0.4 to 0.6 in) long. Adult beetles have dark to almost black heads with the top of the reddish thorax having three black spots that form a triangle. The yellow-to-tan elytra have a thin black line along the middle edge and a wider black line along the outer edge or side (Baker, 1994; Carter et al., 1980). In early to mid-June, adult female dogwood twig borer individuals make two encircling bands of punctures about 13 to 25 mm (0.5 to 1.0 in) apart near the branch tip. Females then make a vertical slit between the rings and insert a single egg beneath the bark flap (Solomon, 1995). After eggs hatch, larvae chew through the bark and enter the branch, then begin tunneling toward the branch tip. After a short distance, larvae turn around and bore down the center of the branch toward the main trunk (Solomon, 1995). Along the way, larvae cut a line of small, closely spaced holes in the branch so that frass can be pushed out. In fall, branch portions that contain larval tunnels will die and larvae overwinter in the hollowed out branch between two plugs of frass (Solomon, 1995). In spring, mature larvae girdle the branch from the inside out, which weakens the branches leading to visible breaks in spring. Larvae plug the openings of broken branches with frass then pupate within small chambers during April and May (Table 4.6). There is one generation per year in the south (Solomon, 1995).

Management

Tunneling by the larvae should cause tip die-back in the summer. Look for tip die-back and for sawdust-like frass being pushed out of the small holes in the branch by the larva. Prune several inches below where the larva is tunneling. Alternatively, a thin metal wire can be inserted or drop of horticultural oil can be applied into the feeding channel to destroy or suffocate the larva (Culin et al., 1993). Dispose of the branch containing the larva because the larva should continue to tunnel inside and expel the frass. Protective insecticide applications to the bark are seldom needed for this pest because damage levels rarely exceed economic-damage thresholds. If the amount of damage from this pest was objectionable over the prior year, one of the insecticides listed under wood boring beetles in Table 4.7 can be applied to the bark in the spring just prior to when egg laying is expected in your area.

Rhododendron Clearwing Borer

Synanthedon rhododendri (Beutenmüller) clearwing moth borer (Figure 4.21) is native to the U.S. and is widely distributed across the eastern half of it. *Synanthedon rhododendri* adult moths have a 10 to 15 mm (0.4 to 0.6 in) long wingspan (Solomon, 1995). The brownish black abdomen has thin yellow bands on segments 2, 4 and 5 with a wedge shaped, black anal tuft that has white scales at the margins. Brown-black legs have pale yellow scales at the joints and on the tarsi (Eichlin and Duckworth, 1988). Adult moths are active from about May through August (Snow et al., 1985). Depending upon host plant quality and environmental effects, larvae can lengthen developmental cycles to include an entire second season (Neal, 1984). Late instar larvae are about 10 mm long and larvae overwinter under bark (Figure 4.22) (Neal, 1984).

Figure 4.22 Clearwing borer larvae feed beneath the bark within rhododendron stems. Frass may be extruded from the hole.

Host plants include ericaceous mountain laurel (*Kalmia* spp.), azaleas, and rhododendrons (*Rhododendron* spp.). Large-leaf rhododendrons seem to be more preferred than azaleas (Solomon, 1995). Engelhardt (1946) reported that *S. rhododendri* infestation was most common on stems 30 cm above the soil line. Female moths deposit eggs directly on host plants and neonate larvae enter wound tissues or penetrate directly into the bark. Entire plants or infested portions of plants may become girdled, leading to initial foliar chlorosis, followed by shoot wilt, loss of plant vigor, and eventually branch or plant death (Engelhardt, 1946; Johnson and Lyon, 1991; Solomon, 1995). At the base of infested plants, scout for frass that is packed within feeding galleries beneath the bark and within woody tissues (Figure 4.23). Loose or split bark, wet spots from exuded sap, sunken lesions with callus tissues and cast pupal skins may also be visible on stems and branches.

Figure 4.21 An adult male rhododendron clearwing borer, *Synanthedon rhododendri.*

Table 4.6 Seasonal activities of the major arthropod pests of azalea and rhododendron in the mid-southern U.S., and unless otherwise noted, represent occurrence in USDA Plant Hardiness Zone 7[Z].

Arthropod Pest	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Oberea longhorned beetle borers												
Synanthedon rhododendri clearwing moth borer												
Xylosandrus germanus ambrosia beetle												
Broadnecked *Prionus* longhorned beetle borer												
Azalea bud larvae (moth)												
Cranberry rootworm												
Black vine weevil					P							
Fuller rose beetle												
Twolined Japanese weevil												
Leaf-feeding caterpillars												
Azalea leafminer (moth)												
Oleander scale						P		P		P		
Peony scale												
Tea scale			P			P		P		P		
Azalea bark scale												
Cottony azalea scale							P					
Cottony camellia scale					P							
Azalea and rhododendron lace bugs												
Graphocephala leafhoppers												
Four-lined plant bug												
Greenhouse thrips[Y]												
Azalea and rhododendron whiteflies												
Southern red mite			P									
Twospotted spider mite												

[Z] Depicted activity may be early or later than shown depending on location. Activities represented in the table are scale insect crawler emergence, as well as adult or nymphal activity of the most common insect and mite pests. Peaks in arthropod pest abundance, when reported, are denoted by 'P'.
[Y] Greenhouse thrips are primarily encountered as pests in protected culture, thus potentially have season-long activity.

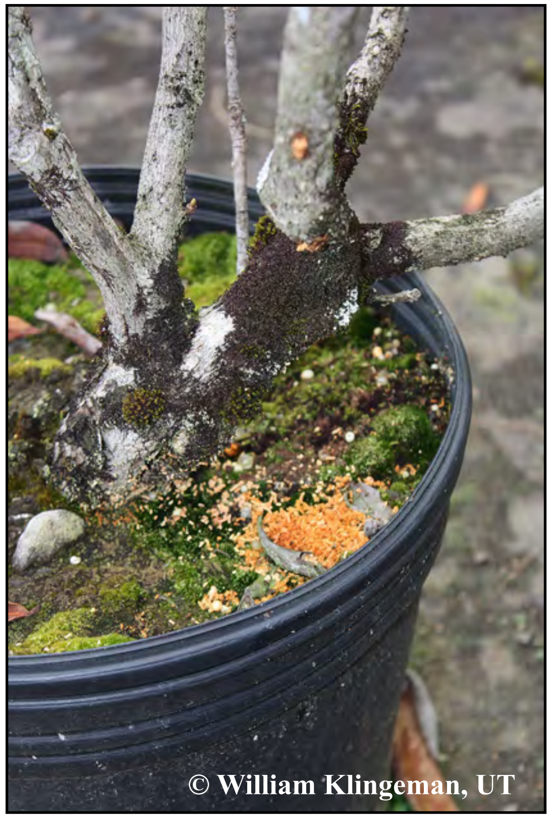


Figure 4.23 Frass from larvae will indicate that larval borers are present. Scout for signs of frass below stems and on ground fabric as part of an active scouting program.

Management.

In nurseries and managed landscapes where azalea and rhododendron are actively growing and healthy, clearwing moth pests are unlikely to be present in large numbers. Scout nurseries and landscapes for evidence of frass beneath stems of infested host plants. Pupae and eggs are seldom found while scouting. When frass indicating active wound sites are detected, presence of live larvae can be confirmed by direct extraction from bark or stems. The thin-skinned larvae are frequently destroyed during removal, making further identification impossible. Once actively feeding larvae are confirmed, other infested stems can often be pruned out and discarded. Adult male clearwing moths of both species can also be trapped and identified using pheromone lures paired with bucket (e.g. Multipher or Universal style, Shaver et al., 1991) or sticky boards within delta or wing traps. Woodpeckers and at least two parasitoid wasp species help limit natural populations of *S. rhododendri* larvae. For these reasons, active efforts to control clearwing borers are seldom necessary (Solomon, 1995). When population levels and past injury to ornamental crops warrant insecticidal controls, several options are available (Table 4.7).

Black Stem Borer and Other Ambrosia Beetles

The black stem borer, *Xylosandrus germanus* (Blandford) (Figure 4.24), is occasionally a damaging pest on azaleas and rhododendrons in nursery and landscape habitats. Native to Asia, the black stem borer, which was introduced to the U.S. in the 1930s, is currently distributed across the eastern and midwestern U.S. and has been found in Oregon (Rabaglia et al., 2006). *Xylosandrus crassiusculus* (Motschulsky), the granulate ambrosia beetle (Figure 4.25) is a closely related ambrosia beetle species that may occasionally attack *Rhododendron* spp. and is the predominant ambrosia borer affecting nursery grown trees in the southeastern U.S. (Gill, 2013). Regardless, the black stem borer appears to be more aggressive in its northern range. The biology of the two species is similar. Adult females become active as early as March in South Carolina (JH Chong, unpublished data), mid-March in Virginia and Tennessee, and late-April in Ohio (Reding et al., 2010). Males, which are small and flightless, remain in the gallery. Adult female beetle colonizers bore into the branches and trunk of susceptible plants. As females excavate galleries (Figure 4.26), they push out sawdust forming a frass tube, or pencil. These frass extrusions can be used as a monitoring and diagnostic characteristic, yet are also fragile and can be lost to rain and wind. Colonizing *X. germanus* introduce ambrosia fungi into host plant galleries as a food resource for their developing beetle larvae. Black stem borers can be collected with ethanol-baited traps throughout the year. Attacks to susceptible plants typically occur following the spring flights.

Figure 4.24 Adult black twig beetle.

Figure 4.25 Granulate ambrosia beetle adult and frass tubes extruded from infested plant material.

Management

Ambrosia beetle flight activity can be monitored with ethanol-baited traps beginning in late-February in the southeastern U.S. Homemade soda bottle traps, Lindgren funnel traps, and modified Japanese beetle traps all work (Oliver et al., 2004), and can be baited with 70% ethanol liquids and suspensions of hand sanitizing gels. Ethanol is released using wicks of cotton string or cotton balls. Slow-release ethanol pouches can also be purchased. Traps should be hung 0.5 to 1.7 m (1.5 to 5.6 ft) above ground (Reding et al., 2010) and checked regularly. Once attacked, shrubs become more attractive to ambrosia beetles, thus may be left as trap crops for 3-4 weeks after initial early season attacks, in order to draw beetles away from other shrubs. If this approach is taken, it will be critical to remove and discard these reservoir hosts within a month to protect other healthy trees and shrubs. The attacked plants are best buried or burned. No effective biological control option is currently

Figure 4.26 Black twig beetle gallery and injury found within an infested stem.

available. Systemic neonicotinoids are not effective in preventing ambrosia beetles attacks and will not kill ambrosia beetles that are already in galleries (Chong, unpublished data). Chemical management of black stem borer beetles relies on preventive treatments. Pyrethroids, particularly bifenthrin and permethrin, can repel ambrosia beetle attacks for up to 10 days; longest when ambient temperatures are moderate (Mizell and Riddle, 2004). Insecticide solutions should be uniformly sprayed to cover all trunk and branch surfaces.

Figure 4.27 An adult broadneck prionus longhorned beetle (*Prionus laticollis*).

Root, Bud- and Leaf-chewing Pests

Broadnecked Prionus *Longhorned Beetles*

Larvae of broadnecked prionus longhorned beetles, *Prionus laticollis* (Drury), are root-feeding pests of many native, ornamental and fruit tree host plant species, including apples and occasionally, azalea and rhododendron species (Benham and Farrar, 1976; Solomon, 1995). Native and widespread across the eastern U.S., *Prionus laticollis* larvae may take 3 to 4 years to complete their life cycle (Benham and Farrar, 1976). Adult beetles (Figure 4.27) are short-lived and range from 22 to 44 mm (0.87 to 1.7 in) long. Females are much larger than males. Beetles emerge in late June to early July, lay eggs on soil adjacent to host plants, and then larvae tunnel into root tissues to feed (Benham and Farrar, 1976). Cream-colored larvae are about 9 cm long when mature (Solomon, 1995). Larvae exit roots to pupate up to 84 cm deep in soil (Benham and Farrar, 1976).

Management

A tachinid fly parasite has been observed attacking prepupae and entomopathogenic bacteria have been identified (Benham and Farrar, 1976). Within roots, larvae may encounter and cannibalize each other (Benham and Farrar, 1976). A general attractant comprised of synthesized 3,5-dimethyldodecanoic acid will serve as a monitoring tool to detect male activity by this and other *Prionus* species in the environment (Barbour et al., 2011).

Azalea Bud Larvae

Azalea bud larvae, *Orthosia hibisci* Guenée, also called speckled green fruitworm (Figure 4.28), is a native, minor pest of native azaleas and is widespread across the U.S. This moth species is primarily a foliar and fruit-feeding pest of deciduous fruit and ornamental trees. Larval feeding damage to petals and flower buds will yield unattractive flowers once buds expand. There is one generation per year with adult moths active in April and May.

Figure 4.28 An azalea bud larvae (Speckled green fruitworm, *Orthosia hibisci*) pictured on oak.

Management

Insecticide sprays should be applied in early spring as soon as *O. hibisci* larvae are found (Galle, 1987; Judd and Gardiner, 1997; Alston, 2010).

Cranberry Rootworm

Cranberry rootworm (*Rhadopterus picipes* Olivier) is a chrysomelid leaf-feeding beetle widely distributed east of the Mississippi River. Adults (Figure 4.29) and larvae of this nursery and landscape pest have an extremely broad host plant range. In addition to azalea and rhododendron species, cranberry rootworm feed on camellia (*Camellia* spp.), cherry laurel (*Prunus laurocerasus* L.), golden raintree (*Koelreuteria paniculata* Laxm.), Japanese holly (*Ilex crenata* Thunb.), Chinese holly (*Ilex cornuta* Lindl. & Paxton), magnolia (*Magnolia* spp.), oaks (*Quercus* spp.), redtips (*Photinia* spp.), *Rhododendron* spp., roses (*Rosa* spp.), silver maple (*Acer saccharinum* L.), sycamore (*Platanus* spp.), sumac (*Rhus* spp.), sassafras (*Sassafras albidum* (Nutt.) Nees.), viburnum (*Viburnum* spp.), and Virginia creeper (*Parthenocissus quinquefolia* (L.) Planch.). Adult beetles are about 0.51 cm (0.2 in) long, dark brown, and shiny. Adults bear one brood per year and emerge from late April to mid-May in Mississippi (Harman, 1931; Johnson and Lyon, 1991; Oliver and Chapin, 1980). Adults are nocturnal feeders, create c-shaped curving holes in leaves (Figure 4.30), and hide in leaf litter and debris during the day. Adults feed for about 2 weeks after emergence, and then seek refuge in leaf litter where they deposit eggs. Larvae (Figure 4.31) are active root feeders (Oliver and Chapin, 1980).

Figure 4.29 An adult cranberry rootworm shown on the face of a penny for scale.

Figure 4.30 Typical curving c-shaped feeding injury caused cranberry rootworm adult beetles.

Figure 4.31 Cranberry rootworm larvae.

Altica Flea Beetles

Altica sp. flea beetles may also occasionally be a leaf-feeding beetle pest on broadleaf rhododendrons. Adult beetles are about 0.5cm (0.2 in) long with metallic blue or blue green thorax and elytra (Figure 4.32). When disturbed, beetles use expanded femoral muscles to spring away from the host plant. Larvae (Figure 4.33) cause the majority of foliar injury, primarily by skeletonizing narrow channels along lower surfaces.

Management

Pesticides may provide control when leaf beetles are actively feeding (Table 4.7). A fraction of applications can also be directed toward leaf litter and debris beneath the affected plant where nocturnally active beetles will hide. Entomopathogenic nematodes including *Heterorhabditis bacteriophora* Poinar and *Steinernema scarabei* (Stock & Koppenhöfer) can control cranberry rootworm larvae (Polavarapu, 1999; van Tol and Raupp, 2005). When disturbed, beetles use expanded femoral muscles to spring away from the host plant. Larvae (Figure 4.33) cause the majority of foliar injury, primarily by skeletonizing narrow channels along lower surfaces of newly emerged leaves. Upper leaf portions of affected leaves may become transparent and then decompose leaving empty shot holes and channels that often have a thin necrotic margin (Figure 4.34).

Figure 4.32 An adult *Altica* flea beetle feeding on *Rhododendron catawbiense* foliage.

Figure 4.33 A larva of an *Altica* flea beetle feeding shallowly on the underside of a rhododendron leaf; a casebearer moth larvae is also seen below.

Figure 4.34 Skeletonized portions of leaf tissues consumed by *Altica* flea beetles may decompose and fall away, leaving window-paned areas with necrotic margins.

Table 4.7 Insecticides available to control common insect pests of *Rhododendron* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Armored scales	Soft scales	White-flies	Thrips	Leafhoppers &/or planthoppers[7]	Lace-bugs	Weevils	Leaf-feeding beetles	Cater-pillars	Leaf-miner (moth)	Clear-wing borers	Ambrosia beetles	Long-horned (round-headed) borers	Spider mites
1A	Acetylcholinesterase inhibitors	Carbamates	carbaryl[8]	Sevin SL	L, N, G	12	x	x	x	x	x[lh]	x	x	x	x	x			x	
			methiocarb	Mesurol 75W	N, G	24				x										x
1B[20]	Acetylcholinesterase inhibitors	Organophosphates	acephate[8,9]	Orthene T&O[9]	L, N, G	24	x	x	x	x	x[lh]	x	x	x	x	x				
				Lepitect[10]	L, N, G	24	x	x	x	x	x[lh]	x	x	x	x	x				x
				Precise GN[9]	G, N	12						x	x	x						
			chlorpyrifos	Dursban 50W[9]	N	24	x	x	x	x	x[lh]	x	x	x	x	x	x	x	x	x
				DuraGuard ME[9]	N, G	24	x	x		x	x[lh]		x	x	x					x
			dicrotophos	Inject-A-Cide B[11]	L	N/A	x	x			x[lh]			x	x		x		x	x
			dimethoate	Dimethoate 4E[9], 4EC[9]	N	14 days[21]	x	x		x	x[lh]	x		x	x	x				x
			malathion	Malathion 5EC[9,10]	L	12	x	x		x	x[lh]	x			x	x				x
			methidathion	Supracide 2E[9,19]	N	30 days[21]	x	x												
			oxydemeton methyl	Harpoon[11,9]	L	0	x	x						x	x		x		x	x
				MSR Spray Concentrate[12,19]	N	10 days[21]				x			x		x	x				x
			trichlorfon	Dylox 420 SL	L	N/A									x					
3A[20]	Sodium channel modulators	Pyrethroids / Pyrethrins	bifenthrin[8]	Attain TR	G	12	x	x		x					x					x
				Menace GC	L, N, G	12	x	x		x	x[lh]	x	x	x	x			x		x
				Onyx	L	N/A	x	x		x	x[lh]	x		x	x	x	x	x		x
				OnyxPro	L, N, I	12	x	x		x	x[lh]	x		x	x	x	x	x		x
				Talstar S Select	N, G	12	x	x		x	x[lh]	x	x	x	x					x
				Talstar Nursery G	N	12							x							
			cyfluthrin	Decathlon[14]	L, N, G, I	12	x	x		x	x[lh]	x	x	x	x					
			beta-cyfluthrin	Tempo Ultra WP[14]	L, I	N/A	x	x		x	x[lh]	x	x	x	x					
				Tempo SC Ultra[14]	L, I	N/A	x	x		x	x[lh]	x	x	x	x					

Table 4.7 (*continued*) Insecticides available to control common insect pests of *Rhododendron* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Armored scales	Soft scales	White-flies	Thrips	Leafhoppers &/or planthoppers[7]	Lace-bugs	Weevils	Leaf-feeding beetles	Cater-pillars	Leaf-miner (moth)	Clear-wing borers	Ambrosia beetles	Long-horned (round-headed) borers	Spider mites
3A[20]	Sodium channel modulators	Pyrethroids / Pyrethrins	*lambda*-cyhalothrin	Demand	L	N/A	x	x	x	x	x	x	x	x	x	x				x
				Scimitar CS; Scimitar GC	L ; L, N, G	24; 24	x	x		x	x[lh]	x	x	x	x	x				x
			cypermethrin	Demon WP	L, I	N/A								x	x					
			deltamethrin	DeltaGard G[14]	L, I	N/A					x[lh]		x		x					
			fenpropathrin	Tame 2.4 EC[9]	N, G, I	24	x			x	x[lh]	x	x		x	x				x
			tau-fluvalinate	Mavrik Aquaflow	L, N, G, I	12				x	x[lh]		x	x	x					x
			permethrin	Astro	L, G, I	12				x	x[lh]	x	x		x	x	x			
				Permethrin Pro	L, I	N/A				x	x[lh]	x	x		x	x	x			
				Perm-Up 3.2 EC	L, N, G, I	12				x	x[lh]	x	x		x	x	x			
			pyrethrins	Tersus	N, G	12	x	x		x	x[lh, ph]	x	x	x	x	x	x	x	x	x
				Pyganic	N, G	12	x	x		x	x[lh]			x	x					x
			pyrethrum	Pyrethrum TR	N, G	12	x	x		x	x[lh]			x	x					x
3A + 4A	Sodium channel modulators	Pyrethroids + Neonicotinoids	bifenthrin + clothianidin	Aloft LC G, LC SC	L	N/A	x	x		x	x[lh]	x	x		x	x				
			bifenthrin + imidacloprid	Allectus SC	L, I	N/A	x	x		x	x[lh]	x	x	x	x	x			x	x
			cyfluthrin + imidacloprid	Discus N/G	N, G, I	12	x	x		x	x[lh]	x	x	x	x	x			x	
			lambda-cyhalothrin + thiamethoxam	Tandem	L	N/A	x	x		x	x[lh]	x	x	x	x	x				
			zeta-cypermethrin + bifenthrin + imidacloprid	Triple Crown T&O	L, I	N/A	x	x	x	x	x[lh]	x	x	x	x			x		x
3A + 27A	Sodium channel modulators	Pyrethroids + Piperonyl - butoxide (PBO)	pyrethrins + piperonyl butoxide[8]	Pyreth-It	N, G	12				x	x[lh]			x	x					
4A[25]	Nicotinic acetylcholine receptor agonists	Neonicotinoids	acetamiprid	TriStar 8.5 SL[9,10,14]	L, N, G	12	x	x	x	x	x[lh]		x	x	x	x				
			clothianidin	Arena 0.25 G[10]	L, I	12			x											
				Arena 50 WDG[10]	L, I	12			x											
			dinotefuran	Safari 2G[10]	L, N, G, I	12	x	x	x	x[15]	x[lh]	x	x	x					x	
				Safari 20 SG[10]	L, N, G, I	12	x	x	x	x[15]	x[lh]	x	x	x					x	
				Zylam Liquid[10]	L	N/A	x	x	x	x[15]	x[lh]	x	x	x	x	x			x	
				Transtect 70 WSP[10]	L	N/A	x	x	x	x[15]	x[lh]	x	x	x	x	x	x		x	

Table 4.7 (*continued*) Insecticides available to control common insect pests of *Rhododendron* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Armored scales	Soft scales	White-flies	Thrips	Leafhoppers &/or planthoppers[7]	Lace-bugs	Weevils	Leaf-feeding beetles	Cater-pillars	Leaf-miner (moth)	Clear-wing borers	Ambrosia beetles	Long-horned (round-headed) borers	Spider mites
4A[25]	Nicotinic acetylcholine receptor agonists	Neonicotinoids	imidacloprid[11]	Xytect 75WSP; 2F[10]	L, N, G, I	12	x[15]	x	x	x[15]	x[lh, ph]	x	x	x					x	
				Marathon II[10]	N, G, I	12	x[15]	x	x	x		x	x	x		x			x	
				Marathon 60WP[10]	N, G, I	12	x[15]	x	x	x	x[lh]	x	x	x		x			x	
			thiamethoxam	Merit[10]	L, I	N/A	x[15]	x	x	x[15]	x[lh, ph]	x	x	x		x			x	
				CoreTect[10]	L, I	N/A	x[15]	x	x	x	x[lh, ph]	x	x	x		x			x	
				Discus Tablets[10]	N, G, I	12	x[15]	x	x	x	x[lh]	x	x	x		x			x	
				Flagship 25WG[10]	N, G, I	12		x	x	x	x[lh]	x	x	x		x				
				Meridian 0.33G[10]	L, I	N/A		x	x	x	x[lh]	x	x	x	x	x				
				Meridian 25WG[10]	L, I	N/A		x	x	x	x[lh]	x	x	x	x	x				
4C + 5[20]	Nicotinic acetylcholine receptor agonists	Sulfoxaflor + Spinosyns	sulfoxaflor + spinetoram	XXpire[16,19]	L, N, G	12	x		x	x		x		x	x					x
5	Nicotinic acetylcholine receptor allosteric activators	Spinosyns	spinosad	Conserve SC	L, N, G	4				x				x	x					x
				Entrust	L, N, G	4				x				x	x					x
6[20]	Chloride channel activators	Avermectins, Milbemycins	abamectin[9]	Lucid, Avid[9]	L, N, G	12			x	x										x
				Aracinate TM[11]	L, N, G, I	N/A	x	x		x		x	x	x	x	x	x			x
			emamectin benzoate	Arbormectin	L	N/A	x								x	x		x	x	x
				Enfold	N	12									x	x				x
				Tree-äge	L	N/A	x								x	x	x		x	
			milbemectin	Ultiflora	N	12														x
6 + 20D	Chloride channel activators	Avermectins, Milbemycins	abamectin + bifenazate	Sirocco[14]	L, N, G, I	12			x	x										x
7A	Juvenile hormone mimics	Juvenile hormone analogues	s-kinoprene	Enstar AQ	G, I	4	x	x	x	x	x[lh]					x				
7B	Juvenile hormone mimics	Fenoxycarb	fenoxycarb	Preclude TR	G	12	x	x	x	x			x		x					x
7C	Juvenile hormone mimics	Pyriproxyfen	pyriproxifen	Distance IGR[9]	L, N, G, I	12	x	x	x											
				Fulcrum	L, N, G, I	12	x	x	x											
8C	Misc. non-specific (multi-site) inhibitors	Fluorides	cryolite (sodium alumino-fluoride)	Kryocide[5]	L	N/A							x	x	x					
8D	Misc. non-specific (multi-site) inhibitors	Borates	sodium tetraborohydrate decahydrate	Prev-AM Ultra	N, G	12	x	x	x											x

Table 4.7 (*continued*) Insecticides available to control common insect pests of *Rhododendron* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Armored scales	Soft scales	White-flies	Thrips	Leafhoppers &/or planthoppers[7]	Lace-bugs	Weevils	Leaf-feeding beetles	Cater-pillars	Leaf-miner (moth)	Clear-wing borers	Ambrosia beetles	Long-horned (round-headed) borers	Spider mites
9B[20]	Selective homopteran feeding blockers	Pymetrozine	pymetrozine	Endeavor[9,14]	L, N, G, I	12			x											
		Pyrifluquinazon	pyrifluquin-azon	Rycar	G	12			x	x	x[lh]									
10A[20]	Mite growth inhibitors	Clofentezine	clofentazine	Ovation SC[9]	N, G	12														x
		Hexythiazox	hexythiazox	Hexygon DF[9,14]	L, N, G, I	12														x
10B[20]	Chitin biosynthesis inhibition	Etoxazole	etoxazole	Beethoven TR	G	24			x											x
				TetraSan 5 WDG	L, N, G, I	12														x
11	Microbial disruptors of insect midgut membranes	*Bacillus thuringiensis* (Bt) & insecticidal proteins	*Bt* subsp. *aizawai*	XenTari	L, N, G, I	4									x					
			Bt subsp. *kurstaki*[8]	Dipel Pro DF	L, N, G, I	4									x					
			Bt subsp. *tenebrioni*	Novodor	L, N	4								x						
12B	Inhibitors of mitochondrial ATP synthase	Organotin miticides	fenbutatin-oxide	Meraz, ProMite 50WP[9]	L, N, G	48														x
13	Uncouplers of oxidative phospho-rylation via disruption of the proton gradient	Chlorfenapyr	chlorfenapyr	Pylon[9]	G	12				x					x					x
15 [25]	Inhibitors of chitin biosynthesis, type 0	Benzoylureas	diflubenzuron	Adept[9]	G	12			x						x	x				
				Dimilin 25W	L, N	12							x		x	x				
				Dimilin 4L	L, N	12							x		x	x				
			novaluron	Pedestal[9]	N, G	12			x	x					x	x[15]				
16	Inhibitors of chitin biosynthesis, type 1	Buprofezin	buprofezin	Talus 70DF	L, N, G	12	x	x	x		x[lh, ph]									
18	Ecdysone receptor agonists	Diacylhydrazines	metho-xyfenozide	Intrepid 2F	L, N, G, I	4									x					
			tebufenozide	Confirm 2F[17]	N	4									x					
20B[25]	Mitochondrial complex III electron transport inhibitors	Acequinocyl	acequinocyl	Shuttle 15 SC	L, I	12														x
				Shuttle-O[9]	N, G	12														x
20D[25]	Inhibition of complex III at the Qo site	Bifenazate	bifenazate	Floramite SC	L, N, G, I	12														x
				Floramite SC/LS	L	N/A														x
21A	Mitochondrial complex I electron transport inhibitors	METI acaricides and insecticides	fenazaquin	Magus[9]	L, N, G, I	12			x											x
			fenpyroximate	Akari 5SC	N, G, I	12														x
			pyridaben	Sanmite	N, G	12			x		x[lh (juvenile)]									x
			tolfenpyrad	Hachi-Hachi SC[9]	G	12	x	x	x	x	x[lh]				x					

Table 4.7 (*continued*) Insecticides available to control common insect pests of *Rhododendron* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Armored scales	Soft scales	White-flies	Thrips	Leafhoppers &/or planthoppers[7]	Lace-bugs	Weevils	Leaf-feeding beetles	Cater-pillars	Leaf-miner (moth)	Clear-wing borers	Ambrosia beetles	Long-horned (round-headed) borers	Spider mites
22A	Voltage-dependent sodium channel blockers	Indoxacarb	indoxacarb	Provaunt	L	N/A					x[lh]				x					
23	Inhibitors of acetyl CoA carboxylase	Tetronic and Tetramic acid derivatives	spiromesifen	Forbid 4F	L	N/A			x											x
				Judo (Savate)[9]	N, G	12			x											x
				Kontos[9]	N, G, I	24	x	x	x	x	x[lh]									x
25	Mitochon-drial complex II electron transport inhibitors	Beta-ketonitrile derivatives	cyflumetofen	Sultan	L, N, G, I	12														x
28	Ryanodine receptor modulators	Diamides	chlorantraniliprole	Acelepryn	L, I	N/A		x[18]				x			x		x			
				Acelepryn G	L, I	N/A									x					
			cyantraniliprole	Mainspring	G, I, L	4		x	x	x		x		x	x					
29	Chorodontal organ modulators (selective homopteran feeding blockers	Flonicamid	flonicamid	Aria[9,16]	L, N, G	12	x	x	x	x	x[lh]									
Unknown[20]	Unknown	Azadirachtin	azadirachtin[8]	Azatin O	L, N, G, I	4	x	x	x	x	x[lh]		x	x	x	x	x		x	
				Azatin XL	N, G, I	4			x	x	x[lh]		x	x	x	x				
				Azatrol EC	L, N, G, I	4	x	x	x	x	x[lh]	x	x	x	x	x			x	x
				Ornazin 3% EC	L, N, G, I	12	x	x	x	x	x[lh]	x	x	x	x	x	x		x	
				TreeAzin[11]	L, N, G	until dry								x	x	x				
		Dicofol	dicofol	Kelthane[19]	L, N, G	48														x
		Pyridalyl	pyridalyl	Overture 35 WP	G	12				x					x					
Not classified	Various		*Beauveria bassiana*[8]	BotaniGard ES; Mycotrol ESO; Mycotrol WPO	L, N, G, I	4			x	x	x[lh]	x	x	x						x
				BotaniGard 22 WP	L, N, G, I	4			x	x		x	x							
				Naturalis-L	L, N, G	4			x	x	x[lh]		x	x	x					x
			Chromobacterium subtsugae	Grandevo PTO	L, N, G	4			x	x		x			x					x

Table 4.7 (*continued*) Insecticides available to control common insect pests of *Rhododendron* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Armored scales	Soft scales	White-flies	Thrips	Leafhoppers &/or planthoppers[7]	Lace-bugs	Weevils	Leaf-feeding beetles	Cater-pillars	Leaf-miner (moth)	Clear-wing borers	Ambrosia beetles	Long-horned (round-headed) borers	Spider mites
Not classified	Various		*Heterorhabditis bacteriophera*	LarvaNem	L	N/A							x							
				Exhibitline H	N	N/A							x[13]							
				NemaShield HB	L, N, G, I	N/A							x		x					
			Isaria formosorosea	NoFly	G	12			x	x	x[lh]			x						
				Preferal	L, N, G	4			x	x			x		x	x				x
			Metarhizium anisopliae	Met52, Tick-EX	L, N, G	4				x			x							
			Steinernema sp.	TigraNem	L, N, G, I	N/A						x								
			Steinernema carpocapsae	Millenium	L, N, G, I	N/A							x		x		x			
				Nematac C	L, N, G, I	N/A				x					x					
				Exhibitline SC	L, N, G, I	N/A									x					
				Carpocapsae-System	L, N, G, I	N/A									x					
				Capsanem	L, N, G, I	N/A									x					
			Steinernema feltiae	NemaSys, *Steinernema*-System	L, N, G, I	N/A				x										
				NemaShield	L, N, G, I	N/A				x										
			Steinernema feltiae	EntoNem	L, N, G, I	N/A				x			x							
			Steinernema kraussei	Nemasys L	L, N, G, I	N/A							x							
				Kraussei-System	N	N/A							x							
			horticultural oil[8,9]	Ultra-Pure Oil, TriTek	L, N, G, I	4	x	x	x	x	x[lh]	x		x	x	x				x
			insecticidal soap[8,9]	M-Pede[9]	L, N, G, I	12	x	x	x	x	x[lh]	x		x	x					x
			neem oil[8]	Trilogy	L, N	4		x	x	x										x
				Triact 70[9]	L, N, G, I	4	x	x	x		x[lh]									
			capsicum extract, garlic oil, soybean oil	Captivia	L,N	4			x	x	x[lh]				x					x

Table 4.7 (*continued*) Insecticides available to control common insect pests of *Rhododendron* spp. in commercial production environments.

Footnotes and Supplemental Information:

1. Pests listed on pesticide labels are subject to change. Within states and counties, products may have additional permitted uses by 2(ee) and Special Local Need allowances. Consult your County Extension Agent or State Agency to determine if other uses are allowable. The label should always be consulted to confirm that chart-listed pests still appear on the label. Check the product labels for specific site restriction information, notes on application, sensitive plant species and specific target pest species.
2. Use site information is provided for reference only: L = landscape; N = nursery; G = greenhouse; I = interiorscape. t/s*Products listed for use in L, N, G or I sites may include active ingredients in differently labeled products that are designated for use in turfgrass or sod production use sites. Mention of those products is beyond the scope of this resource guide. Products listed herein may not be legal or appropriate for site uses in turfgrass or sod production.
3. IRAC Code designations and Related Modes of Action are explained at the Insecticide Resistance Action Committee Database 2016. (IRAC 2016; http://www.irac-online.org/teams/mode-of-action/).
4. Trade names of products are provided as examples only. No endorsement of mentioned product nor criticism of unmentioned products is intended.
5. Products may not be registered or renewed for use in all southeastern U.S. states. Consult your state's Department of Agriculture to confirm legal use of products in your state.
6. Re-Entry Interval (REI) designations apply to agricultural (nursery) uses. Within REI column, 'N/A' is used to indicate products with Landscape and/or Interiorscape site uses and that present Non-Agricultural Use Requirements. Consult product labels for specific details required for compliance within those application conditions and use sites, including those for which REI listings do not apply. Additionally, some product labels may list site uses that are beyond the scope of this publication (e.g., sod farms, silvicultural & Christmas tree nurseries, pastures, rights-of-way, etc.). These site uses may involve Agricultural Use Requirements that list an REI not presented here.
7. lh = leafhopper; ph = planthopper (juv. = juveniles, or nymphs)
8. Multiple formulations and trade names of the same active ingredients are available. Labels among these products often differ in legally allowable uses (regarding sites, pest, REI, etc.); representative example(s) presented.
9. See product label for information about potential crop phytotoxicity, known plant sensitivity, and how to test for phytotoxicity.
10. Check label for additional restrictions (e.g., on the number of times the product can be applied in a growing season or year, or additional application restrictions within permitted use sites; and crop types or flowering status that may not be legally treated.)
11. Product formulated for tree or shrub injection; specialized equipment may be required.
12. Use of handheld application equipment is prohibited. For use only on seedling trees and non-bearing fruit trees in commercial nurseries.
13. Label includes white grub and weevil larval control in nursery containers.
14. Intended for commercial/professional use only. Not intended for homeowner use.
15. Label-indicated efficacy against target pest group is primarily by population suppression.
16. Product labels may restrict landscape uses only to commercial landscapes. Residential landscapes are not a permitted application site.
17. Only for use on Christmas trees.
18. On the Acelepryn Section 2ee label.
19. Product uses cancelled or in process of Federal cancellation or review. Use of existing stocks of any end-use product may be allowable beyond the cancellation date. Consult your extension agent or state agency for information about allowable use and conditions for proper disposal.
20. Applications of products from these footnoted IRAC groups are optimized when not preceded by/or followed by products in subsequently noted IRAC Group(s), particularly when managing the indicated pest. Suggested rotation combinations to avoid include serial treatments with a.i. products from: IRAC 1B, 3A, 6 (western flower thrips); IRAC 4A, 4B, 4C (green peach aphid); IRAC 4A and 9B/9C (glasshouse and silverleaf whiteflies); IRAC 10A, 10B, 15 (Panonychus [red and citrus] spider mites); IRAC 20B and 20D (= bifenazate) (twospotted spider mite). Previously unclassified within IRAC, bifenazate has been moved to IRAC 20D and has similar mode of action as other mitochondrial complex III electron transport inhibitors.
21. Some REI periods are in days, rather than hours. Please consult the insecticide label for specific REI period.

Root- and Foliar-Feeding Weevils

Several different weevil species (Coleoptera: Curculionidae) feed on roots and foliage of azalea and rhododendron species and are common pests in outdoor nurseries and landscapes across the southern U.S.

Figure 4.35 Adult black vine weevils are nocturnal and move up from the ground to feed on foliage at night.

Black Vine Weevil

Otiorhynchus sulcatus (F.), the black vine weevil, was introduced to the U.S. from northern Europe in 1835 (Smith, 1932) and feeds on more than 150 other plant species including yews, camellias, euonymus, and hollies (Reding, 2008). Adult beetles are 9.5 mm (0.38 in) long with a short broad snout. The elytral wing covers on adult beetles are dull gray to black and include numerous small golden spots that are visible with magnification as pits surrounded by short golden hairs (Figure 4.35). Flightless adult females become active at night, climb onto the host plant and cause mainly aesthetic damage to nursery crops by creating hemispherical or elongated notches on the edges of the leaves (Figure 4.36). Damage can become severe (Figure 4.37). Legless, white, C-shaped larvae with brown heads also feed on roots, eventually girdling the roots and stem. Older larvae overwinter in the soil; there is only one generation per year. In spring, adults emerge from puparia in soil and immediately begin feeding (Cowels, 2005). Black vine weevil populations consist solely of females; reproduction is by parthenogenesis, with unfertilized eggs becoming new females. Egg laying occurs about one month after adult emergence and continues until winter, with each female depositing up to an average of 880 eggs depending on the host plant species (Cowles, 2003; Fisher, 2006). Adult females drop the eggs on the soil surface or insert them into plant crevices (Cowles, 2005; Smith, 1932).

Figure 4.36 Small numbers of black vine weevil adults may cause only minimal aesthetic injury.

Figure 4.37 Severe black vine weevil feeding injury to *Rhododendron* leaves.

Twobanded Japanese Weevil

Pseudocneorhinus (formerly *Callirhopalus*) *bifasciatus* (Roehlofs), is a non-native pest that has become widely distributed throughout the eastern and midwestern U.S. following its importation from Asia in about 1914 (Wheeler and Boyd, 2005). This species feeds on azaleas as well as a wide variety of ornamental and invasive plants (Marrone and Zepp, 1979; Baker, 1994; Boyd and Wheeler, 2004). Flightless, adult twobanded Japanese weevils are about 5 mm (0.2 in) long with a pear-shaped, light brown body that has indistinct white-lined bands on the fused elytra (Figure 4.38). Adult beetles emerge from the soil pupal cells between May and early July, then feed on new leaves or shoots during the day and drop to the ground when disturbed (Allen, 1959; Baker, 1994). Up to five eggs are deposited into folded leaf margins of grass blades and other plant foliage that is pressed closed through action of the legs (Zepp, 1978). Adult host plant influences female fecundity, with up to 355 eggs possible with *Rosa multiflora* (Thunb.) used as a host plant (Maier, 1983). Larvae are about 7.5 to 8.5 mm long and feed on host plant roots. There is one generation per year (Allen, 1959).

Fuller Rose Beetle

Naupactus (formerly *Pantormorus*) *cervinus* Boheman, is another polyphagous weevil species (Chadwick 1965) that is distributed throughout the U.S. Fuller rose beetle adults are 6 to 8.5 mm (0.2 to 0.3 in) long, with a short broad snout and a brown-grey body speckled with white patches (Figure 4.39). There is generally one generation per year with larvae overwintering in soil. In Florida, however, two generations have been reported (Woodruff and Bullock, 1979). Adults emerge in May and feed on buds and cutting notches on the leaves (Woodruff and Bullock, 1979). During the 3 to 8 month lifespan, individual females lay more than 200 eggs that are adhered in groups to plant materials and habitat substrates. After hatching, the larvae drop to the ground, burrow up to 61 cm (2 ft) deep in soil and feed on plant roots (Masakim et al., 1996).

Figure 4.38 Twobanded Japanese weevil adults are flightless and about 5 mm long.

Management of Weevils

Because weevil species that attack azaleas and rhododendrons can feed and sustain their populations on many other ornamental plant

Figure 4.39 A Fuller rose beetle adult.

species, management efforts should extend across the entire nursery or landscape system. Prevention should begin with close inspection of plant material shipped into nurseries, retail, or wholesale receiving yards. Infested plants or shipments should be quarantined away from susceptible host plants until treated. Larvae may be present but difficult to detect within containers and root systems of ornamental nursery stock. Where sufficient volumes and nutrition from roots exists, larvae may not move far from these resources, so may not encounter lethal levels of chemical control products (Cowles, 2001). Nocturnally active weevils also become active within shipping containers and be active or apparent on foliage upon inspection of newly delivered plant stock (Cowles, 2003).

Isolated plants in landscapes may benefit from barriers and bands that use sticky materials, like Tanglefoot, to keep beetles away from foliage (Helm, 2001). When observed, adults should be removed by hand and destroyed (Masiuk, 2003). Regardless, these cultural and physical methods are likely to be too labor intensive for commercial nurseries. Good cultural practices should also include sanitation efforts to remove leaf litter and crop debris that provides places for weevils to hide, pupate and lay eggs.

Landscapes and nurseries can be scouted for adult weevils by monitoring susceptible plant species for foliar leaf notching, particularly on the lower branches and leaves. Because weevils are mostly nocturnal, adults may not be directly observed during monitoring activity. Adult emergence in spring and seasonally active populations can be detected using 1:1 mixture of (Z)-2-pentenol and methyl eugenol, which will attract black vine weevil to traps (van Tol et al., 2012). In Ohio, adult black vine weevils emerge when black locust and yellowwood are in full flower and mountain laurel and multiflora rose are first flowering (Herms, 2004).

Adult and larval ground beetles (Coleoptera: Carabidae) are effective predators of adult and larval weevils and weevil eggs and pupae in nursery and landscape habitats. Chemical pesticides severely impact beneficial carabid beetle survival, so should be limited in use to help conserve existing populations (Cowles, 2003).

Chemical management of black vine weevils is challenged by ability of this and other weevil species populations to tolerate or avoid insecticides containing acephate,

Figure 4.40 A late instar azalea caterpillar (*Datana major*) larvae feeding on foliage.

bendiocarb, bifenthrin, carbaryl, chlorpyrifos, endosulfan and fenpropathin (Cowles, 2003). Where still effective and necessary to limit crop injury, chemical applications should begin after adult emergence in May. For adult control, topical applications of pyrethroids and azadirachtin should be applied at least twice, about two to three weeks apart, following adult emergence. Systemic neonicotinoid insecticides applied as a soil drench to containerized or field-grown plants are effective in reducing the numbers of larvae as well as feeding damage by the adults (Reding and Persad, 2009; Reding and Ranger, 2011). Bifenthrin can provide three or more years of efficacy when incorporated into potting media and can be used as a replacement for the more hazardous chlorpyrifos for root ball dipping (Cowles, 2001; 2003).

Entomophathogenic nematodes are among the more effective tools for managing weevil larvae in soils and potting substrates. Drenches using commercially available *Steinernema carpocapsae, Steinernema feltiae, Steinernema kraussei* and *Heterorhabditis bacteriophora* species can be made to thoroughly wetted soil or substrates in May and late August (Cowles, 1997; Grewal, 2012). *Hetetorhabditis* species disperse readily within potting media, so may be more efficient at reducing the abundance of black vine weevil larvae (Ansari and Butt, 2011). *Beauveria bassiana* is an entomopathogenic fungus that can also be applied to moist soil and substrates and will provide adult and larvae control (Anon., 2004).

Caterpillar Defoliators

A few caterpillar pests, including azalea caterpillars (*Datana major* Grote & Robinson) (Figure 4.40) and yellownecked caterpillars [*Datana ministra* (Drury)] on azaleas and obliquebanded leafrollers [*Choristoneura rosaceana* (Harris)] on rhododendrons, can occasionally defoliate small portions of shrub foliage (Johnson and Lyon, 1991). Other caterpillars are occasional pests of rhododendrons (Figure 4.41; Figure 4.42). Early caterpillar instars start out feeding on the top layer of the leaf or chewing small holes in the leaves during their early stages. As they grow, they will often devour the entire leaf or only leave sections of the midvein. The obliquebanded leafroller rolls the leaf into a tube and feeds inside the leaf. Damage from caterpillars can occur throughout the growing season (Table 4.6).

Management

In commercial production systems, feeding by caterpillar defoliators is generally limited to small portions of the canopy on individual plants. In turn, preventative chemical

Figure 4.41 A larva of an unidentified caterpillar feeding on *Rhododendron catawbiense* foliage.

Figure 4.42 An unidentified caterpillar feeding on deciduous azalea foliage. Note the small, black fecal pellets betraying activity of the feeding larva and can be used as a scouting aid.

management is seldom necessary. Periodic scouting will be sufficient to enable managers to detect caterpillar feeding damage early enough to be managed by hand. Where pest populations are atypically large or aesthetic decline warrants active treatment, infested plants can be scouted and infested specimens spot sprayed with insecticides (Table 4.7).

Azalea Leaf Miner

Azalea leaf miner, *Caloptilia azaleela* (Brants) is an Asian azalea moth pest introduced into the U.S. from Europe in about 1910 (Valley, 1975) that has become widespread across the range of evergreen azaleas (Johnson and Lyon, 1991). In Florida, small yellow- and purple-marked adult moths (Figure 4.43) are active all season (Deckle, 2013) with activity peaking between January and March (Mizell and Schiffhauer, 1991). Individual females live less than a week. Moths lay about 5.3 eggs per day along mid-veins on undersides of leaves (Mizell and Schiffhauer, 1991). Injury to azaleas is caused by larvae feeding within leaf layers, exiting after the second or third instar (Davis, 1987) then continuing to feed while protected within rolled and tied tips of single leaves. Larvae spin a pupal case tied to the lower leaf surface after up to five instars (Mizell and Schiffhauer, 1991).

Figure 4.43 An adult azalea leaf miner (*Caloptilia azaleela*) will lay eggs on azalea foliage. Larvae hatch and feed within rolled and tied azalea leaf

Management

A limited trial, which examined 15 evergreen azalea cultivars suggested that smaller-leaved 'Coral Bell', 'Gumpo White' and 'Gumpo Pink' azaleas are least susceptible hosts, while 'Delaware Valley White', 'George Lindley Taber' 'White Cascade', 'Mrs. G. G. Gerbing' and 'Red Ruffle' azaleas are susceptible to azalea leaf miner (Mizell and Schiffhauer, 1991). In addition, nine different hymenopteran parasitoid species were found to be active against larvae and pupae in Florida. Several effective chemical control options are available (Mizell and Schiffhauer, 1991) (Table 4.7).

Piercing-sucking arthropod pests

Armored Scale Insects

Oleander scale, *Aspidiotus nerii* Bouché (Figure 4.44), is a polyphagous armored scale pest reported to feed on representative plants in at least 86 plant families (Miller and Davidson, 2005), including azaleas and rhododendrons (Johnson and Lyon, 1991). It is widespread in the U.S., persisting across seasons in the southern U.S. At least three generations per year occur with scales overwintering primarily as newly mature females. Crawlers emerge in about May and settle to feed on both upper and lower leaf surfaces (Miller and Davidson, 2005). *Aphytis* spp. parasitoid wasps may help to control *Aspidiotus nerii* populations and a parthogenetic strain is used to rear commercial numbers of *Aphytis melinus* wasps (Hymenoptera: Aphelinidae) for biological control efforts in citrus (González-Zamora et al., 2012). Larvae and adult predaceous thrips, *Aleurodothrips fasciapennis* (Franklin) were found on Cumberland Island in Georgia feeding on adult oleander scale infesting sago palm (*Cycas revoluta* Thunb.) (Beshear and Nakahara, 1975).

Figure 4.44 An oleander scale, *Aspidiotus nerii*.

Peony scale, *Pseudaonidia paeoniae* (Cockerell), sometimes called the Japanese camellia scale (Figure 4.45), is a small (2.5 mm; 0.1 in) grayish brown scale that feeds on twigs and branches of rhododendron species, causing sunken bark and eventually branch dieback. Peony scale occurs in the eastern U.S. and also in California (Garcia Morales et al., 2016). In the southern U.S., it is also a pest of camellias and hollies (Nash, 1973; Baker, 1980). Crawler emergence of the peony scale was reported in April in Georgia (Tippins et al., 1977). A single generation per year is typical (Miller and Davidson, 2005), while overlapping generations of all life stages can be found in greenhouses in Maryland (Stoetzel, 1975).

Figure 4.45 A close up of a peony scale, Pseudaonidia paeoniae, on a camellia twig. The grey test, or covering, has been flipped up to show the insect feeding beneath.

Figure 4.46 Moderate (A) to severe (B) yellow blotching on camellia leaves after an infestation (C) of tea scale on the underside of camellia leaves. All life stages of tea scale overview (D) and closeup (E), and tea scale predation by lady beetle larvae (F).

Tea scale*, Fiorinia theaea* Green was introduced to the U.S. from Asia (Cooper and Oetting, 1987), and has become widespread from Florida to Massachusetts, west to Texas with populations in California (Garcia Morales et al., 2016). Beside rhododendrons, tea scale also infests boxwood (*Buxus* spp.), camellia (*Camellia* spp.), euonymus (*Euonymus* spp.) and hollies (*Ilex* spp.) (Garcia Morales et al., 2016). Nymphs and adults of tea scale (Figure 4.46) feed on leaf undersides, removing cell contents with stylet mouthparts and causing stipples and yellow blotches where cell walls remain around emptied cells (Figure 4.46). All life stages of tea scales are present on host plants throughout the year (Munir and Sailer, 1985). Generations require 60-70 days to complete development, depending on temperature (English and Turnipseed, 1940), so up to three overlapping generations may co-occur in southern locations. Tiny yellow crawlers (Figure 4.46) emerge from the first generation during the months of about February in Georgia (Hodges and Braman, 2004) and April in Maryland (Gill, 2011). After emerging from beneath the female tests, crawlers disperse to new feeding sites on old and new leaves. The first crawler stadium is about 14 days, then the second instar lasts about 6 days before nymphs mature to adult females. Males develop through three additional instars, taking about 20 days to become becoming adults (Figure 4.46) (Munir and Sailer, 1985). Adult females are about 15 mm (0.6 in) long with yellow bodies covered by elongated, brown, single-ridged tests. Each female produces 17 to 43 eggs within a month and eggs hatch in about 2 weeks (Chiu and Kouskolekas, 1980; Munir and Sailer, 1985).

Soft Scale Insects

Scale insects use piercing-sucking mouthparts to feed on the phloem of plants. Different from the armored scales, soft scales (as well as mealybugs and aphids) produce large volumes of carbohydrate-rich honeydew as they feed. Feeding by a large number of nymphs can cause slight yellowing of the leaves. Sooty mold associated with honeydew from soft scales reduces the aesthetic value of plants and also reduces plant photosynthesis. Under heavy infestations, plant growth rate is reduced and leads to crown thinning and branch dieback (Baker, 1980).

Azalea bark scale, *Acanthococcus azaleae* (Comstock) (formerly *Eriococcus azaleae*), is a soft scale pest that feeds in the crotch of azalea and rhododendron branches (Figure 4.47) and also infests andromeda (*Pieris japonica*), and other plant species (Rosetta, 2006). Adult female azalea bark scales are tiny, red and have short legs and antennae and long slender sucking mouthparts. Female scales are covered by a white egg sac, or ovisac, made of matted waxy filaments (Baker, 1980) that cannot be separated from the body without killing the insect. Azalea bark scale has two generations in the southeastern U.S. (Baker, 1980). In North Carolina, the adult females fill the ovisac with red eggs in late April and eggs hatch in about 3 weeks (Table 4.6). Nymphs mature during summer, producing eggs for a second generation in September. Azalea bark scales overwinter as late instar nymphs that mature into adults in spring (Baker, 1980).

Figure 4.47 Azalea bark scale, *Acanthococcus azaleae*, on stems of azalea.

Cottony azalea scale (*Pulvinaria ericicola* McConnell) has been found on azalea and rhododendrons, and blueberries (*Vaccinium* sp.) (Polavarapu et al., 2000) and appears to be widespread across the eastern U.S. (Johnson and Lyon, 1991). Female scales produce eggs in June in Maryland, and scales overwinter as fertilized immature females (Johnson and Lyon, 1991).

Cottony camellia scale, *Pulvinaria floccifera* (Westwood), is a soft scale (Hemiptera: Coccidae) that feeds on camellia, holly, yew (*Taxus* spp., thus also called cottony yew/taxus scale), maple (*Acer* spp.) and other plant species from 35 plant families, including *Rhododendron* species (Garcia Morales et al., 2016). Cottony camellia scale is distributed throughout the eastern and midwestern U.S. and west to Texas, with populations in California and the Pacific Northwest (Garcia Morales et al., 2016). Before egg production, adult females (Figure 4.48) are about 3 mm (0.12 in) long, elongated oval, yellowish brown in the back, with dark brown margin. Adults produce hundreds of reddish or purplish eggs in white, waxy, fluffy ovisacs (about twice the length of female) on the underside of leaves

Figure 4.48 Cottony Camellia scale adult female overwintering.

in May (Figure 4.49). Crawlers emerge in June in Virginia (Day, 2009). There is only one generation per year.

Management of Scale Insects
Scale management is most successful when contact insecticide applications coincide with crawler activity. Scout trees by surveying the trunk and scaffold branches, especially at branch collars. Use double-sided sticky tape on branches to detect crawler activity.

Natural enemies that include insect pathogens, predators and parasitoids, usually maintain scale insect populations at low population levels. Cooper and Oetting (1987) reported three parasitoids of tea and camellia scales. Hodges and Braman (2004) reported five and six predator and parasitoid species of Indian wax scale and tea scale, respectively, in Georgia. In Florida, about 13.5 percent of the tea scale population is lost to parasitoids and predators annually (Munir and Sailer, 1985).

Populations and activities of the arthropod predators and parasitoids can be disrupted, however, by use of broad-spectrum, contact insecticides like acephate, carbaryl, chlorpyrifos and pyrethroids. Contact insecticide use can often result in scale outbreaks because these broad-spectrum pesticides often kill natural enemies while not controlling mature armored scales (McClure, 1977; Merritt et al., 1983; Raupp et al., 2001). Reduced-risk insecticides such as insect growth regulators and some neonicotinoids are alternatives to pyrethroid and organophosphate insecticides (Table 4.7) (Rebek and Sadof, 2003; Raupp et al., 2006; Frank and Sadof, 2011). Soft scale species can be controlled with imidacloprid, whereas imidacloprid does not kill armored scale and can actually increase its abundance (Rebek and Sadof, 2003; Sadof and Sclar, 2000).

Figure 4.49 Cottony camellia scale adult female laying the white ovisac.

Systemic insecticides including neonicotinoids are more effective against nymphs and adult scale insects that are feeding on leaves (e.g., tea scale and cottony camellia scale) than those feeding on the twigs, branches or trunks. For scale insects that are feeding on woody plant tissues (e.g., azalea bark scale and peony scale), chemical management is best combined with repeated topical sprays of horticultural oil. Applications of dormant oil after leaf drop and before bud swell is also effective in reducing the severity of infestations, particularly of armored scales. Insect growth regulators (e.g., buprofezin and pyriproxyfen), spirotetramat and the neonicotinoids can be timed to coincide with, or shortly after, peak crawler emergence.

Azalea and Rhododendron Lace Bugs
Native to Asia and imported into New Jersey on nursery stock from Japan in about 1915 (Weiss, 1916; Drake and Ruhoff, 1965), azalea lace bug (*Stephanitis pyrioides* Scott) is the most economically significant pest of azaleas (Raupp et al., 1985; Nair and Braman, 2012). The species is widespread across the eastern U.S. and southern Canada (Johnson and Lyon, 1991; Nair and Braman, 2012). Since 2008, *S. pyrioides* has also become established in parts of the Pacific Northwest (Rosetta, 2013). The indigenous rhododendron lace bug, *Stephanitis rhododendri* (Horváth), is widely distributed across the eastern and central U.S. and southern Canada (Drake and Ruhoff, 1965), and is found in Washington and Oregon (Johnson and Lyon, 1991).

Figure 4.50 Adult male and female rhododendron lace bugs on leaf undersides. Eggs are deposited into leaf tissues beneath the black tarry frass spots.

Adults of both species have opaque, brown-patched wings latticed with lace-like patterns (Figure 4.50; Figure 4.51). Adults are about 2.8 to 3.3 mm long, with females slightly larger (Shen et al., 1985). Spiny dark nymphs pass through 4 (*S. rhododendri*) or 5 (*S. pyrioides*) instars ranging from 0.1 to 1.8 mm (0.004 to 0.07) long and wing pads become visible upon the fourth molt (Johnson, 1936; Shen et al., 1985; Shaefer and Panizzi, 2000).

Figure 4.51 Adult azalea lace bugs look very similar to rhododendron lace bugs, yet are more prevalent on azalea shrubs in the eastern U.S.

Stephanitis species overwinter primarily as eggs inserted into host plant leaves, generally inserted along the leaf mid- and secondary veins (Bailey, 1951; Johnson and Lyon, 1991; Balsdon et al., 1993; Neal and Bentz, 1997), yet *S. pyrioides* may persist in winter as active adults from North

Carolina southward (Horn et al., 1979; Mead, 1967; Braman et al., 1992; Nalepa and Baker, 1994). Eggs are deposited beneath a tar-like, black cap of fecal material (Johnson and Lyon, 1991; Balsdon et al., 1993; Neal and Bentz, 1997). At high population densities, newly hatched *Stephanitis* nymphs may become stuck in excremental residues and die (Dunbar, 1977; Neal, 1988).

When calculating degree-days for azalea lace bug development, threshold temperatures differ for eggs (10.2°C; 50.4°F) and nymphs (12.2°C; 54.0°F), versus time to complete development (11.2°C; 52.2°F) (Braman et al., 1992). Consequently, eggs require an accumulated total of 213.1 degree-days to hatch, nymphal development requires 179.2 degree-days, and completed development, from oviposition to newly eclosed adult, requires 394 degree-days. Plus, female azalea lace bugs required an additional 75 degree-days as a pre-ovipositional period required to achieve egg viability (Braman et al., 1992). Field-collected populations of azalea lace bugs present a 2.7:1 female/male sex ratio (Braman et al., 1992) and lace bugs can live as long as 250 days at 20.6°C (69°F) (Neal and Douglass, 1988). Mated individual female lace bugs can lay an average of 350 eggs in 73 days at 26.1°C (80°F), and 350 eggs in 121 days at 20.6°C (69°F) (Neal and Douglass, 1988). *Stephanitis rhododendri* have just one generation per year in more northern parts of their range, with two generations possible from New Jersey southwards (Dickerson, 1917; Johnson, 1936). Bailey (1951) reported two to three annual generations of *S. pyrioides* in the New England states. In Maryland, four annual generations are known (Neal and Bentz, 1997; Neal and Douglass, 1988) and this pattern persists into north and central Georgia (Braman et al., 1992). Regardless, the number of generations achieved within a season may be influenced by plant stress, in addition to seasonal temperature patterns (e.g., Neal, 1985).

Figure 4.52 Severe feeding injury by adult and nymph azalea and rhododendron lace bugs can cause overall bleaching of leaf tissues caused by direct removal of cell contents by the piercing-sucking insects.

Foliar injury to rhododendrons and azaleas occurs when nymphal and adult lace bugs insert stylet mouthparts into stomates on lower leaf surfaces and remove leaf cellular contents during feeding (Figure 4.52) (Mead, 1967; Ishihara and Kawai, 1981; Johnson and Lyon, 1991). Most injury on plants is caused by feeding of second and third generations of lace bug nymphs (Galle, 1987). Azalea lace bugs prefer to feed on mature older leaves, and either avoid or seldom move onto young, tender foliage (Johnson, 1936; Bailey, 1951). Chlorotic stipples result when palisade parenchymal cells in the leaf mesophyll are drained of both chloroplasts and cellular contents (Ishihara and Kawai, 1981; Buntin et al., 1996). Both photosynthesis in the remaining cells and leaf respiration are also reduced, likely due to stomatal closure in injured leaves (Buntin et al., 1996). Female azalea lace bug feeding causes more injury than that of individual adult males or nymphs (Neal and Douglass, 1988; Buntin et al., 1996). Measured across two years, neither azalea flower number, leaf or stem dry mass, new shoot length or number, nor leaf or stem dry mass among new shoots differed when up to 14 percent of shrub canopy was injured by azalea lace bug feeding (Klingeman et al., 2001b). Although long-term effects of azalea lace bug feeding injury have not been adequately quantified (Buntin et al., 1996), leaves may drop prematurely, plant vigor may be reduced leading to potential plant death (Bailey, 1951; Mead, 1967; Nalepa and Baker, 1994).

Management

Consumers perceive and relate to azalea lace bug feeding differently at hypothetical point-of-purchase versus if established in the landscape. Survey and live-plant assessments of lace bug injury to 'Buccaneer' azaleas in #3 containers confirmed that retail consumers associate higher levels of plant feeding injury with diminished plant quality, yet are not able to attribute the cause of stippling to azalea lace bugs (Oliver and Alverson, 1990). A follow-up study validated grower and consumer recognition of injury to 'Delaware Valley White' azaleas when less than 2 percent of foliar leaf surfaces are stippled (Klingeman et al., 2000a; b), and half of respondents indicated that they would be unwilling to purchase the plants once more than 10 percent of an azalea canopy has achieved *S. pyrioides* foliar feeding injury levels of two percent or more leaf area. Still, about half state they would not treat injured azaleas in their landscapes until more than 43 percent of the shrub canopy displays foliage having at least 2 percent foliar stippling (Klingeman et al., 2000a).

A majority of rhododendrons and azaleas will support lace bug populations when directly challenged (Braman and Pendley, 1992; Schultz, 1993; Wang et al., 1998; Kirker et al. 2008). An evaluation of host plant acceptance among 20 evergreen azalea types indicated that cuttings from all tested cultivars would provide food and egg deposition resources to azalea lace bugs. Stippling injury to leaves of all cultivars affected about 40 percent of foliar surface area, with 'Hino Crimson' and 'Tradition' cultivar injuries exceeding 70 percent of the leaf surface area. 'Macrantha Pink' had leaf injury to just 12 percent of the leaf surface and lower numbers of eggs than all but cultivars 'Elsie Lee' and 'Kathy'. Neither flower color nor abaxial leaf texture explained host plant preference by azalea lace bug (Schultz, 1993).

In both choice and no-choice laboratory bioassays run with 33 different evergreen azalea cultivars, 'Watchet' Robin Hill hybrid, 'Fashion' Glenn Dale hybrid, and 'Kelly Marie' Tom Dodd hybrid azalea were consistently rated among the top ten cultivars with the highest level of fecal production and eggs laid per lace bug per day, indicating that these are competent hosts plants for *S. pyrioides*. Conversely, 'Fourth of July' azalea is a wild-type evergreen azalea of unknown parentage and foliage had the least feces and egg tallies across choice and no-choice studies. Encore Autumn™ 'Twist' azalea was also consistently among the three least preferred azalea cultivars. Encore Autumn™ 'Twist' and Encore Autumn™ 'Sangria' cultivars were developed as branch sports from Encore Autumn™ 'Royalty', a 'Georgia Giant' by 'Fourth of July' hybrid. All three of these cultivars were consistently among the ten least preferred azaleas (Kirker et al., 2008). Although other cultivars containing 'Fourth of July' as a parent demonstrated variable levels of feeding preference and egg deposition, presence of dense foliar trichomes and a sticky leaf surface in 'Fourth of July', which are absent in 'Watchet', suggests that these characteristics, and not interaction with leaf stomatal size, may help confer resistance to azalea lace bug (Kirker et al., 2008). Deciduous azalea species, present a full range in resistance with *Rhododendron canescens* (Michaux) Sweet and *R. periclymenoides* (Michaux) Shinners most resistant to feeding (Wang et al, 1998; Chappell and Robacker, 2006). Evidence indicates that leaf pubescence, in combination with plant exudates and epicuticular wax composition, (including ursolic and oleanic acids and α– and β–amyrin) likely function to exclude or deter feeding and egg deposition by azalea lace bugs (Balsdon et al., 1995; Wang et al.,1998; Chappell and Robacker, 2006). These chemical constituents vary within season (Wang et al., 1999), and their presence and function among evergreen azalea cultivars has not been thoroughly assessed.

Plant nutrition, for example when azalea growth is altered by use of plant growth regulators (Coffelt and Schultz, 1988), may influence host plant preference (Casey and Raupp, 1999a; b) by altering epicuticular lipid composition (Clark, 2000), yet has not influenced lace bug fitness (Casey and Raupp, 1999a; b). Azaleas transplanted into areas with high sunlight exposure have greater incidence of lace bug injury (White, 1933; Trumbule et al., 1995; Trumbule and Denno, 1995; Shrewsbury and Raupp, 2006; Linderman and Benson, 2014) and azaleas grown in shade are more tolerant of lace bug infestation (Bentz, 2003). When sun versus shade condition was evaluated for azaleas grown in containers, however, levels of oviposition, egg hatch, and foliar feeding by azalea lace bugs did not differ (Kintz and Alverson, 1999).

Lace bugs are preyed upon by many different natural enemy arthropods. A complex landscape habitat structure including annuals, perennials, shrubs, and both understory and canopy trees, provides shade yet also supports greater numbers of generalist predators including natural enemies of azalea lace bugs (Hatley and McMahon, 1980; Riechert and Bishop, 1990; Trumbule et al., 1995; Leddy, 1996; Raupp et al., 2001; Stewart et al., 2002a; Raupp and Shrewsbury, 2005; Shrewsbury and Raupp, 2006).

Japanese plant bug, *Stethoconus japonicus* Scott, was first reported in Beltsville, Maryland in 1985 (Henry et al., 1986) and is a predatory mirid that feeds on azalea lace bugs (Neal et al., 1991). When *S. japonicus* eggs are deposited into leaf midveins, higher leaf moisture yields faster egg maturation than for eggs that are laid in stem tissues of new shoots (Neal et al., 1991; Neal and Haldemann, 1992). Consequently, asynchronous egg hatch results in later-maturing *S. japonicus* individuals that emerge in time to feed on second-generation *S. pyrioides,* thus avoiding pesticides used to control first-generation lace bugs (Neal and Haldemann, 1992). Another predaceous mirid, *Rhinocapsus vanduzeei* Uhler, ranges from Ontario south to North Carolina and west to Missouri (Wheeler and Herring, 1979).

Rhinocapsus vanduzeei nymphs and adults feed on rhododendron stamens, damaging pollen and causing filaments to atrophy (Wheeler and Herring, 1979). Yet, fifth instar *R. vanduzeei* consumed 37% of azalea lace bugs presented in no-choice consumption tests (Braman and Beshear, 1994). In central Georgia, *R. vanduzeei* emergent nymphs were detected in deciduous azaleas in mid-April and peak adult occurrence was observed in mid-May (Braman and Beshear, 1994).

Green lacewings and spiders are important generalist predators that limit lace bug populations in landscape and nursery habitats. *Chrysoperla carnea* and *C. rufilabris* are effective landscape predators of plant pests including azalea lace bugs (Shrewsbury and Smith-Fiola, 2000; Stewart et al., 2002a; Rinehart and Boyd, 2006). Anyphaenid leaf foraging spiders, *Anyphaena celer* (Hentz), and thomisid crab spiders were among the predators most frequently encountered in complex landscapes (Leddy, 1996; Stewart et al., 2002b; Shrewsbury and Raupp, 2006).

Anagrus takeyanus Gordh is a mymarid wasp and egg parasite to *Stephanitis* lace bug species including andromeda lace bug, *S. takeyai* Drake and Maa and *S. pyrioides* (Gordh and Dunbar, 1977; Braman et al., 1992; Balsdon et al., 1993). Wasps are encountered in the eastern U.S. from Maryland to Florida (Balsdon et al., 1993; Balsdon et al.,1996; Leddy, 1996). In Georgia, *A. takeyanus* parasitism rates varied from 3 to 48% of eggs, with wasps persisting after treatments with chemical pesticides (Balsdon et al., 1993; Braman et al., 1992).

Many chemical pesticides are registered for use against azalea lace bugs on azaleas and rhododendrons and can be used to manage lace bugs before they reach adulthood and lay more eggs (Table 4.7). Insecticidal soaps, horticultural oils, neem oil extract, and sharp streams of water have also been recommended as safer alternatives to traditional chemical control of lace bugs (White, 1933; Balsdon et al., 1993; Gill and Raupp, 1988; Gill and Raupp, 1989). Spray foliage undersides with water to dislodge lace bugs. Azaleas should be sprayed as the lace bugs are hatching. A second spray should be applied in seven days and then as needed throughout the summer (Galle, 1987).

Leafhoppers

Redbanded leafhopper, *Graphocephala coccinea* Forster, and rhododendron leafhopper, *G. fennahi* Young, are U.S. native and widespread pests that feed on azaleas and rhododendrons throughout their range (Wheeler and Valley, 1980a; Wheeler and Valley, 1980b; Johnson and Lyon, 1991). Both species are brightly colored, with green and red stripes extending from markings on the head across forewings that cover the entire pale yellow abdomen (Figure 4.53). Final instar nymphs (Figure 4.54) and adults of both species are about 8 mm (0.31 in) long (Johnson and Lyon, 1991).

Figure 4.53 A redbanded leafhopper adult (*Graphocephala coccinea*).

Figure 4.54 Leafhopper nymphs on the underside of *Rhododendron catawbiense* foliage.

Although *G. fennahi* appears more host restricted preferring azaleas and rhododendrons (Wheeler and Valley, 1980b), *G. coccinea* also feeds on *Kalmia latifolia*, forsythia, privet, rose, and about 50 other plant species (Wheeler and Valley, 1980a; Johnson and Lyon, 1991). Leafhoppers insert eggs into the spongy mesophyll of lower leaf surfaces, leaving circular oviposition scars about 4 mm (0.16 in) in circumference (Johnson and Lyon, 1991). Feeding injury occurs when stylet mouthparts pierce cells from undersides of leaves enabling cellular contents to be withdrawn and leaving cream- to silver-colored stipples that coalesce with time. Cellular damage and perhaps salivary toxins inserted during expansion of newly formed foliage can cause foliar distortion with leaves becoming brittle. Evidence remains unclear if leafhoppers can vector bud blight pathogens to *Rhododendron* spp., for example in the Pacific Northwest U.S. (Glawe and Hummel, 2006), however *G. coccinea* can vector bacterial leaf scorch, *Xylella fastidiosa* Wells et al. to other host plants in nurseries and landscapes (Huang, 2007).

In a Maryland evaluation of red maple trees, *G. coccinea* was most abundant in June (Bentz and Townsend, 2005). There are two generations per year with adults in Pennsylvania appearing in late April and mid July (Wheeler and Valley, 1980a; b) and remaining active in landscapes until first frost (Johnson and Lyon, 1991).

Management

Leafhoppers can be monitored using sticky cards and sweep netting. Look for cast skins from nymphs attached to leaf undersides. Nymphs are strong jumpers and will spring from host plants when disturbed. Pyrethroids and contact pesticides work, though may need to be re-applied at 2 week intervals while *Graphocephala* species are confirmed to be feeding and causing aesthetic injury to rhododendrons and azaleas. A drench with systemic neonicotinoids will provide extended season control and will help conserve natural enemies.

Four-lined Plant Bug

Figure 4.55 Characteristic damage from four-lined plant bug feeding on leaves (A) with adult (inset) and (B) older instar with black wing pads each with a yellow stripe.

The four-lined plant bug, *Poecilocapsus lineatus* (Fabricius) (Heteroptera: Miridae) (Figure 4.55), is a U.S. native and widespread generalist feeder with host plants including more than 250 herbaceous and woody ornamental plants species across 57 plant families. In addition to azaleas, deutzia (*Deutzia* spp.), dogwood (*Cornus* spp.), forsythia (*Forsythia* spp.), Amur maple (*Acer ginnala* Maxim.), rose (*Rosa* spp.), sumac (*Rhus* spp.),weigela (*Weigela florida* Thunb.) and viburnum (*Viburnum* spp.), are occasionally attacked (Johnson and Lyon, 1991). Foliar injury is caused by lacerate-flush feeding, in which barbed stylet mouthpart tips are used to slice and tear plant cells beneath the leaf surface. Softer leaf, bud and flower parts are preferred, but seed, stem and root tissues may also be affected (Schaefer and Panzini 2000; Schuh and Slater, 1995). The feeding pocket in a leaf is flooded with saliva and digestive enzymes that liquefy rigid parts of the ruptured cells before fluids are ingested. Injury may take several days to become widely apparent. Feeding points darken then are transformed into small, nearly transparent “windows” of just clear upper and lower leaf tissues. With time, clear windows coalesce into a necrotic patch. Late stage injury to foliage can be misidentified as shot holes caused by fungal pathogens.

Adult four-lined plant bugs are readily recognizable from the black and yellow stripes on the hemelytra. Newly hatched four-lined plant bug nymphs are reddish-orange with black spots. Older instars have black wing pads with a yellow stripe on each pad (Figure 4.55). Adults and nymphs are extremely mobile. Nymphs hatch in mid- to late-spring from banana-shaped eggs deposited the previous fall. Eggs are deposited at right angles in 50 to 75 mm (2 to 3 in.) long vertical slits along host plant stems (Johnson and Lyon, 1991; Wheeler and Miller, 1981). Clusters of six or more four-lined plant bug eggs can be laid in cinquefoil (*Potentilla* sp.), loosestrife (*Lythrum* sp.), and rose campion (*Lychnis coronaria* (L.) Murray) that may serve as refuge resources (Wheeler and Miller, 1981). Nymphs stay near hatching sites and within about one month, complete metamorphosis to adults. There is typically only one generation of *P. lineatus* per season.

Management

Deciduous plants can be scouted once they lose their leaves in fall. Infested plant portions can be manually pruned and discarded (Johnson and Lyon, 1991). Trap crops, including mints, can be used in crop borders to protect sensitive crops and landscape beds. Scout for feeding injury and live four-lined plant bugs in late May or early June. Insecticides for managing four-lined plant bugs include both broad-spectrum, persistent pesticides that can eliminate beneficial arthropod predators in the garden and landscape, as well as alternatives less toxic to natural enemies (Table 4.7).

Greenhouse Thrips

Greenhouse thrips, *Heliothrips hemorrhoidalis* (Bouche) (Thysanoptera), may occasionally feed on azalea flowers and foliage (Figure 4.56), particularly in glasshouse production (Denmark and Fasulo, 2013). Both upper and lower surfaces of leaves become stippled and silvery, and affected foliage may drop prematurely. Thrips typically cause only minor injury to plants in nursery production and in landscapes.

Figure 4.56 Greenhouse thrips infesting the underside of host plant foliage.

Management

Egg parasites and predatory thrips that feed on greenhouse thrips may also be present in protected culture systems and may be obtained commercially. *Heliothrips hemorrhoidalis* can be controlled with several pesticides labeled for greenhouse use (Table 4.7).

Azalea and Rhododendron Whiteflies

Two whitefly species cause injury to azaleas and rhododendrons, primarily in protected culture. Azalea whitefly, *Pealius azaleae* (Baker & Moles), was imported from Holland and Belgium in about 1910 (Baker and Moles 1920). Adult whiteflies are about 1.5 mm (0.6 in.) long with all-white wings and light yellow bodies (Baker and Moles, 1920). Adults and nymphs feed by sucking sap from cells on leaf undersides eventually causing leaves to become mottled and chlorotic. Leaves low in the canopy may also become covered with honeydew leading to sooty mold growth. Whiteflies overwinter as nymphs (Figure 4.57 and 4.58) on evergreen foliage and reach adulthood in spring as new foliage appears (Figure 4.59) (Baker, 1994).

Rhododendron whitefly, *Dialeuroides chittendeni* Laing, was intercepted entering the U.S. from England in 1932, then subsequently found in a Seattle, WA landscape in 1933, Long Island, NY in 1934, and on *R. maximum* L. in the wild near Johnson City, TN in 1935 (Latta, 1937). Feeding injury by *D. chittendeni* is similar to that of *P. azaleae*, and in some cases, may result in curled leaf margins (Latta, 1937). Long-term feeding results in generally unthrifty plants with an overall reduction in aesthetic quality (Latta, 1937; Baker, 1994). A single generation per year was observed in Washington state with adults active mid May through July and pupae observed at low levels from November through late March, peaking between early April and early June (Latta, 1937).

Figure 4.57 A cluster of whitefly nymphs, and an individual nymph, infesting the underside of Rhododendron foliage.

Figure 4.58 An individual whitefly nymph on the underside of Rhododendron foliage.

Figure 4.59 Cluster of whiteflies on underside of deciduous azalea foliage.

Management

Azalea and rhododendron whiteflies can be controlled by applying horticultural oils and insecticidal soaps. Insecticides should be directed at leaf undersides at about seven-day intervals. In a greenhouse setting, whiteflies can be detected and trapped using yellow sticky cards. Several commercially-available natural enemies are also effective whitefly predators.

Spider mites

Two spider mite species are pests of azaleas and rhododendrons that feed on leaf undersides causing mottled, bronze foliage. Spider mites cause the most damage in the spring and in the fall (Galle, 1987). Advance spider mite injury will cause azalea foliage to turn silvery to brown, then drop prematurely (Figure 4.60).

Southern red mite (*Oligonichus ilicis* McGregor) is a widely distributed, cool-season mite that feeds on many broad-leaved ornamental host plants in the eastern US. In addition to rhododendron and azalea species, susceptible hosts include camellia, clethra (*Clethra alnifolia* L.), eleagnus (*Eleagnus* spp.), eucalyptus (*Eucalyptus* spp.), hibiscus (*Hibiscus* spp.), holly (*Ilex* spp.), photinia (*Photonia* spp.), pyracantha (*Pyracantha* spp.), laurel (*Kalmia* spp.) and viburnum (*Viburnum* spp.) (Carter et al., 1980; Johnson and Lyon, 1991). Adult female southern red mites are about 0.5 mm (0.02 in.) long, rounded and reddish brown body, have no clear division between body segments, and have four pairs of legs. In New Jersey, the first peak in mite activity was observed in April (Mague and Streu, 1980). Southern red mite populations (Figure 4.60) build during spring and fall and mites become quiescent, resting during hotter summer temperatures. Despite this cycle, southern red mites achieve multiple generations per year.

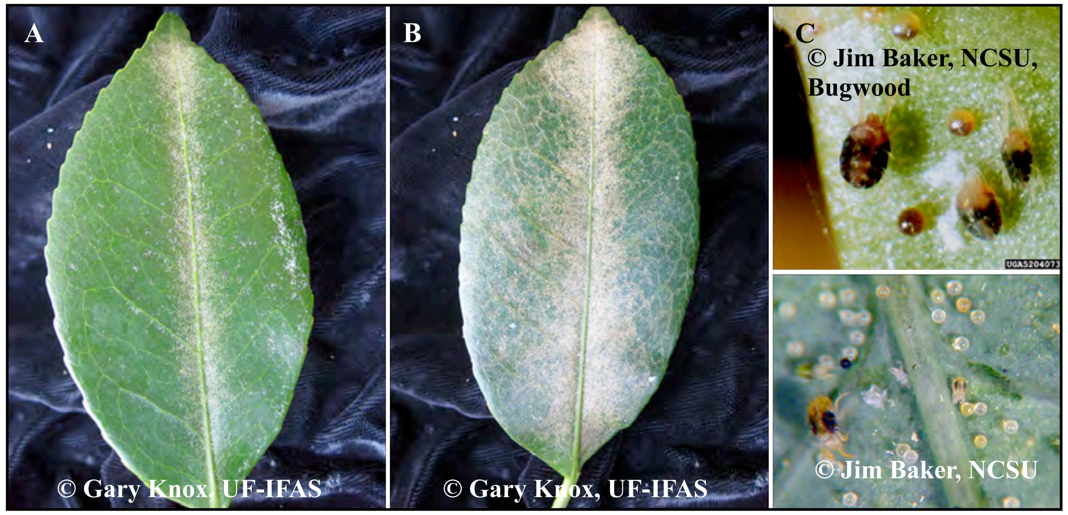


Figure 4.60 Camellia foliage helps illustrate the characteristic stippling observed early (A) and late (B) in a typical southern red mite infestation. Southern red mites can be observed with all life stages, as well as cast skins (C), present on leaf surfaces.

Twospotted spider mite, *Tetranychus urticae* Koch, is one of the most common and destructive mite species in both production and landscape settings and can attack over 200 ornamental plant species (Johnson and Lyon, 1991), including azaleas and rhododendrons. Adult and immature twospotted spider mites (Figure 4.61) are 0.5 mm (0.02 in) long. Both life stages are pale-green to yellow or cream colored and have two dark green-black patches of spotting on the body. Eggs are spherical and translucent. Foliar feeding injury becomes evident on susceptible host plants as twospotted spider mites feed on the undersides of leaves, and empty leaf cell contents to affect a silvery stippling damage visible on leaves (Figure 4.62). Twospotted spider mites spin silken threads that can aid in dispersal, and webbing can cover branches when infestations become severe.

Twospotted spider mites overwinter as adult females either in bark crevices or in the soil (Johnson and Lyon, 1991), and may persist throughout the season within protected nursery structures, provided that plant food resources, including weed species, remain present. Eggs of subsequent seasonal populations are deposited directly on leaf undersides. Spider mite populations begin to expand in April and pass through several generations per year, with peak activities occurring in June and July (Potter, 2008). Twospotted spider mites become increasingly active as localized conditions become hot and dry, and during warmer seasons of the year (Table 4.6). In tree fruit systems, twospotted spider mites migrate from groundcovers onto trees, which may help trigger higher levels of summer activity (Gotoh, 1997). Development of twospotted spider mite on raspberry (*Ribes idaeus*) can be completed in about 7 days at 86°F (30°C), 14 days at 77°F (25°C), 16 days at 60.8°F (16°C), and 25 days at 59°F (15°C) (Bounfour and Tanigoshi, 2001). Each female can produce 38 to 125 eggs (Bounfour and Tanigoshi, 2001). Populations remain active as long as environmental conditions and plant quality are favorable for reproduction and growth.

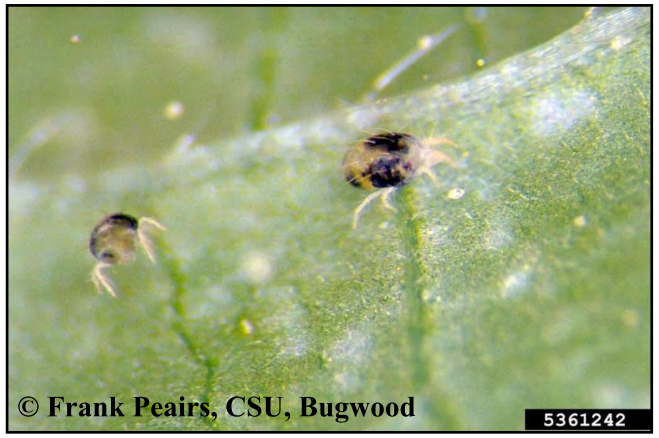


Figure 4.61 Adult twospotted spider mites.

Figure 4.62 Foliar feeding injury becomes evident on susceptible host plants as twospotted spider mites feed on the undersides of leaves, and empty leaf cell contents to affect a silvery stippling damage visible on leaves.

Management

Incipient spider mite populations are easy to overlook due both to their small size and their overwintering habits. Managers should proactively scout seasonal hot spots (e.g., dry areas, next to dusty roadways, and close to exhaust fans and doorways), and monitor susceptible crops and weedy plant refuges by looking for the characteristic stippling injury to leaf surfaces. Remove weeds, debris, and dehisced plant material to maintain a clean environment. Susceptible crop plants can be repositioned away from dry, dusty roads and also away from doorways and exhaust fans within production structures. In outdoor container operations and landscapes, overhead irrigation and hand watering can help restrict mite population growth.

Many natural enemy organisms will feed on twospotted spider mites, including *Phytoseiulus persimilis* Athias-Henriot and *Amblyseius andersoni* (Chant) predatory and *Neoseiulus fallacis* (no official common name) (Garman) predatory mites. In Oregon, *N. fallacis* suppressed twospotted spider mites in ornamental nurseries (Pratt, 1999). Once introduced, some predatory organisms can persist by foraging on prey present on alternate host plants [e.g., crabapple (*Malus* sp.)] including ground covers (Stanyard et al., 1997). Conserve these and other beneficial arthropods by selecting miticides that are compatible with the natural enemies that are already present. For example, insecticidal oils and soaps have more limited contact and residual efficacy than chemical miticides. Consequently, these products have less long-term impact on beneficial arthropods, but need to be reapplied more frequently to prevent mite populations from rebounding.

Several factors should be considered when using chemical miticides for population management. Select a miticide that is most effective against the life stages detected during scouting. Because twospotted spider mites reproduce rapidly and can quickly develop pesticide resistance, it is critical to rotate between different modes of action (Table 4.7). Pyrethroids that are registered for spider mite management are generally not effective. Finally, adding a surfactant or penetrant may increase penetration of spray solutions into hard to reach crevices or between bud scales.

SECTION 4

Disease Management

Figure 4.63 *Phytophthora* root rot on rhododendron appears with severe wilt and leaf rolling.

COMMON DISEASE PESTS

1. Root Diseases
2. Stem and Branch Diseases
3. Fungal Leaf Spot Diseases
4. Flower and Leaf Bud Diseases

There are numerous plant diseases that affect roots, branches, leaves and flowers of azaleas and rhododendrons and can be problematic in production (Table 4.8). Most diseases affect both azaleas and rhododendrons. However, there are few differences, such as Rhizoctonia web blight and Cylindrocladium blight and root rot that almost exclusively affects azaleas. Where there are differences, they are noted in the text.

Plant disease development and management is governed by the "disease triangle" concept, which requires the presence of a susceptible host, a virulent pathogen, and a favorable environment for pathogen growth in order for disease to occur. To successfully manage plant diseases, at least one part of the disease triangle needs to be removed. Host resistance, sanitation to remove sites of pathogen survival, preventive fungicide applications and reducing the duration of leaf wetness and soil saturation are commonly recommended disease management practices that correspond to the host, pathogen and environment components of the disease triangle. Another important concept in disease management is that plant diseases cannot be cured. Successful disease management relies on disease prevention. Once disease symptoms are present, it is often too late to stop a disease epidemic. Fungicide applications work to protect new or non-infected tissues from infection and have little to no effect on the already infected tissues. Knowing which diseases are likely to occur throughout the production cycle, as well as scouting plants for early symptoms needs to be included in pest management programs.

Root Diseases

Phytophthora and Pythium Root Rots

The most important root and crown disease on rhododendron is Phytophthora root rot. Azaleas are also affected by Phytophthora root rot, but to a lesser degree. Pythium root rot can also occur on both rhododendron and azalea and at least 10 *Pythium* species have been implicated (Linderman and Benson, 2014). Phytophthora root rot is caused by several species including *Phytophthora cactorum* (Lebert & Cohn) Schröt., *P. cambivora* (Petri) Buisman, *P. cinnamomi* Rands, *P. citricola* Sawada, *P. cryptogea* Pethybr. & Laff., and *P. nicotianae* Breda de Haanof. The predominant species in southeastern U.S. ornamental plant nurseries are *P. cinnamomi* and *P. nicotianae* (Linderman and Benson, 2014). Symptoms of Phytophthora root rot on rhododendron and azalea are similar. Infected roots turn brown and necrotic. Pythium root rot is more frequently a problem on small feeder roots, causing the outer root surface to soften and slough off. Infection often progresses into the crown causing a rust-brown colored canker on the lower stem at the soil line. Symptoms visible above ground vary on rhododendron and azalea. On rhododendron, the first symptom is often wilting and downward rolling of the foliage (Figure 4.63). Wilting of azaleas is uncommon. On both rhododendron and azalea, leaves on infected plants turn from yellow to brown as plants are killed. Infection can occur year-round in southeastern U.S. plant nurseries; however, plant decline symptoms are most commonly seen during the

Table 4.8 Plant diseases affecting *Rhododendron* spp. in horticultural production systems (adapted from Daughtrey and Benson 2001; Benson and Williams-Woodward 2001).

Disease	Crop Type	Regional Occurrence [z]	Season	Frequency [y]	Disease Severity [x]	Treatment Frequency [w]	Cultural Controls [v]	Sanitation Efforts [u]	Resistant Cultivars [t]
Root rot diseases									
Phytophthora root rot	Azalea	Z 6-10	Apr-Sep	5	4	3	3	2	3
	Rhododendron	Z 5-8	Apr-Sep	5	5	3	3	3	2
Cylindrocladium root rot	Azalea	Z 6-10	Apr-Sep	1	3	2	2	3	1
	Rhododendron	Z 4-8	Mar-Oct	2	4	2	2	3	1
Stem & branch diseases									
Botryosphaeria dieback	Rhododendron	Z 4-8	Mar-Nov	2	3	1	3	1	1
Phomopsis dieback	Azalea	Z 7-10	Dec-May	1	3	1	1	1	1
Phytophthora dieback	Azalea	Z 8-10	Jul-Aug	1	5	2	1	1	3
	Rhododendron	Z 5-8	Jun-Aug	4	4	2	3	3	2
Foliar diseases									
Anthracnose (*Colletotrichum*)	Azalea	Z 7-10	May-Oct	1	1	1	1	2	1
Botrytis gray mold	Azalea	Z 6-10	Dec-May	1	2	2	2	2	1
Discula leaf spot	Rhododendron	Z 6-8	Apr-Jun	1	2	2	2	2	3
Powdery mildew	Azalea	Z 6-9	Jul-Sep	1	1	2	1	1	2
	Rhododendron	Z 6-8	Apr-Nov	5	2	2	2	1	3
Rhizoctonia web blight	Azalea	Z 7-10	May-Sep	3	3	2	3	3	1
Flower & leaf bud diseases									
Exobasidium leaf & flower gall	Azalea	Z 6-7	Apr-Jun	4	2	2	2	3	2
	Rhododendron	Z 4-8	Apr-Jun	3	2	2	2	3	1
Ovulinia petal blight	Azalea	Z 6-10	Mar-May	4	2	3	2	2	1

Table 4.8 (***Continued***) Plant diseases affecting *Rhododendron* spp. in horticultural production systems (adapted from Daughtrey and Benson 2001; Benson and Williams-Woodward 2001).

[z] USDA Plant Hardiness Zones in which the disease is most likely to occur.
[y] Regularity with which the disease may affect the crop: 5 = annual, to 1 = rare.
[x] Extent to which the disease is likely to damage the crop: 5 = plants killed, to 1 = very little damage.
[w] Frequency at which chemical treatments are needed to protect the crop: 3 = used every year, to 1 = not used.
[v] Value of cultural practices used to mitigate pathogen impact on the crop: 3 = very important, to 1 = not important.
[u] Value of sanitation efforts when practiced to limit pathogen spread to crop plants: 3 = very important, to 1 = very important.
[t] Availability of cultivars demonstrated to have disease resistance: 3 = many cultivars, to 1 = no [known] resistance.
[s] Phytophthora dieback caused by *P. syringae* occurs only in winter in mild climates (Daughtrey and Benson 2001).

Phytophthora and Pythium Root Rots (text continued)
summer months (June-August) when high temperatures and heat injury to container-grown roots further stress the disease-compromised root system. Moderate temperatures of 20-28°C (68-82°F) favor *P. cinnamomi* growth; whereas, temperatures higher than 32°C (90°F) tend to limit or suppress growth (Eggers et al., 2012). Optimal temperatures for growth of *P. nicotianae* are similar to *P. cinnamomi*, except that isolates of *P. nicotianae* can continue to grow at 35°C (95°F) (Erwin and Ribeiro, 1996; Taylor et al., 2008). Targeting chemical control when foliage symptoms are observed in the summer is too late and most likely misses the infection period of the pathogen.

Phytophthora and *Pythium* are "water mold" pathogens. These are oomycete pathogens that are more closely related to brown algae than a fungus (Gunderson et al., 1987). For this reason, chemical management options are different for Phytophthora than for other root-infecting fungal pathogens. Oomycete pathogens require a wet environment to infect and spread. *Phytophthora* spp. produce motile spores (zoospores) within a sac-like structure (sporangium) that have two flagella that propel and direct the spore in water-saturated conditions toward plant roots. Phytophthora zoospores can be recovered from irrigation retention ponds, streams, puddles, and nursery effluent (Bush et al., 2003). Spread of Phytophthora or Pythium is often from water-splashed sporangia and zoospores to foliage and growing substrate of adjacent plants (Kuske and Benson, 1983). *Phytophthora* spp. can also survive as chlamydospores and oospores depending upon the species. Chlamydospores and oospores survive within infected roots, plant residue and in soil.

Management
Limiting the severity of Phytophthora root rot requires prevention, cultural and chemical approaches. Most rhododendron plants in nurseries are propagated by stem cuttings. *Phytophthora* spp. can be introduced into propagation and production areas via infected cuttings. Phytophthora chlamydospores and oospores can survive for years within plant and soil debris (Linderman and Davis, 2006; Zentmyer and Mircetich, 1966). Germinating sporangia, zoospores and hypha can infect root tips when containers are placed on infested soil or nursery pads containing debris. Nursery pads constructed of gravel can reduce water-splash and puddling under containers compared to pads covered in polyethylene ground cloth. Saturated rooting substrate from over-irrigation or poorly draining mixes will increase Phytophthora root rot disease (Ownley et al., 1990). Fungus gnats, shore flies, and even snails can spread oomycetes like Pythium and Phytophthora around nursery and greenhouse production areas (Hyder et al., 2009), so management of these pests should be integrated into a comprehensive disease control program.

Of the rhododendron cultivars screened for resistance to Phytophthora root rot, caused by *P. cinnamomi*, between 6% (Hoitink and Schmitthenner, 1974) and 23% (Krebs and Wilson, 2002) have resistance. The cultivars 'Anna H. Hall', 'Bali', 'Crete', 'English Roseum' (Grootendorst), 'Hawaii', 'Peter Tigerstedt' and 'Samoa' were determined to be moderately resistant to *P. cinnamomi* and 'Brittany', 'Caroline', 'Ginny Gee', 'Ingrid Mehlquist', 'Martha Isaacson', Normandy', 'Pink Trumpet', 'Professor Hugo de Vries', 'Red Head', 'Rocket' and 'Venus' were resistant (Hoitink and Schmitthenner, 1974; Krebs and Wilson, 2002). Some of the more popular and common rhododendron cultivars within the nursery trade, including 'Roseum Elegans', 'Nova Zembla', 'Chionoides', and 'Bessie Howells' are very susceptible to root rot. Evergreen azaleas cultivars are less susceptible to Phytophthora root rot, of 73 cultivars screened, 64 percent were rated as resistant to moderately resistant, including the cultivars 'Formosa', 'Merlin', 'Glacier', 'Chimes', 'Alaska', and 'Pink Gumpo' (Benson and Cochran, 1980). The popular cultivars, 'Hershey Red', 'Catawba', 'Snow', 'Mrs. 'G. G. Gerbing', 'Coral Bells', 'Purple Splendour', and 'Pink Cloud' were susceptible and developed severe root rot (Table 4.9) (Benson and Cochran, 1980).

Much of the Phytophthora root rot resistance evaluations have been conducted on older cultivars. There is a need to evaluate newer cultivars and hybrids for root rot resistance and incorporate resistance into breeding lines. A few of the Encore Azalea® series cultivars have been evaluated for susceptibility to *Phytophthora cinnamomi*. All eight cultivars screened were less susceptible than the highly susceptible cultivar 'Hinodigiri'. Four Encore Azalea® cultivars ('Roblen,' 'Roblee,' 'Roblem,' and 'Roblel' corresponding to the trademark names of Encore Autumn Sunset®, Encore Autumn Sangria®, Encore Autumn Starlite® and Encore Autumn Debutante®, respectively) showed a high degree of resistance to Phytophthora root rot. The cultivars 'Conlea' (Encore Autumn Rogue®), 'Conled' (Encore Autumn Coral®), and 'Conles' (Encore Autumn Empress®) were susceptible, with 'Conler' (Encore Autumn Ruby®) being the most susceptible (Warfield and Parra, 2004)

Phytophthora and Pythium inoculum survives within contaminated substrate, soil, host material, and water. Care should be taken to prevent introduction of these pathogens into propagation and production areas. Practice good nursery sanitation that includes: immediately removing infected plants; not re-using container substrates or containers unless they have been cleaned, disinfested or steam sanitized; removing fallen leaf litter and clippings as much as possible and at a minimum between crops; and storing container substrates on a concrete pad to reduce contact with contaminated soil and runoff water from the nursery. Limiting overhead sprinkler irrigation to times when the plants will dry quickly or using drip emitters can reduce Phytophthora and Pythium infection and spread. Placing containers on gravel beds can also reduce water splash of the pathogen. Container substrates must be porous and allow for good water drainage. Water management to avoid saturated substrates is the primary means of reducing Phytophthora disease development.

Fungicides are an important component in reducing Phytophthora and Pythium infections. Root rot susceptible plants cannot be successfully grown within nurseries without fungicides. The oldest and most commonly used fungicides are fosetyl-Al and mefenoxam. Resistance to mefenoxam has been reported in both Phytophthora and Pythium examined within ornamental nursery and greenhouse systems (Hwang and Benson, 2005; Moorman et al., 2002; Olson and Benson, 2011; Olson et al., 2013). Newer and very effective fungicides were introduced into the ornamental market within the past decade including cyazofamid, dimethomorph, fenamindone, fluopicolide and mandipropamid. Also, a myriad of formulations of phosphorus acid derivatives (e.g. phosphonate and phosphite fungicides) are available. Multiple applications are needed and none of the fungicides are therapeutic. Disease management relies on preventing infection. Fungicides are best applied as a drench to wet the root zone. The phosphorous acid derivatives, including fosetyl-Al, are phloem-mobile and can move downward in the plant, which allows for foliar spray applications. However, foliar applications are most effective when plants are small, such as in propagation. Foliar application of these fungicides to larger plants can effectively reduce Phytophthora stem and leaf blight, but their effectiveness in reducing root disease may be limited.

Fungicides need to be applied whenever conditions are favorable for disease development. Drenches should be applied beginning in the spring when container substrate temperatures reach 15-20°C (59-68°F) and Phytophthora infection occurs. A second drench 4 to 6 weeks later can protect roots and lessen root rot development. Mefenoxam fungicide also has a granular formulation that can be incorporated into container substrates prior to transplanting on Phytophthora-susceptible crops. Waiting until symptoms develop in the warmer, summer months is too late to effectively manage this root disease.

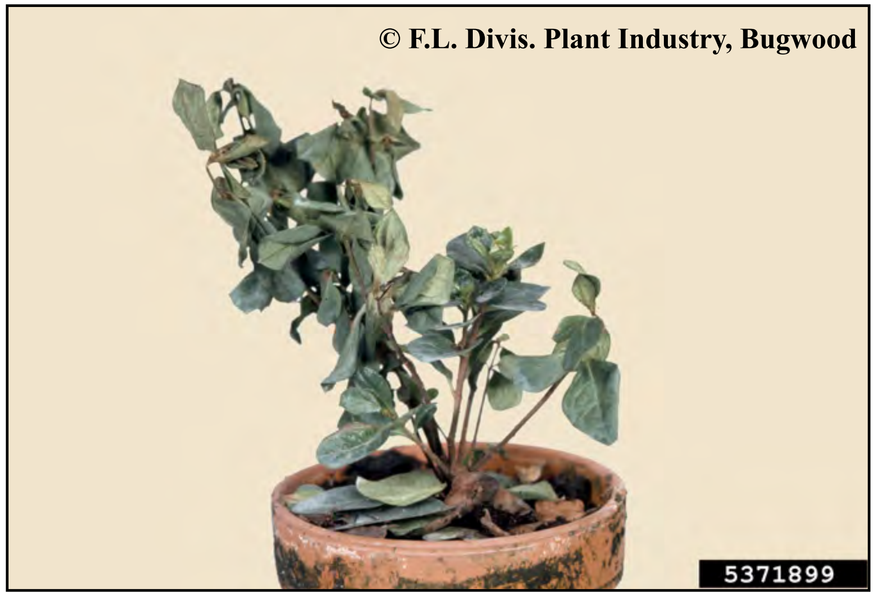


Figure 4.64 *Cylindrocladium* blight will cause infected azalea leaves to drop prematurely. Fallen leaves then become a primary source of pathogen spread among adjacent un-infected plants.

Cylindrocladium Blight and Root Rot

Cylindrocladium blight and root rot, caused by the fungi *Cylindrocladium scoparium* Morgan and *C. theae* (Petch) Subram, was a very damaging disease of potted florist azaleas (Cox, 1969; Timonin and Self, 1955). However, in more recent years, the disease occurs only sporadically and often in locations that have been unsuccessful in eradicating the pathogen from production houses. Cylindrocladium causes a leaf spot, root rot, and wilt disease. All are connected and are just different phases of the same disease.

Cylindrocladium infection causes circular, dark brown leaf spots and brown flecking on azalea blooms. Roots are often symptomless in early stages of the disease, which is one of the reasons why the disease can be readily spread on propagative material. Eventually, roots become dark with discoloration extending into the lower stem. Infected plants yellow, wilt, and die. The disease is favored by warmer, humid, and wet conditions. Cylindrocladium disease severity is greatest at 27-32°C (81-90°F) with near 100% relative humidity (Linderman and Benson, 2014). Leaf spot symptoms are often absent under less humid conditions. Cylindrocladium does not sporulate on attached leaves. Infected leaves will drop prematurely (Figure 4.64) where the fungus readily produces microsclerotia within the leaf tissue and abundant asexual conidia. Fallen leaf litter is a primary source of inoculum for this disease where conidia may be water-splashed to adjacent plants or washed into the soil where they may contact plant roots (Linderman, 1974). Plant wetness is required for spore germination. Sexual reproduction does occur and dark orange, round, spore-producing structures (perithecia) containing ascospores can be produced on infected leaves. Microsclerotia within leaf litter and infected roots are the primary means of long-term survival of the pathogen.

Management

Few cultivars have been evaluated for their susceptibility to Cylindrocladium blight and root rot. However, in several limited evaluations, no resistance was identified (Alfieri et al.,

Table 4.9 Greenhouse evaluation of root rot resistance among evergreen azalea species, hybrid groups, and cultivars following inoculation with *Phytophthora cinnamomi*[z] on a one to five scale [y].

Hybrid Group & Cultivar	Root Rot Rating[y]
Species type azalea	
R. poukhanense	1.6
R. mucronatum	
'Delaware Valley White'	2.4
'Back Acres' type	
'Corrine Murrah'	1.9
'Rachel Cunningham'	2.3
'Margaret Douglas'	2.5
'White Jade'	2.7
'Marian Lee'	3.1
'Pat Kraft'	3.3
'Saint James'	3.4
Gable Hybrids	
'Rose Greeley'	2.2
'China Seas'	2.8
'Herbert'	3.0
'Royalty'	3.1
'Rosebud'	3.2
'Purple Splendour'	3.5

Hybrid Group & Cultivar	Root Rot Rating[y]
Glenn Dale Hybrids	
'Fakir'	1.9
'Merlin'	2.1
'Glacier'	2.1
'Polar Seas'	2.2
'Gaiety'	2.5
'Copperman'	2.7
'Martha Hitchcock'	2.8
'Robinhood'	3.0
'Catawba'	3.0
'Treasure'	3.3
'Pinocchio'	3.5
Indian Hybrids	
'Formosa'	1.8
'Redwing'	2.2
'Chimes'	2.2
'New White'	2.3
'Pink Supreme'	2.4
'Prince of Orange'	2.6
'California Sunset'	2.9
'Pride of Summerville'	2.9
'Mrs. G. G. Gerbing'	3.2

Table 4.9 (***continued***) Greenhouse evaluation of root rot resistance among evergreen azalea species, hybrid groups, and cultivars following inoculation with *Phytophthora cinnamomi*[z] on a one to five scale [y].

Hybrid Group & Cultivar	Root Rot Rating[y]
Kurume Hybrids	
'Morning Glow'	2.4
'Hexe'	2.7
'Massasoit'	2.8
'Hinodegiri'	2.9
'Hershey Red'	3.0
'Snow'	3.1
'Coral Bells'	3.2
'General MacArthur'	3.5
'Pink Pearl'	3.6
'Hino Crimson'	3.9
NCSU Selection	
'Sunglow'	3.8
'Carror'	3.4
'Elaine'	4.1
'Emily'	4.3
'Pink Cloud'	4.5
'Adelaide Pope'	4.6
'Jane Spaulding'	5.0

Hybrid Group & Cultivar	Root Rot Rating[y]
Pericat Hybrids	
'Hampton Beauty'	2.1
'Sweetheart Supreme'	2.4
'Barbara Gail'	2.4
'Pink Hiawatha'	2.5
'Sensation'	2.6
'Flanders Field'	2.9
'Fortune'	3.0
Rutherford Hybrids	
'Alaska'	2.2
'Dorothy Gish'	2.5
'White Gish'	2.5
'Gloria'	2.6
Satsuki Hybrids'	
'Higasa'	2.1
'Shin-ki-gen'	2.3
'Pink Gumpo'	2.3
'Eikan'	2.3
'White Gumpo'	2.4
'Amaghasa'	2.9
'Kow-ko-ku'	3.2
'Johga'	3.7

Hybrid Group & Cultivar	Root Rot Rating[y]
Whitewater Hybrids	
'Rentschler's Rose'	2.5
'Kingfisher'	2.6
'White Christmas'	2.6
'Warbler'	2.9

[z] Data derived from Benson and Cochran, 1980.

[y] Visual estimate of mean root rot severity observed among plants wherein 1 = healthy roots, 2 = fine roots necrotic, 3 = coarse roots necrotic, 4 = crown rot, 5 = dead plant.

1972; Sobers and Crane, 1971). Because of the long-term survival of *Cylindrocladium* spp. in leaf litter and roots, good sanitation to remove leaf litter and any declining and adjacent plants can reduce disease development in subsequent crops. Cuttings should not be taken from any plants suspected of being infected. Cuttings can be dipped into chlorine dioxide or 10% bleach solution to reduce surface contamination; however, this will have no effect on internally colonized tissues. Cylindrocladium diseases are difficult to control chemically. Fungicides containing chlorothalonil, fludioxonil, and triflumizole provide better control followed by the QoI (strobilurin) fungicides of azoxystrobin, pyraclostrobin, and trifloxystrobin (Table 4.10).

Figure 4.65 Phytophthora blight results in rapid shoot death and premature leaf drop.

Stem and Branch Diseases

Phytophthora Leaf Blight and Dieback

No other pathogen has been the focus of as much attention and concern as Phytophthora leaf blight and dieback in rhododendrons. In part, much of the emphasis has centered around discovery of the non-U.S.-native pathogenic species, *Phytophthora ramorum* Werres, De Cock, & Man in 't Veld, that is the cause of sudden oak death and Phytophthora leaf blight (Werres et al., 2001). In addition to *P. ramorum*, 19 additional *Phytophthora* spp., including *P. cactorum*, *P. citricola*, and *P. nicotianae* (syn. *P. parasitica*), have been implicated as causal agents of plant disease in rhododendron and azalea (Linderman and Benson, 2014). In the southeastern U.S., *P. ramorum* is still relatively rare due to federal quarantines that have reduced the risk of *P. ramorum* introduction on infected nursery stock (Jeffers et al., 2010; Olson et al., 2013).

Figure 4.66 Young foliage on shoot tips and terminal stems wilt become necrotic.

Visible disease symptoms in infected host plants appear as shoot blight, or rapid shoot death with dropping foliage (Figure 4.65), resulting in leafless shoots, to leaf blight evidenced by rapid foliar discoloration. Young leaves and shoots are most susceptible to *Phytophthora* spp. infection (Figure 4.66). Foliar symptoms first appear with brown margins on leaf undersides, expanding into the midrib with dark brown to black, irregularly shaped necrotic lesions (Figure 4.67). From the leaves, the pathogen invades the petiole (Figure 4.68) and then shoots causing dieback and eventual death of the plant. Infection of the crown and lower stem appears as defoliation of the lower stem

Figure 4.67 Phytophthora dieback, including decline caused by *P. ramorum*, will present irregularly shaped necrotic lesions.

Figure 4.68 *Phytophthora ramorum* may be evident at the petiole where the leaf meets the stem.

followed by rapid wilt and death of the plant. Often, roots look healthy, even when they are infected. Spores, also called sporangia, may also move through wind-driven rain or water splash during rains or overhead irrigation. Humans and equipment can also move the pathogen under shoes, tires, or other equipment. Long distance movement occurs though movement of infected plant material (Dailey at al. 2004; ODA 2013). Phytophthora leaf and shoot blight ultimately leads to plant death (Figure 4.69) (Sinclair and Lyon, 2005).

Generally, infections occur during the spring when climactic conditions favor spore development and mycelial growth. For most infectious *Phytophthora* spp. the optimum temperature for sporulation and infection is 78-86°F (25-30°C), but infection by some species may occur at temperatures as low as 50°F (10°C) (Linderman and Benson, 2014). Rainfall or high humidity is also necessary for infection, as pathogen growth and sporulation typically occur under high moisture conditions (Tooley and Browning, 2015). Symptoms usually develop within two days of infection.

Figure 4.69 Left untreated, Phytophthora dieback can cause extensive loss across a broad area in production settings.

Table 4.10 Fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of rhododendron and azalea[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Pythium root rot[X]	Phytopthora root rot[X]	Cylindrocladium	Phytopthora leaf blight	Botryospheria	Phomopsis	Anthracnose	Botrytis	Cercospora & Leaf Scorch	Powdery mildew	Rhizoctonia	Leaf rust	Exobasidium gall	Petal blight
1: MBC Benzimidazoles: Upwardly systemic. Broad spectrum fungicide for various fungi. Fungicide resistance risk high. Tank mix with fungicides from a different fungicide group (FRAC) to prevent or delay resistance development. Do not mix with copper.														
thiophanate methyl: Cleary's 3336 F; Allban 50 WSP; OHP 6672 50WP, 4.5F			x			x	x	x	x	x	x			x
1 + 2: MBC Benzimidazoles + Dicarboximides: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.														
thiphanate methyl + iprodione: 26/36[W]			x					x						
1 + 3: MBC Benzimidazoles + DMI or SI Triazoles: Systemic. Long protection during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.														
thiophanate methyl + propiconazole: Protocol[W]						x			x	x	x	x		x
1 + 14: MBC Benzimidazoles + Aromatic Hydrocarbons: Systemic. Effective against water molds, or oomycetes. Long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.														
thiophanate methyl + etridiazole: Banrot 40WP[W]	x	x		x							x			
1 + M3: MBC Benzimidazoles + Multi-site inhibitor: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.														
thiophanate methyl + manxozeb: Zyban WSB[W]						x	x		x					x
1 + M5: MBC Benzimidazoles + Multi-site inhibitor: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.														
thiophanate methyl + chlorothalonil: Spectro 90 WDG[W]			x				x		x	x	x			x
2: Dicarboximides: Locally systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.														
iprodione: Chipco 26019 FLO, N/G; 26GT; Raven[V]			x					x			x			
3: DMI or SI Triazoles: This group was formerly known as De-Methylation Inhibitors (DMI) and are now classified as Sterol Biosynthesis Inhibitors (SBI or SI). Upwardly systemic. Rain-fast in 2 hours. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.														
fenarimol: Rubigan AS										x				
imazalil: Fungaflor TR								x		x				
metconazole: Tourney[W]						x	x		x	x				
myclobutanil: Eagle 20W; Rally 40WSP; Siskin							x		x	x				
propiconazole: Banner Maxx; Banner Maxx II; Strider			x			x			x	x	x			x

Table 4.10 (*continued*) Fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of rhododendron and azalea[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Pythium root rot[X]	Phytopthora root rot[X]	Cylindrocladium	Phytopthora leaf blight	Botryospheria	Phomopsis	Anthracnose	Botrytis	Cercospora & Leaf Scorch	Powdery mildew	Rhizoctonia	Leaf rust	Exobasidium gall	Petal blight
3: DMI or SI Triazoles: This group was formerly known as De-Methylation Inhibitors (DMI) and are now classified as Sterol Biosynthesis Inhibitors (SBI or SI). Upwardly systemic. Rain-fast in 2 hours. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.														
tebuconazole: Torque										x	x	x		x
triadimefon: Bayleton 50, FLO; Strike 50 WDG[V]														x
triflumizole: Terraguard			x							x				
triticonazole: Trinity, Trinity TR									x	x				
3 + 11: SI Triazole + QoI Q: Upwardly systemic. Rain-fast in 2 hours. Some curative activity. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance.														
triadimefon + trifloxystrobin: Trigo							x			x				
4: Phenylamides: Systemic. Effective against diseases caused by oomycetes, or water molds, including damping-off, root and stem rots, and foliar diseases. Use as soil drench or foliar application.														
mefenoxam: Subdue Maxx; Subdue GR	x	x		x										
4 + 12: Phenylamides + Phenylpyrroles/Osmotic signal transducers: Systemic. Broad spectrum drench-applied fungicide.														
mefenoxam + fludioxonil: Hurricane, Hurricane WDG[U,V,W]	x	x	x	x							x			
5: Morpholine: Inhibits sterol biosynthesis in membranes. For use only in greenhouse and similar enclosed structures. Do not use after flower buds visible.														
piperalin: Pipron										x				
7: Carboxamides/Succinate Dehydrogenase Inhibitors (SDHI): Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Spray apply for leaf disorders.														
utolanil:Prostar 70WP											x	x		
7 + 11: Carboxamides/Succinate Dehydrogenase Inhibitors (SDHI) + QoI Q: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.														
benzovindiflupyr + azoxystrobin: Mural [W]			x					x	x					
fluxapyroxad + pyraclostrobin: Orkestra Intrinsic [W]							x	x		x				
pyraclostrobin + boscalid: Pageant; Pageant Intrinsic [W]	x	x	x	x			x	x	x	x	x			
9 + 12: Anilino-Pyrimidines/Methionine biosynthesis inhibitors + Phenylpyrroles: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.														
cyprodinil + fludioxonil: Palladium[W]			x			x	x		x	x				

Table 4.10 (*continued*) Fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of rhododendron and azalea[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Pythium root rot[X]	Phytopthora root rot[X]	Cylindrocladium	Phytopthora leaf blight	Botryospheria	Phomopsis	Anthracnose	Botrytis	Cercospora & Leaf Scorch	Powdery mildew	Rhizoctonia	Leaf rust	Exobasidium gall	Petal blight
11: Quinone outside Inhibitors (QoI): These fungicides are also known as strobilurins. Locally systemic. Effective on mildews, foliar pathogens, and most fungi. Some control of oomycetes, or water molds. Fungicide resistance risk high.														
azoxystrobin: Heritage; Strobe 50WG	x	x		x		x	x		x	x	x			
fenamidone: FenStop	x	x		x						x				
fluoxastrobin: Disarm O		x					x		x	x	x			
kresoxim methyl: Cygnus							x		x	x				
pyraclostrobin: Insignia; Insignia SC; Empress Intrinsic	x	x		x							x			
trifloxystrobin: Compass O 50 EDG[V]		x	x				x		x	x	x			
12: Phenylpyrroles/Osmotic signal transducers: Non-systemic but good residual protection. Broad spectrum fungicide, not effective against oomycetes, or water molds.														
fludioxonil: Emblem; Medallion; Medallion WDG; Mozart			x						x		x			
13: Uncouplers of oxidative phosphorylation via disruption of the proton gradient: Absorbed rapidly and systemic. Good coverage and wetting of the foliage are necessary. Rainfall or sprinkler irrigation after one hour does not decrease effectiveness. Control may be less effective on plants suffering from drought stress.														
triadimefon: Bayleton 50									x					
14: Aromatic Hydrocarbons: Locally systemic. Effective against water molds, or oomycetes.														
etridiazole: Terrazole 35% WP, CA, L; Truban 25EC, 30WP[V]	x	x		x										
PCNB: Terraclor WP, 400[W]														x
17: Hydroxyanilides: Locally systemic. For use in outdoor and greenhouse nursery crops.														
fenhexamid: Decree 50 WDG[V]								x						
19: Polyoxins: Locally systemic chitin synthase inhibitor. For use in outdoor and greenhouse nursery crops. Resistance risk high.														
polyoxin D zinc salt: Affirm; Veranda O; Endorse WP [W]							x	x		x	x			
21: Quinone inside Inhibitors: Locally systemic. Effective against water molds, or oomycetes. Resistance risk unknown but presumed to be medium to high.														
cyazofamid: Segway, Segway O, Segway SC	x	x		x										
28: Carbamates: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes.														
propamocarb: Banol[V]	x	x		x										

Table 4.10 (*continued*) Fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of rhododendron and azalea[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Pythium root rot[X]	Phytopthora root rot[X]	Cylindrocladium	Phytopthora leaf blight	Botryospheria	Phomopsis	Anthracnose	Botrytis	Cercospora & Leaf Scorch	Powdery mildew	Rhizoctonia	Leaf rust	Exobasidium gall	Petal blight
33: Phosphonates: Fully systemic; when applied to leaves, product can translocate to lower parts. Effective against water molds, or oomycetes, such as Phytophthora, Pythium, and downy mildew pathogens. Low risk for fungicide resistance development.														
phosphorous acid: Alude		x		x						x				
phosphorous acid: K-Phite T/O, 7LP				x						x				
potassium phosphite: Vital		x		x						x				
fosetyl-Al: Aliette WDG	x	x		x						x				
40: Carboxylic Acid Amides: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes.														
mandipropamid: Micora		x		x										
dimethomorph: Stature DM, SC	x	x		x										
43: Pyridinemethyl-benamides Delocalisation of spectrin-like proteins: Locally systemic, translaminar. Control of oomycetes, or water molds. Medium to high resistance risk.														
fluopicolide: Adorn	x	x		x										
44: Bacillus subtilis: Microbial disruptor of cellular membranes. Spray apply for leaf disorders, soil drench for phytophthora.														
Bacillus subtilis: Cease							x		x	x				
45 + 40: Quinone outside Stigmatellin-binders (QoS) + Carboxylic Acid Amides: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes.														
ametoctradin + dimethomorph: Orvego[W]		x		x										
49: Piperidinyl-thiazole- isoxazolines: Locally systemic. Control of oomycetes, or water molds. Rain fast. Medium to high resistance risk.														
oxathiapiprolin: Segovis		x		x										
M: Chemicals with multi-site activity: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.														
(M1) copper hydroxide: Champ DP, Champ Formula 2; Kocide 2000; CuPro 5000							x		x	x				
(M1) copper octanoate: Camelot O							x	x	x	x				
(M1) copper sulfate: Basicop							x							x
(M1) copper sulphate pentahydrate: Phyton 27; Phyton 35								x		x				
(M1) tribasic copper sulfate: Cuproxat										x				
(M1 + M3) copper hydroxide + mancozeb: Junction[W]							x		x	x			x	x
(M3) mancozeb: Dithane 75DF; Fore 80WP; Mancozeb 4F, Floable with Zinc; Pentathlon DF, LF; Protect DF					x	x	x	x	x	x	x			x
(M4) captan: Captan, 50WP, W, 4L, 80WDG									x					x

Table 4.10 (*continued*) Fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of rhododendron and azalea[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Pythium root rot[X]	Phytopthora root rot[X]	Cylindrocladium	Phytopthora leaf blight	Botryospheria	Phomopsis	Anthracnose	Botrytis	Cercospora & Leaf scorch	Powdery mildew	Rhizoctonia	Leaf rust	Exobasidium gall	Petal blight
M: Chemicals with multi-site activity: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.														
(M5) chlorothalonil: Daconil Ultrex, Zn Flowable, Weather Stik[T]			x				x	x	x	x				x
(M5) chlorothalonil: Manicure 6FL, Manicure Ultra; Thalonil 90DF, Thalonil 6L							x	x	x		x			
M3 + 3: Multi site inhibitor + Sterol Biosynthesis Inhibitor: Effective as protectants (and upwardly systemic) on broad spectrum including most fungi and mildews. Fungicide resistance risk low.														
mancozeb: + myclobutanil: Clevis[W]					x	x	x		x	x				x
M5 + 3: Multi site inhibitor + SI Triazoles: Upwardly systemic. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.														
propiconazole + chlorothalonil: Concert II [X]							x		x	x				x
NC: Not a Classified substance: Contact fungicide for greenhouse and nursery use. Low risk for resistance.														
potassium bicarbonate: MilStop						x	x			x				

Footnotes:

[Z] Fungicide Resistance Action Committee (FRAC) 2016.

[Y] Check current products for labeled pesticides, sites for control and plant safety and efficacy on fungal species. This table reports information on fungicide labels and does not necessarily reflect product efficacy. Refer to fungicide labels for rates and usage, specific host information, possible phytotoxicity, re-entry intervals and resistance management. Within columns, products indicated by "Y" are labeled for use against the listed pathogen type.

[X] Including the causal agent of sudden oak death, *Phytophthora ramorum*.

[W] Chemical contains more than one active ingredient, thus product may be listed within more than one FRAC code designation (FRAC 2016).

[V] Not for use in residential landscapes; commercial use only.

[U] Greenhouse use only.

[T] Do not apply with mist blowers or high pressure spray equipment in greenhouses.

In the nursery, infected potting substrate is a likely source of new infections and a means of long-term pathogen survival. Water splashed from infected substrate onto new foliage causes most leaf and shoot infections (Kuske and Benson, 1983; Linderman and Benson, 2014). Drainage water from containers seems to be the main route by which new root infections occur (Heungens et al., 2010; Linderman and Benson, 2014). Plants that appear healthy may be asymptomatic carriers of the disease and have infected roots yet present no foliar symptoms. Substrate from containers holding these plants can be splashed and will readily infect leaves of adjacent plants (Bienapfl and Balci, 2014). Root infections can eventually manifest into foliar symptoms, further increasing the likelihood of transmission (Loyd et al., 2014). Soil, gravel and landscape fabric materials beneath containers can also harbor Phytophthora propagules, thus provide a source of infection (Parke et al., 2010). Spores and sporangia of some infectious *Phytophthora* spp. may survive in the soil or potting substrate for up to three years in the absence of a host, even when soil temperature is near freezing (Babadoost and Pavon, 2013; Linderman and Davis, 2006; Tjosvold et al., 2009; Vercauteren et al., 2013).

Management

To prevent the introduction and spread of Phytophthora, incoming plant material should be thoroughly inspected for symptoms, and current stock should be monitored regularly. Any material showing signs of infection should be removed and destroyed, and nearby plants should be quarantined (Heungens et al., 2010). Cuttings should be propagated only from plants that are known to be pathogen-free. Take steps to employ sanitary measures and precautions when moving susceptible nursery plants, and walking or driving through areas with susceptible species or suspected symptoms (Dailey et al., 2004). Sanitization of tools and surfaces which may have come into contact with infected plant material or used potting substrate will help prevent the spread of Phytophthora within the nursery. Commonly used disinfectants, including 15% bleach solution, are effective in preventing the growth and sporulation of *P. ramorum* (James et al., 2012). Reuse of containers should be avoided since spores from infected substrate may remain on container surfaces (Parke et al., 2010).

Since potting substrate is a source of infection, even when symptoms are not apparent in plants, irrigation practices which minimize splashing are most effective in preventing the spread of Phytophthora from substrate to leaves. Maximizing the distance between plants is an effective preventative measure against splashing as well as shoot-to-shoot transmission. Placing gravel or absorbent weed cloth beneath containers can prevent pathogen spread between containers via puddling and provide a barrier between the plants and potentially infected base material (Linderman and Benson, 2014).

Contaminated substrate can be effectively sterilized by steaming at 122°F (50°C) for at least 2 hours. This treatment was effective at completely destroying *P. ramorum* in infested nursery soil (Schweigkofler et al., 2014). Composting can also be an effective means of destroying Phytophthora in infected plant material as long as adequate temperatures are maintained for prolonged periods (Noble et al., 2011). Nonetheless, removal and destruction of contaminated or potentially infested material may be the most effective means of preventing Phytophthora spread and future outbreaks.

Fertilization practices can influence the incidence and severity of Phytophthora infections. Providing elevated nitrogen levels to young plants may lead to greater susceptibility of young succulent leaf and stem tissues to Phytophthora infection (Hoitink et al., 1986). Both the risk of infection and the severity of leaf blight are increased when plants are fertilized at higher rates and when foliar nitrogen concentration is greater (Hummel et al., 2013; Linderman and Benson, 2014).

Recirculated irrigation water reservoirs may become a source of Phytophthora infection, as spores can survive there for prolonged periods of time and may be continually introduced throughout the year (Linderman and Benson, 2014; Parke et al., 2014; Ridge et al., 2012). Pathogenic *Phytophthora* spp. have been recovered from retention basins, and chemical decontamination may be necessary to prevent further spread and infection (Ghimire et al., 2011; Werres et al., 2007). However, the risk of infection from recirculated irrigation water is likely minimal. Pathogenic *Phytophthora* spp. have been shown to account for only a small percentage of the Phytophthora population in irrigation reservoirs, and the concentration of propagules that reaches plants in the re-used water is likely too small to cause infection (Copes et al., 2015; Loyd et al., 2014). Overhead irrigation should be avoided, particularly in the early morning and late afternoon, when water on foliage is likely to evaporate slowly. Close plant spacing is also likely to increase spread of oospores among plants. Good cultural practices include spacing plants to prevent foliage overlap. Aisles between blocks of plants are recommended at 2-meter (6.6 ft.) widths (USDA APHIS-PPQ, 2014a). Senescent leaf tissues should be removed from containers, landscape fabric beneath containers, and from under plantings in the landscape. A base layer of 5 to 7 cm (2.0 to 2.8 in.) of gravel can be used in container production areas to limit splash of water onto foliage (Kuske et al., 1983).

Fungicides suppress but do not cure Phytophthora leaf and shoot blights. Fungicide applications can only mask disease, thus once routine applications cease symptoms may resume and again become noticeable (Dailey et al., 2004). Healthy plants should be protected with regular applications of fungicides like cyazofamid (Segway) or dimethomorph (Stature), particularly when new growth is present and when confirmed infections are reported nearby (van Tol and Raupp, 2005). Mefenoxam (metalaxyl-M) is highly effective in suppressing the growth of all *Phytophthora* spp. commonly found in the southeastern U.S. However, frequent applications can lead to the development of resistant strains, so robust and rotational fungicide applications should be used to suppress this pathogen (Olson, 2013). Commercial biofungicides effectively suppress *P. ramorum*, and are likely effective against other infective *Phytophthora* spp., especially when applied as a drench (Bailey et al., 2012; Linderman and Davis, 2006).

The USDA Animal and Plant Health Inspection Service Plant Protection and Quarantine (USDA APHIS-PPQ) has published several protocols for managing cases of *Phytophthora* *ramorum*, a federally regulated plant pathogen, which are used to guide inspection and sampling in residential and commercial landscapes (USDA APHIS-PPQ, 2013), at nurseries not known to contain *P. ramorum*-infected plants (USDA APHIS-PPQ, 2014a), as well as nurseries known to contain *P. ramorum*-infected plants (USDA APHIS-PPQ, 2014b). These protocols are periodically updated and can be accessed online (https://www.aphis.usda.gov/plant_health/).

Botryosphaeria Dieback

Botryosphaeria dieback is primarily a problem on evergreen rhododendron within landscapes. It is of minor importance within plant nurseries on both rhododendron and azalea. It can occur; however, infection often follows stem wounding. The fungus, *Neofusicoccum ribis* (Slippers, Crous & M.J. Wingf.) Crous, Slippers & A.J.L. Phillips (syn. *Botryodiplodia ribis* (Fuckel) Petr.), is the primary cause of Botryosphaeria dieback. There is quite a bit of confusion surrounding the reevaluation of *Botryosphaeria* spp. taxonomy (Slippers et al., 2004; Crous et al., 2006). The related fungus, *Botryosphaeria dothidea* (Moug.) Ces. & De Not. (syn. *Fusicoccum aesculi* Corda), has also been described as the primary causal agent (Linderman and Benson, 2014; Wills and Lambe 1986). A survey of rhododendron stem dieback may help elucidate the fungal species involved in this Botryosphaeria disease complex in the southeastern U.S.

Figure 4.70 Botryosphaeria dieback may present a canker low on the stem. First symptoms in rhododendron may be wilt and tight leaf curling.

Symptoms of Botryosphaeria dieback resemble that of Phytophthora infection, which is a much more common disease in nurseries because of overhead irrigation and wet growing environments. Leaves on Botrytopshaeria-infected stems wilt and roll inward parallel to the midrib (Figure 4.70). It is usually seen scattered through landscapes affecting individual branches. The fungus enters into the stem at wound sites such as fresh leaf scars, pruning wounds, deep cracks from freeze injury or below spent flower clusters. Sunken, reddish-brown stem cankers are often evident and when the stems are cut horizontally, V-shaped discoloration can be seen extending from the cankered area toward the center of the stem. Infection can move downward and kill large sections of the affected plants (Figure 4.71). The fungus produces abundant spore-producing pycnidia along the dead stem tissue. Spores are spread by water-splashing from precipitation or sprinkler irrigation. Spores may also be spread on pruning tools if cuts are made when the plants are wet (Schrieber, 1964).

Figure 4.71 Later in disease progression, Botryosphaeria dieback can kill large sections of a rhododendron crown.

Management

Botryosphaeria dieback is difficult to control because the pathogen is ubiquitous in the southeastern U.S. infecting numerous other plant species. Although infection most often occurs in the spring and early summer, it can occur at wounds at any time of the year. Furthermore, *Botryosphaeria* spp. are also known to have long incubation periods and survive within asymptomatic tissues (Creswell and Millholland, 1988; Schreiber, 1964). Symptoms may develop within a few weeks to several months following infection. The best approach to managing this disease is to minimize stem wounding and unnecessary pruning. Providing irrigation during periods of drought and growing rhododendron in partial shade can help alleviate plant stress. Pruning dying branches several inches below the discolored wood can help reduce disease spread. Few fungicides are labeled for use on this disease. By the time symptoms are seen, fungicides are ineffective. The value of applying fungicides preventively is uncertain and most likely uneconomical. If fungicides are to be used to reduce disease development, an application immediately following any pruning activities can help protect the fresh pruning wounds.

Few cultivars have been evaluated for resistance to Botryosphaeria dieback, and none are known to be resistant. Several cultivars were found to be less susceptible to infection having less than 10% dieback, including ‘Boursault’, ‘Cunningham’s White’, ‘English Roseum’, ‘LeBar’s Red’, ‘Roseum Elegans’, and ‘Roseum 2’; whereas, ‘Nova Zembla’ averaged 10-50% dieback (Benson et al., 1990).

Phomopsis Dieback

Phomopsis dieback, caused by the fungus, *Phomopsis* spp., generally occurs more commonly on azalea, particularly the Southern Indica hybrids, than on rhododendron. It is also more common in landscapes than in nurseries. The Phomopsis species causing the dieback has not been identified. Studies on its epidemiology and management are limited, which is likely due to it generally being considered a weak pathogen that infects wounds.

Phomopsis dieback looks similar to Botryosphaeria dieback. Infected stems may show slight chlorosis after flowering, then browning, wilting and necrosis (Figure 4.72) (Miller and Baxter, 1970). Drought stress often contributes to symptom development, and

Figure 4.72 Browning, wilting and necrosis evident in Phomopsis-infected azalea leaves.

symptoms are most commonly seen in the mid- to late-summer months. Eventually, leaves on affected stems drop prematurely, leaving only bare stems evident by fall and winter months. Infection can occur up to 8 days after wounding at natural sites (leaf scars, frost cracks) or due to pruning activities (Miller and Baxter, 1970). There is also a lengthy incubation period with symptoms often not becoming evident until at least two months following infection. When dieback is present, affected stems exhibit reddish brown streaking in the wood from the site of infection, and similar to Botryosphaeria dieback, a V-shaped, brown discoloration extending from the cambial tissues to the center of the stem can be seen when the stem is cut crosswise.

Management

Management of Phomopsis dieback is the same as Botryosphaeria dieback. Reducing plant stress and stem wounding can reduce Phomopsis infection. The fungus produces spores within pycnidia on infected stems that can be water-splashed or spread on pruning tools to new wound sites. Prune affected stems several inches below where the dieback is evident to reduce inoculum and pathogen spread. Disinfesting pruning tools between cuts can also reduce infection at pruning sites. Fungicides are generally not needed nor has fungicide efficacy been studied on this disease within landscapes or nurseries.

Limited studies have been conducted on cultivar susceptibility to Phomopsis infection. It's been noted that the cultivars 'Delaware Valley White', 'Hershey Red', 'Pink Gumpo' and 'Snow' were least susceptible to Phomopsis dieback. 'Coral Bells,' 'Orange Cup,' 'Pink Ruffles,' and 'Pride of Mobile' were moderately susceptible and 'Copperman,' 'Fashion,' 'Hinodegiri,' 'Southern Charm,' 'Tradition,' and 'Treasure' were the most susceptible (Linderman and Benson, 2014). Additional cultivars evaluations that include newer hybrids are needed.

Fungal Leaf Spot Diseases

There are several fungi associated with leaf spot diseases on rhododendron and azalea. Most are of minor importance and occur sporadically, often on stressed or injured tissues.

Anthracnose

The fungus, *Glomerella cingulata* (Stoneman) Spauld. & H. Schrenk [syn. *Colletotrichum gloeosporioides* (Penz.) Penz. & Sacc.], causes leaf spots and defoliation of Indica and Kurume hybrid azalea cultivars. Damage is most often seen during warmer summer months with frequent rainfall. Leaf spots on azalea are small, circular, and olive to rust-brown (Figure 4.73), appear "greasy", and may be found on both the upper or lower leaf surfaces. Anthracnose is not a significant concern on rhododendron. Infection will cause larger, rust-colored spots that may expand to blight larger leaf areas. The fungus produces numerous acervuli (spore-producing structures) on fallen leaf litter and exposure to 24-48 hours of wetness. Conidia are produced on the leaf litter and water-splashed to new growth.

Figure 4.73 Anthacnose leaf spots on azalea are small, circular and olive colored.

Botrytis Leaf and Petal Blight

Botrytis leaf and petal blight, caused by the fungus *Botrytis cinerea* Pers.:Fr., is primarily a problem during propagation where it can cause blighting and death of stem cuttings under humid, moist, shaded or overcast conditions. It is not a significant problem on mature rhododendron or azalea. Botrytis is a weak pathogen that infects mechanically or environmentally-stressed tissues or senescing flowers. Infected flowers develop brown spots (Figure 4.74) and blighting of the petals. Zonate leafs spots with alternating light and dark zones are characteristic of Botrytis infection. Infected tissues produce abundant conidia that are water-splashed or wind dispersed to adjacent flowers or leaves.

Figure 4.74 Botrytis blight is characteristic in appearance; shown growing on dead rose tissue.

Botrytis can grow at temperatures ranging between 38°F (4°C) to 90°F (32°C), with optimal conidial germination from 59-88°F (15-30°C) when tissues are wet for at least 6 hours (Sirjusingh and Sutton, 1996). Relative humidity above 93% favors sporulation and disease development. At lower humidity levels, the fungus persists as hyphae within plant tissues. Since plant wetness and high humidity are essential for spore germination and infection, keep

Figure 4.75 Necrosis caused by gray mold (*Botrytis*) in flats of seedlings of deciduous

plants as dry as possible and reduce humidity levels by increasing air circulation, if possible. Botrytis also preferentially colonizes dead or dying plant tissue, including fallen leaves and clippings (Figure 4.75). Therefore, sanitation that includes eliminating decaying leaves, flowers, and plant debris is important in disease prevention. Fungicides can reduce Botrytis infection; however, fungicides are usually not needed except in humid propagation houses.

Cercospora Leaf Spot

The fungus, *Pseudocercospora handelii* (Bubák) Deighton (syn. *Cercospora handelii* Bubák), is a widespread pathogen of both rhododendron and azalea. Infection causes reddish to dark brown, angular leaf spots (McArthur, 1959). As the leaf spots age, the center of the spot turns gray (Figure 4.76). Infection is often seen on new growth in the spring and fall under warmer, wet conditions. The fungus produces olivaceous clusters of elongated conidia on short conidiophores mostly on the upper leaf surface that are water-splashed to adjacent leaves. Severe blighting can occur on rhododendron and evergreen azalea in southeastern U.S. nurseries that can lead to defoliation and unmarketable crops (Figure 4.77). Research is limited, and much needed, on the biology of this pathogen. Fungicides can reduce infection when applied in the spring to protect new growth and continued on a regular schedule through the summer.

Figure 4.76 Dark brown, angular Cercospora leaf spots turn gray with age.

Gray Blight

The fungus, *Pestalotiopsis sydowiana* (Bres.) B. Sutton, is a weak pathogen and saprophyte of azalea and rhododendron (Linderman and Benson, 2014). The fungus often invades leaves injured by winter damage, sunscald, or other wounds. As a secondary invader, it causes brown to gray leaf spots that may coalesce and blight large sections of the leaf (Figure 4.78). Within the graying leaf spot, black pustules or spore-containing acervuli are easily seen. Gray Blight generally does not cause significant damage to mature plants. The affected leaves would likely drop from the plant; however, secondary infection by *Pestalotiopsis* quickens the defoliation. The disease can be a problem on stem cuttings in propagation because the fungus can infect stem wounds, girdling, and killing it. Fungicides are not regularly applied to manage this disease.

Figure 4.77 Left untreated, leaf spotting by Cercospora can become severe across rhododendron and azalea crops, leading to significant aesthetic injury and economic loss.

Figure 4.78 Rhododendron foliage affected by *Pestalotiopsis*, causal agent of gray blight in rhododendrons and azaleas.

Leaf Scorch

Leaf scorch, caused by the fungus *Phloeospora azaleae* (Voglino) Priest (syn. *Septoria azaleae* Voglino), only affects evergreen azalea cultivars under wet, humid conditions. Infected leaves develop irregular, angular, brown leaf spots with red to purple margins and yellow halo (Figure 4.79). Lower leaves are more likely to be infected. Under severe disease pressure, infected leaves prematurely defoliate, which can affect plant growth (McArthur, 1959; Raabe and Lenz 1958). The fungus produces black, spore-producing pycnidia on the fallen leaves that are water-splashed onto nearby plants.

Management

Management of fungal leaf spot diseases should begin with removal of fallen leaf litter from the nursery beds and where practical, on the surface of the container substrate. Prolonged periods of leaf wetness are required for infection and spread of fungal spores. Therefore, practices that allow plant foliage to dry quickly, such as watering earlier in the day, increasing plant spacing to allow for more air circulation around plants, and avoiding wetting the foliage in the evening hours can lessen disease development. Fungicides are often not needed for some leaf spot diseases with the exception of Cercospora leaf spot and anthracnose on evergreen azaleas in the southeastern U.S. Fungicide applications in the

Figure 4.79 Leaf scorch, caused by the fungus *Phloeospora azaleae* manifests as irregular, angular, brown leaf spots with red to purple margins and yellow halo.

spring beginning at bud break and continuing through the summer can help manage these leaf spot diseases and indirectly reduce the other leaf diseases of relatively minor importance.

Powdery Mildew

Powdery mildew occurs on rhododendron and deciduous and evergreen azaleas worldwide. The disease has increased in importance and incidence over the past 30 years in nurseries and landscapes in the U.S. and Europe (Basden and Helfer, 1995). It is generally not a significant problem within commercial nurseries in the southeastern U.S. where sprinkler irrigation and wet conditions reduce powdery mildew growth on leaf surfaces. Powdery mildew is caused by several different *Erysiphe* spp., most commonly including *E. azaleae* (U. Braun) U. Braun & S. Takam (syn. *Microsphaera azaleae*); *E. digitata* (A. J. Inman & U. Braun) A. J. Inman & U. Braun; and *E. vaccinii* Schwein. (syn. *M. vaccinii*) (Linderman and Benson, 2014). Other species have been reported; however, identification was based on asexual stages or obsolete morphological taxonomic classification and are likely in error.

Powdery mildew is recognizable because of the white growth consisting of fungal hyphae and asexual conidia (spores) on upper and lower leaf surfaces (Figure 4.80). When the white fungal growth is not present, infected leaves may show pale, chlorotic spots with diffuse margins with or without purple-brown leaf spotting directly opposite on the lower leaf surface, reddish to purple-tinted foliage, and purplish vein discoloration (Helfer, 1994; Linderman and Benson, 2014). On deciduous azalea, infected new growth may be distorted and crinkled. Symptoms vary with cultivar and powdery mildew resistance in known.

Figure 4.80 Typical appearance of powdery mildew on rhododendron leaves.

The disease is favored by high humidity (>85% RH) and low light (Kenyon et al., 2002). It is active over a wide range of temperatures 41-86°F (5-30°C); however, it is most active under warm 77°F (25°C) day and cool 59°F (15°C) conditions during the late spring and early fall months (Benson and Williams-Woodward, 2001). Optimum germination and hyphal growth is between 15-20°C (Kenyon et al., 1998). It is most often seen either on lower, shaded leaves or on succulent, new growth when grown in shaded environments. Powdery mildew fungi have both asexual and sexual reproduction. Growth and reproduction during the growing season is primarily via asexual conidia. Conidia are wind or mechanically dispersed. Conidia germinate with hyphae growing superficially across the leaf surface and only penetrating leaf epidermal cells to form haustoria as a means of absorbing nutrients from the cytoplasm of living cells. Conidia are produced singularly or in chains, depending upon the powdery mildew species, 10 to 12 days after infection (Kenyon et al., 1998). Late in the growing season, as temperatures cool, tan to black, enclosed sexual spore-producing structures (chasmothecia) are formed on the leaf surface. Sexual spores (ascospores) are released from the chasmothecia on fallen leaf litter in the early spring to infect succulent new growth during wet periods. In the southeastern U.S. and in other areas with mild winters, powdery mildew fungi may overwinter as dormant mycelium in leaf buds of evergreen hosts.

Management

Management of powdery mildew is best achieved through the use of resistant cultivars. Evaluations of deciduous azalea cultivars have been conducted in gardens and artificially inoculating plants under controlled conditions within growth chambers. Susceptibility has varied among the garden evaluations due to changes in environmental conditions and disease incidence (Long et al., 2010). The cultivars 'Fragrant Star', 'Garden Party', 'June Flame', 'Late Lady', 'Lollipop', 'Magic', 'Millennium', 'Parade', 'Pink and Sweet', 'Popsicle' and 'Snowbird' were found to be resistant to highly resistant across two locations and multiple years (Long et al., 2010). The most susceptible cultivars in that study included 'Arneson Gem', 'Arneson Ruby', 'Cannon's Double', 'Cheerful Giant', 'Gibraltar', 'Irene Koster', 'Klondyke', 'Mount Saint Helen', 'Orange Jolly', 'Orchid Lights', 'Rosy Lights', 'Strawberry Ice', 'Western Lights' and 'Yellow Cloud'. Of the rhododendrons, the least susceptible species are *R. degronianum* subsp. *yakushimanum* (Nakai) H. Hara (and its hybrids), *R. albiflorum* Hook., and *R. macrophyllum* D. Don ex G. Don) (Linderman and Benson, 2014). Highly susceptible rhododendrons are those hybridized with *R. cinnabarinum* Hook. f. and *R. campylocarpum* Hook. f. (Linderman and Benson, 2014).

Powdery mildew disease development is dependent on environmental conditions. Changing the growing sites, such as growing plants in full sun or light shade, reducing relative humidity by increasing plant spacing to facilitate air circulation, and watering early in the day to allow foliage to dry and humidity to lessen during the evening hours. High rates of nitrogen fertilizer may increase susceptibility of young, succulent growth. Scouting plants regularly for the first sign of powdery mildew growth and prompt application of fungicides can reduce disease development and spread. Demethylation inhibiting (DMI) fungicides (FRAC 3) containing metaconazole, myclobutanil, propiconizole, tebuconazole and triadimefon are very effective in reducing powdery mildew development. Other quinone outside [site] inhibiting (QoI) fungicides (FRAC 11) such as azoxystrobin, pyraclostrobin and trifloxystrobin, as well as potassium bicarbonate and copper-containing fungicides are also effective.

Rhizoctonia Web Blight

Web blight, caused by binucleate *Rhizoctonia* spp., is common on azaleas in southern and southeastern U.S. ornamental plant nurseries. Although, *Rhizoctonia* spp. can affect and cause dieback of rhododendron stem cuttings during propagation, it usually does not present a major problem for rhododendron production. Azalea cultivar susceptibility varies. Compact, evergreen azaleas are the most susceptible, particularly cultivars in the Glenn Dale, Kurume, Robin Hill, and Satsuki groups (Wehler and Cox, 1966). In addition to azalea, web blight also may affect compact plants, such as Japanese holly (*Ilex crenata*), juniper (*Juniperus chinesis*), *Cotoneaster* sp., Indian hawthorn (*Rhaphiolepis indica*), *Pittosporium* sp., crape myrtle (*Lagerstroemia indica*), and arborvitae (*Thuja occidentalis*) (Benson and Jones, 2001).

Rhizoctonia infection is favored by warmer temperatures (between 77-86°F (25-30°C) for 6-8 hr per day), high relative humidity (>95% for >8 hr per day), and moist conditions (>6 hr of leaf wetness) (Copes and Scherm, 2010). These conditions are widely prevalent within southern nurseries during the summer months. However, *Rhizoctonia* spp. can be recovered year-round from asymptomatic azalea stem tissue in southern nurseries (Copes et al., 2011). Symptoms and recovery of Rhizoctonia may also be prevalent during spring and fall months in propagation areas or in heated glass- or plastic-covered houses wherever warmer, humid conditions exist.

Figure 4.81 Rhizoctonia blight may be difficult to detect early in infection. Leaves in the canopy interior are the first to become leaf spotted, necrotic, and then drop prematurely.

Much research has occurred in recent years identifying the *Rhizoctonia* spp. involved in web blight disease of azalea. *Rhizoctonia solani* J. G. Kuhn [syn. *Thanatephorus cucumeris* (A.B. Frank) Donk], *Rhizoctonia ramicola* W. A. Weber & D. A. Roberts (syn. *Ceratobasidium ramicola* C.C. Tu, Roberts & Kimbr.) and several anatamosis groups (AG) of binucleate *Rhizoctonia* (syn. *Ceratobasidium* D. P. Rogers) have been recovered from infected leaf tissue (Frisina and Benson, 1987; Frisinia and Benson, 1989; Rhinehart et al., 2007; Wehler and Cox, 1966). In Alabama and Mississippi nurseries, the predominant species has been binucleate Rhizoctonia AG-U (syn. AG-P) (Rhinehart et al., 2007).

Symptoms of Rhizoctonia web blight begin within the inner canopy and are often overlooked. Individual leaves within the lower and inner canopy may be develop leaf spotting and necrosis (Figure 4.81). The blight progresses outward and upward through the canopy until much of the foliage is necrotic. Individual leaves will drop from the plant, but may appear to remain attached to the stems because Rhizoctonia hyphae mats the leaves together and to the stems thereby holding the leaves in place. Often this results in leaves dangling from stems by thread-like hyphae. As temperatures cool during fall months, the fungus stops growing and the affected leaves drop from the plant. The fallen leaf litter within containers and on gravel or polypropylene-covered nursery pads can serve as a source of inoculum for infections the following year. Small-diameter stems can be killed by Rhizoctonia infection resulting in stem dieback.

Rhizoctonia spp. do not produce spores. The fungus spreads via hyphal growth across rooting substrate and plant tissues. Colonized leaf debris and soil or medium particles can be water splashed to adjacent plants and containers. Leaf debris may also be wind-blow to new locations. Although one study did not identify Rhizoctonia spreading from contaminated propagation house floors into rooted azalea stem cuttings, Rhizoctonia will survive within colonized organic material such as leaf debris and organic fines surrounding gravel or on polypropylene fabric used in propagation houses for weeks (Copes, 2015b).

Management. Management of web blight begins with sanitation. Rhizoctonia can survive within contaminated plant debris on gravel or polypropylene plastic covered nursery pads, within container growing substrate, and along the stems of infected plants (Copes et al., 2011). Fallen leaf litter should be removed to reduce Rhizoctonia survival. Rhizoctonia may also be introduced into propagation areas on infected cuttings. Submerging azalea stem cuttings in hot water (122°F; 50°C) for 21 minutes was shown to eliminate Rhizoctonia from the cuttings without damaging the plant and reducing the need for fungicide applications (Anon., 2010; Copes and Blythe, 2009). Rhizoctonia hyphae can grow from plant to plant and along the surface of growing media within propagation trays (Copes et al., 2011). It is logical to think that increasing plant spacing will reduce disease spread. Although increasing plant spacing can delay the onset of web blight symptoms compared to non-spaced plants, spacing has not been shown to reduce disease spread (Copes and Scherm, 2005). Furthermore, production practices that reduce humidity, such as irrigating at midday rather than early morning or late evening can help slow, but not reduce, disease onset (Copes and Scherm, 2010).

Fungicides are effective and necessary to manage web blight in azalea. Attempts to identify environmental factors that can be used to model disease development and target fungicide applications have been unsuccessful. Because many plant nurseries use overhead sprinkler irrigation that wets the foliage canopy daily, weather information does not accurately predict disease onset (Copes and Scherm, 2010) nor do weather-based models provide greater disease control or ease of use over calendar sprays (Copes, 2015a). The most effective means of managing Rhizoctonia web blight is by applying fungicides on a calendar basis (Copes and Scherm, 2010; Copes et al., 2012). Two to three applications of azoxystrobin, chlorothalonil, fludioxonil, flutolanil, pyraclostrobin, thiophanate methyl (higher labeled rate), or trifloxystrobin fungicides applied beginning the first week of July and continuing at 14 to 21-day intervals depending upon the product used through the late August will effectively control web blight disease development. Fungicides are also recommended prior to covering azaleas under winter-protective covers.

Leaf Rusts

Several leaf rusts have been described on *Rhododendron* spp. in the southeastern US, including *Chrysomyxa roanensis* (Arthur) Arthur affecting *R. catawbiense*, *C. nagodhii* P. E. Crane on cultivated rhododendrons, and *Thekopsora minima* Syd. & Syd. (syn. *Pucciniastrum minimum* Arthur) on numerous *Rhododendron* spp. and hybrids. Rusts have complicated lifecycles and often require two unrelated hosts to complete it. *Chrysomyxa roanensis* produces uredinia and telia on *Rhododendron* spp. and spermogonia and aecia on *Picea* spp. (Spruce). *Thekopsora minima* produces uredinia and telia on Ericaceae hosts, including *Rhododendron* spp., and aecia on *Tsuga* spp. (Hemlock) (Farr and Rossman, n.d.).

Figure 4.82 *Piccuiastrum vaccinii* on deciduous azalea foliage.

Leaf rusts produce similar symptoms on rhododendron and azalea making causal rust species identification difficult. Leaf rusts (Figure 4.82) are characterized by golden, orange or brown-colored spore masses arising from pustules on the leaf underside (Figure 4.83). Upper leaf surfaces may exhibit light green to yellow spotting with the white rust pustules forming directly beneath the spot on the leaf underside (Figure 4.84). Rust infection usually does not cause significant damage to rhododendron or azalea; however, if disease pressure is high, photosynthesis is impaired and leaves often prematurely defoliate.

Most rust pustules on rhododendron are evident during the summer to early fall. *Thekopsora* spores produced on rhododendron are wind-blown to hemlock where the rust fungus overwinters and produces spores on the needles that are wind-blown back to rhododendron the following summer. *Chrysomyxa* rusts infect rhododendron leaves during the spring and summer. Often rust pustules do not develop on rhododendron during the first season following infection. Rather, the rust fungus grows intercellularly and overwinters as dormant mycelia within the leaves (Linderman and Benson, 2014). The following spring, spores are produced that are wind-blown to spruce species. Spores produced on the spruce needles are wind-blown back to rhododendron hosts in the summer to complete the rust lifecycle. One spore stage, urediniospores, is often referred to as the summer-repeating stage because this spore stage produced on rhododendrons can re-infect rhododendron throughout the growing season. This leads to an increase in disease severity and premature rhododendron leaf drop.

Figure 4.83 Closeup of *Pucciniastrum vaccinii* on the underside of deciduous azalea foliage.

Management

Although leaf rusts don’t cause significant damage to rhododendron, they can make the crop unmarketable. Management practices such as removing fallen leaf litter and infected leaves can reduce rust survival, but they are impractical within larger ornamental plant nurseries. Leaf wetness should be kept at a minimum by irrigating by emitters rather than overhead sprinklers and irrigating early in the day so plants do not go through the evening hours wet. Fungicide sprays applied in the spring to protect new growth and continuing through the

Figure 4.84 Closeup of *Pucciniastrum vaccinii* on the top of deciduous azalea foliage.

summer can reduce rust infection. Flutolanil and tebuconazole are labeled for use on rust diseases on rhododendron (Table 4.10).

Flower and Leaf Bud Diseases

Exobasidium Leaf and Flower Gall

Leaf and flower gall (Figure 4.85) is mostly an aesthetic disease in landscapes and nurseries that can interfere with flowering and reduce plant growth over time. It is typically not detrimental to plant health or growth. However, flower gall formation on native flame azalea [*R. calendulaceum* (Michx.) Torr.] negatively impacted flowering, seed set and the potential survival of the species in the Appalachian Mountains of western Virginia (Wolfe and Rissler, 2000). Leaf and flower gall on rhododendron and azalea is caused by the fungus, *Exobasidium vaccinii* (Fuckel) Woronin. A similar *Exobasidium* sp., *E. burtii* Zeller, is believed to cause circular white to yellow leaf spots both on the underside (Figure 4.86) and top of the leaves (Figure 4.87), rather than gall-like symptoms.

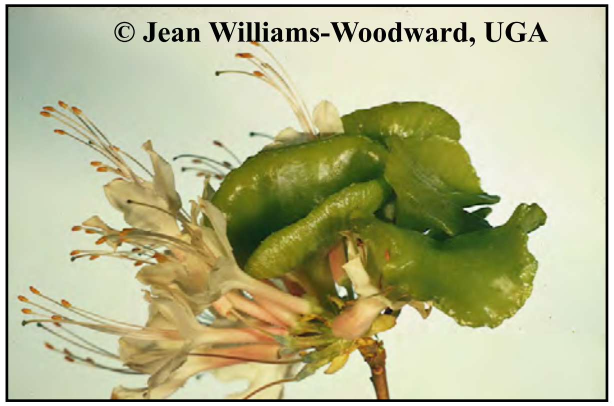


Figure 4.85 A fleshy Exobasidium gall on leaf tissues adjacent to azalea flowers. Galls can also form on floral tissues.

Symptoms of *E. vaccinii* infection include a thickening of developing shoots and flowers. The galls may range from small swellings on the edges of leaves to larger, fleshy amorphous galls from colonization of the entire leaf or floral parts on evergreen azalea and rhododendron to bell-shaped, fleshy galls extending from flowers or leaf tips of *R. canescens*. The galled tissue is initially pale green to pinkish, but turns white as it becomes covered with a hymenial layer of fungal spores (basidia) (Figure 4.88). As the galls age, they turn brown, hard and shrink. Hardened older galls may remain on the plant or may drop as leaves and flowers senesce. Yellow leaf spot, caused by *E. burtii*, does not produce a gall, but rather a white to yellow circular leaf spot on native rhododendron, particularly *R. maximum*. The spot is yellow on the upper leaf surface with a white hymenial layer and basidia directly beneath the spot on the leaf underside. As the leaf spot ages, it turns reddish brown from the center outward.

The lifecycle of *E. vaccinii*, particularly the overwintering of the pathogen, is not completely understood. It is believed that basidiospores produced on the galls overwinter within bud scales on host plants because infection occurs in the spring as leaf and flower buds break bud under cooler, wet conditions (Graafland, 1960). Overhead irrigation and shaded growing areas within nurseries provide ideal conditions for disease development. After infection, the fungus grows intercellularly as the gall forms due to hormonal

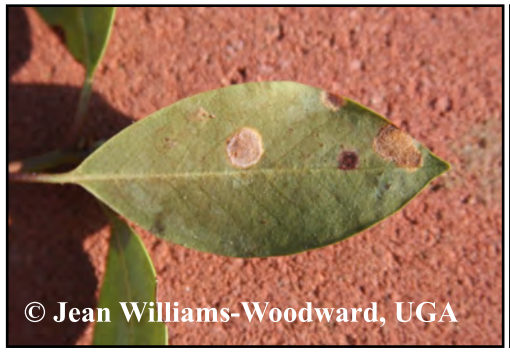


Figure 4.86 Exobasidium leaf gall evident on rhododendron as a yellowish leaf spot on the lower leaf surface.

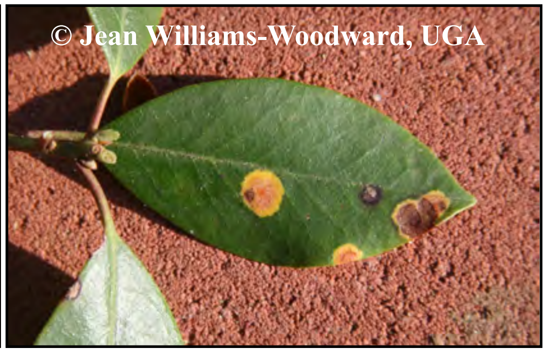


Figure 4.87 A yellow leaf spot on rhododendron caused by Exobasidium leaf gall (upper leaf surface).

changes within the plant. New basidiospores are produced on the young galls that may be water-splashed or wind-dispersed to newly developing buds as secondary infection or may lay dormant within bud scales until bud break the following year. Older growth is immune to infection. Only newly expanding buds are susceptible. Most azalea and rhododendron cultivars, as well as other ericaceous hosts, are susceptible to exobasidium infection with Southern Indica azaleas, *R. maximum*, *R. catawbiense*, and their hybrids are very susceptible (Saville, 1959).

The lifecycle of *E. burtii*, causing a leaf spot on *R. maximum*, is also not completely understood. Infection occurs in the spring and continues until leaves mature. Infection does not appear to significantly affect plant growth or flower formation. Incidence of *Exobasidium* leaf spot increases in springs with abundant rainfall.

Figure 4.88 This fleshy exobasidium gall on *Rhododendron catawbiense* foliage has become whitish as fungal spores mature.

Management

Because infection is only on newly developing leaf and flower buds, control is directed at protecting new growth in the spring. Applications are not always successful if applied after bud break. Fungicides are not needed unless severe infection has been seen in the past. Where practical, removing galled tissue before it turns white and releases basidiospores can reduce infection and disease development the following year.

Ovulinia Petal Blight

Ovulinia petal blight is a damaging disease affecting the flowers of primarily azalea cultivars, but rhododendron flowers are also affected. It is caused by the fungus, *Ovulinia azaleae* F. A. Weiss. The disease only affects the flowers. Symptoms appear as small, translucent spots on flower petals (Figure 4.89). The spots enlarge rapidly under cool, wet conditions until the entire petal becomes blighted and slimy. Infected flowers easily disintegrate when rubbed between fingers. This differentiates the disease from freeze injury in which the injured flowers will not break apart.

Figure 4.89 Small spots appear translucent on azalea petals infected with Ovulinia petal blight fungus.

Ovulinia azaleae overwinters as hardened, black sclerotia on diseased flowers littering the soil or nursery be surface or on those still clinging to the plant (Plakidas, 1949). The sclerotia germinate producing a very small, cup-like ascocarp (apothecium) from which ascospores are ejected that contact lower flowers or are wind-dispersed to adjacent plants. Ascospores germinate on the flowers causing initial infection from which secondary, asexual conidia are produced on the flower surface. Conidia may be spread by water-splash, air currents or by insects to adjacent flowers. Epidemics of the disease can occur within days following initial infection, which can shorten and destroy the blooms. Infection is favored by high humidity, plant wetness, and cooler temperatures of 55-70°F (10-22°C) with 64°F (18°C) being optimal during flowering (Bertus, 1974). Early and late flowering cultivars and species may escape infection.

Management
The primary means of reducing Ovulinia petal blight is to protect flowers by applying fungicide sprays at weekly intervals beginning at initial flower bud break and continuing throughout the flowering period (Peterson and Davis, 1977). The fungicides, mancozeb, myclobutanil, propiconazole, and thiophanate methyl are effective in reducing petal blight disease development. Removing fallen, dried, flowers harboring sclerotia can help reduce initial infection; however, it is often not practical nor reliable. Pruning infected flower trusses can help reduce survival of the sclerotia in the area and can reduce disease in future years.

Nematodes
Nematode diseases are of minor importance. Root-infecting nematodes such as root-knot (*Meloidogyne* spp.) and stunt (*Tylenchorhynchus claytoni* Steiner) nematodes are not common within containerized rhododendron and azaleas grown in soil-less container substrates. The most commonly seen nematode on evergreen azalea within nurseries is the foliar nematode [*Aphelenchoides fragariae* (Ritzema Bos) Christie]. Foliar nematode feeding causes yellow to dark brown lesions that are restricted by the leaf veins (Kohl, 2011). A diagnostic feature of foliar nematode lesions that distinguished it from fungal or bacterial leaf diseases is the occurrence of a light to dark color gradient for individual lesions. Foliar nematodes readily move in and out of wet leaf surfaces. Keeping the foliage as dry as possible can reduce foliar nematode spread and infestation. Foliar nematodes reside within infested leaves. Pruning and discarding infested branches can help manage foliar nematode populations. Chemical nematicides are not labeled for use on ornamental plants.

Tissue Proliferation
Like other ericaceous plants (e.g., *Kalmia* and *Pieris*), rhododendron cultivars, like 'Montego' and others, can develop abnormal proliferation of dwarfed shoots or callus-like tissues, particularly at the crown of micropropagated plants (Brand and Kiyomoto, 1992; LaMondia et al., 1997). Resulting growths look like *Agrobacterium*-induced crown gall, but unlike crown gall, tissues are not contagious to other plants in culture. The cause of this abnormal growth is not well understood and may result from epigenetic changes to plantlets induced by tissue culture processes and accelerated growth conditions in nursery production (Linderman and Benson, 2014). Tissues exhibiting characteristic symptoms appear to be overproducing cytokinins (Brand and Kiyomoto, 1992).

References

Allen, H.W. 1959. The Japanese weevil *Pseudocneorhinus bifasciatus* Roelofs. J. Econ. Entomol. 52:586-58.

Alfieri, S. A., Linderman, R. G., Morrison, R.H., and Sobers, E. K. 1972. Comparative pathogenicity of *Calonectria theae* and *Cylindrocladium scoparium* to leaves and roots of azalea. Phytopathology 62:647-650.

Alston, D. 2010. Speckled Green fruitworm (*Orthosia hibisci*). Utah Pests Fact Sheet ENT-141-10. http://utahpests.usu.edu/ipm/htm/fruits/fruit-insect-disease/fruitworm10. Accessed 15 May 2015.

Anonymous. 2004. Black vine weevil. *In*: IPM of Midwest Landscapes, pp. 77-78. V. Krischik and J. Davidson, eds. Minnesota Agricultural Experiment Station Publication SB-07645.

Anonymous. 2010. Research Summary: Eliminating *Rhizoctonia* from azalea cuttings. J. Amer. Rhododendron Soc. 65:17.

Anonymous. 2015. Henning's Rhododendron and Azalea page. www.rhodyman.net/rhodyhy.html Accessed 30 Sept 2015.

Ansari, M.A. and T.M. Butt. 2011. Effect of potting media on the efficacy and dispersal of entomopathogenic nematodes for control of black vine weevil, *Otiorhynchus sulcatus* (Coleoptera: Curculionidae). Biol. Contr. 58:310-318.

Appleton, B.L. 1989. Evaluation of nursery container designs for minimization or prevention of root circling. J. Environ. Hort. 7:59-61.

Appleton, B.L. and S.C. French. 1999. Improving root systems on container-grown azaleas. J. Amer. Rhododendron Soc. 53:6-7.

Arora, R., Y. Peng, D. Karlson, J.L. Reyes, and A.A. Covarrubias. 2008. Physiology of cold-hardening in *Rhododendron*: role of a dehydrin protein from *R. catawbiense* (RcDhn5) in cryoprotection and improving freezing tolerance. J. Amer. Rhododendron Soc. 62:153-158.

Babadoost, M. and C. Pavon. 2013. Survival of oospores of *Phytophthora capsici* in soil. Plant Disease 97:1478-1483.

Bailey, N.S. 1951. The Tingoidea of New England and their biology. Entomol. Am. 31:1-140.

Bailey, K.L. J. Derby, S.M. Boyetchko, K. Sawchyn, E. Becker, G. Sumampong, S. Shamoun, D. James, S. Masri, and A. Varga. 2012. *In vivo* studies evaluating commercial biofungicide suppression of blight caused by *Phytophthora ramorum* in selected ornamentals. Biocontrol Science and Technology 22:1268-1283.

Baker, A.C. and M.L. Moles. 1920. A new species of Aleyrodidae found on Azalea (Hom.). Proc. Entomol. Soc. Wash. 22:81-83.

Baker, J.R. 1994. Azalea whitefly. *In*: Insects and Related Pests of Flowers and Foliage Plants. NC Coop. Ext. Serv. AG-136. 106 p. http://ipm.ncsu.edu/AG136/index.html Accessed 15 May 2015.

Baker, J.R. (ed.) 1980. Insect and Related Pests of Shrubs. North Carolina State University Cooperative Extension Services, Bulletin AG-189. Raleigh, NC. 199 pp.

Balsdon, J.A., S.K. Braman, A.F. Pendley and K.E. Espelie. 1993. Potential for integration of chemical and natural enemy suppression of azalea lace bug (Heteroptera: Tingidae). J. Environ. Hort. 11:153-156.

Balsdon, J.A., K.E. Espelie, and S.K. Braman. 1995. Epicuticular lipids from azalea (*Rhododendron* spp.) and their potential role in host plant acceptance by azalea lace bug, *Stephanitis pyrioides* (Heteroptera: Tingidae). Biochem. Syst. Ecol. 23:477-485.

Banko, T.J. and M. Stefani. 1996. Do chemical growth regulators stimulate new shoot growth and improve overwintering of deciduous azalea cuttings? J. Amer. Rhododendron Soc. 50:32-33.

Barbour, J.D., J.C. Millar, J. Rodstein, A.M. Ray, D.G. Alston, M. Rejzek, J.D. Dutcher and L.M. Hanks. 2011. Synthetic 3,5-dimethyldodecanoic acid serves as a general attractant for multiple species of *Prionus* (Coleoptera: Cerambycidae). Ann. Entomol. Soc. Amer. 104:588-593.

Basden, N. and Helfer, S. 1995. World survey of *Rhododendron* powdery mildews. J. Amer. Rhodo. Soc. 49:147-56.

Benham, G.D., Jr. and R.J. Farrar. 1976. Notes on the biology of *Prionus laticollis* (Coleoptera: Cerambycidae). Can. Entomol. 108:569-576.

Benson, D. M. and F. D. Cochran. 1980. Resistance of evergreen hybrid azaleas to root rot caused by *Phytophthora cinnamomi*. Plant Dis. 64:214-215.

Benson, D. M., Daughtrey, B. I., and Jones, R. K. 1990. Botryosphaeria dieback in hybrid rhododendron, 1986-1990. Biol. Cult. Tests Control Plant Dis. 6:108.

Benson, D. M. and Jones, R. K. 2001. Rhizoctonia web blight. In: Diseases of Woody Ornamentals and Trees in Nurseries. Eds: D. M. Benson and R. K. Jones. American Phytopathological Society. St. Paul, MN. pp: 63-64.

Benson, D. M. and Williams-Woodward, J. L. 2001. Azalea Diseases. In: Diseases of Woody Ornamentals and Trees in Nurseries. Eds: D. M. Benson and R. K. Jones. American Phytopathological Society. St. Paul, MN. pp: 81-88.

Bentz, J.A. 2003. Shading induced variability in azalea mediates its suitability as a host for the azalea lace bug. J. Amer. Soc. Hort. Sci. 128:497-503.

Bentz, J. and A.M. Townsend. 2005. Diversity and abundance of f species (Homoptera: Cicadellidae) among red maple clones. J. Ins. Conserv. 9:29-39.

Bertus, A. L. 1974. Azalea petal blight – Its life cycle and control. Proc. Int. Plant Prop. Soc. 24:274-279.

Beshear, R.J. and S. Nakahara. 1975. *Aleurodothrips fasciapennis* (Thysanoptera: Tubulifera) a predator of *Aspidiotus nerii* (Homoptera: Diaspididae). J. Georgia Entomol. Soc. 10:223-224.

Bienapfl, J.C. and Y. Balci. 2014. Movement of *Phytophthora* spp. in Maryland's nursery trade. Plant Disease 98:134-144.

Bir, R.E. 1992. Growing and propagating showy native woody plants. University of North Carolina Press, Chapel Hill, NC.

Bir, R.E. and J.L. Conner. 1998. Increasing flowers in container grown hybrid rhododendron. Proc. SNA Research Conference 43:282-285.

Blazich, F.A. and D. B. Rowe. 2008. *Rhododenron* L. Woody Plant Seed Manual. USDA FS Agriculture Handbook 727. pp. 943-951. http://www.nsl.fs.fed.us/Q&R%20genera.pdf Accessed 30 Sept 2015.

Bounfour, M. and L.K. Tanigoshi. 2001. Effect of temperature on development and demographic parameters of *Tetranychus urticae* and *Eotetranychus carpini borealis* (Acari: Tetranychidae). Proc. Annu. Entomol. Soc. Amer. 94:400-404.

Braman, S.K. and R.J. Beshear. 1994. Seasonality of predaceous plant bugs (Heteroptera: Miridae) and phytophagous thrips (Thysanoptera: Thripidae) as influenced by host plant phenology of native azaleas (Ericales: Ericaceae). Environ. Entomol. 23:712-718.

Braman, S.K., A.F. Pendley, B. Sparks and W.G. Hudson. 1992. Thermal requirements for development, population trends, and parasitism of azalea lace bug (Heteroptera: Tingidae). J. Econ. Entomol. 85:870-877.

Brand, M. and Kiyomoto, R. 1992. Abnormal growths of micropropagated elepidote rhododendrons. Proc. Int. Plant Prop. Soc. 42: 530-534.

Buntin, G.D., S.K. Braman, D.A. Gilbertz, and D.V. Phillips. 1996. Chlorosis, photosynthesis, and transpiration of azalea leaves after azalea lace bug (Heteroptera: Tingidae) feeding injury. J. Econ. Entomol. 89:990-995.

Carter, C.C., K.F. Horn, D. Kline, J.R. Baker, J. Scott, and H. Singletary. 1980. with the collaboration of D. L. Stephan. Insect and Related Pests of Shrubs. http://ipm.ncsu.edu/AG189/html/Dogwood_Twig_Borer.HTML Accessed 29 Mar. 2014.

Casey, C.A. and M.J. Raupp 1999a. Effect of supplemental nitrogen fertilization on the movement and injury of azalea lace bug (*Stephanitis pyrioides* (Scott) in container-grown azaleas. J. Environ. Hort. 17:95-98.

Casey, C.A. and M J. Raupp 1999b. Supplemental nitrogen fertilization of containerized azaleas does not affect performance of azalea lace bug (Heteroptera: Tingidae). Environ. Entomol. 28:998-1003.

Chadwick, C.E. 1965. A review of Fuller's rose weevil (*Pantomorus cervinus* Boheman) (Coleoptera, Curculionidae). J. Entomol. Soc. Australia (N.S.W.) 2:10-20.

Chappell, M. and C. Robacker. 2006. Leaf was extracts of four deciduous azalea genotypes affect azalea lace bug (*Stephanitis pyrioides* (Scott) survival rates and behavior. J. Amer. Soc. Hort. Sci. 131:225-230.

Chiu, C.-H. and C.A. Kouskolekas. 1980. Observations on reproductive biology of tea scale, *Fiorinia theae* Green. J. Georgia Ento. Soc. 15:317-327.

Chong, J.-H., S.A. White, and N. Ward. 2012. Cherry – *Prunus* spp., p. 79-108. *In*: A.F. Fulcher and S.A. White (eds.). IPM for select deciduous trees in southeastern US nursery production. Southern Nursery IPM Working Group, Knoxville, TN.

Clark, M.B. 2000. A study involving epicuticular leaf waxes and nitrogen nutrition, and their effects on the resistance of two deciduous azaleas to the azalea lace bug. M.S. Thesis. Univ. Georgia, Athens, GA.

Coffelt, M.A. and P.B. Schultz. 1988. Influence of plant growth regulators on the development of the azalea lace bug (Hemiptera: Tingidae). J. Econ. Entomol. 81:290-292.

Cole, D.M., J.L. Sibley, E.K. Blythe, D.J. Eakes, and K.M. Tilt. 2005. Effect of cotton gin compost on substrate properties and growth of azalea under differing irrigation regimes in a greenhouse setting. HortTechnology 15:145-148.

Cooper, R.M. and R.D. Oetting. 1987. Hymenopterous parasitoids of tea scale and camellia scale in Georgia. J. Ento. Sci. 22:297-301.

Copes, W.E. 2015a. Weather-based forecasting of Rhizoctonia web blight development on container-grown azalea. Plant Dis. 99:100-105.

Copes, W.E. 2015b. Spread potential of binucleate *Rhizoctonia* from nursery propagation floors to trays containing azalea stem cuttings and sanitary control options. Plant Dis. 99:842-847.

Copes, W.E. and E.K. Blythe. 2009. Chemical and hot water treatments to control *Rhizoctonia* AG P infesting stem cuttings of azalea. HortScience 44:1370-1376.

Copes, W.E. and E.K. Blythe. 2011. Rooting response of azalea cultivars to hot water treatment used for pathogen control. HortScience 46:52-56.

Copes, W.E. and E.K. Blythe. 2012. Response of azalea cuttings to leaf damage and leaf removal. Proc. SNA Research Conference 57:287-290.

Copes, W.E., M. Garcia-Rodriguez-Carres, T. Toda, T.A. Rinehart, and M.A. Cubeta. 2011. Seasonal prevalence of species of binucleate *Rhizoctonia* fungi in growing medium, leaf litter, and stems of container-grown azalea. Plant Dis. 95:705-711.

Copes, W.E., A. Hagan, and J. Olive. 2012. Timing of fungicides in relation to calendar date, weather, and disease thresholds to control Rhizoctonia web blight on container-grown azalea. Crop Prot. 42:273-280.

Copes, W.E. and H. Scherm. 2005. Plant spacing effects on microclimate and Rhizoctonia web blight development in container-grown azalea. HortScience 40:1408-1412.

Copes, W.E. and H. Scherm. 2010. Rhizoctonia web blight development on container-grown azalea in relation to time and environmental factors. Plant Dis. 94-891-897.

Copes, W.E., X. Yang, and C.X Hong. 2015. *Phytophthora* species recovered from irrigation reservoirs in Mississippi and Alabama nurseries and pathogenicity of three new species. Plant Dis. 99:1390-1395.

Cowles, R.S. 1995. Black vine weevil biology and management. J. Amer. Rhododendron Soc. 49:83-85, 94-97.

Cowles, R.S. 1997. Several methods reduce insecticide use in control of black vine weevils. Frontiers Plant Sci. 49:2-4.

Cowles, R.S. 2001. Protecting container-grown crops from black vine weevil larvae with bifenthrin. J. Envir. Hort. 19:184-189.

Cowles, R.S. 2003. Practical black vine weevil management. J. Amer. Rhododendron Soc. 57:219-222.

Cox, R.S. 1969. *Cylindrocladium scoparium* on azalea in south Florida. Plant Dis. Rep. 53:139.

Creswell, T.C. and R.D. Milholland. 1988. Spore release and infection periods of *Botryosphaeria dothidea* on blueberry in North Carolina. Plant Dis. 72:342-346.

Cresswell, C.G. and R.G. Weir. 1997. Plant Nutrient Disorders 5.: Ornamental Plants and Shrubs. NSW Agriculture. Inkata Press, Sydney, Australia. 233p.

Crous, P.W., B. Slippers, M.J. Wingfield, J. Rheeder, W.F.O. Marasas, A.J.L. Philips, A. Alves, T. Burgess, P. Barber, and J.Z. Groenewald. 2006. Phylogenetic lineages in the Botryosphaeriaceae. St. Mycology 55:235-253.

Culin, J.D., C.S. Gorsuch, and T.M. Pizzuto. 1993. Natural history and recommendations for control of the rhododendron stem borer, *Oberea myops*. J. Amer. Rhododendron Soc. 47:206-209.

Dailey, A., M. Toohey, J. Parke, R. Linderman, and J. Pscheidt. 2004. *Phytophthora ramorum*: A guide for Washington nurseries. Oregon State University Extension Service publication. http://agr.wa.gov/PlantsInsects/Diseases/SOD/docs/PhytophthoraRamorumGuide.pdf Accessed 16 May 2015.

Day, E. 2009. Scale insects. http://pubs.ext.vt.edu/2808/2808-1012/2808-1012.html. Accessed 21 May 2015.

Deckle, G.K. 2013. Azalea leafminer, *Caloptilia azaleella* (Brants) (Insecta: Lepidoptera: Gracillaridae). Univ. FL IFAS-EDIS, EENY-379 (IN736). http://edis.ifas.ufl.edu/in736 Accessed 16 May 2015.

Denmark, H.A. and T.R. Fasulo. 2013. Greenhouse thrips, *Heliothrips haemorrhoidalis* (Bouche) Insecta: (Thysanoptera: Thripidae). Univ. FL IFAS-EDIS, EENY-075. http://edis.ifas.ufl.edu/in732 Accessed 16 May 2015.

Dickerson, E.L. 1917. Notes on *Leptobrysa rhododendri* Horv. J. N.Y. Entomol. Soc. 25:105-112.

Dirr, M.A. and C.W. Heuser, Jr. 2006. The Reference Manual of Woody Plant Propagation. Varsity Press, Cary, NC.

Drake, C.J., and F.A. Ruhoff. 1965. Lace bugs of the world: a catalogue (Hemiptera: Tingidae). Bull. U. S. Nat. Hist. Mus. 243:1-634.

Dunbar, D.M. 1977. Bionomics of the andromeda lacebug, *Stephanitis takeyai*. pp. 277-289. *In:* R. L. Bearl (ed.). Memoirs of the Conneticut Entomological Society (25th Anniversary). Conn. Entomol. Soc., New Haven, CT.

Edwards, L., C.H. Gilliam, G.B. Fain, and J.L. Sibley. 2012. Eastern red cedar as an alternative substrate in nursery production. Proc. SNA Research Conference 57:11-15.

Eggers, J.E., Y. Balci, and W.L. MacDonald. 2012. Variation among *Phytophthora cinnamomi* isolates from oak forest soils in the eastern United States. Plant Dis. 96:1608-1614.

Eichlin, T.D. and W.D. Duckworth. 1988. The Moths of America North of Mexico. Fasc. 5.1: Sesioidea: Sesiidae. The Wedge Entomological Research Foundation, Washington, DC, 176 p.

Engelhardt, G.P. 1946. The North American clearwing moths of the family Ageriidae. U.S. National Museum Bulletin 190:1-222.

English, L.L. and G.F. Turnipseed. 1940. Insect pests of azaleas and camellias and their control. Alabama Polytechnic Institute Agricultural Experiment Station Circular 84. 18 pp.

Erwin, D.C. and O.K. Ribeiro. 1996. *Phytophthora* Diseases Worldwide. St. Paul, MN: The American Phytopathological Society.

Farr, D.F. and A.Y. Rossman. Fungal Databases, Systematic Mycology and Microbiology Laboratory. Agricultural Research Service, U.S. Department of Agriculture. Available online at http://nt.ars-grin.gov/fungaldatabases

Fisher, J.R. 2006. Fecundity, longevity and establishment of *Otiorhynchus sulcatus* (Fabricius) and *Otiorhynchus ovatus* (Linnaeus) (Coleoptera Curculionidae) from the Pacific North-west of the United States of America on selected host plants. Agric. For. Entomol. 8:281-287.

Frank, S.D. and C.S. Sadof. 2011. Reducing insecticide volume and non-target effects of ambrosia beetle management in nurseries. J. Econ. Entomol. 104:1960-1968.

Frisina, T.A. and D.M. Benson. 1987. Characterization and pathogenicity of binucleate *Rhizoctonia* spp. from azaleas and other woody ornamental plants with web blight. Plant Dis. 71:977-981.

Frisina, T.A. and D.M. Benson. 1989. Occurrence of binucleate *Rhizoctonia* spp. on azalea and spatial analysis of web blight in container-grown nursery stock. Plant Dis. 73:249-254.

Fulcher, A., N. Ward Gauthier, W. Klingeman, F. Hale, and S.A. White. 2015. Blueberry culture and pest, disease, and abiotic disorder management during nursery production in the southeastern U.S.: A review. J. Envir. Hort. 33:33-47.

Galle, F.C. 1987. Azaleas. Timber Press, Portland, OR, 519 p.

García Morales, M., B.D. Denno, D.R. Miller, G.L. Miller, Y. Ben-Dov, and N.B. Hardy. 2016. ScaleNet: A literature-based model of scale insect biology and systematics. Database. doi: 10.1093/database/bav118. http://scalenet.info.

Ghimire, S.R., P.A. Richardson, P. Kong, J.H. Hu, J.D. Lea-Cox, D.S. Ross, G.W. Moorman, and C.X. Hong. 2011. Distribution and diversity of *Phytophthora* species in nursery irrigation reservoir adopting water recycling system during winter months. J. Phytopathology 159:713-719.

Gilkey, R. 1996. Correlation between weather conditions and rhododendron flower bud formation. J. Amer. Rhododendron Soc. 50:38-40.

Gill, S. and M. Raupp. 1988. Insecticidal soap as an azalea lacebug control. J. Am. Rhododendron Soc. 42:103-104.

Gill, S. and M. Raupp. 1989. Control of azalea lace bug using insecticidal soap and neem. J. Am. Rhododendron Soc. 43:216-217.

Gill, S. 2011. TPM/IPM Weekly Report for Arborists, Landscape Managers & Nursery Managers, April 8, 2011. http://www.ipmnet.umd.edu/landscape/LndscpAlerts/2011/11Apr08L.pdf. Accessed 23 July 2013.

Gill, S. 2013. TPM/IPM Weekly Report for Arborists, Landscape Managers & Nursery Managers, Univ. MD Extension. https://www.extension.umd.edu/sites/default/files/_docs/programs/ipmnet/13Apr12L.pdf Accessed 16 May 2015.

Glawe, D.A. and R.L. Hummel. 2006. New North American host records for *Seifertia azaleae*, cause of Rhododendron bud blight disease. Pacific Northwest Fungi 1(5):1-6. DOI: 10.2509/pnwf.2006.001.005

Goetsch, L.A., A.J. Eckert, and B.D. Hall. 2005. The molecular systematics of Rhododendron (Ericaceae): a phylogeny based upon RPB2gene sequences. Systematic Bot. 30:616–626. DOI:10.1600/0363644054782170. Accessed 3 July 2015.

González-Zamora, J.E., M.L. Castillo, and C. Avilla. 2012. Assessment of life history parameters of *Aspidiotus nerii* (Hemiptera: Diaspididae) to improve the mass rearing of *Aphytis melinus* (Hymenoptera: Aphelinidae). Biocontr. Sci. Tech. 22:791-801.

Gordh, G. and D.M. Dunbar. 1977. A new *Anagrus* important for the biological control of *Stephanitis takeyai* and a key to North American species. Fla. Entomol. 60:85-95.

Gotoh, T. 1997. Annual life cycles of populations of the two-spotted spider mite, *Tetranychus urticae* Koch (Acari: Tetranychidae) in four Japanese pear orchards. Appl. Entomol. Zoology 32:207-216.

Graafland, W. 1960. The parasitism of *Exobasidium japonicum* Shir. On azalea. Acta. Bot. Neerl. 9:347-379.

Grewal, P.S. 2012. Entomopathogenic nematodes as tools in integrated pest management. In: Integrated Pest Management: Principles and Practice. D. P. Abrol and U. Shankar, eds. CABI Publishing, Wallingford, England.

Gunderson, D.E., H. Elwood, A. Ingold, K. Kindle, and M.L. Sogin. 1987. Phylogenetic relationships between chlorophytes, chrysophytes and oomycetes. Proc. Nat.Aca. Sci. 84:5823-5827.

Harden, W. 1990. Growing rhododendrons in warmer climates. J. Amer. Rhododendron Soc. 44:140-142.

Harman, S.W. 1931. The cranberry rootworm as an apple pest. J. Econ. Entomol. 24:180-182.

Harrington, J.L. 1990. Rhododendron propagation. J. Amer. Rhododendron Soc. 44:154-160, 173-176.

Hatley, C.L. and J.A. McMahon. 1980. Spider community organization, seasonal variation and the role of vegetation architecture. Environ. Entomol. 9:632-639.

Helfer, S. 1994. Rhododendron powdery mildews. Acta Hortic. 364:155-159.

Helm, H. 2001. Root weevils: troublesome rhododendron pests. J. Amer. Rhododendron Soc. 55:195-198.

Henry, T.J., J.W. Neal, Jr., and K.M. Gott. 1986. *Stethoconus japonicus* (Heteroptera: Miridae): a predator of *Stephanitis* lace bugs newly discovered in the United States, promising in the biocontrol of azalea lace bug (Heteroptera: Tingidae). Proc. Entomol. Soc. Wash. 88:722-730.

Herms, D.A. 2004. Using degree-days and plant phenology to predict pest activity. In: IPM of Midwest Landscapes, pp. 49-59. V. Krischik and J. Davidson, eds. Minnesota Agricultural Experiment Station Publication SB-07645.

Heungens, K. I. De Dobbelaere, B. Gehesquiere, A. Vercauteren, and M. Maes. 2010. Within-field spread of *Phytophthora ramorum* on rhododendron in nursery settings. *In*: Proceedings of the Sudden Oak Death 4th Science Symposium (S.J. Frankel, J.T. Klienjuna, and K.M. Palmieri, eds.). USDA-FS Gen. Tech. Report Pacific Northwest 229:72-75.

Hodges, G.S. and S.K. Braman. 2004. Seasonal occurrence, phenological indicators and mortality factors affecting five scale insect species (Hemiptera: Diaspididae, Coccidae) in the urban landscape setting. J. Entomol. Sci. 39:611-622.

Hoffman, H., J. Eakes, C. Robinson, J. Sibley, S. Ditchkoff, and C. Coker. 2012. Impact of landscape plant species and repellent type on white-tailed deer browse. Proc. SNA Research Conference 57:233-237.

Hoitink, H.A.J. and A.F. Schmitthenner. 1974. Resistance of *Rhododendron* species and hybrids to *Phytophthora* root rot. Plant Dis. Rpt. 58:650-653.

Hoitink, H.A.J., M.E. Watson, and W.R. Faber. 1986. Effect of nitrogen concentration in juvenile foliage of rhododendron on *Phytophthora* dieback severity. Plant Dis. 70:292-294.

Horn, K.F., C.G. Wright, and M.H. Farrier. 1979. The lace bugs (Hemiptera: Tingidae) of North Carolina and their hosts. N. C. Agric. Stn. Bull. 257:1-22.

Huang, Q. 2007. Natural occurrence of *Xylella fastidiosa* in a commercial nursery in Maryland. Can. J. Plant Pathol. 29:299-303.

Hummel, R.L., M. Elliott, G. Chastagner, R.E. Riley, K. Riley, and A. DeBauw. 2013. Nitrogen fertility influences growth and susceptibility of rhododendrons to *Phytophthora ramorum*. HortScience 48:601-607.

Hwang, J. and D.M. Benson. 2005. Identification, mefenoxam sensitivity, and compatibility type of *Phytophthora* spp. attacking floriculture crops in North Carolina. Plant Dis. 89:185-190.

Hyder, N., M.D. Coffey, and M.E. Stanghelllini. 2009. Viability of oomycete propagules following ingestion and secretion by fungus gnats, shore flies, and snails. Plant Dis. 93:7200-726.

Irving, T. 2000. Pruning of rhododendrons and azaleas. J. Amer. Rhododendron Soc. 54:35-36.

Ishihara, R. and S. Kawai. 1981. Feeding habits of the azalea lace bug, *Stephanitis pyrioides* (Scott) (Hemiptera: Tingidae). Jap. J. Appl. Entomol. Zool. 25:200-202.

James, D., A. Vargo, E. Beckner, G. Sumampong, K. Bailey, M. Elliott, S. Masri, and S.F. Shamoun. 2012. Screening of several disinfectants to assess their efficacy in controlling mycelial growth, sporangia germination, and recovery of viable *Phytopthora ramorum*. Crop Protection 42:186-192.

Jeffers, S.N., H. Hwang, Y.A. Wamishe, and S.W. Oak. 2010. Detection of *Phytophthora ramorum* at retail nurseries in the southeastern United States. *In*: Proceedings of the Sudden Oak Death 4th Science Symposium (S.J. Frankel, J.T. Klienjuna, and K.M. Palmieri, (eds.). USDA-FS Gen. Tech. Report Pacific Northwest 229:69-71.

Johnson, C.G. 1936. Biology of *Leptobrysa rhododendri* Horvath (Hemiptera: Tingidae), the rhododendron lace bug. Ann. Appl. Biol. 23:342-368.

Johnson, W.T. and H.H. Lyon. 1991. Insects that feed on trees and shrubs, 2nd Ed. Cornell Univ. Press, Ithaca, NY. 560 p.

Jones, J., A.V. LeBude, and T.G. Ranney. 2010. Vegetative propagation of oconee azalea (*Rhododendron flammeum*) by stem cuttings and mound layering. J. Environ. Hort. 28:69-73.

Jones, J.R., T.G. Ranney, and T.A. Eaker. 2008. A novel method for inducing polyploidy in *Rhododendron* seedlings. J. Amer. Rhododendron Soc. 62:130-134.

Judd, G.J.R. and M.G.T. Gardiner. 1997. Forecasting phenology of *Orthosia hibisci* Guenée (Lepidoptera: Noctuidae) in British Columbia using sex-attractant traps and degree-day models. Can. Entomol. 129:815-825.

Kenyon, D.M., G.R. Dixon, and S. Helfer. 1998. The effect of temperature on colony growth by *Erysiphe* sp. infecting *Rhododendron*. Plant Pathol. 47:411-416.

Kenyon, D.M., G.R. Dixon, and S. Helfer. 2002. Effects of relative humidity, light intensity and photoperiod on the colony development of *Erysiphe* sp. on *Rhododendron*. Plant Pathol. 51:103-108.

Kintz, J.L. and D.R. Alverson. 1999. The effects of sun, shade, and predation on azalea lace bug populations in containerized azaleas. HortTechnology 9:638-641.

Kirker, G.T., B.J. Sampson, C.T. Pounders, J. M. Spiers, and D.W. Boyd. 2008. The effects of stomatal size on feeding preference of azalea lace bug, *Stephanitis pyrioides* (Hemiptera: Tingidae), on selected cultivars of evergreen azalea. HortScience 43:2098-2103.

Klingeman, W.E., S.K. Braman, and G.D. Buntin. 2000a. Evaluating grower, landscape manager, and consumer perceptions of azalea lace bug (Hemiptera: Tingidae) feeding injury. J. Econ. Entomol. 93:141-148.

Klingeman, W.E., S.K. Braman, and G.D. Buntin. 2000b. Feeding injury of the azalea lace bug, *Stephanitis pyrioides* (Scott) (Hemiptera: Tingidae). J. Entomol. Sci. 35:213-219.

Klingeman, W.E., S.K. Braman, and G.D. Buntin. 2001a. Azalea growth in response to azalea lace bug (Hemiptera: Tingidae) feeding. J. Econ. Entomol. 94:129-137.

Klingeman, W.E., G.D. Buntin and S. K. Braman. 2001b. Using aesthetic assessments of azalea lace bug (Hemiptera: Tingidae) feeding injury to provide thresholds for pest management decisions. J. Econ. Entomol. 94:1187-1192.

Klingeman, W.E., S.A. White, A. LeBude, A. Fulcher, N. Ward-Gauthier, and F. Hale. 2014. A review of arthropod pests, plant diseases and abiotic disorders and their management on *Viburnum* species in the southeastern U.S. J. Environ. Hort. 32:84-102.

Knox, G.W., W.E. Klingeman, M. Paret, and A. Fulcher. 2012. Management of pests, plant diseases and abiotic disorders of *Magnolia* species in the Southeastern U.S.: A review. J. Environ. Hort. 30:223-234.

Knox, G., M. Chappell, J-H. Chong, J. Williams-Woodward and A.V. LeBude. 2014. Camellia – *Camellia* spp., p. 31-62 *In*: S.A. White and W.E. Klingeman (eds.). IPM for Shrubs in Southeastern US Nursery Production. Vol. 1. Southern Nursery IPM Working Group, Clemson, SC.

Kohl, L.M. 2011. Foliar nematodes: A summary of biology and control with a compilation of host range. Plant Health Progress. Doi:10.1094/PHP-2011-1129-01-RV.

Krebs, S.L. and Wilson, M.D. 2002. Resistance to Phytophthora root rot in contemporary Rhododendron cultivars. HortScience 35:790-792.

Kuske, C.R. and Benson, D.M. 1983. Survival and splash dispersal of *Phytophthora parasitica*, causing dieback of rhododendron. Phytopathology. 73:1188-1191.

Kuske, C.R., D.M. Benson, and R.K. Jones. 1983. A gravel container base for control of Phytophthora dieback in rhododendron nurseries. Plant Dis. 67:1112-1113.

LaMondia, J.A., V.L. Smith, and T.M. Rathier. 1997. Tissue proliferation in rhododendron: lack of association with disease and effect on plants in the landscape. HortScience 32:1001-1003.

Latta, R. 1937. The rhododendron whitefly and its control. USDA Circ. No. 429, Washington, D.C. 8 pp.

Leach, D.G. 1961. Rhododendrons of the world. Charles Scribner's Sons, New York, New York. 544 pp.

Leddy, P.M. 1996. Factors influencing the distribution and abundance of azalea lace bug, *Stephanitis pyrioides*, in simple and complex landscape habitats. Ph.D. Dissertation, Univ. Maryland, College Park, MD, 159 p.

Linderman, R.G. 1974. The role of abscised *Cylindrocladium*-infected azalea leaves in the epidemiology of Cylindrocladium wilt of azalea. Phytopathology 64:481-485.

Linderman, R.G. and D.M. Benson. 2014. Compendium of Rhododendron and Azalea Diseases and Pests, Second Edition. APS Press, St. Paul, MN. 136 pp.

Linderman, R.G. and E.A. Davis. 2006. Survival of *Phytophthora ramorum* compared to other species of *Phytophthora* in potting media components, compost, and soil. HortTechnol. 16:502-506.

Long, M.C., S.L. Krebs, and S.C. Hokanson. 2010. Field and growth chamber evaluation of powdery mildew disease on deciduous azaleas. HortScience 45:784-789.

Loyd, A.L., D.M. Benson, and K.L. Ivors. 2014. *Phytophthora* populations in nursery irrigation water in relationship to pathogenicity and irrigation frequency of *Rhododendron* and *Pieris*. Plant Dis. 98:1213-1220.

Mague, D. and H.T. Streu. 1980. Life history and seasonal population growth of *Oligonychus ilicis* infesting Japanese holly in New Jersey. Environ. Entomol. 9:420-424.

Maier, C.T. 1983. Influence of host plants on the reproductive success of the parthenogenetic twobanded Japanese weevil, *Callirhopalus bifasciatus* (Roelofs) (Coleoptera: Curculionidae). Envir. Entomol. 12:1197-1203.

Marrone, P.G. and D.B. Zepp. 1979. Descriptions of the larva and pupa of *Callirhopalus* (subg. *Pseudocneorhinus*) *bifasciatus*, the twobanded Japanese weevil, with new host plant records. Ann. Entomol. Soc. Amer. 72:833-838.

Masakim M., M. Kadoi, and M. Yoneda. 1996. Effects of temperature on development of Fuller's rose weevil, *Pantomorus cervinus* (Boheman) (Coleoptera: Curculionidae). Res. Bull. Plant Protection Serv., Japan 32:7-13.

Masiuk, M. 2003. Black vine weevil fact sheet. Penn State Cooperative Extension. http://woodypests.cas.psu.edu/factsheets/InsectFactSheets/html/Black_Vine_Weevil.html Accessed 26 July 2013.

McAleese, A.J. and D.W. Rankin. 2000. Growing rhododendrons on limestone soils: is it really possible? J. Amer. Rhododendron Soc. 54:126-134.

McArthur, G. 1959. Cercospora leaf spot on Rhododendron (*Cercospora handelii* Bubak). NZ J. of Ag. Res. 2:86-89.

McClure, M.S. 1977. Resurgence of the scale, *Fiorinia externa* (Homoptera: Diaspididae) on hemlock following insecticide application. Environ. Entomol. 6:480-484.

Mead. F.W. 1967. *Stephanitis* lace bugs of the United States (Hemiptera: Tingidae). Fl. Dept. Agric. Div. Pl. Ind. Ent. Cir. No. 62.

Meijona, M., R. Rodrigueza, M.J. Canala, and I. Feitoc. 2009. Improvement of compactness and floral quality in azalea by means of application of plant growth regulators. Scientia Horticulturae 119:169-176.

Merritt R.W., M.K. Kennedy, and E.E Gersabeck. 1983. pp 277-299. *In*: G.W. Frankie and C.S. Koehler (eds.). Urban entomology: Interdisciplinary perspectives. Preager, New York, NY.

Miller, S.B. and L.W. Baxter. 1970. Dieback in azaleas caused by Phomopsis species. Phytopathology 60:387-388.

Miller, D.R. and J.A. Davidson. 2005. Armored scale insect pests of trees and shrubs. Cornell Univ. Press, Ithaca, NY, 442 p.

Million, J., T. Yeager, and C. Larsen. 2007. Water use and fertilizer response of azalea using several no-leach irrigation methods. HortTechnol. 17:21-25.

Mills, H.A. and J.B. Jones Jr. 1996. Plant analysis handbook II. MicroMacro Publishing, Athens, GA. p. 159.

Mizell, R.F. and T.C. Riddle. 2004. Evaluation of insecticides to control the Asian ambrosia beetle, *Xylosandrus crassiusculus*. Proc. Southern Nursery Assoc. Res. Conf. 49:152-155.

Mizell, R.F. III and D.E. Shiffhauer. 1991. Biology, effect on hosts, and control of the azalea leafminer (Lepidoptera: Gracillaridae) on nursery stock. Environ. Entomol. 20:597-602.

Moorman, G.W., S. Kang, D.M. Geiser, and S.H. Kim. 2002. Identification and characterization of *Pythium* species associated with greenhouse floral crops in Pennsylvania. Plant Dis. 86:1227-1231.

Morsink, W. 2005. Why do some Hymenanthes rhododendron in Toronto raise and lower their leaves seasonally? J. Amer. Rhododendron Soc. 59:21-31.

Munir, B. and R.I. Sailer. 1985. Population dynamics of the tea scale, *Fiorinia theae* (Homoptera: Diaspididae), with biology and life tables. Environ. Entomol. 14:742-748.

Nair, S. and S.K. Braman. 2012. A scientific review of the ecology and management of the azalea lace bug Stephanitis pyriorides (Scott) (Tingidae: Hemiptera). J. Entomol. Sci. 47:247-263.

Nalepa, C.A. and J.R. Baker. 1994. Winter oviposition of the azalea lace bug (Hemiptera: Tingidae) in North Carolina. J. Entomol. Sci. 29:482-489.

Nash, R.F. 1973. Control of the peony scale, *Pseudaonidia paeoniae*, and a wax scale, *Ceroplastes ceriferus*, with granular systemic insecticides on *Camellia japonica*. J. Georgia Entomol. Soc. 8:149-151.

Neal, J.W., Jr. 1984. Bionomics and instar determination of *Synanthedon rhododendri* (Lepidoptera: Sesiidae) on rhododendron. Ann. Entomol. Soc. Amer. 77:552-560.

Neal, J.W., Jr. 1988. Unusual oviposition behavior on evergreen azalea by the andromeda lace bug *Stephanitis takeyai* (Drake and Maa) (Heteroptera: Tingidae). Proc. Entomol. Soc. Wash. 90:52-54.

Neal, J.W., Jr. and J. Bentz. 1997. Physical dynamics of the noncleidoic egg of *Stephanitis pyrioides* (Heteroptera: Tingidae) during development. Environ. Entomol. 26:1066-1072.

Neal, J.W., Jr. and L.W. Douglass. 1988. Development, oviposition rate, longevity, and voltinism of *Stephanitis pyrioides* (Heteroptera: Tingidae), an adventive pest of azalea, at three temperatures. Environ. Entomol. 17:827-831.

Neal, J.W., Jr. and R.L. Haldemann. 1992. Regulation of seasonal egg hatch by plant phenology in *Stethoconus japonicus* (Heteroptera: Miridae), a specialist predator of *Stephanitis pyrioides* (Heteroptera: Tingidae). Environ. Entomol. 21:793-798.

Neal, J.W., Jr., R.L. Haldemann, and T.J. Henry. 1991. Biological control potential of a Japanese plant bug, *Stethoconus japonicus* (Heteroptera: Miridae), an adventive predator of the azalea lace bug (Heteroptera: Tingidae). Ann. Entomol. Soc. Amer. 84:287-293.

Noble, R., A. Dobrovin-Pennington, S. Pietravalle, and C.M. Henry. 2011. Composting of rhododendron and bilberry wastes to contain spread of exotic plant pathogens *Phytophthora kernoviae* and *Phytophthora ramorum*. Compost Sci. Utilization 19:219-225.

ODA (Oregon Department of Agriculture). 2013. Sudden Oak Death. http://www.oregon.gov/ODA/CID/PLANT_HEALTH/pages/sod_index.aspx Accessed 1 Aug 2015.

Oliver, A.D. and J.B. Chapin. 1980. The cranberry rootworm: adult seasonal history and factors affecting its status as a pest of woody ornamentals in Louisiana. J. Econ. Entomol. 73:96-100.

Oliver, J.B. and C.M. Mannion. 2001. Ambrosia beetle (Coleoptera: Scolytidae) species attacking chestnut and captured in ethanol-baited traps in middle Tennessee. Environ. Entomol. 30:909-918.

Olson, H.A. and D.M. Benson. 2011. Characterization of *Phytophthora* spp. on floriculture crops in North Carolina. Plant Dis. 95:1013-1020.

Olson, H.A., S.N. Jeffers, K.L. Ivors, K.C. Steddom, J.L. Williams-Woodward, M.T. Mmbaga, D.M. Benson, and C.X. Hong. 2013. Diversity and mefenoxam sensitivity of *Phytophthora* spp. associated with the ornamental horticulture industry in the southeastern United States. Plant Dis. 97: 86-92.

Ownley, B.H., D.M. Benson, and T.E. Bilderback. 1990. Physical properties of container media and relation to severity of Phytophthora Root Rot of Rhododendron. J. Amer. Soc. Hort. Sci. 115:564-570.

Parke, J.L., N. Grunwald, C. Lewis, and V. Fieland. 2010. A systems approach for detecting spores of *Phytophthora* contamination in nurseries. *In*: Proceedings of the Sudden Oak Death 4th Science Symposium (S.J. Frankel, J.T. Klienjuna, and K.M. Palmieri, eds.). USDA-FS Gen. Tech. Report Pacific Northwest 229:67-68.

Parke, J.L., B.J. Knaus, V.J. Fieland, C. Lewis, and N.J. Grünwald. 2014. *Phytophthora* community structure analyses in Oregon nurseries inform systems approaches to disease management. Phytopathology 104:1052-1062.

Pellet, H., S. Moe, and W. Mezitt. 1986. Flower bud hardiness of rhododendron taxa. J. American Rhododendron Society 40(4). http://scholar.lib.vt.edu.prox.lib.ncsu.edu/ejournals/JARS/v40n4/v40n4-pellett.htm, Accessed 30 Sept 2015.

Peterson, J.L. and S.H. Davis Jr. 1977. Effect of fungicides and application timing on control of azalea petal blight. Plant Dis. Rep. 61:209-212.

Plakidas, A.G. 1949. Effect of sclerotial development on incidence of azalea flower blight. Plant Dis. Rep. 33:272-273.

Polavarapu, S. 1999. Insecticidal nematodes for cranberry pest management, pp. 79-90. Proceedings at the National Workshop on Optimal Use of Insecticidal Nematodes in Pest Management. Rutgers Univ. Chatsworth, NJ.

Polavarapu, S., J.A. Davidson, and D.R. Miller. 2000. Life history of the Putnam Scale, *Diaspidiotus ancylus* (Putnum) (Hemiptera: Coccoidea: Diaspididae) on blueberries (*Vaccinium corymbosum*, Ericaceaes) in New Jersey, with a world list of scale insects on bluebrrries. Proc. Entomol.. Soc. Wash. 102:549-560.

Potter, M.F. 2008. Spider Mites on Landscape Plants. Univ. Ky. ENTFACT-438. http://www2.ca.uky.edu/entomology/entfacts/ef438.asp. Accessed 21 May 2015.

Pratt, P.D. 1999. Biological control of spider mites by the predatory mite *Neoseiulus fallacis* (Acari: Phytoseiidae) in ornamental nursery systems. Or. State Univ., Corvallis, PhD Diss.

Preece, J.E., M.R. Imel, and A. Shevade. 1993. Regeneration of *Rhododendron* PJM Group plants from leaf explants. J. Amer. Rhododendron Soc. 47:68-71.

Raabe, R.D. and J.V. Lenz. 1958. Septoria leaf scorch of azalea. California Agriculture 12:11.

Rabaglia, R.J., S.A. Dole, and A.I. Cognato. 2006. Review of American *Xyloborina* (Coleopteraa: Curculionidae: Scolytinae) occurring North of Mexico, with an illustrated key. Ann. Entomol. Soc. Amer. 99:1034-1056.

Raupp, M.J., A.B. Cumming, and E.C. Raupp. 2006. Street tree diversity in Eastern North America and its potential for tree loss to exotic borers. Arboric. Urban For. 32:297-304.

Raupp, M.J. and P.M. Shrewsbury. 2005. The role of plant diversity in reducing pest outbreaks in landscapes. *In*: Proceedings, 2005 ESA Annual Meeting and Exhibition, 15-18 December 2005. Fort Lauderdale, FL. Entomol. Soc. Amer., Lanham, MD.

Raupp, M.J., J.A. Davidson, J.J. Holmes and J.L. Hellman. 1985. The concept of key plants in integrated pest management for landscapes. J. Arboric. 11:317-322.
Raupp, M.J., J.J. Holmes, C.S. Sadof, P.M. Shrewsbury, and J.A. Davidson. 2001. Effects of cover sprays and residual pesticides on scale insects and natural enemies in urban forests. J. Arboric. 27:203-214.

Rebek, E.J. and C.S. Sadof. 2003. Effects of pesticide applications on the euonymus scale (Homoptera: Diaspididae) and its parasitoid, *Encarsia citrina* (Hymenoptera: Aphelinidae). J. Econ. Entomol. 96:446-452.

Reding, M.E. 2008. Black vine weevil (Coleoptera: Curculionidae) performance in container- and field-grown hosts. J. Entomol. Sci. 43:300-310.

Reding, M., J. Oliver, P. Schultz, and C. Ranger. 2010. Monitoring flight activity of ambrosia beetles in ornamental nurseries with ethanol-baited traps: Influence of trap height on captures. J. Environ. Hort. 28:85-90.

Reding, M.E. and A.B. Persad. 2009. Systemic insecticides for control of black vine weevil (Coleoptera: Curculionidae) in container- and field-grown nursery crops. J. Econ. Entomol. 102:927-933.

Reding, M.E. and C.M. Ranger. 2011. Systemic insecticides reduce feeding, survival, and fecundity of adult black vine weevils (Coleoptera: Curculionidae) on a variety of ornamental nursery crops. J. Econ. Entomol. 104:405-413.

Reichert, S.E. and L. Bishop. 1990. Prey control by an assemblage of generalist predators: spiders in garden test systems. Ecology 71:1441-1450.

Ridge, G., S.A. White, I.M. Meadows, and S.N. Jeffers. 2012. Developing a method to evaluate plants used in constructed wetlands for susceptibility to five species of *Phytophthora*. Proc. SNA Res. Conf. 57:251-256.

Riley, E.D., H.T. Krauss, T.E. Bilderback, and B.E. Jackson. 2012. Cotton waste: a new spin on southeastern substrates. Proc. SNA Res. Conf. 57:63-67.

Rinehart, T.A. and D.W. Boyd, Jr. 2006. Rapid, high-throughput detection of azalea lace bug (Heteroptera: Tingidae) predation by *Chrysoperla rufilabris* (Neuroptera: Chrysopidae), using fluorescent-polymerase chain reaction primers. J. Econ. Entomol. 99:2136-2141.

Rinehart, T.A., W.E. Copes, T. Toda, and M.A. Cubeta. 2007. Genetic characterization of binucleate *Rhizoctonia* species causing web blight on azalea in Mississippi and Alabama. Plant Dis. 91:616-623.

Rosetta, R. 2006. Azalea Bark Scale. Pacific Northwest Nursery IPM, Oregon State University, Corvallis. http://oregonstate.edu/dept/nurspest/azalea_bark_scale.htm. Accessed 21 May 2015.

Rosetta, R. 2013. Azalea lace bug: Biology and management in commercial nurseries and landscapes. Oregon State University Extension Service, EM 9066, 6 p.

Ryan, G. 1990. Phosphorus and nitrogen nutrition of rhododendrons. J. Amer. Rhododendron Soc. 42:84-87, 118-119.

Sadof, C.S. and D.C. Sclar. 2000. Effects of horticultural oil and foliar or soil-applied systemic insecticides on Euonymus scale in pachysandra. J. Arboric. 26:120-125.

Salley, H.E., and H.E. Greer. 1986. Rhododendron Hybrids, 2nd Ed., Timber Press, 442 p.

Saville, D.B.O. 1959. Notes on *Exobasidium*. Can. J. Bot. 37:641-656.

Scagel, C.F., G. Bi, D.R. Bryla, L.H. Fuchigami, and R.P. Regan. 2014. Irrigation frequency during container production alters rhododendron growth, nutrient uptake, and flowering after transplanting into a landscape. HortScience 49:955-960.

Scagel, C.F., G. Bi, L.H. Fuchigami, and R.P. Regan. 2008. Nitrogen availability alters mineral nutrient uptake and demand in container-grown deciduous and evergreen rhododendron. J. Environ. Hort. 26:177–187.

Scagel, C.F., G. Bi, L.H. Fuchigami, and R.P. Regan. 2011. Effects of irrigation frequency and nitrogen fertilizer rate on water stress, nitrogen uptake, and plant growth of container-grown Rhododendron. HortScience 46:1598–1603.

Schaefer, C.W. and A.R. Panizzi. 2000. Heteroptera of Economic Importance. CRC Press LLC, Boca Raton, FL. 856 p.

Schreiber, L.R. 1964. Stem canker and die-back of Rhododendron caused by *Botryosphaeria ribis* Gross. & Dug. Plant Dis. Reporter. 48:207-210.

Schultz, P.B. 1993. Host plant acceptance of azalea lace bug (Heteroptera: Tingidae) for selected azalea cultivars. J. Entomol. Sci. 28:230-235.

Schuh, R.T. and J.A. Slater. 1995. True Bugs of the World (Hemiptera: Heteroptera): Classification and Natural History. Cornell Univ. Press, Ithaca, NY. 416 pp.

Schweigkofler. W. K. Kosta, V. Huffman, S. Sharma, and S. Ghosh. 2014. Steaming inactivates *Phytophthora ramorum*, causal agent of sudden oak death and ramorum blight, from infested nursery soils in California. Plant Health Progress. PHP-RS-13-0111.

Shen, H.W., W.J. Wu, and P.S. Yang. 1985. The biology of the azalea lace bug, *Stephanitis pyrioides* (Scott). I. The morphology of the azalea lace bug, *Stephanitis pyrioides* (Scott), Mem. Coll. Agric. Nat. Taiwan Univ. 25:143-154.

Shrewsbury, P.M. and M.J. Raupp. 2006. Evaluation of components of the vegetational texture for predicting abundance of azalea lace bug, *Stephanitis pyrioides* (Scott) (Heteroptera: Tingidae), in managed landscapes. Environ. Entomol. 29:919-926.

Shrewsbury, P.M. and D.C. Smith-Fiola. 2000. Evaluation of green lacewings for suppressing azalea lace bug populations in nurseries. J. Environ. Hort. 18:207-211.

Sinclair, W.A. and H.H. Lyon. 2005. Diseases of trees and shrubs. 2nd ed. Cornell University Press. Ithaca, NY. 680 pp.

Sirjusingh, C. and J.C. Sutton. 1996. Effects of wetness duration and temperature on infection of geranium by *Botrytis cinerea*. Plant Dis. 80:160-165.

Slippers, B., P.W. Crous, S. Denman, T.A. Coutinho, B.D. Wingfield, and M.J. Wingfield. 2004. Combined multiple gene genealogies and phenotypic characters differentiate several species previously identified as *Botryosphaeria dothidea*. Mycologia 96:83-101.

Smith, F.F. 1932. Biology and control of the black vine weevil. USDA Technical Bulletin 325. 46 p.

Snow, J.W., T.D. Eichlin, and J.H. Tumlinson. 1985. Seasonal captures of clearwing moths (Sesiidae) baited with various octadecadienyl acetates and alcohols in central Georgia during 1983-1985. Environ. Entomol. 18:216-222.

Sobers, E.K. and G.L. Crane. 1971. Susceptibility of azalea varieties to *Calonectria theae* and *Cylindrocladium scoparium*. Fla State Hort Soc Proc. No. 4207:357-359.

Solomon, J.D. 1995. Guide to insect borers of North American broadleaf trees and shrubs. Agric. Handbook 706. Washington, DC. U.S. Dept. Agriculture, Forest Service. 735 p.

Sommerville, E.A. 1998. Propagating native azaleas. J. Amer. Rhododendron Soc. 52:126-127.

Stanyard, M.J., R.E. Foster, and T.J. Gibb. 1997. Effects of orchard ground cover and mite management options on the population dynamics of European red mite (Acari: Tetranychidae) and *Amblyseius fallacis* (Acari: Phytoseiidae) in apple. J. Econ. Entomol. 90:595-603.

Stewart C.D., S.K. Braman, and A.F. Pendley. 2002a. Functional response of the azalea plant bug (Heteroptera: Miridae) and a green lacewing, *Chrysoperla rufilabris* (Neuroptera: Chrysopidae), to two predators of the azale lace bug, (Heteroptera: Tingidae). Environ. Entomol. 31:1184-1190.

Stewart C.D., S.K. Braman, and B.L. Sparks. 2002b. Abundance of beneficial arthropods on woody landscape plants at professionally-managed landscape sites. J. Environ. Hort. 20:67-72.

Stoetzel, M.B. 1975. Seasonal history of seven species of armored scale insects of the Aspidiotini (Homoptera: Diaspididae). Ann. Entomol. Soc. Amer. 68:489-492.

Taylor, R.J., J.S. Pasche, C.A. Gallup, H.D. Shew, and N.C. Gudmestad. 2008. A foliar blight and tuber rot of potato caused by *Phytophthora nicotianae*: New occurrences and characterization of isolates. Plant Dis. 92:492-503.

Ticknor. R.L. and M. Skinner. 1991. Chemical pruning of rhododendrons. J. Amer. Rhododendron Soc. 45:93.

Timonin, M.L. and R.L. Self. 1955. *Cylindrocladium scoparium* Morgan on azaleas and other ornamentals. Plant Dis. Rep. 39:860-865.

Tippins, H.H., H. Clay, and R.M. Barry. 1977. Peony scale: a new host and biological information. J. Georgia Entomol. Soc. 12:68-71.

Tjosvold, S.A., D.L. Chambers, E.J. Fichtner, S.T. Koike, and S.R. Mori. 2009. Disease risk of potting media infested with *Phytophthora ramorum* under nursery conditions. Plant Disease 93:371-376.

Tooley, P.W. and M. Browning. 2015. Temperature effect on the onset of sporulation by *Phytophthora ramorum* on *Rhododendron* 'Cunningham's White'. J. Phytopathology 163:908-914.

Towe, L.C. 2004. American Azaleas. Timber Press. 188 p.

Trumbule, R.B. and R.F. Denno. 1995. Light intensity, host-plant irrigation, and habitat-related mortality as determinants of the abundance of azalea lace bug (Heteroptera: Tingidae). Environ. Entomol. 24:898-908.

Trumbule, R.B., R.F. Denno, and M.J. Raupp. 1995. Management considerations for the azalea lace bug in landscape habitats. J. Arboric. 21:63-68.
USDA APHIS-PPQ, 2013. Official regulatory protocol for *Phytophthora ramorum* detections in residential or landscaped commercial settings. Confirmed Residential Protocol, Vers. 2.0, Rev. Jan 15, 2013. 29 p. https://www.aphis.usda.gov/plant_health/plant_pest_info/pram/downloads/pdf_files/ResidentialProtocol.pdf Accessed 16 Dec. 2015.

USDA APHIS-PPQ, 2014a. 2014 *Phytophthora ramorum* inspection and sampling protocol for nurseries under DA-2014-02 compliance. Rev. Aug. 6, 2014. 19 p. https://www.aphis.usda.gov/plant_health/plant_pest_info/pram/downloads/pdf_files/ConfirmedNurseryProtocol.pdf Accessed 16 Dec. 2015.

USDA APHIS-PPQ, 2014b. Official regulatory protocol for nurseries containing plants infected with *Phytophthora ramorum* . Confirmed Nursery Protocol, Vers. 8.2, Rev. Aug. 6, 2014. 54 p. https://www.aphis.usda.gov/plant_health/plant_pest_info/pram/downloads/pdf_files/ConfirmedNurseryProtocol.pdf Accessed 16 Dec. 2015.

U.S. Department of Agriculture (USDA). 2009. 2007 Census of Agriculture, Washington, D.C.

Valley, K. 1975. Azalea leafminer, *Gracillaria azaleella* Brants, p. 11-12. *In*: Pennsylvania Dept. of Agric., Bur. Plant Ind. Regulatory Hort. Entomol. Circ. 6:2.

van Tol, R.W.H.M. and M.J. Raupp. 2005. Nursery and tree application, *In*: Grewal, P.S., R.-U. Ehlers, and D.I. Shapiro-Ilan (eds.), Nematodes as Biocontrol Agents. CAB Intl. London, UK, 528 p.

van Tol, R.W.H.M., D.J. Bruck, F.C. Griepink, and W.J. De Kogel. 2012. Field attraction of the vine weevil *Otiorhynchus sulcatus* to kairomones. J. Econ. Entomol. 105:169-175.

Vercauteren, A., M. Riedel, M. Maes, S. Werres, and K. Heungens. 2013. Survival of *Phytophthora ramorum* in rhododendron root balls and in rootless substrates. Plant Pathol. 62:166-176.

Wang, Y.F., C.D. Robacker, and S.K. Braman. 1998. Identification of resistance to azalea lace bug among deciduous taxa. J. Amer. Soc. Hort. Sci. 123:592-597.

Wang, Y.F., S.K. Braman, C.D. Robacker, J.G. Latimer, and K.E. Espelie. 1999. Composition and variability of epicuticular lipids of azaleas and their relationship to azalea lace bug resistance. J. Amer. Soc. Hort. Sci. 124:239-244.

Warfield, C.Y. and G.R. Parra. 2004. Comparison of Encore Azalea® cultivar resistance to Phytophthora root rot, 2003. B&C Tests Vol 19:O001.
Wehlbur, C. and R.S. Cox. 1966. Rhizoctonia leaf blight of azalea. Plant Dis. Rep. 50:354-355.

Weiss, H.B. 1916. Foreign pests recently established in New Jersey. J. Econ. Entomol. 9:212-216.

Werres, S., R. Marwitz, W.A. Man in't Veld, A.W.A.M. Cock, P.J.M. de Bonants, M. Weerdt, M. K. Themann, E. Ilieva, and R.P. Baayen. 2001. *Phytophthora ramorum* sp. nov., a new pathogen on Rhododendron and Viburnum. Mycological Research. 105:1155-1165.

Werres, S., S. Wagner, T. Brand, K. Kaminski, and D. Siepp. 2007. Survival of *Phytophthora ramorum* in recirculating irrigation water and subsequent infection of *Rhododendron* and *Viburnum*. Plant Disease 91:1034-1044.

Wheeler, A. and G. Miller. 1981. Fourlined plant bug, a reappraisal: life history, host plants, and plant response to feeding. Great Lakes Entom. 14:23-35.

Wheeler A.G. and D.W. Boyd Jr.. 2005. Southeastern U.S. distribution of an invasive weevil, *Pseudocneorhinus bifasciatus* Roelofs (Coleoptera: Curculionidae). J. Entomol. Sci. 40:25-30.

Wheeler, A.G., Jr. and J.L. Herring. 1979. A potential insect pest of azaleas. J. Am. Rhododendron Soc. 33:12-14.

Wheeler, A.G., Jr. and K.R. Valley. 1980a. *Graphocephala coccinea*: seasonal history and habits on ericaceous shrubs with notes on *G. fennahi* (Homoptera: Cicadellidae). Melsheimer Entomol. Ser. 29:23-27.

Wheeler, A.G., Jr. and K.R. Valley. 1980b. A rhododendron leafhopper: field recognition and habits. Bull. Amer. Rhododendron Soc. 34:202-205.

White, R.P. 1933. The insects and diseases of rhododendrons and azaleas. J. Econ. Entomol. 26:631-640.

Wills, W.H. and R.C. Lambe. 1986. Studies on rhododendron dieback caused by *Botryosphaeria dothidea*. J. Amer. Rhododendron Soc. 40(4). http://scholar.lib.vt.edu/ejournals/JARS/v40n4/v40n4-wills.htm Accessed 31 March 2017.

Wright, R.D. 1992. Influence of nitrogen nutrition on flower bud initiation of rhododendron. J. Amer. Rhododendron Soc. 46:99-100.

Wolfe, L.M. and L.J. Rissler. 2000. Reproductive consequences of a gall-inducing fungal pathogen (*Exobasidium vaccinii*) on *Rhododendron calendulaceum* (Ericaceae). Can. J. Bot. 77:1454-1459.

Woodruff, R.E. and R.C. Bullock. 1979. Fuller's rose weevil, *Pantomorus cervinus* (Boheman), in Florida (Coleoptera: Curculionidae). Division of Plant Industry Entomology Circular No. 207. http://www.freshfromflorida.com/pi/enpp/ento/entcirc/ent207.pdf. Accessed 26 July 2013.

Yeager, T. and J. Million. 2014. Effect of container spacing and plant species on water use. Proc. SNA Research Conference 59:267-269.

Zentmyer, G.A. and S.M. Mircetich. 1966. Saprophytism and persistence in soil by *Phytophthora cinnamomi*. Phytopathology 56:710-712.

Zepp, D.B. 1978. Egg pod formation by *Callirhopalus* (subg. *Pseudocneorhinus*) *bifasciatus* (Roelofs) (Coleoptera: Curculionidae: Eremninae). Coleopterists Bull. 32:311-313.

Rhaphiolepis - Rhaphiolepis spp. - *Rhaphiolepis* spp.

Sarah A. White, Clemson University

Juang-Horng (JC) Chong, Clemson University

Alan Windham, University of Tennessee

Section 1

History, Culture and Management

Introduction

1. History and Landscape Value
2. Management and Production Practices
3. Abiotic Stressors

Figure 5.1 Examples of popular cultivars of *Rhaphiolepis* grown in the Southeastern U.S. (a) 'Clara', (b) Eleanor Tabor^{TM}, (c) Gulf GreenTM, (d) 'Snow White' , and the (e) form and (f) flower of Spring Sonata^{TM}.

History and Landscape Value

The genus *Rhaphiolepis* contains nearly 15 species of evergreen, small (2 feet tall) to large (10+ feet tall), flowering shrubs. The genus *Rhaphiolepis* belongs to the Rosaceae family and these species were introduced into the U.S. from temperate and subtropical eastern and southeastern Asia. More than 250 nurseries in the Southeastern U.S. sell 27 cultivars of *Rhaphiolepis* (Table 5.1, Figure 5.1). Indian (*R. indica*) and Yeddo (*R. umbellata*) hawthorns are valued in landscapes because the glossy evergreen foliage forms attractive mounds that are covered with white or pink

Figure 5.2 Spring Sonata^{TM} Indian hawthorn (a) flowering in the landscape, (b) foliage in fall, and (c) cluster of berries.

flowers in the spring (mid-April to May in the Southeastern U.S.), followed by formation of blue berries in late summer that persist through early winter (Figure 5.2). New cultivars, with resistance to Entomosporium leaf spot, have enhanced the landscape viability of this genus.

Figure 5.3 *Rhaphiolepis* flowers range from (a) white to (b) pink in the landscape. White flowering cultivars seem to be more cold tolerant, likely due to parentage.

Species and Cultivar Characteristics

Confusion surrounds the species of *Rhaphiolepis* marketed by the trade and grown within the southeastern U.S. Dirr (2009) states that most of the species present in the southeastern U.S. are actually *R. umbellata*, rather than *R. indica*, because the physical characteristics of most cultivars available in the trade favor the technical description of *R. umbellata* or *R. ×delacourii*, rather than *R. indica. Rhaphiolepis umbellata* has leathery leaves with rounded margins, is native to temperate regions (more cold-tolerant), and has white flowers. *Rhaphiolepis indica* is native to sub-tropical regions in Asia, is freeze-tender, and can have either white or pink flowers (Figure 5.3). Hybrid crosses of *R. indica* and *R. umbellata* are desired because of their increased cold hardiness, disease resistance, and flower color. Accurate species identification of plants grown and sold in the trade was not feasible for this book chapter; thus the cultivars listed in Table 5.2 are listed as marketed by the green industry or as specified by industry experts. The spelling of *Rhaphiolepis* is also sometimes debated (as the authors found in their efforts to compile horticulturally relevant data about the crop); it is also sometimes spelled *Raphiolepis.*

If growing *Rhaphiolepis* in more northern areas of the Southeastern U.S., select those cultivars that are most cold tolerant (Table 5.2). The white-flowered cultivars (Figure 5.3) may prove more cold hardy, as their genetic parentage may more closely align with *R. umbellata* (Klingaman, 2012).

Management and Production Practices

Propagation

Propagate from softwood and semi-hardwood cuttings from June to August or layering in autumn to ensure retention of desired physical characteristics. Ninety to 100% rooting of cuttings was reported for Springtime® and 'Enchantress' cuttings when wounded and dipped in either 4,000 ppm IBA-talc, 1 to 2% IBA-quick dip, or 8,000 ppm + 2,500 ppm NAA-solution. After hormone dip, place under shade and mist in a greenhouse with temperature maintained between 70 to 75°F (21 to 24°C) (Dirr and Heuser, 1989). If the hormone is applied via incorporation in the substrate the cuttings are stuck into, rooting can also occur. Blythe et al. (2005) reported successful rooting with a 600 ppm K-IBA substrate treatment for cuttings of *Rhaphiolepis indica* 'Alba.'

Pruning

Without pruning, *Rhaphiolepis* plants tend to branch sparsely and are not marketable. Thus, either mechanical or chemical pruning is required for production of a marketable crop. *Rhaphiolepis* grown in #1 containers (Figure 5.4) require at least one pruning event and 12 months in production from liner to finished crop (Chappell, 2015); while *Rhaphiolepis* grown in #3 containers may require two additional pruning events and 16+ months (liner to marketable crop) of production time. Three or more weeks of active growth are lost after each pruning event, increasing the time to produce a finished crop (Oates et al., 2004).

Chemical pruning is less labor intensive than physical pruning and results in less time lost to inhibition of active growth (Oates et al., 2005a). Foliar application of benzyladenine (BA) at 2,500 ppm three times at one or two week intervals resulted in desired branching of Olivia™ and Eleanor Tabor™ crops. Phytotoxicity symptoms (leaf curling) were noted when applications were made to new growth (Oates et al., 2004). To avoid potential for phytotoxicity, make applications after new growth flushes have hardened off (Oates et al., 2005b). Foliar (spray) application of cyclanilide (CYC) at 100 ppm resulted in adequate branching of both Eleanor Tabor™ and Olivia™ (Farris et al., 2010). Williamson et al. (2009) reported that substrate drenches of cyclanilide from 10 to 40 ppm induced desired branching with minimal phytotoxicity, and the drench application was more persistent mitigating the need for additional treatments.

Figure 5.4 *Rhaphiolepis* grown in #1 containers is common in the nursery industry.

Table 5.1 Number of production nurseries growing various *Rhaphiolepis* spp. reported [plant & supply locator] for twelve southern states.

Species	Number of Nurseries	States
Rhaphiolepis indica	144	AL, FL, GA, LA, MS, NC, SC, TX
'Alba'	50	AL, FL, GA, LA, NC, SC
'Ballerina' ('Dancing Girl')	2	FL, TX
'Bay Breeze'	3	LA, TX
'Clara'	33	GA, LA, MS, NC, SC, TX
Eleanor Tabor™ ('Conor')	22	TX, NC, GA, FL, TN, SC, LA, MS
'Elizabeth'	3	FL, LA, TX
'Enchantress' ('Pinkie')	6	TX
'Jack Evans'	1	TX
'Pink Clara'	2	FL, TX
'Pink Dancer'	1	NC
'Regal Rose'	1	FL
Rosalinda®	3	AL, FL, SC
'Rosea'	1	TX
'Snow'	8	AL, FL, GA, LA, NC, SC
Snow Pink™ ('Sopink')	1	LA
'Spring Sonata'	2	AL, SC
White Enchantress® ('Monant')	1	FL
Rhaphiolepis umbellata	5	FL
Blueberry Muffin'	1	TX
Minor' - Dwarf	11	AL, FL, SC, NC, TX, VA
RutRhaph1'	4	FL, NC, SC
Rhaphiolepis ×delacourii	-	-
Coats Crimson'	3	FL
Georgia Petite'	2	NC, SC
Indian Princess™	1	MS
Majestic Beauty™ ('Montic')	21	FL, NC, SC, TN, TX
Snowcap®	4	FL, TN, TX
Snow White'	27	AL, FL, GA, LA, MS, NC, SC, TN, TX
Springtime® ('Pink Lady')	6	AL, TX

Irrigation

Rhaphiolepis are drought tolerant, and thus can tolerate managed irrigation deficits (of 60 to 100% of regularly applied water), with no decrease in crop quality (Beeson, 2006; Figure 5.5). Managed irrigation deficits may produce the same quality of crop, but at a slower rate because crop growth will be reduced. *Rhaphiolepis* should not be irrigated with saline water (EC > 2.0 dS/m^2) as growth will slow (Valdez-Aguilar et al., 2011).

Figure 5.5 *Rhaphiolepis* tolerates managed irrigation deficits and therefore is well suited to container production, which has limited water holding capacity due to container capacity.

Fertilization

Tillman et al. (2008) evaluated a range of liquid fertilizer application rates and reported that fertilization levels of 145 mg/L nitrogen (with 4:1:2 ratio of N:P:K) resulted in rapid growth of rooted *Rhaphiolepis ×delacourii* 'Snow White' cuttings, while maintaining foliar nutrition levels (Table 5.3) similar to those recommended by Bryson et al. (2014). Others recommend a controlled release or coated product that can slowly release nitrogen and other nutrients to the plants over time. Coated or controlled-release products can potentially reduce environmental impact and costs associated with fertilizer losses to leaching.

Weed control

For general weed management practices, see Chapter 6. Specific weed control studies in *Rhaphiolepis* are uncommon, but some work has been conducted. Post-emergence control of yellow nutsedge (*Cyperus esculentus*) and warm-season broadleaf weeds is possible with minimal foliar injury or reduction in growth of *Rhaphiolepis indica* 'Alba', even if high rates of selected herbicide are applied [e.g. bentazon (3-isopropyl-1H-2,1,3-benzothiadiazin-(4)-3H-one) sprayed over the top in 2 successive weeks at both the 1.1 or 2.2 kg/ha rate] (Norcini and Aldrich, 1992). Also, a 1 to 2-inch layer of coarse organic mulch (Figure 5.6) covering the potting mix, controlled weed emergence for 6 months with plant growth either improved or similar to traditionally grown (control) *Rhaphiolepis* (Wilen et al., 1999).

Figure 5.6 Weed control options are limited in broadleaf evergreen crops, and for that reason more and more growers are turning to mulches (such as rice hulls) to provide weed suppression.

Mammalian pests

White-tailed deer browsing damage is common for *Rhaphiolepis* both in production nurseries and in landscapes. *Rhaphiolepis* and hosta were the most commonly browsed species in a survey of 223 members of the Alabama nursery and landscape association (Witcher et al., 2013). Of methods used to deter deer browsing in commercial settings – electric fencing, Liquid Fence®, and a high fence were the most common responses with moderately effective control – but none were ranked as completely effective deterrents (Witcher et al., 2013).

Abiotic Stressors

Rhaphiolepis is generally known as a resilient shrub that is capable of tolerating many abiotic stress factors. Some research has been conducted to identify abiotic stress factors that impact growth of hawthorn.

Winter burn

Foliage of evergreen shrubs is commonly damaged by winter burn or scorch. Winter burn appears as desiccated leaf margins and is most pronounced on the sunward or windward sides of the plant (Klingeman et al., 2014). Because evergreen foliage continues to lose moisture during winter, injury may be particularly extensive following clear, cold sunny winter days when the ground is frozen and plant roots and interior stems cannot absorb water to replace that which is lost through transpiration (Relf and Appleton, 2009). Many *Rhaphiolepis* cultivars lack winter-hardiness, symptomology includes damaged foliage (Figure 5.7), canopy dieback, or death if temperatures become too low. Some cultivars have been released that are more cold hardy; Table 5.2 shows the minimum temperature tolerances for a range of cultivars currently in the trade.

Management

In areas where winter burn has been a problem, *Rhaphiolepis* should be planted in early fall, allowing additional time during fall for roots to become established and the plant hydrated prior to the onset of freezing conditions and drying winds (Adkins et al., 2010). Managers should plan for regular irrigation in the fall, in the absence of rainfall, to increase water availability to newly planted shrubs. Also select cultivars from Table 5.2 that exhibit the desired degree of cold-tolerance.

Figure 5.7 Spring Sonata™ Indian hawthorn in the landscape with foliage exhibiting winter burn damage.

Nutrient deficiency and toxicity

Nutrient deficiencies occasionally affect hawthorn (Figure 5.8). Typical ranges of foliar nutrient concentrations may be found in Table 5.3. *Rhaphiolepis indica* plants are tolerant of 4 to 6 ppm boron in irrigation water, but if boron levels exceed 6 ppm, severe leaf injury can occur (Wu and Dodge, 2005). Symptoms of boron injury occur first on older leaves include development of yellow to orange spots on the leaf tips of new foliage and leaf margins along older leaves within the canopy. If symptoms advance, due to continued irrigation with water containing concentrations of boron > 6 ppm, leaves may curl inward and prematurely senesce, and the terminal growing bud may die (Western Plant Health Association, 2010).

Figure 5.8 Hawthorn foliage, the new growth exhibiting chlorosis associated with iron (Fe) deficiency.

Rhaphiolepis is considered moderately salt tolerant (Myamoto et al., 2004) but salt injury can occur, appearing as marginal necrosis and tip burn. Typically, it results from applications of excess fertilizer that kills root tips and prevents sufficient water absorption. Salt damage can also occur if water with high salinity concentration is used for irrigation. Tolerance ratings for *R. indica* plants irrigated with saline water were developed. Soil salinity ratings were based on plant response (no symptoms of salt-related stress) to irrigation with saline water. *R. indica* tolerance of soil salinity ranged from moderate (EC of 6 – 8 dS m^{-1}) to highly (EC of 8-10 dS m^{-1}) tolerant. *Rhaphioleis indica* plant response (shoot growth reduced 25% or more than 25% of leaves damaged) to salinity in overhead irrigation spray were also ranked; plant tolerance ranged from moderate (water with EC of 2-3 dS m^{-1} with less than 425 ppm Na and less than 590 ppm Cl) to high (EC greater than 3 dS m^{-1}) (Myamoto et al, 2004). No information was available in the literature related to either soil salinity or salt spray tolerance of *R. umbellata*.

Management

Nutrient deficiencies and salt burn may be managed by fertilizing appropriately as indicated by soil analysis, plant symptoms or foliar analyses. Soil pH testing may provide guidance for potential nutrient deficiencies. Managing salinity in irrigation water involves monitoring soil pH and EC and managing irrigation volumes, timing, and leaching to limit accumulation of salt in the plant root zone. *Rhaphiolepis* spp. are relatively tolerant of salt and thus may be good species to produce if only poor quality water is available.

Table 5.2 Growth characteristics of selected *Rhaphiolepis* spp. and cultivars currently in production in southeastern United States nurseries.

Species	Cultivar / Trademark	USDA Plant Hardiness Zones	Habit	Flower color	Foliage	Pest resistance	Reference
R. indica	species	7b - 10					
	'Alba'	7b - 11	Compact-mound, 4' x 6'	White, spring & fall flowering	Bronze green in winter	-	-
	'Ballerina' ('Dancing Girl')	8 - 10	Rounded habit, 2-3' x 4'	Rose-pink, intermittent	Foliage reddish color in winter	-	Dirr 2009
	'Bay Breeze'	7 - 10	Broad-mounded habit, 2.5' x 6'	Deep-pink	New leaves bronze, mature leaves slightly twisted	Susceptible to *Entomosporium* leaf spot, better performance in drier area	Dirr 2009; Hagan et al., 2002
	'Clara'	7b - 10	Rounded low, 4' x 4'	White	Red growth matures to dark green	Moderate susceptibility to *Entomosporium* leaf spot	Dirr 2009; Hagan et al., 2002
	Eleanor Tabor™ ('Conor')	7 - 10	Vigorous, mounded, 3' x 4'	Pinkish	Large, dark green	Resistant to *Entomosporium* leaf spot, fire blight	Dirr 2009; Hagan et al., 2002
	'Enchantress' (aka 'Pinkie', 'Moness')	7 - 10	Compact, 3' x 5'	Rose-pink	Glossy, dark green	Susceptible to *Entomosporium* leaf spot	Dirr 2009; Hagan et al., 2002
	'Jack Evans'	7b - 10	Upright, 4-5' x 4'	Vivid pink panicles	Bronze-burgundy new leaves	Susceptible to *Entomosporium* leaf spot	Dirr 2009; Hagan et al., 2002
	'Pink Dancer'	7b - 10	1.3' x 3'	Deep pink	Shiny with undulating margins, maroon in winter	Resistant to *Entomosporium* leaf spot	-
	Rosalinda® ('Conda')	8 - 10	Tree, 10-14' x 8-10'	Dark pink, fragrant	Bronzy-red new foliage matures to dark green	Moderate susceptibility to *Entomosporium* leaf spot	Hagan et al., 2002
	'Rosea'	8 - 10	Loose, mound, 3-5' x 5-6'	Light pink	Bronze new leaves		Dirr 2009

Table 5.2 (*continued*) Growth characteristics of selected *Rhaphiolepis* spp. and cultivars currently in production in southeastern United States nurseries.

Species	Cultivar / Trademark	USDA Plant Hardiness Zones	Habit	Flower color	Foliage	Pest resistance	Reference
R. indica	Snow Pink™ ('Sopink')	8 - 11	Compact, 3' x 3'	Brilliant pink	Red new foliage matures to dark green	Moderate resistance to *Entomosporium* leaf spot	Dirr 2009
	Spring Sonata™	7 - 10	Dense, 4' x 5'	White (late season), dark purple fruit	Dark green	- (Dirr notes as disease resistant)	Dirr 2009
	White Enchantress® ('Monant')	8 - 11	Low, 3' x 5'	White	Reddish-tinge in winter	Highly susceptible to *Entomosporium* leaf spot	Hagan et al., 2002
R. umbellata	species	7b - 10	Rounded, 4-6'	White to light pink	Dark green	Susceptible to *Entomosporium* leaf spot and fireblight. Scale and aphids can be troublesome.	
	'Blueberry Muffin'	7a - 10	Dense, rounded habit, 4-6' x 4-5'	White, slightly fragrant, blue berries	Foliage purplish in winter	Resistant to *Entomosporium* leaf spot	Dirr 2009; Hagan et al., 2002
	Gulf Green™ ('Minor')	8 - 10	Upright, 3-4' x 2-3'	White, purple-black berries	Smaller, dark green	Resistant to *Entomosporium* leaf spot	Dirr 2009; Hagan et al., 2002
	Southern Moon® ('RutRhaph1')	7 - 10	Compact-mound, 4' x 6'	White	Glossy, dark green with wavy margins	Highly resistant to *Entomosporium* leaf spot	Ruter (personal communications)
R. ×delacourii	hybrid species	7 - 10					
	'Coats Crimson'	8a - 10	Compact, 2-4' x 2-4'	Crimson-pink	Glossy, dark green	-	Dirr 2009
	'Georgia Petite'	7 - 10	Compact, 2.5' x 3'	Light pink, fade to white	Dark green with light green underside	Resistant to *Entomosporium* leaf spot	Dirr 2009; Corley and Lindstrom 1995
	Indian Princess™ ('Monto')	7 - 11	Broad, mounded 3' x 5'	Broad, bright pink, fade to white	Tomentose new leaves mature to smooth, glossy green	Highly resistant to *Entomosporium* leaf spot	Dirr 2009; Hagan et al., 2002
	Majestic Beauty™ ('Montic')	7 - 11	Upright, 10 - 20' x 10'	Pinkish, fragrant	Bronze 4" leaves mature to dark green	Moderate resistance to *Entomosporium* leaf spot	Dirr 2009; Hagan et al., 2002

Table 5.2 (*continued*) Growth characteristics of selected *Rhaphiolepis* spp. and cultivars currently in production in southeastern United States nurseries.

Species	Cultivar / Trademark	USDA Plant Hardiness Zones	Habit	Flower color	Foliage	Pest resistance	Reference
R. ×delacourii	Snowcap® ('Corleyscourii')	7 - 10	Rounded-mound, 4' x 4'	Pink buds, pale pink flowers	Burgundy-red in winter	Highly resistant to *Entomosporium* leaf spot	Dirr 2009
	'Snow White'	7a - 10	Compact, 3' x 5'	White	Red new foliage matures to dark green	Moderate resistance to *Entomosporium* leaf spot	Hagan et al., 2002
	Springtime® ('Pink Lady')	8 - 10	4 - 6' x 6 - 8'	Deep pink	Glossy, green	Susceptible to *Entomosporium* leaf spot	Dirr 2009; Hagan et al., 2002

Table 5.3 Average range of foliar concentrations reported for macronutrients (A) and micro-nutrients (B) measured in recently mature leaves collected at mid-season from that current season's growth of select *Rhaphiolepis* spp.

(A) Macronutrient % dry weight	N	P	K	Ca	Mg	S
Rhaphiolepis indica 'Bay Breeze' [z]	1.45 - 2.22	0.15 - 0.25	1.25 - 1.75	1.50 - 2.50	0.35 - 0.60	0.15 - 0.28
Rhaphiolepis umbellata Gulf Green™ ('Minor') [z]	1.61 - 2.22	0.12 - 0.13	1.44 - 1.53	2.29 - 2.49	0.30 - 0.33	0.07 - 0.09
Rhaphiolepis ×delacourii 'Snow White' [y]	1.87 - 2.86	0.21 - 0.70	1.12 - 1.45	0.94 - 1.29	0.29 - 0.39	0.78 - 1.19

(B) Micronutrient ppm (µg/g)	Fe[W]	Mn[X]	B	Cu[X]	Zn[X]	Mo	Al	Na
Rhaphiolepis indica 'Bay Breeze' [z]	36 - 55	36 - 75	30 - 45	6 - 15	64 - 75	0.01 - 0.65	37	116
Rhaphiolepis umbellata Gulf Green™ ('Minor') [z]	4 - 16	36 - 122	31 - 37	5 - 10	64 - 112	0.12 - 1.32	21 - 25	106 - 256
Rhaphiolepis ×delacourii 'Snow White' [y]	-	-	-	4.5 - 6.5	-	-	-	-

[z] Bryson et al., 2014

[y] Tillman et al., 2008

[x] Leaf tissue analyses of Cu, Mn, and Zn may not be reliable indicators of plant nutritional status because foliage in commercial nurseries may be exposed to fungicides and nutrient solutions containing trace elements, and surface level contamination may persist even after leaves are washed.

[w] Surface contamination of foliage from soil and presence of unavailable (physiologically inactive) and immobile iron in plant tissues limits the informative value of iron measured by foliar analyses.

Arthropod Pest Management

Figure 5.9 Spirea aphid adult and nymphs.

COMMON ARTHROPOD PESTS

1. Aphids
2. Soft Scales
3. Thrips
4. Weevils

Aphids

Aphids (Hemiptera: Aphididae) are frequent pests of various ornamental plants. However, they are only occasional pests of *Rhaphiolepis*. Aphids typically attack new growth on *Rhaphiolepis*, removing plant sap with their syringe-like sucking mouthparts. Feeding results in deposition of honeydew, an excellent growing medium for black sooty mold. Affected plants are often shiny and sticky on the upper canopy and black in the lower canopy and branches. Extensive feeding can also cause stunted or distorted leaves and buds. The combination of aphids, shed skins, shiny and sticky honeydew, black sooty mold, and distorted tissues makes plants unsalable.

Spirea aphid, *Aphis spiraecola* (Figure 5.9), is the most common aphid species on *Rhaphiolepis*. Spirea aphid is native to North America and has an extremely wide host range, including many fruit and flowering fruit trees. Spirea aphid prefers fruit trees (such as citrus, apple, pear, pomegranate, quince and others) and can spread to nearby *Rhaphiolepis* through dispersal of winged adults. Spirea aphid overwinters as small, shiny, black eggs deposited on the bark of host trees. Aphids of the first generation hatch in early spring and are often called stem mothers. The stem mothers are greenish to yellowish with a pair of black cornicles or tailpipes. The stem mothers produce live nymphs and many of the nymphs develop into winged adults, which then disperse to other hosts or another part of the host tree. The populations remaining on the host trees continue to develop and reproduce wingless individuals, which are pear-shaped, green, and with a pair of black tailpipes. Each aphid completes development in 10 days and produces on average 12-43 offspring in its lifetime (Tsai and Wang, 2001; Wang and Tsai, 2000). There are several generations from spring to fall (Table 5.4). In the fall, winged adults are produced and they disperse to a new host or part of the host plant to deposit overwintering eggs.

Management

Spirea aphid population does not typically remain on *Rhaphiolepis* for extended periods, nor does it cause significant long-term damage. Therefore, infestation by aphids does not always require management.

Figure 5.10 Aphids often escape treatment by residing on the boarders of production areas, in this case on a milkweed. Such populations can quickly re-infest areas after a contact insecticide is applied.

The first step in developing an aphid management program is to develop good weed management and sanitation programs where weeds, debris, and infested plants are removed regularly (Figure 5.10). Growing attractive plant species, such as flowering quince and fruit trees, downwind from the *Rhaphiolepis* can also help to reduce the dispersal of aphids.

When the aphid population is small, spraying with a forceful jet of water can also dislodge many from the infested plant. This method, however, may not be practical when a large number of densely packed *Rhaphiolepis* shrubs are infested.

No study has investigated resistance in various *Rhaphiolepis* species and cultivars against aphids.

Aphid populations are constantly attacked by natural enemies, such as lady beetles, lacewings, syrphid flies and parasitoid wasps. In warm and wet weather, aphid populations are often decimated by outbreaks of naturally existing entomopathogenic fungi. The use of broad-spectrum insecticides (such as organophosphates and pyrethroids) should be limited to conserve natural enemies. Ants are known to collect aphid honeydew and interfere with the actions of natural enemies. Therefore, it is advisable to also develop an ant management program to reduce their population in order to improve biological control.

Table 5.4 Seasonal activity of the major arthropod pests of *Rhaphiolepis* species in southern US. The depicted activity may be early or later than shown depending on location. Activity represented in the table are scale insect crawler emergence, as well as adult and/or nymphal activity of aphids, weevils, thrips and flatheaded borers. The peaks in arthropod abundance, when known, are denoted by 'P.'

Pest	Jan	Feb	Mar	Apr	May	Jun	Jul	Aug	Sep	Oct	Nov	Dec
Spirea aphid, *Aphis spiraecola*												
Flatheaded borer, *Chrysobothris mali*						P	P					
Florida wax scales, *Ceroplastes floridensis*					P		P					
Chilli thrips, *Scirtothrips dorsalis*												
Black vine weevil, *Otiorhynchus sulcatus*					P							
Fuller's rose beetle, *Naupactus cervinus*					P							
Sri Lankan weevil, *Myllocerus undecimpustulatus undatus* (Florida only)	P	P	P								P	P

Aphid populations are seldom large enough to warrant chemical management. When the need arises, insecticides registered for the management of aphids in a production nursery include carbaryl, acephate, pyrethroids, neonicotinoids (acetamiprid, dinotefuran, imidacloprid and thiamethoxam), pymetrozine, azadiractin, abamectin, spirotetramat, neem oil, horticultural oil and insecticidal soap (Table 5.5). The entomopathogenic fungus *Beauveria bassiana* is also very effective. Systemic insecticides, such as the neonicotinoids, could be applied at topical sprays, soil drench, broadcast granules, or trunk sprays. The neonicotinoids applied through indirect method may require days or weeks to move into the plant tissues. Therefore, these chemicals should be applied before the population increases. They can provide long residual protection against aphid infestation. For existing populations, topical applications of listed insecticides to the infested terminals on a weekly or biweekly basis can be very effective in eliminating an infestation and preventing re-infestation during the current growing season.

Soft scales

Growers and landscape professionals frequently find Florida wax scale**,** *Ceroplastes floridensis* Comstock, infesting *Rhaphiolepis* (Figure 5.11). The infestation is often at a low density. Florida wax scale is a polyphagous soft scale species, infesting more than 250 plant species. Hollies (*Ilex* spp.) are the most frequently attacked ornamental plants. The chance of infestation is higher for *Rhaphiolepis* grown near infested hollies, likely via wind

Figure 5.11 Florida wax scales feeding on *Rhaphiolepis*.

dispersal of crawlers (Yardeni and Rosen, 1990). The pinkish females are oval, about 1/8 inch in length, and covered with a thick layer of gummy wax deposit (Figure 5.12). Nymphs are also oval; the wax deposit does not cover the entire body but appears as blooms of wax on the fringe and top of body (Figure 5.12). Both adults and nymphs feed on twigs and branches, and along the leaf veins. Feeding by Florida wax scale does not cause plant death or dieback, but the presence of the scale insects, honeydew and sooty mold reduce the esthetic value of the shrubs. There are two generations per year in the southern range of this scale insect (FL to TX and NC); one generation may occur north of VA, but the population often fails to overwinter (Kosztarab, 1996). Crawler emergence typically occurs in May and July in the southern range (Chong, 2015) (Table 5.4). Nymphs develop through 3 instars over a 2-month period. A nymph can move from leaves to branches (and vice versa) throughout its life. Each female is capable of producing 150 reddish to pinkish eggs.

Figure 5.12 Adult Florida wax scales.

Management
No information is available on the resistance of *Rhaphiolepis* species and cultivars to the Florida wax scale.

Scale insect management begins with prevention. Sanitation practices, such as removal of debris, old plants and infested plant tissues (through pruning), can remove the source of inoculum and prevent the build up of a scale insect population. Crawlers hatch from debris, leaves, and pruned tissues and can crawl back onto the *Rhaphiolepis* cuttings or shrubs.

Figure 5.13 Parasitoids, such as *Coccophagus lycimnia* seen here, are significant predators of scale.

Florida wax scale populations often do not cause significant damage because they are kept at a low density by their natural enemies (mainly predators and parasitoids). Parasitoids, such as *Coccophagus lycimnia* (Figure 5.13), are major mortality factors of Florida wax scale (Gordh, 1979). However, the populations and activities of the arthropod predators and parasitoids can be disrupted by the use of broad-spectrum insecticides (such as acephate, carbaryl, chlorpyrifos and pyrethroids); therefore, avoid making foliar applications of these broad-spectrum insecticides.

Heavy infestations may require chemical management. The most effective timing for topical application of insecticides is during or shortly after the emergence of crawlers (Table 5.4). The scales (tests or shells) are made of wax, cast skin and fecal materials, and function as protection against the environment and natural enemies. Once the shells are formed, insecticide solutions are not effective in penetrating the protective layer and contacting the bodies of scale insects. Crawlers have little or no waxy shells and so they are more susceptible.

Crawler emergence can be monitored with:

1. Visual inspection: Inspect infested plants or leaves weekly near the reported time of crawler emergence. Hand lens and magnifying glass are great aids. Look for reddish or pinkish crawlers that are dispersing or emerging from the females.
2. Sticky trap: A sticky trap can be easily constructed by wrapping a double-sided sticky tape around an infested branch. Construct and check the trap weekly a few weeks before the reported emergence time of the crawlers of each species. Look for reddish or pinkish crawlers that are stuck to the sticky trap.

Unfortunately, no degree-day or plant phenological indicator model is available for the Florida wax scale.

Insecticides registered for use against soft scales in nurseries include acephate, carbaryl, chlorpyrifos, dimethoate, flonicamid, pyrethroids, buprofezin, azadirachtin, pyriproxyfen, spirotetramat, neonicotinoids (acetamiprid, dinotefuran, imidacloprid and thiamethoxam), entomopathogenic fungi (*Beauvaria bassiana*), insecticidal soap and horticultural oil (Table 5.5). Except for the neonicotinoids, all insecticides should be applied as foliar sprays at or near the time of crawler emergence. A second application is required 14 or 21 days after the first application because the crawlers often emerge over an extended period of time. Often the addition of adjuvant or spreader-sticker can increase the efficacy. Neonicotinoids may be applied directly via foliar spray, or indirectly via soil or substrate drench, soil injection, trunk injection, trunk spray or granule broadcast application. Foliar sprays of neonicotinoids should follow the rule of application at or near the time of crawler emergence. Indirect applications should be applied before crawler emergence to allow translocation of the insecticide throughout the plant.

Thrips
Before the introduction of the chilli thrips, *Scirtothrips dorsalis* Hood (Figure 5.14), *Rhaphiolepis* was a group of relatively low-maintenance shrubs (except for occasional aphid and Florida wax scale infestations). Also known as the yellow tea thrips, chilli thrips are widely distributed in the world, inflicting serious damage to vegetables, ornamentals, and fruit crops. The first successfully established chilli thrips population was detected in

Florida in 2005 (Edwards et al., 2005). Currently, the chilli thrips are found in Florida, Georgia and Texas. Holtz (2006) suggested that the distribution of chilli thrips may be restricted to the southern states and the Pacific states.

Figure 5.14 Chilli thrips are widely distributed in the world, inflicting serious damage to vegetables, ornamentals and fruit crops. However, in the U.S. they are predominantly located along the gulf coast (as of 2016).

Chilli thrips prefers to feed on growing plant tissues, such as leaf, fruit and flower buds. However, it will attack all above-ground plant tissues indiscriminately. Thrips feed by puncturing cell walls and removing cell contents. As the damaged buds grow and expand, the feeding damage manifests as distortion of foliage (often curling upward), flowers and shoots, as well as bronzing of foliage and flowers (Figure 5.15). Older leaves can be severely scarred and necrotic blotches or bronzing may appear. Heavily infested plants are stunted, often resulting in disfigured, unsalable plants. No study has investigated the potential resistance of various *Rhaphiolepis* species and cultivars to chilli thrips.

Figure 5.15 Damage on *Rhaphiolepis indica* by the chilli thrips.

The adult chilli thrips is a small insect [less than 1 mm (4/100 of an inch)] and is smaller than typical flower thrips. The adult chilli thrips has a pale yellow or almost white body banded with small blotches on the abdominal segments (Figure 5.14) (Hodges et al., 2009). A female can insert 60 to 200 tiny eggs into plant tissues over her lifetime. Nymphs feed on the foliage immediately after hatching. Duration of development was 18-21 days when reared on bean, eggplant, pepper, rose, squash and tomato at 78.8°F (26°C) (Seal et al., 2010). In southern Florida, 14 to 18 generations of chilli thrips are possible per year (Holtz, 2006), where the chilli thrips is expected to remain active for most of the year (Table 2.4).

Management

Management of chilli thrips should begin with sanitation. Remove weeds, debris, and old or infested plants within and around the production areas and propagation greenhouses.

Chilli thrips and other flower thrips can be monitored with yellow or blue sticky cards. Install the cards vertically just above the canopy and along the edge of plots, or near vents, doors and windows. Examine the cards at least weekly to determine if thrips are captured. Also check the surrounding plants for feeding damage to new leaves and growing buds (necrotic scars and distortion). Alternatively, tap foliage or flowers on a white background (a piece of paper or board) and look for pale yellow or white cigar-shaped thrips. If infestation by chilli thrips is suspected, thrips should be collected and sent to state extension specialists or diagnostic services for identification.

Predatory mites (*Neosieuslus cucumeris, Amblyseius swirskii* and *Hypoaspis miles*) and bugs (*Orius* species) are commercially available for biological control of thrips. Arthurs et al. (2009), Dogramaci et al. (2011) and others have demonstrated the potential of these biological control agents to reduce chilli thrips populations. Entomopathogenic nematodes (*Steinernema feltiae*) and fungi (*Beauveria bassiana*) can be effective alternatives to insecticides. When biological control program is being implemented, it is important to understand the potential negative interactions selected insecticides, miticides, and fungicides have with the biological control agents. This information can usually be found on the suppliers websites.

Insecticides are most effective against thrips population at lower densities, before damage becomes apparent. Reapplication on at least a biweekly basis may be necessary to reduce the thrips population that is constantly been supported by individuals migrating from the surrounding areas. To delay the development of insecticide resistance, applicators should rotate among insecticides of different modes of action or IRAC numbers with each treatment. Chemicals registered for treatment against thrips in the nurseries include acephate, abamectin, acetamiprid, azadirachtin, dinotefuran, flonicamid, novaluron, pyrethroids, spinosad, thiamethoxam, insecticidal soap and horticultural oil. Most of these chemicals are evaluated against the western flower thrips but efficacy data against chilli thrips are also available (Table 5.5; Bethke et al., 2011).

Weevils

Weevils are occasional pests of *Rhaphiolepis*. Adult weevils are easily distinguished by their elongated snouts. Adults feed on the leaves of *Rhaphiolepis* and cause hemispherical or elongated notches, often starting from the edges, on the leaves. Larvae are deposited by the females on the shrubs, or on the ground. The larvae drop to the ground after hatching and feed on the roots. Larvae of most weevil species have brown or reddish heads, are soft-bodied, white, C-shaped and legless. Feeding by a large number of larvae can eventually reduce root mass and girdle the main roots and stems.

Figure 5.16 Adult black vine weevil.

Black vine weevil, *Otiorhynchus sulcatus* (F.), is a common pest of many ornamental plant species, including yews, rhododendrons, euonymus and hollies (Reding, 2008). It is an occasional but at times severely damaging pest of *Rhaphiolepis* in the Northeastern and Midwestern U.S. The black vine weevil is introduced from northern Europe in 1835 (Smith, 1932). Adult weevil is 3/8 inches long, dull gray to black, with short broad snout, and numerous small golden spots on the elytra (Figure 5.16). Adults (all of them females that reproduce through parthenogenesis) do not fly, but climb onto the host plant to feed at night. They return to the soil or mulch during the day. Larvae feed on the roots. There is one generation per year, with larvae overwintering in the soil and adults emerging in the spring. Each female deposits up to an average of 880 eggs (Fisher, 2006).

As the black vine weevil is the predominant weevil pest of *Rhaphiolepis* in the Northeastern and Midwestern U.S., Fuller's rose beetle, *Naupactus cervinus* Boheman, is similarly important in the southern U.S. This species was introduced from South America, first detected in California in 1879 (Chadwick, 1965) followed by Florida in 1916 (Woodruff and Bullock, 1979). It is now widely distributed east of the Rocky Mountains, but infestations can also be found along the Pacific Coast. This species has an extremely wide host range (Chadwick, 1965). The adult Fuller's rose beetle is about 1/3 inch long, with a short broad snout, and a brownish grey body speckled with white patches (Figure 5.17). There is one generation per year in most of the U.S., while there may be two generations in Florida (Woodruff and Bullock, 1979). Adults emerge in May (August for the second generation in Florida) and active feeding occurs on the leaves and buds throughout the summer (Table 5.4). Similar to the black vine weevil, the Fuller's rose weevils are all females and reproduce through parthenogenesis. Eggs are deposited on plants, and larvae drop to the ground to feed on roots. This species overwinters as larvae in the soil.

Figure 5.17 Adult Fuller's rose beetle.

Sri Lankan weevil, *Myllocerus undecimpustulatus undatus* Marshall, is a pest of *Rhaphiolepis* only in Florida. First detected in 2000, the Sri Lankan weevil is now distributed mainly in central and southern Florida (Neal, 2013). This weevil species is native to India and Sri Lanka, but it is not clear how it was introduced to Florida. In Florida, adult Sri Lankan weevils feed on the foliage of 150 plant species (O'Brien et al., 2006; Neal, 2013), whereas larvae feed on roots. The Sri Lankan weevil adult is about 1/5 to 3/10 inch in length, it has a whitish body with black mottles, yellowish head, broad snout, broad and strongly angled humeri (which give the weevil an appearance of having head and thorax sitting on a broad 'shoulder'), and spines on the femora (Figure 5.18). Each female may produce as many as 360 eggs (Neal, 2013). The life history of the Sri Lankan weevil is relatively known. Adult emergence from the soil appears to occur predominantly from November to March, whereas adult feeding on foliage can occur at any time of the year in Florida (Epsky et al., 2009).

Management

The black vine weevil, Fuller's rose weevil, and Sri Lankan weevil are polyphagous species that could feed and develop on many ornamental plant species grown in the same nursery or landscape. A management program should therefore take a systematic approach by managing weevil population not only on the ornamental plant species the weevils are currently impacting, but also on other potential host species. Plants on which typical adult damage have been observed (i.e. notched leaf margin) should be treated for both adults and larvae, or removed from the production area so that they do not become a source of infestation. In addition, by laying down a nursery mat or by removing leaf litter or other debris, growers can deny hiding, pupating, and egg deposition sites for the weevils.

When only a few plants are impacted, a sticky material (Tanglefoot) can be applied to the trunk, and leaves and branches touching the ground should be removed, to eliminate routes the weevils can use to climb up the plants. Also, hand removal of adults can also be effective (Masiuk, 2003). However, these cultural and physical methods may be too labor intensive for treating a large number of plants in commercial nurseries.

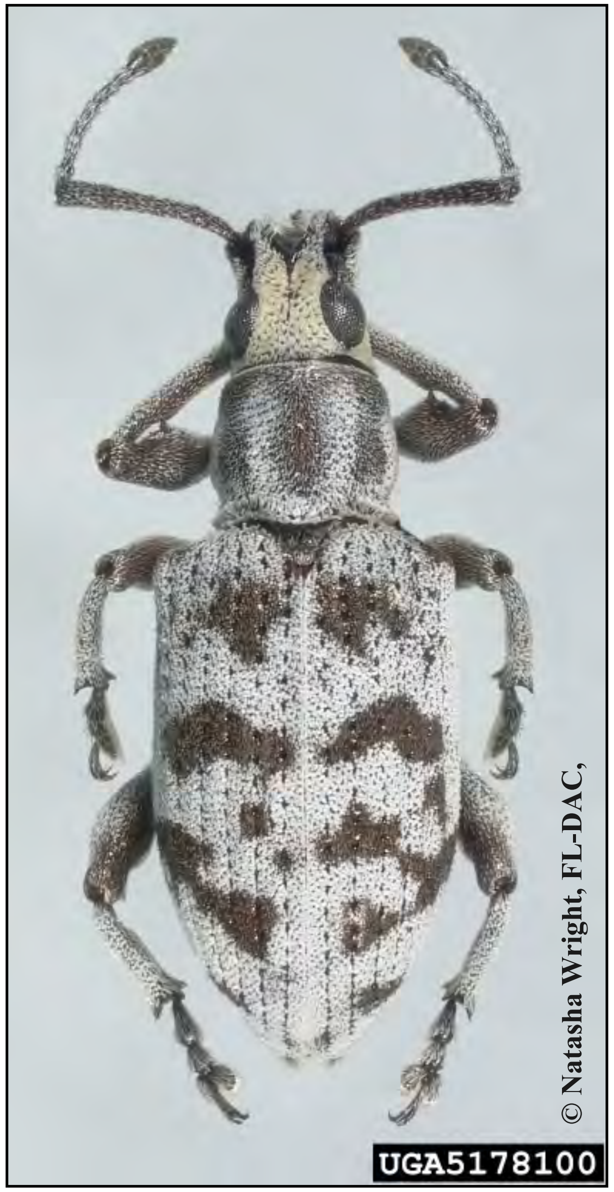


Figure 5.18 Adult Sri Lankan weevil.

The presence of weevils in nurseries is determined by monitoring for damage. Visual monitoring, however, does not provide growers with information on the spring emergence of adults so that timely application of control methods can begin before feeding and reproduction. A recent study showed that a 1:1 mixture of (Z)-2-pentenol and methyl eugenol was effective in attracting the black vine weevils in Oregon (van Tol et al., 2012). However, the effectiveness of such an attractant for monitoring black vine weevils in commercial tree and shrub nurseries remains to be tested. In Ohio, adult emergence of the black vine weevil was found to begin when black locust and yellowwood are in full bloom, and mountain laurel and multiflora rose are in first bloom (Herms, 2004). Emergence traps resembling the pyramid traps used for pecan weevils may be used to detect emergence of the Sri Lankan weevil and the Fuller’s rose weevil, whereas the beat sheet method can be useful in finding adults feeding in the canopy (Epsky et al., 2009). About 99% of Fuller’s rose weevil eggs hatch when 351 DDC at a base temperature of 10.2°C (50.2°F) have been accumulated (Lakin and Morse, 1989).

Chemical management of pest weevils should begin soon after adult emergence (Table 5.5). For adult control, topical applications of acephate, carbaryl, chlorpyrifos, pyrethroids and azadirachtin should be applied at least twice (2-3 weeks between the applications) after adult emergence. Systemic neonicotinoid insecticides applied as a soil drench to containerized or field-grown plants are effective in reducing the numbers of larvae as well as feeding damage by the adults (Reding and Persad, 2009; Reding and Ranger, 2011). Bifenthrin can be used as a replacement for the more hazardous chlorpyrifos for root ball dipping (Cowles, 2001).

Few effective biological control agents are available for the management of weevils. Drenching the soil and substrate with entomophathogenic namatodes (*Steinernema carpocapsae, Steinernema feltiae, Steinernema kraussei* and *Heterorhabditis bacteriophora*) and entomopathogenic fungi (*Beauveria bassiana, Isaria formosorosea* and *Metarhizium anisopliae*) may reduce adult and larvae population. There are indications that *Hetetorhabditis* species are more effective in reducing the abundance of black vine weevil larvae due to its higher dispersal within potting substrate (Ansari and Butt, 2011). The soil or substrate should be thoroughly wet before application of nematodes or fungi to increase their efficacy.

Table 5.5 Insecticides available to control common insect pests of *Rhaphiolepis* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Soft scales	Thrips	Weevils	Aphids
1A	Acetylcholinesterase inhibitors	Carbamates	carbaryl[8]	Sevin SL	L, N, G	12	x	x	x	x
			methiocarb	Mesurol 75W	N, G	24		x		x
1B[20]	Acetylcholinesterase inhibitors	Organophosphates	acephate[8,9]	Orthene T&O[9]	L, N, G	24	x	x	x	x
				Lepitect[10]	L, N, G	24	x	x	x	x
				Precise GN[9]	G, N	12			x	x
			chlorpyrifos	Dursban 50W[9]	N	24	x	x	x	x
				DuraGuard ME[9]	N, G	24	x	x	x	x
			dicrotophos	Inject-A-Cide B[11]	L	N/A	x			x
			dimethoate	Dimethoate 4E[9], 4EC[9]	N	14 days [21]	x	x		x
			malathion	Malathion 5EC[9,10]	L	12	x	x		x
			methidathion	Supracide 2E[9,19]	N	30 days [21]	x			
			oxydemeton methyl	Harpoon[11,9]	L	0	x			x
				MSR Spray Concentrate[12,19]	N	10 days [21]		x	x	x
3A[20]	Sodium channel modulators	Pyrethroids / Pyrethrins	bifenthrin[8]	Attain TR	G	12	x	x		x
				Menace GC	L, N, G	12	x	x	x	x
				Onyx	L	N/A	x	x		x
				OnyxPro	L, N, I	12	x	x		x
				Talstar S Select	N, G	12	x		x	x
				Talstar Nursery G	N	12			x	
			cyfluthrin	Decathlon[14]	L, N, G, I	12	x	x	x	x
			beta-cyfluthrin	Tempo Ultra WP[14]	L, I	N/A	x	x	x	x
				Tempo SC Ultra[14]	L, I	N/A	x	x	x	x
			lambda-cyhalothrin	Demand	L	N/A	x	x	x	x
				Scimitar CS; Scimitar GC	L ; L, N, G	24; 24	x	x	x	x
			cypermethrin	Demon WP	L, I	N/A				x
			deltamethrin	DeltaGard G[14]	L, I	N/A			x	
			fenpropathrin	Tame 2.4 EC[9]	N, G, I	24		x	x	x
			tau-fluvalinate	Mavrik Aquaflow	L, N, G, I	12		x	x	x

Table 5.5 (***continued)*** Insecticides available to control common insect pests of *Rhaphiolepis* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Soft scales	Thrips	Weevils	Aphids
3A[20]	Sodium channel modulators	Pyrethroids / Pyrethrins	permethrin	Astro	L, G, I	12		x	x	x
				Permethrin Pro	L, I	N/A		x	x	x
				Perm-Up 3.2 EC	L, N, G, I	12		x	x	x
			pyrethrins	Tersus	N, G	12	x	x	x	x
				Pyganic	N, G	12	x	x		x
			pyrethrum	Pyrethrum TR	N, G	12	x	x		x
3A + 4A	Sodium channel modulators	Pyrethroids + Neonicotinoids	bifenthrin + clothianidin	Aloft LC G, LC SC	L	N/A	x	x	x	x
			bifenthrin + imidacloprid	Allectus SC	L, I	N/A	x	x	x	x
			cyfluthrin + imidacloprid	Discus N/G	N, G, I	12	x	x	x	x
			lambda-cyhalothrin + thiamethoxam	Tandem	L	N/A	x	x	x	x
			zeta-cypermrthrin + bifenthrin + imidacloprid	Triple Crown T&O	L, I	N/A	x	x	x	x
3A + 27A	Sodium channel modulators	Pyrethroids + Piperonyl butoxide (PBO)	pyrethrins + piperonyl butoxide[8]	Pyreth-It	N, G	12		x		x
4A[25]	Nicotinic acetylcholine receptor agonists	Neonicotinoids	acetamiprid	TriStar 8.5 SL[9,10,14]	L, N, G	12	x	x	x	x
			clothianidin	Arena 0.25 G[10]	L, I	12				x
				Arena 50 WDG[10]	L, I	12				x
			dinotefuran	Safari 2G[10]	L, N, G, I	12	x	x[15]	x	
				Safari 20 SG[10]	L, N, G, I	12	x	x[15]	x	x
				Zylam Liquid[10]	L	N/A	x	x[15]	x	x
				Transtect 70 WSP[10]	L	N/A	x	x[15]	x	x
			imidacloprid	Xytect 75WSP; 2F	L, N, G, I	12	x	x	x	x
				Marathon II	N, G, I	12	x	x	x	x
				Marathon 60WP	N, G, I	12	x	x	x	x
				Merit	L, I	N/A	x	x	x	x
				CoreTect	L, I	N/A	x	x	x	x
				Discus Tablets	N, G, I	12	x	x	x	x

Table 5.5 (***continued)*** Insecticides available to control common insect pests of *Rhaphiolepis* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Soft scales	Thrips	Weevils	Aphids
4A[25]	Nicotinic acetylcholine receptor agonists	Neonicotinoids	thiamethoxam	Flagship 25WG	N, G, I	12	x	x	x	x
				Meridian 0.33G	L, I	N/A	x	x	x	x
				Meridian 25WG	L, I	N/A	x	x	x	x
4C + 5 [17]	Nicotinic acetylcholine receptor agonists	Sulfoxaflor + Spinosyns	sulfoxaflor + spinetoram	XXpire[13,16]	L, N, G	12		x		x
5	Nicotinic acetylcholine receptor allosteric activators	Spinosyns	spinosad	Conserve SC	L, N, G	4		x		
				Entrust	L, N, G	4		x		
6 [20]	Chloride channel activators	Avermectins, Milbemycins	abamectin[9]	Lucid, , Minx, Avid[9]	L, N, G	12		x		x
				Aracinate TM[11]	L, N, G, I	N/A	x	x	x	x
			emamectin benzoate	Arbormectin	L	N/A				x
6 + 20D	Chloride channel activators	Avermectins, Milbemycins	abamectin + bifenazate	Sirocco[14]	L, N, G, I	12		x		x
7A	Juvenile hormone mimics	Juvenile hormone analogues	s-kinoprene	Enstar AQ	G, I	4	x	x		x
7B	Juvenile hormone mimics	Fenoxycarb	fenoxycarb	Preclude TR	G	12	x		x	x
7C	Juvenile hormone mimics	Pyriproxyfen	pyriproxifen	Distance IGR[9]	L, N, G, I	12	x	x		
				Fulcrum	L, N, G, I	12	x			x
8C	Misc. non-specific (multi-site) inhibitors	Flourides	cryolite (sodium alumino-fluoride)	Kryocide[4]	L	N/A			x	
8D	Misc. non-specific (multi-site) inhibitors	Borates	sodium tetraborohydrate decahydrate	Prev-AM Ultra	N, G	12	x			x
9B[20]	Selective homopteran feeding blockers	Pymetrozine	pymetrozine	Endeavor[9,14]	L, N, G, I	12				x
		Pyrifluquinazon	pyriflquinazon	Rycar	G	12				x
13	Uncouplers of oxidative phospho-rylation via disruption of the proton gradient	Chlorfenapyr	chlorfenapyr	Pylon[9]	G	12		x		
15 [25]	Inhibitors of chitin biosynthesis, type 0	Benzoylureas	diflubenzuron	Dimilin 25W	L, N	12			x	
				Dimilin 4L	L, N	12			x	
			novaluron	Pedestal[9]	N, G	12		x		
16	Inhibitors of chitin biosynthesis, type 1	Buprofezin	buprofezin	Talus 70DF	L, N, G	12	x			
21A	Mitochondrial complex I electron transport inhibitors	METI acaricides and insecticides	tolfenpyrad	Hachi-Hachi SC[9]	G	12	x	x		x
23	Inhibitors of acetyl CoA carboxylase	Tetronic and Tetramic acid derivatives	spirotetramat	Kontos[9]	N, G, I	24	x	x		x

Table 5.5 (***continued)*** Insecticides available to control common insect pests of *Rhaphiolepis* spp. in commercial production environments.

IRAC Code[3]	Mode of Action	Chemical subgroup	Active Ingredient	Selected Trade Names[4,5]	Use Site[2]	REI (hrs)[6]	Soft scales	Thrips	Weevils	Aphids
28	Ryanodine receptor modulators	Diamides	chlorantraniliprole	Acelepryn	L, I	N/A	x[18]			x
				Acelepryn G	L, I	N/A				x
			cyantranili-prole	Mainspring	G, I, L	4	x	x		x
29	Chordotonal organ Modulators	Flonicamid	flonicamid	Aria[9,16]	L, N, G	12	x	x		x
Unknown[20]	Unknown	Azadirachtin	azadirachtin[8]	Azatin O	L, N, G, I	4	x	x	x	x
				Azatin XL	N, G, I	4		x	x	x
				Azatrol EC	L, N, G, I	4	x	x	x	x
				Ornazin 3% EC	L, N, G, I	12	x	x	x	x
		Pyridalyl	pyridalyl	Overture 35WP	G	12		x		
Not classified	Various		*Beauveria bassiana*[10]	BotaniGard ES	L, N, G, I	4		x	x	x
				BotaniGard 22 WP	L, N, G, I	4		x	x	x
				Naturalis-L26	L, N, G	4		x	x	x
			Chromobacterium subtsugae	Grandevo PTO[5]	L, N, G	4		x		x
			Heterorhabtditis bacteriophera	LarvaNem	L	N/A			x	
				ExhibitlineH	N	N/A			x	
				NemaShield	L, N, G, I	N/A			x	
			Isaria formosorosea	NoFly, Preferal	L, N, G	12		x	x	x
			Metarhizium anisopliae	Met52, Tick-EX	L, N, G	4		x	x	
			Steinernema carpocapsae	Millenium, EntoNem	L, N, G, I	N/A		x	x	
			Steinernema feltiae	NemaSys, Steinernema-System	L, N, G, I	N/A		x		
				NemaShield	L, N, G, I	N/A		x		
				EntoNem	L, N, G, I	N/A		x	x	
			Steinernema kraussei	Nemasys L	L, N, G, I	N/A			x	
				Kraussei-System	N	N/A			x	
			horticultural oil[8,9]	Ultra-Pure Oil, TriTek	L, N, G, I	4	x	x		x
			insecticidal soap[8,9]	M-Pede[9]	L, N, G, I	12	x	x		x
			neem oil[8]	Trilogy	L, N	4	x	x		x
				Triact 70[9]	L, N, G, I	4	x			x
			capsicum extract	Captivia	L, N	4		x		

Table 5.5 (***continued)*** Insecticides available to control common insect pests of *Rhaphiolepis* spp. in commercial production environments.

Footnotes and Supplemental Information:

1. Pests listed on pesticide labels are subject to change. Within states and counties, products may have additional permitted uses by 2(ee) and Special Local Need allowances. Consult your CountyExtension Agent or State Agency to determine if other uses are allowable. The label should always be consulted to confirm that chart-listed pests still appear on the label. Check the product labels for specific site restriction information, notes on application, sensitive plant species and specific target pest species.
2. Use site information is provided for reference only: L = landscape; N = nursery; G = greenhouse; I = interiorscape. t/s*Products listed for use in L, N, G or I sites may include active ingredients in differently labeled products that are designated for use in turfgrass or sod production use sites. Mention of those products is beyond the scope of this resource guide. Products listed herein may not be legal or appropriate for site uses in turfgrass or sod production.
3. IRAC Code designations and Related Modes of Action are explained at the Insecticide Resistance Action Committee Database 2016 (IRAC 2016; http://www.irac-online.org/teams/mode-of-action/).
4. Trade names of products are provided as examples only. No endorsement of mentioned product nor criticism of unmentioned products is intended.
5. Products may not be registered or renewed for use in all southeastern U.S. states. Consult your state's Department of Agriculture to confirm legal use of products in your state.
6. Re-Entry Interval (REI) designations apply to agricultural (nursery) uses. Within REI column, 'N/A' is used to indicate products with Landscape and/or Interiorscape site uses and that present Non-Agricultural Use Requirements. Consult product labels for specific details required for compliance within those application conditions and use sites, including those for which REI listings do not apply. Additionally, some product labels may list site uses that are beyond the scope of this publication (e.g., sod farms, silvicultural & Christmas tree nurseries, pastures, rights-of-way, etc.). These site uses may involve Agricultural Use Requirements that list an REI not presented here.
7. lh = leafhopper; ph = planthopper (juv. = juveniles, or nymphs)
8. Multiple formulations and trade names of the same active ingredients are available. Labels among these products often differ in legally allowable uses (regarding sites, pest, REI, etc.); representative example(s) presented.
9. See product label for information about potential crop phytotoxicity, known plant sensitivity, and how to test for phytotoxicity.
10. Check label for additional restrictions (e.g., on the number of times the product can be applied in a growing season or year, or additional application restrictions within permitted use sites; and crop types or flowering status that may not be legally treated.)
11. Product formulated for tree or shrub injection; specialized equipment may be required.
12. Use of handheld application equipment is prohibited. For use only on seedling trees and non-bearing fruit trees in commercial nurseries.
13. Label includes white grub and weevil larval control in nursery containers.
14. Intended for commercial/professional use only. Not intended for homeowner use.
15. Label-indicated efficacy against target pest group is primarily by population suppression.
16. Product labels may restrict landscape uses only to commercial landscapes. Residential landscapes are not a permitted application site.
17. Only for use on Christmas trees.
18. On the Acelepryn Section 2ee label.
19. Product uses cancelled or in process of Federal cancellation or review. Use of existing stocks of any end-use product may be allowable beyond the cancellation date. Consult your extension agent or state agency for information about allowable use and conditions for proper disposal.
20. Applications of products from these footnoted IRAC groups are optimized when not preceded by/or followed by products in subsequently noted IRAC Group(s), particularly when managing the indicated pest. Suggested rotation combinations to avoid include serial treatments with a.i. products from: IRAC 1B, 3A, 6 (western flower thrips); IRAC 4A, 4B, 4C (green peach aphid); IRAC 4A and 9B (glasshouse and silverleaf whiteflies); IRAC 10A, 10B, 15 (Panonychus [red and citrus] spider mites); IRAC 20B and 20D bifenazate (twospotted spider mite).
21. Some REI periods are in days, rather than hours. Please consult the insecticide label for specific REI period.

SECTION 3

Disease Management

Figure 5.19 Near complete defoliation (leaf drop) on *Rhaphiolepis* caused by Entomosporium leaf spot.

COMMON DISEASE PESTS

1. Entomosporium leaf spot
2. Fire Blight
3. Anthracnose
4. Rhizoctonia

Entomosporium leaf spot

Entomosporium leaf spot caused by the fungus *Entomosporium mespili* is the most common disease of *Rhaphiolepis* in the Southeastern U.S. It is found in container nurseries and continues to be a problem in landscapes. The pathogen is most active in late fall through winter and spring (Hagan et. al., 2003). Cultivars of *Rhaphiolepis* that are moderately to highly susceptible to Entomosporium may suffer significant defoliation by mid-summer (Figure 5.19; Table 5.2).

Figure 5.20 Close-up of entomosporium leaf spot symptomlogy.

Symptoms begin as small red, leaf lesions and expand to form lesions up to 1 cm (0.4 in.) in diameter (Figure 5.20). As lesions mature they may be have red-to-brown borders with brown to gray centers. Black fruiting structures of the fungus are often visible in the center of lesions. As lesions coalesce, large portions of the leaf may be killed. Mature leaves that are infected in late spring may exhibit light green-to-white leaf spots called ghost spots. Warmer temperatures slow or stop the infection process leading to ghost spots (Figure 5.21).

Entomosporium mespili (teleomorph, *Diplocarpon mespili*) spores (conidia) are produced in fruiting bodies (acervulii) just underneath the leaf epidermis. The fungus survives on twigs and fallen leaves. Mild, wet weather promotes infection. Temperatures of 15-25°C (59-77°F) and 9 to 12 hours of leaf wetness favor disease development (Sinclair and Lyon, 2005). In sub-tropical areas, the fungus may remain active all winter. The host range of *E. mespili* includes: apple, cotoneaster, crabapple, hawthorn, loquat, medlar, mountain ash, pear, photinia, quince and serviceberry (Colbaugh et al., 2001).

Figure 5.21 Ghost spot symptoms on mature *Rhaphiolepis* leaves infected in late spring.

Management
Integrated disease management of Entomosporium leaf spot includes cultural practices, sanitation, disease resistance and/or fungicide sprays.

Cultural practices and sanitation - Incoming plants should be inspected for symptoms of disease. Cuttings should only be taken from disease free plants. Stock plants should be isolated from production areas. Diseased leaves should be removed from production areas. Irrigation intervals should be scheduled to minimize leaf wetness.

Host resistance – *Rhaphiolepis* cultivars resistant to Entomosporium leaf spot in Alabama include: Indian Princess™, 'Clara', 'Snow White', 'Olivia' and Eleanor Tabor™ ('Conor') (Hagan et al., 2003). In 2004, Ruter reported that the *Rhaphiolepis* cultivars 'Minor', 'Georgia Petite', 'Olivia', 'Georgia Charm', 'Eleanor Taber' and 'Pink Pearl' were resistant to Entomosporium leaf spot.

Rhaphiolepis cultivars susceptible to Entomosporium leaf spot in Alabama include: 'Spring Rapture', White Enchantress®, 'Enchantress', 'Heather', 'Harbinger of Spring', 'Pinkie' and 'Bay Breeze' (Hagan et al., 2003); while in Georgia, Ruter (2004) that 'Snow White', 'Clara', 'Ballerina', 'Bay Breeze', 'Cameo', 'Elizabeth', and 'Kathy' were susceptible. As there is some disagreement as to the resistance and susceptibility of *Rhaphiolepis* cultivars to Entomosporium leaf spot, it is possible that different strains of the fungus exist in various locales. Use resistance/susceptible terminology as a tentative guide to evaluate cultivars in your area.

Fungicides should be used as preventative treatments prior to disease development (Table 5.6). In the lower south, fungicide sprays may be needed as early as November. Fungicide sprays should be continued at 10-14 day intervals (as directed by the product's label).

Fire Blight
Fire blight is an occasional bacterial disorder in *Rhaphiolepis.* The causal agent, *Erwinia amylovora*, is a bacterial pathogen of plants in the Rosaceae. Fire blight has been reported in Alabama, Florida, Georgia and Louisiana in landscapes and container nurseries (Hagan et al., 2001; Holcomb, 1998; Momol et al., 2000). Fire blight may cause a blossom blight and extensive shoot blight during mild, wet weather. New growth may wilt as large portions of the plant are killed. Infected plants are often unsalable.

As in apple, *E. amylovora* may overwinter on infected twigs or cankers on larger branches. During cool, wet weather in the spring, the bacterium can be found in amber colored ooze on infected tissue. Bacteria may be splash dispersed to adjacent leaves, flowers and plants. Pollinating insects may move the bacteria among flowers. High nitrogen fertilization of quickly available nitrogen that stimulates excessive new growth may make plants more susceptible to disease.

Management
To minimize damage from fire blight. Use cultural practices such as slow release fertilizer. Consider using split applications of fertilizer when potting plants and later as a top-dressed application later in the season to avoid tender, succulent growth. If pruning or shearing in the spring during bloom, disinfect tools as often as practical. Discard severely infected plants as soon as they are spotted. Antibiotics such as streptomycin have been used during bloom to minimize damage; during the shoot blight phase they are of little value. There are reports of *Erwinia* resistance to antibiotics and copper materials (Sinclair and Lyons, 2005; Table 5.6). Hagan et al, (2001) reported that *Rhaphiolepis* cultivars resistant to fire blight are (Table 5.2): 'Bay Breeze', 'Clara', 'Dwarf Yedda', Eleanor Tabor™ ('Conor'), 'Enchantress', 'Harbinger of Spring', 'Heather', Indian Princess™, Majestic Beauty™, 'Pinkie', Rosalinda®, 'Snow White', 'Spring Rapture', Springtime® and 'White Enchantress'; susceptible cultivars 'Jack Evans' and 'Janice'. Also, 'Olivia' was reported to be susceptible to fire blight in Louisiana by Holcomb (1998).

Anthracnose
Anthracnose is a fungal disease of *Rhaphiolepis* caused by *Colletotrichum gloeosporioides.* Symptoms include leaf spots, leaf drop and twig dieback. While there are many anthracnose diseases of ornamental plants, little is known about the importance and occurrence of anthracnose on *Rhaphiolepis*. Hagan et al. (2001) reported that 'Majestic Beauty' was moderately susceptible to anthracnose.

It is most prevalent in dense plantings and develops under wet, hot conditions (75 to 95°F; 24 to 35°C). Symptoms occur as large necrotic brown spots which appear simultaneously on leaves and flowers, especially during periods of heavy rainfall. The infected spots are a lighter brown in the center, with darker discoloration on the outer edges and have a "bull's-eye" appearance. Spores are spread by water.

Management.
Remove infected leaves and flowers, and take cuttings from disease-free plants to avoid future infections. Preventative applications of fungicides can be used to avoid anthracnose infections, but repeated and frequent applications are required (Table 5.2).

Rhizoctonia
Other diseases of *Rhaphiolepis*, include diseases such as Rhizoctonia leaf spot (Figure 5.22) and web blight (Figure 5.23) caused by *Rhizoctonia solani* and other *Rhizoctonia* spp. (J. Olive, personal communication). Web blight occurs in hot, humid climates. Cobweb-like mycelium of Rhizoctonia grows throughout the canopy of infected plants,

Figure 5.22 Zonate (target-like) spots on *Rhaphiolepis* leaves indicate Rhizoctonia leaf spot infection.

blighting leaves in the interior of the canopy. Initial symptoms of Rhizoctonia web blight within the inner canopy are often overlooked. Individual leaves within the lower and inner canopy may develop leaf spotting and necrosis. The blight progresses outward and upward through the canopy until much of the foliage is necrotic. Individual leaves will drop from the plant, but may appear to remain attached to the stems because *Rhizoctonia* hyphae mats the leaves together and to the stems, thereby holding the leaves in place. Often this results in leaves dangling from stems by thread-like hyphae. As temperatures cool during fall months, the fungus stops growing and the affected leaves drop from the plant. The fallen leaf litter within containers and on gravel or polypropylene-covered nursery pads can serve as a source of inoculum for infections the following year. Small-diameter stems can be killed by Rhizoctonia infection resulting in stem dieback.

Fungicides labeled for Rhizoctonia diseases are generally the primary means of prevention in areas where the disease is endemic (Table 5.6). *Rhizoctonia solani* may also be found in zonate leaf spots on *Rhaphiolepis* leaves. This is similar to leaf spots found on other crops called target spot, zonate leaf spot or bull's eye leaf spot. Fungicides labeled for leaf spot diseases of ornamentals may be used for prevention of this disease.

Figure 5.23 Web blight infection by *Rhizoctonia solani* on container grown *Rhaphiolepis* plants, blighted leaves in interior of canopy have senesced and fallen to container surface and surface of production area.

Table 5.6 Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Rhaphiolepis* spp.[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Entomosporium Leaf Spot	Fire Blight	Rhizoctonia	Anthracnose
1: MBC Benzimidazoles: Upwardly systemic. Broad spectrum fungicide for various fungi. Fungicide resistance risk high. Tank mix with fungicides from a different fungicide group (FRAC) to prevent or delay resistance development. Do not mix with copper.				
thiophanate methyl: Cleary's 3336 F, EG; Allban 50 WSB; OHP 6672 50WP, 4.5F			x	x
1 + 2: MBC Benzimidazoles + Dicarboximides: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.				
thiphanate methyl + iprodione: 26/36[W]			x	x
1 + 3: MBC Benzimidazoles + DMI or SI Triazoles: Systemic. Long protection during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.				
thiophanate methyl + propiconazole: Protocol[W]			x	
1 + 14: MBC Benzimidazoles + Aromatic Hydrocarbons: Systemic. Effective against water molds, or oomycetes. Long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.				
thiophanate methyl + etridiazole: Banrot 8G, 40WP[W]			x	
1 + M3: MBC Benzimidazoles + Multi-site inhibitor: Systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Do not mix with copper.				
thiophanate methyl + chlorothalonil: Spectro 90 WDG, Zyban[W]	x		x	x
2: Dicarboximides: Locally systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.				
iprodione: Chipco 26019 FLO, N/G; 26GT; Iprodione Pro 2SE; Raven[V]			x	
3: DMI or SI Triazoles: This group was formerly known as De-Methylation Inhibitors (DMI) and are now classified as Sterol Biosynthesis Inhibitors (SBI or SI). Upwardly systemic. Rain-fast in 2 hours. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.				
metconazole: Tourney[W]			x	x
myclobutanil: Eagle, 20W; Siskin	x			x
propiconazole: Banner Maxx, Banner Maxx II, Strider	x		x	
tebuconazole: Torque			x	x
triadimefon: Bayleton 50, Flo; Strike 50 WDG[V]	x		x	
triflumizole: Terraguard SC, SC/LS			x	
triticonazole: Trinity , Trinity TR	x			

Table 5.6 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Rhaphiolepis* spp.[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Entomosporium Leaf Spot	Fireblight	Rhizoctonia	Anthracnose
3 + 11: DMI or SI Triazoles + Quinone outside Inhibitors (QoI): Rain-fast in 2 hours and locally systemic. Some curative activity. There is wide variation in activity within this group. Effective on powdery mildew. Not effective on downy mildew. Medium risk for resistance.				
trifloxystrobin + triadimefon: Trigo				x
4: Phenylamides: Systemic. Effective against diseases caused by oomycetes, or water molds, including damping-off, root and stem rots, and foliar diseases. Use as soil drench or foliar application.				
mefenoxam: Subdue Maxx, Subdue GR			x	
4 + 12: Phenylamides + Phenylpyrroles/Osmotic signal transducers: Systemic. Broad spectrum drench-applied fungicide.				
mefenoxam + fludioxonil: Hurricane, Hurricane WDG[U,V,W]			x	
7: Carboxamides/Succinate Dehydrogenase Inhibitors (SDHI): Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Spray apply for leaf disorders.				
utolanil:Prostar 70WP			x	
7 + 11: Carboxamides/Succinate Dehydrogenase Inhibitors (SDHI) + QoI Q: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom. Spray apply for leaf disorders, soil drench for phytophthora.				
pyraclostrobin + boscalid: Orkestra Intrinsic, Mural, Pageant, Pageant Intrinsic[W]			x	x
9 + 12: Anilino-Pyrimidines/Methionine biosynthesis inhibitors + Phenylpyrroles: Upwardly systemic, long protection period during wet conditions. Broad spectrum fungicide for greenhouse and nursery use. Medium to high risk for resistance. Toxic to honey bees; do not apply during bloom.				
cyprodinil + fludioxonil: Palladium[W]			x	x
11: Quinone outside Inhibitors (QoI): These fungicides are also known as strobilurins. Locally systemic. Effective on mildews, foliar pathogens, and most fungi. Some control of oomycetes, or water molds. Fungicide resistance risk high. Apply as a soil drench for phytophthora root rot.				
azoxystrobin: Heritage; Palladium; Disarm O	x		x	x
azoxystrobin: Strobe 50WG			x	x
kresoxim methyl: Cygnus				x
pyraclostrobin: Insignia; Insignia SC			x	x
pyraclostrobin: Empress Intrinsic			x	
trifloxystrobin: Compass; Compass O 50 EDG[V]			x	x
12: Phenylpyrroles/Osmotic signal transducers: Non-systemic but good residual protection. Broad spectrum fungicide, not effective against oomycetes, or water molds.				
fludioxonil: Emblem, Medallion; Medallion WDG			x	

Table 5.6 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Rhaphiolepis* spp.[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Entomosporium Leaf Spot	Fireblight	Rhizoctonia	Anthracnose
14: Aromatic Hydrocarbons: Locally systemic. Effective against water molds, or oomycetes.				
etridiazole: Terrazole 35% WP, CA, L; Truban 25EC, 30WP[V]			x	
PCNB: Terraclor WP, 400[W]			x	
triflumizole: Terraguard SC, 50W, SC/LC[V]			x	
19: Octopamine Receptor Agonists: Locally systemic. For use in outdoor and greenhouse nursery crops.				
polyoxin D: Affirm, Veranda O, Endorse			x	x
21: Quinone inside Inhibitors: Locally systemic. Effective against water molds, or oomycetes. Resistance risk unknown but presumed to be medium to high. Apply as a soil drench for phytophthora root rot.				
cyazofamid: Segway, Segway O, Segway SC			x	
25: Glucopyranosyl Antibiotics: Bacterial protectant. Broad spectrum bacterial control.				
streptomycin sulfate: Agri-Mycin 17		x		
28: Carbamates: Locally systemic. Control of oomycetes, or water molds. Not for use in landscapes.				
propamocarb: Banol[V]			x	
33: Phosphonates: Fully systemic; when applied to leaves, product can translocate to lower parts. Effective against water molds, or oomycetes, such as Phytophthora, Pythium, and downy mildew pathogens. Low risk for fungicide resistance development. Apply as a soil drench for phytophthora root rot.				
phosphorous acid: K-Phite T/O, 7LP				x
fosetyl-Al: Aliette WDG		x		
44: *Bacillus subtilis*: Microbial disruptor of cellular membranes. Spray apply for leaf disorders, soil drench for phytophthora.				
Bacillus subtilis: Cease	x		x	x
M: Chemicals with multi-site activity: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.				
(M1) copper hydroxide: Champ DP, Champ Formula 2		x		x
(M1) copper hydroxide: Kocide 2000		x		x
(M1) copper hydroxide: CuPro 5000, T/N/O; Nu-Cop 50DF, HB, 3L		x		x
(M1) copper octanoate: Camelot O	x	x	x	x
(M1) copper sulfate: Basicop		x		x
(M1 + M3) copper hydroxide + mancozeb: Junction	x	x		x

Table 5.6 (*continued*) Labeled fungicidal activity arranged by Fungicide Resistance Action Committee (FRAC) codes[Z] to facilitate development of a fungicide rotation plan for managing key plant pathogens of *Rhaphiolepis* spp.[Y].

FRAC code[Z]: Class description (Action and management notes) Active ingredient(s): Brand name(s)	Entomosporium Leaf Spot	Fireblight	Rhizoctonia	Anthracnose
M: Chemicals with multi-site activity: No systemic activity. Effective as protectants on broad spectrum including most fungi and mildews. Fungicide resistance risk low.				
(M3) mancozeb: Mancozeb 4 F, Flowable with Zinc			x	x
(M3) mancozeb: Pentathlon LF				x
(M3) mancozeb: Protect DF				x
(M4) captan: Captan, 50WP, W, 4L, 80WDG			x	
(M5) chlorothalonil: Daconil Ultrex, Zn Flowable, Weather Stik [T]	x		x	x
(M5) chlorothalonil: Manicure 6FL, Manicure Ultra; Thalonil 90 DF, Thalonil 6L				x
M3 + 3: Multi site inhibitor + Sterol Biosynthesis Inhibitor: Effective as protectants (and upwardly systemic) on broad spectrum including most fungi and mildews. Fungicide resistance risk low.				
mancozeb: + myclobutanil: Clevis[W]	x			x
M5 + 3: Multi site inhibitor + Sterol Biosynthesis Inhibitor: Effective as protectants (and upwardly systemic) on broad spectrum including most fungi and mildews. Fungicide resistance risk low.				
propiconazole + chlorothalonil: Concert II	x			x
NC: Not a Classified substance: Contact fungicide for greenhouse and nursery use. Low risk for resistance.				
potassium bicarbonate: MilStop				x

[Z] (FRAC, 2017).
[Y] Check current products for labeled pesticides, sites for control and plant safety and efficacy on fungal species. This table reports information on fungicide labels and does not necessarily reflect product efficacy. Refer to fungicide labels for rates and usage, specific host information, possible phytotoxicity, re-entry intervals and resistance management. Within columns, products indicated by "x" are labeled for use against the listed pathogen type. Always test product on a sample (small number) of plants before treating an entire crop, as some fungicides listed are for control of the disorder but not specifically labeled for use in *Rhaphiolepis* spp.
[X] Including the causal agent of sudden oak death, *Phytophthora ramorum*.
[W] Chemical contains more than one active ingredient, thus product may be listed within more than one FRAC code designation (FRAC, 2017).
[V] Not for use in residential landscapes; commercial use only.
[U] Only use as a suppressant, does not grant preventative control.
[T] Do not apply with mist blowers or high pressure spray equipment in greenhouses.

References

Adkins, C., G. Armel, M. Chappell, J.H. Chong, S. Frank, A. Fulcher, F. Hale, W. Klingeman III, K. Ivors, A. LeBude, J. Neal, A. Senesac, S.A. White, J. Williams-Woodward, and A. Windham. 2010. Pest Management Strategic Plan for Container and Field-Produced Nursery Crops in Georgia, Kentucky, North Carolina, South Carolina and Tennessee. A. Fulcher (ed.). Southern Region IPM Center. Accessed April 20, 2011 <http://www.ipmcenters.org/pmsp/pdf/GA-KY-NC-SC-TNnurserycropsPMSP.pdf>.

Ansari, M.A. and T.M. Butt. 2011. Effect of potting media on the efficacy and dispersal of entomopathogenic nematodes for control of black vine weevil, *Otiorhynchus sulcatus* (Coleoptera: Curculionidae). Biological Control 58:310-318.

Arthurs, S.P., C.L. McKenzie, J.J. Chen, M. Dogramaci, M. Brennan, K. Houben and L. Osborne. 2009. Evaluation of *Neoseiuslus cucumeris* and *Amblyseius swirskii* (Acari: Phytoseiidae) as biological control agents of chilli thrips, *Scirtothrips dorsalis* (Thysanoptera: Thripidae), on pepper. Biological Control 49:91-96.

Beeson, Jr., R.C. 2006. Relationship of plant growth and actual evapotranspiration to irrigation frequency based on management allowed deficits for container nursery stock. J. American Soc. Hort. Sci. 131:140-148.

Bethke, J., J. Chamberlin, R. Cloyd, J, Dobbs, M. Faver, D. Gilrein, K. Heinz, R. Lindquist, S. Ludwig, C. McKenzie, G. Murphy, R. Oetting, L. Osborne, C. Palmer, M. Parrella, N. Rechcigl and R. Yates. 2011. Thrips Management Program for Ornamental Horticulture. Accessed April 22, 2015 <http://mrec.ifas.ufl.edu/lso/DOCUMENTS/ThripsManagementProgram-February%202011-FINAL.pdf>.

Blythe, E.K., J.L. Sibley, and J.M. Ruter. 2005. Cutting propagation with auxin applied via the rooting substrate. HortScience 40:884.

Brown, L.R., and C.O. Eads. 1965. A technical study of insects affecting the oak tree in southern California. Bulletin 810. University of California, California Agricultural Experiment Station, Riverside, CA. 106 pp.

Bryson, G.M., H.A. Mills, D.N. Sasseville, J.B. Jones, Jr., and A.V. Barker. 2014. Plant Analysis Handbook III. Micro-Macro Publishing, Inc., Athens. GA.

Chadwick, C.E. 1965. A review of the Fuller's rose weevil (*Pantomorus cervinus* Boheman) (Coleoptera, Curculionidae). J. Entomol. Soc. Australia (N.S.W.) 2:10-20.

Chappell, M. 2015. Crop pruning and production timeline. Personal Communication.

Chong, J-H. 2015. Scale insect emergence and life cycle in the southeast. Personal Communication.

Colbaugh, P., A. Hagan, J. Walker, and L. Barnes. 2001. Diseases of Woody Ornamentals and Trees in Nurseries. Jones, R.K. and D.M. Benson, eds. APS Press. St. Paul, MN.

Corley, W.L., and O.M. Lindstrom. 1995. 'Georgia Petite' and 'Georgia Charm' Rhaphiolepis. J. Env. Hort. 14:204.

Cowles, R.S. 2001. Protecting container-grown crops from black vine weevil larvae with bifenthrin. J. Env. Hort. 19:184-189.

Dirr, M. A. 2009. Manual of Woody Landscape Plants: Their Identification, Ornamental Characteristics, Culture, Propagation and Uses. 6th ed. Stipes Publishing L.L.C. Champaign, IL.

Dirr, M.A. and C.W. Heuser, Jr. 1989. The Reference Manual of Woody Plant Propagation: From Seed to Tissue Culture. Varsity Press, Inc. Athens, GA.

Dogramaci, M., S.P. Arthurs, J.J. Chen, C. McKenzie, F. Irrizary and L. Osborne. 2011. Management of chilli thrips, *Scirtothrip dorsalis* (Thysanoptera: Thripidae), on peppers by *Amblysieus swirskii* (Acari: Phytoseiidae) and *Orius insidiosus* (Hemiptera: Anthocoridae). Biological Control 59:340-347.

Dreistadt, S.H., J.K. Clark, and M.L. Flint. 2004. Pests of Landscape Trees and Shrubs: An Integrated Pest Management Guide. University of California, Agricultural and Natural Resources, Publication 3359. 2nd Edition. Oakland, CA.

Edwards, G.B., G. Hodges, and W. Dixon. 2005. Chilli thrips *Scirtothrips dorsalis* Hood (Thysanoptera: Thripidae): a new pest thrips for Florida. Florida Department of Agriculture & Consumer Services, Division of Plant Industry. Accessed Accessed July 29, 2015 <http://www.freshfromflorida.com/pi/pest-alerts/scirtothrips-dorsalis.html>.

Epsky, N.D., A. Walker, and P.E. Kendra. 2009. Samplign methods for *Myllocerus undecimpustulatus undatus* (Coleoptera: Curculionidae) adults. FL Entomologist 92:388-390.

Farris, M.E., G.J. Keever, J.R. Kessler, and J.W. Olive. 2010. Effects of cyclanilide and benzyladenine tank mixes on shoot production and growth of three woody landscape species during production. J. Env. Hort. 28:91-95.

Fisher, J.R. 2006. Fecundity, longevity and establishment of *Otiorhynchus sulcatus* (F.) and *Otiorhynchus ovatus* (L.) (ColeopteraL Curculionidae) from the Pacific North-west of the United States of America on selected host plants. Ag. Forest Entomol. 8:281-287.

Gordh, G. 1979. Family Encyrtidae. In Catalog of Hymenoptera in America North of Mexico, K. V. Krombein, P. D. Hurd, D. R. Smith and B. D. Burk (eds.), pp. 903. Washington, DC.

Hagan, A.K., J.R. Akridge , K.L. Bowen, J.W. Olive, and K.M. Tilt. 2001. Resistance of selected cultivars of Indian hawthorn to entomosporium leaf spot, fire blight and anthracnose. J. Env. Hort. 19:43-46.

Hagan, A.K., J.R. Akridge, K.L. Bowen, J.W. Olive, K.M. Tilt. 2002. Disease resistance of selected cultivars of Indian hawthorn in Alabama. AL Exp. Station. Bulletin 648.

Hagan, A.K., J.R. Akridge, and M.E. Rivas-Davila. 2003. Impact of fungicide inputs on the severity of entomosporium leaf spot on selections of dwarf Indian hawthorn. J. Env. Hort. 21:16-19.

Herms, D.A. 2004. Using degree-days and plant phenology to predict pest activity. In: IPM of Midwest Landscapes, pp. 49-59. V. Krischik and J. Davidson, eds. MN Ag. Exp. Station Publication SB-07645.

Hodges, A., S. Ludwig, L. Osborne, and G.B. Edwards. 2009. Pest Thrips of the United States: Field Identification Guide. Accessed July 29, 2013 <http://www.ncipmc.org/alerts/chili_thrips_deck.pdf>.

Holcomb, G.E. 1998. First report of fire blight on indian hawthorn cultivar Olivia in Louisiana. Plant Disease. 82:1402.

Holtz, T. 2006. NPAG Report: *Scirtothrips dorsalis* Hood. New pest advisory group, center for plant health science and technology, USDA-APHIS, Raleigh, NC. 7 pp.

Klingaman, Gerald. 2012. Plant of the Week: Indian hawthorn. Extension News: Univ. AR Ext. Service. October 5, 2012.

Klingeman, W.E., S.A. White, A.V. LeBude, A. Fulcher, N.W. Gauthier, and F. Hale. 2014. A review of arthropod pests, plant diseases and abiotic disorders and their management on viburnum species in the Southeastern U.S. J. Env. Hort. 32:84-102.

Kosztarab, M. 1996. Scale Insects of Northeastern North America – Identification, Biology, and Distribution. Special Publication No. 3. VA Museum Nat. History, Martinsville, VA. 650 pp.

Lakin, K.R. and J.G. Morse. 1989. A degree-day model for Fuller's rose beetle, *Pantomorus cervinus* (Boheman) (Col., Curculionidae) egg hatch. J. Applied Entomol. 107:102-106.

Masiuk, M. 2003. Black vine weevil fact sheet. Penn State Cooperative Extension. Accessed July 22, 2016 <http://woodypests.cas.psu.edu/factsheets/InsectFactSheets/html/Black_Vine_Weevil.html>.

Momol, M.T., E.A. Momol and W. Dankers. 2000. A severe outbreak of fire blight in woody ornamental *Rosaceae* plants in North Florida and South Georgia. Plant Disease. 84:1153.

Myamoto, S., I. Martinez, M. Padilla, and A. Portillo. 2004. Landscape Plant Lists for Salt Tolerance Assessment. Texas A&M Univ. Res. Center and El Paso Water Utilities.

Neal, A. 2013. Sri Lanka weevil. Publication number EENY-579. UF, Institute Food Ag. Sci., Gainesville, FL. Accessed July 23, 2016 <http://entnemdept.ufl.edu/creatures/orn/sri_lankan_weevil.htm>.

Nelson, G.H., G.C. Walters, R.D. Haines, and C.L. Bellamy. 2008. A Catalog and Bibliography of the Buprestoidea in America North of Mexico. The Coleopterists Society, Special Publication No. 4. The Coleopterists Society, North Potomac, MD. 274 pp.

Norcini, J.G. and J.H. Aldrich. 1992. Phytotoxicity of bentazon to woody and herbaceous landscape plants. HortScience 27:1196-1198.

Oates, J.M., G.J. Keever, and J.R. Kessler, Jr. 2004. BA-induced shoot formation in Indian hawthorn. J. Env. Hort. 22:71-74.

Oates, J.M., G.J. Keever, and J.R. Kessler, Jr. 2005a. BA application frequency and concentration effects on two Indian hawthorn cultivars. J J. Env. Hort. 23:37-41.

Oates, J.M., G.J. Keever, and J.R. Kessler, Jr. 2005b. BA developmental stage influences plant response to benzyladenine. J. Env. Hort. 23:149-152.

O'Brien, C.W., M. Haseeb, and M.C. Thomas. 2006. *Myllocerus undecimpustulatus undatus* Marshall (Coleoptera: Curculionidae), a recently discovered pest weevil from the Indian subcontinent. Entomology Circular No. 412. FL Dept. Ag.Consumer Serv., Gainesville, FL.

Olive, J. 2015. Occurrence of rhizoctonia on *Rhaphiolepis* spp. Personal Communication.

Reding, M.E. 2008. Black vine weevil (Coleoptera: Curculionidae) performance in container- and field-grown hosts. J. Entomol. Sci. 43:300-310.

Reding, M.E. and A.B. Persad. 2009. Systemic insecticides for control of black vine weevil (Coleoptera: Curculionidae) in container- and field grown- nursery crops. J. Econ. Enotmol. 102:927-933.

Reding, M.E. and C.M. Ranger. 2011. Systemic insecticides reduce feeding, survival, and fecundity of adult black vine weevils (Coleoptera: Curculionidae) on a variety of ornamental nursery crops. J. Econ. Enotmol. 104:405-413.

Relf, D. and B. Appleton. 2009. Managing winter injury to trees and shrubs. VA Tech Ext. Pub. 426-500. Accessed January 21, 2014 <http://pubs.ext.vt.edu/426/426-500/426-500.html>.

Ruter, J.M. 2004. Resistance of rhaphiolepis selections to entomosporium leaf spot. XXVI International Horticultural Congress: Nursery Crops: Development, Evaluation, Production and Use. DOI: 10.17660/ActaHortic.2004.630.3

Seal, D.R., W. Klassen and V. Kumar. 2010. Biological parameters of *Scirtothrips dorsalis* (Thysanoptera: Thripidae) on selected hosts. Env. Entomol. 39:1389-1398.

Sinclair, W.A. and H.H. Lyon. 2005. Diseases of Trees and Shrubs. 2nd ed. Comstock Press.

Smith, F.F. 1932. Biology and control of the black vine weevil. USDA Tech. Bulletin 325.

Solomon, J.D. 1995. Guide to Insect Borers in North American Broadleaf Trees and Shrubs. Agriculture Handbook 706. Washington, DC. USDA, Forest Service. 735 p.

Tillman, A.L., S.L. Warren, and F.A. Blazich. 2008. Optimized nitrogen fertilization for production of containerized *Raphiolepis ×delacourii* 'Snow White.' J. Env. Hort. 26:157-163.

Tsai, J.H. and J. J. Wang. 2001. Effects of host plants on biology and life table parameters of *Aphis spiraecola* (Homoptera: Aphididae). Env. Entomol. 30:44-50.

Valdez-Aguilar, L.A., C.M. Grieve, A. Razak-Mahar, M.E. McGiffen, D.J. Merhaut. 2011. Growth and ion distribution is affected by irrigation with saline water in selected landscape species grown in two consecutive growing seasons: spring-summer and fall-winter. HortScience. 46:632-642.

van Tol, R.W.H.M., D.J. Bruck, F.C. Griepink and W.J. De Kogel. 2012. Field attraction of the vine weevil *Otiorhynchus sulcatus* to kairomones. J. Econ. Enotmol. 105:169-175.

Wang, J.J. and J.H. Tsai. 2000. Effect of temperature on the biology of *Aphis spiraecola* (Homoptera: Aphididae). Annals Entomol. Soc. America 93:874-883.

Western Plant Health Association. 2010. Western Fertilizer Handbook. 9th Edition. Waveland Press, Inc. Long Grove, IL.

Wilen, C.A., U.K. Schuch, C.L. Elmore. 1999. Mulches and subirrigation control weeds in container production. J. Env. Hort. 17:174-180.

Williamson, Z., G.J. Keever, J.R. Kessler, J.W. Olive. 2009. Drench application of cyclanilide promotes shoot devleopment in containerized woody landscape species during nursery production. J. Env. Hort. 27:217-222.

Witcher, A.B., C.W. Robinson, C.H. Coker, D. J.Eakes, S.S. Ditchkoff, and G.B. Fain. 2013. Impact of white-tailed deer (*Odocoileus virginianus* Zimmerman) on ornamental plants in Alabama green industries and home landscapes. J. Env. Hort. 31:14-20.

Woodruff, R.E. and R.C. Bullock. 1979. Fuller's rose weevil, *Pantomorus cervinus* (Boheman), in Florida (Coleoptera: Curculionidae). Div. Plant Ind. Entomol. Circular No. 207. Accessed July 26, 2013 <http://www.freshfromflorida.com/pi/enpp/ento/entcirc/ent207.pdf. [Accessed 26 July 2013].

Wu, L. and L. Dodge. 2005. Landscape Plant Salt Tolerance Selection Guide for Recycled Water Irrigation. A Special Report for the Elvenia J. Slosson Endowment Fund. Univ. CA Ag. Natural Res.

Yardeni, A., and D. Rosen. 1990. Wind dispersal and pattern of colonization of *Ceroplastes floridensis* on citrus in Israel. Proceedings of the Sixth International Symposium on Scale Insect Studies, pp. 125-128.

CHAPTER 6

Weed Management in Shrub Production

Joseph Neal, North Carolina State University

Jeff Derr, Virginia Tech

Chris Marble, University of Florida

Weed Management in Production Systems

WEED MANAGEMENT IN PRODUCTION SYSTEMS

1. Common Weed Species Grouped by Their Life Cycles
2. Weed Management in Container Production
3. Weed and Ground Cover Management in Field Nursery Crop Production
4. Preemergence & Postemergence Herbicide Use
5. Alternatives to Herbicides in Field Production

Weed Management in Ilex, Loropetalum, Raphiolepis and Rhododendron Production

Weed management strategies and options in shrub production depend upon the production system, the species being grown, and the predominant weed species at the site. A different set of weed species typically infests container production compared to field production. In containers, growers are primarily faced with summer and winter annual weeds, especially annual broadleaf weeds. Only a few perennial weed species are major production problems in containers. In field production, growers are faced with a wide range of annual, biennial, and perennial weed species. Furthermore, herbicide and cultural weed management practices will differ between container and field production sites. Therefore, the two production systems are discussed separately. Herbicide registrations can differ between field nursery production and landscape maintenance, thus information on landscape uses for several herbicides is included as well. Species specific information are provided for holly, loropetalum, rhododendron/azalea, and Indian hawthorn in separate sections at the end of this chapter. Due to more detailed application and product information, specific information for weed control in hydrangea production can be found in Chapter 1.

Every pest management plan begins with an understanding of the pest species, populations and life cycles. Weed species can be grouped by their life cycle into annuals, biennials, and perennials. Annuals live for only one growing season and reproduce strictly by seed. These weed species can be divided into summer annuals and winter annuals.

Summer annuals, which start to germinate in March, April or May, depending on species and location, and may continue to germinate though the summer, will flower in summer or early fall and generally die with the first killing frost. Summer annuals can be divided into broadleaves, grasses, and sedges. Winter annuals start to germinate in early fall (generally September), and continue to germinate into October and November, depending on species and location, and there may be additional germination in late winter to early spring (February to April). Winter annuals generally flower in spring, and then die off with the onset of hot, dry weather. Winter annual weeds are divided into broadleaves and grasses. Sedges are warm-season plants that do not grow during the winter.

Summer annuals are of greater concern to producers than winter annuals because they compete for water and nutrients with actively-growing trees and shrubs during the growing season. Winter annuals generally do not pose this competition concern while nursery crops are dormant. Winter annuals may actually be beneficial in field production for reducing soil erosion during the winter months. Winter annual weed species that grow tall and thus interfere with crop growth after budbreak, like horseweed, or species that climb up trees, like narrowleaf vetch, however, need to be controlled

Biennials live for two years and also reproduce strictly by seed. They form a rosette during the first year and then flower in the second year. The biennial weeds of concern are all broadleaves.

Perennials live for many years and generally are the hardest species to control. There are two types of perennials – simple and creeping. Simple perennials, like common dandelion, spread only by seed. Creeping perennials spread by seed and also vegetatively through rhizomes, stolons, bulbs, tubers or roots. It is more difficult to eradicate creeping perennials than simple perennials. Some perennials die back to the ground in winter and regrow in spring from rhizomes, tubers or other underground structures, such as yellow nutsedge. Others, like quackgrass, are green throughout the year. Perennial species can also be classified as herbaceous (lacking a woody stem), such as common dandelion, or woody, like poison ivy. Perennials can be divided taxonomically into broadleaves, grasses, sedges, and other monocots. Field horsetail, in a separate taxonomic group, is a non-flowering primitive plant that spreads by spores or vegetatively by rhizomes.

Common Weed Species, Grouped by Their Life Cycles

<u>Summer Annuals</u>

Summer annual grasses – barnyardgrass (*Echinochloa crus-galli*), large crabgrass (*Digitaria sanguinalis,* Figure 6.1*),* goosegrass (*Eleusine indica s,* Figure 6.2), giant foxtail (*Setaria* spp.), fall panicum (*Panicum dichotomiflorum*), witchgrass (*Panicum capillare*)

Figure 6.1 Large crabgrass seedling (A) and growth habit (B). Joseph Neal, NCSU

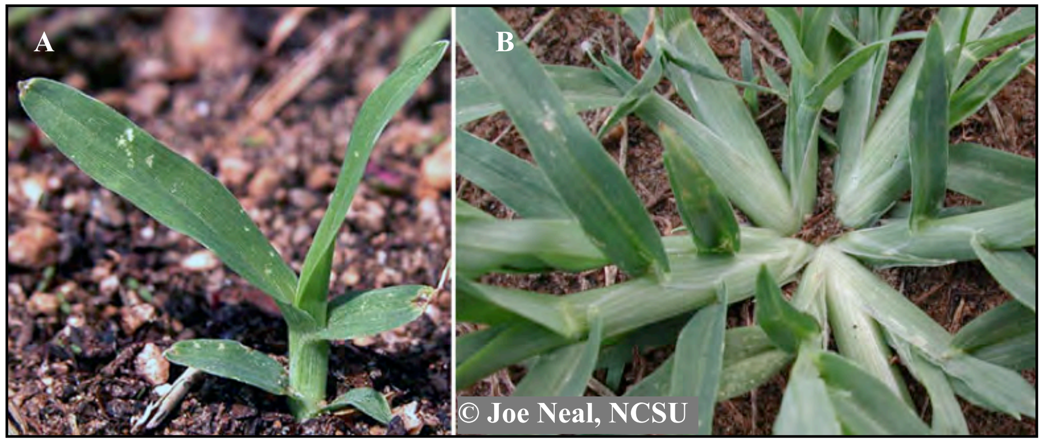


Figure 6.2 Goosegrass seedling (A) and rosette habit (B).

Summer annual broadleaves – wild buckwheat (*Polygonum convolvulus*), cocklebur (*Xanthium strumarium*), hairy galinsoga (*Galinsoga quadriradiata*), jimsonweed (*Datura stramonium)*, ladysthumb (*Polygonum persicaria*), common lambsquarters (*Chenopodium album*), morningglory (*Ipomoea* spp. - several species, Figure 6.3), pigweed (*Amaranthus* spp - several species), purslane (*Portulaca oleracea*), common ragweed (*Ambrosia artemesiifolia*), Pennsylvania smartweed (*Polygonum pennsylvanicum*)

Figure 6.3 Tall morningglory leaves and flowers.

<u>Winter Annuals</u>

Winter annual grasses – downy brome (*Bromus tectorum*), annual bluegrass (*Poa annua*, Figure 6.4), cheat (*Bromus secalinus*), Italian (annual) ryegrass (*Lolium multiflorum*)

Figure 6.4 Annual bluegrass.

Winter annual broadleaves – catchweed bedstraw (*Galium aparine*), bittercress (*Cardamine* spp., Figure 6.5), common chickweed (*Stellaria media)*, redstem filaree (*Erodium cicutarium*), common groundsel (*Senecio vulgaris*), henbit (*Lamium amplexicaule*), horseweed (*Conyza canadensis*), prickly lettuce (*Lactuca serriola*), pennycress (*Thlapsi arvensis*), sowthistle (*Sonchus* spp.) Virginia pepperweed (*Lepidium virginicum*), shepherd's-purse (*Capsella bursa-pastoris*), vetch (*Vicia* spp.)

Biennials

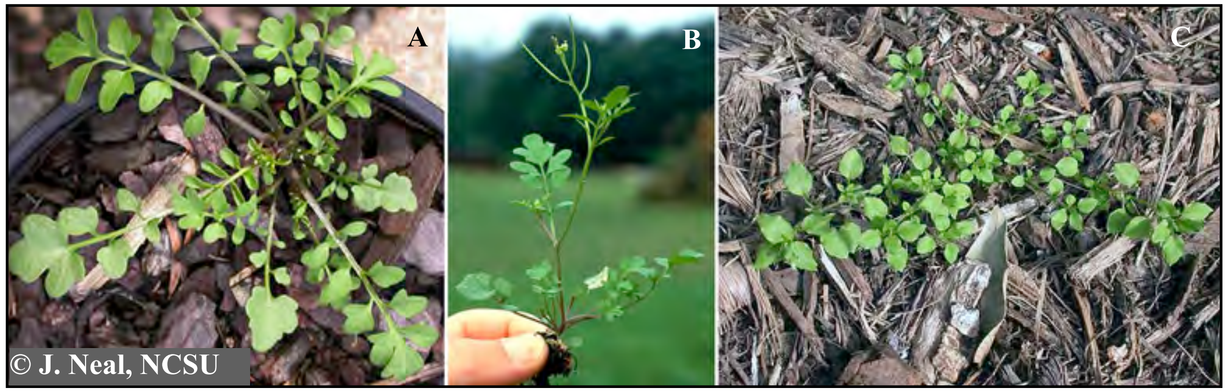


Figure 6.5 Bittercress young rosette (A) and seedhead (B), and common chickweed habit (C)

burdock (*Arctium minus*), wild carrot (*Daucus carota*, Figure 6.6), common eveningprimrose (*Oenothera biennis*), yellow goatsbeard (*Tragopogon dubius*), common mullein (*Verbascum thapsis*), plumeless thistle (*Carduus acanthoides*)

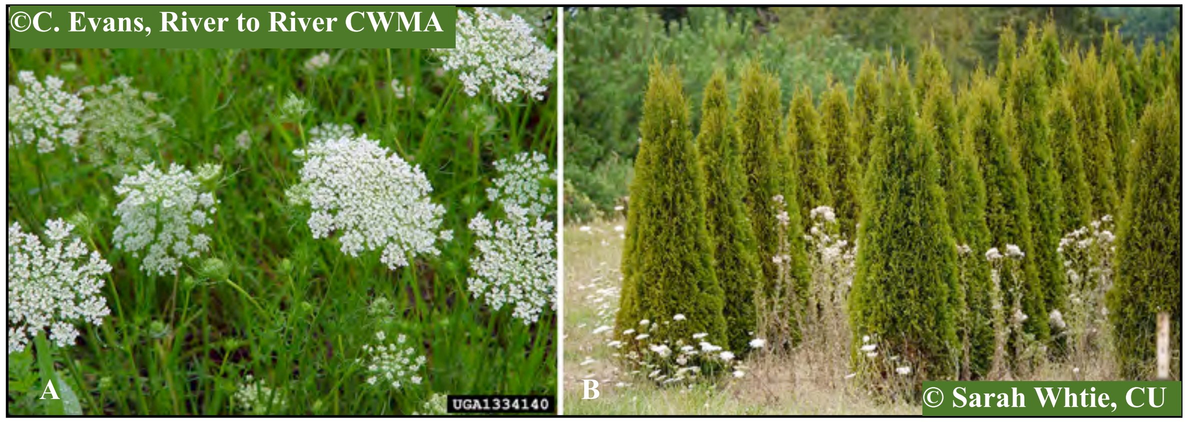


Figure 6.6 Closeup of wild carrot (Queen Anne's lace) flower (A; Chris Evans, River to River CWMA, Bugwood.org) and established infestation in the field (B; Sarah White, CU).

Perennials

Perennial grasses

Clump-type perennial grasses – tall fescue (*Lolium arundinaceum*), orchardgrass (*Dactylis glomerata*, Figure 6.7)

Creeping perennial grasses – bermudagrass (*Cynodon dactylon*), johnsongrass

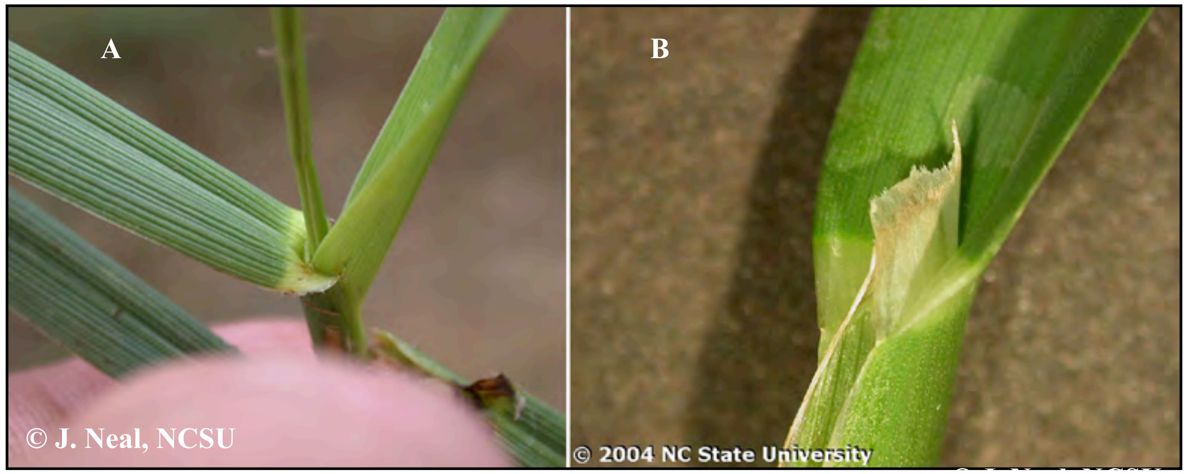


Figure 6.7 Closeup of collar of tall fescue (A) and the ligule of orchardgrass (B).

(*Sorghum halapense*, Figure 6.8), wirestem muhly (*Muhlenbergia frondosa*), quackgrass (*Elymus repens*), timothy (*Phleum pratense*)

Figure 6.8 Johsongrass leaf blade (A) with characteristic white mid-vein, leaf blade attachment to stem (B) and reddish-purple stem base attachment to root system (C).

Perennial sedges – yellow nutsedge (*Cyperus esculentus*, Figure 6.9), purple nutsedge (*Cyperus rotundus*), green kyllinga (*Kyllinga brevifolia*)

Figure 6.9 Yellow nutsedge young plant and seedhead.

Other monocots – wild garlic and wild onion (*Allium* spp.), dayflower (*Commelina* spp.), doveweed (*Murdannia nudiflorum*, Figure 6.10)

Figure 6.10 Doveweed (*Murdannia nudiflorum*) clasping leaf emergence (A) and spreading growth habit (B).

Spore producers – field horsetail (*Equisetum arvense*), liverwort (*Marchantia polymorpha*)

Perennial broadleaves

Simple broadleaf perennials – chicory (*Cichorium intybus*), broadleaf dock (*Rumex obtusifolius*), curly dock (*Rumex crispus*), dandelion (*Taraxacum officinale*), broadleaf plantain (*Plantago major*), buckhorn plantain (*Plantago lanceolata*), pokeweed (*Phytolacca americana*)

Creeping broadleaf perennials – wild blackberry (Rubus spp.), field bindweed (*Convolvulus arvensis*), hedge bindweed (*Calystegia sepium*), Virginia-creeper (*Parthenocissus quinquefolia*), hemp dogbane (*Apocynum cannabinum*), goldenrod (*Solidago* spp.), Japanese honeysuckle (*Lonicera japonica*), horsenettle (*Solanum carolinense*), poison ivy (*Toxicodendron radicans*), common milkweed (*Asclepias syriaca*), mugwort (*Artemesia vulgaris*), red sorrel (*Rumex acetosella*), Canada thistle (*Cirsium arvense*), trumpetcreeper (*Campsis radicans*), woodsorrel (*Oxalis* spp.)

Figure 6.11 Weed management in and around production areas is critical. To lessen weed pressure, actively scout for and manage weeds not only in production beds but also around borders.

Weed Management in Container Production

Figure 6.12 Many nursery weeds, such as bittercress pictured here in loropetalum liners, have mechanisms for self-dispersal. Control weeds in propagation areas to prevent spread and remove emerged weeds before they can go to seed.

Production of shrubs such as azalea, loropetalum (Figure 6.11), hydrangea, and Indian hawthorn is primarily (though not exclusively) in containers. Preventing weed introduction and spread is an important part of any weed management program, but especially in containers. Weed seed and other propagules are introduced into nurseries in the potting substrates, by wind-blown seed, washed in by rain runoff, deposited by birds, and (most importantly) in the root balls of propagated plant materials (Figure 6.12). Consequently, an effective weed management plan starts in propagation.

Weed management in propagation areas

Flexuous bittercress, crabgrass, woodsorrel species (oxalis), mulberryweed, and common groundsel are common weeds in propagation houses. No preemergence herbicides are registered for use during rooting of cuttings. While certain herbicides may be used on liners after they are well rooted, root inhibiting products, such as the dinitroanilines (oryzalin, pendimethalin, prodiamine, trifluralin), indaziflam, and dimethenamid-p, should only be used on plants with an established root system. Since no preemergence herbicides are available for use in containers in greenhouses or similar covered structures, and very limited postemergence herbicide options exist, one must rely primarily on good sanitation practices to limit infestations of weeds during propagation in a covered greenhouse or poly-house (Figure 6.13).

Figure 6.13 Good sanitation practices in propagation houses will limit weed issues. In this picture, note that the floor is covered with a weed barrier.

Start with new containers, trays and flats. If reusing flats or containers, wash them to remove any leftover substrate and weed seeds. Commercially available substrates are generally weed free, but if you determine a load of substrate to be contaminated, steam sterilization (180ºF for at least 30 minutes) will kill most, but not all, weed seeds.

Hand weed flats as weeds emerge to prevent spread. Remember, many common nursery weeds have mechanisms for self-dispersal; oxalis and bittercress can "throw" their seeds many feet. Additionally, many weeds can flower very quickly after emergence. If weeds are allowed to establish and flower, weed populations can increase and spread rapidly. Consequently, one must remove weeds from containers frequently to prevent plants from going to seed. Control weeds on greenhouse floors as well as areas outside the greenhouse to prevent weeds from spreading to the crop. This is particularly important in propagation facilities, as weeds spreading into liner trays will ultimately infest outdoor production areas. Several nonselective postemergence herbicides are labeled for use on greenhouse floors including diquat, glufosinate, glyphosate, and pelargonic acid. It is important to note which products are labeled for use while crops are present. Clethodim can be used for controlling grasses such as crabgrass and annual bluegrass. Indaziflam is the only preemergence herbicide labeled for use on greenhouse floors, but it should not be applied in greenhouses while crops are present, nor should it be applied to crops in covered houses.

Well rooted cuttings, or "liners", are often moved from closed propagation houses to shade houses. It is then possible to use some preemergence herbicides. Residual herbicides that inhibit root growth are to be avoided. A more extensive discussion of weed control in greenhouses and similar covered structures, including a full list of labeled herbicides is available in Horticulture Information Leaflet 570 available on-line at: https://content.ces.ncsu.edu/greenhouse-weed-control.

Common weeds in container production

The major weeds infesting container production include the summer annual broadleaf weeds: spotted spurge, longstalked phyllanthus, eclipta, and mulberry weed; cool season broadleaf weeds such as horseweed (marestail), chickweed, groundsel, sowthistle and flexuous bittercress; summer annual grasses such as large crabgrass and goosegrass, and the cool-season species annual bluegrass; and several species of annual sedges that grow during the summer. Due to the frequent irrigation in container production, which results in a cooler, wetter environment, cool-season weeds such as flexuous bittercress may persist year-round. Yellow and creeping woodsorrel are perennial weeds but generally the infestation in containers starts from seeds. As such, herbicide effectiveness charts generally list most preemergence herbicides as controlling these species. The control, though, is referring to control at germination and not of established plants. Yellow nutsedge can occasionally be a problem in containers but more often is found growing in the gravel underneath containers. If perennial weeds such as yellow nutsedge or mugwort are common, it is important to determine the source of the infestation and implement sanitation practices to prevent its introduction.

Sanitation

The substrate used in containers needs to be kept weed free. In the Southeastern U.S., the growing mix used in containers is generally composed of 100% pine or hardwood bark, or a combination of bark with a small fraction of sand, peat, or other non-soil components. These soilless components initially will have few to no weeds, if proper sanitation is followed. Ideally, piles of bark, sand, peat, or other components need to be kept on concrete pads until needed (Figure 6.14).

Figure 6.14 Piles of bark, sand, peat, or other substrate components need to be kept on concrete pads until needed to reduce weed seed contamination.

Otherwise, field soil, which would contain weed seed, could be introduced into the soil-less mix. Control weeds that germinate in bark piles, and prevent weeds from flowering in areas near substrate piles.

Use of heavy duty fabrics below containers will reduce weed populations around the containers. Weeds such as doveweed, eclipta, and spotted (prostrate) spurge readily grow in gravel or bare soil areas. Seed from these plants can wash into the drain holes of containers or be splashed onto the surface of the substrate. Therefore, application of a preemergence (plus a postemergence herbicide if weeds are already present) to the gravel areas before containers are placed will reduce the source(s) of weed inoculum. Several herbicides are labeled for this purpose. Common treatments include glyphosate, combined with flumioxazin or prodiamine or indaziflam. If yellow nutsedge has been a problem in the gravel area, s-metolachlor or dimethenamid-p can be added to the application. As an alternative, dichlobenil, sold primarily in granular form as Casoron, may be applied to the gravel for preemergence weed control, including nutsedge suppression, but diclobenil should only be used during the winter.

Figure 6.15 Many weeds have the ability to mature and disperse seeds after they have been removed (pulled). This common groundsel plant was pulled 2 days before this photo was taken. Remove weeds from the site to prevent spread.

Hand weeding

No herbicide will control all weed species. Plan to walk container production areas on a weekly or biweekly basis, removing any emerged weeds. Do not leave weeds on the gravel or fabric as this may introduce weed seeds into crop containers. Certain weeds, like common groundsel, may still be able to disperse seed after plants have been pulled up (Figure 6.15). Remove all weeds from the production areas. Certain perennial weeds, though, such as yellow nutsedge, mugwort or yellow fieldcress, are very difficult to control through hand weeding due to rhizomes or other underground structures remaining after hand weeding. Containers infested with such weeds should be discarded to prevent the infestation from spreading (and to avoid sending such weedy products to your customers).

Other nonchemical control options

Besides hand weeding, there are other options for controlling weeds without using chemicals. Various disks have been made out of organic materials (plant fibers, human hair) or synthetic materials (plastics). Weed seedlings are generally unable to push up through the material. Weed growth can occur around the edges of the disks, or through the slit necessary for getting the disk around the stem of the nursery plant (Figure 6.16). Some of the lighter disks have been prone to being blown out of containers.

Pine bark nuggets, wood chips, rice hulls and other mulch materials can be placed on the container substrate surface to prevent or reduce germination of many weed species by excluding light and providing a physical barrier. Typically, these mulch materials will need to be applied at depths of at least 0.5 to 1 inch in order to be effective. Coarse mulch materials are usually more effective than fine-particle materials. Mulch is more expensive than herbicides, but it can be cost effective on larger plant material, longer-term crops, or on chemically sensitive species in which herbicides cannot be used. It should be noted that mulches are less effective in containers that suffer blow-over, as mulches will be spilled from the container(s) in such cases.

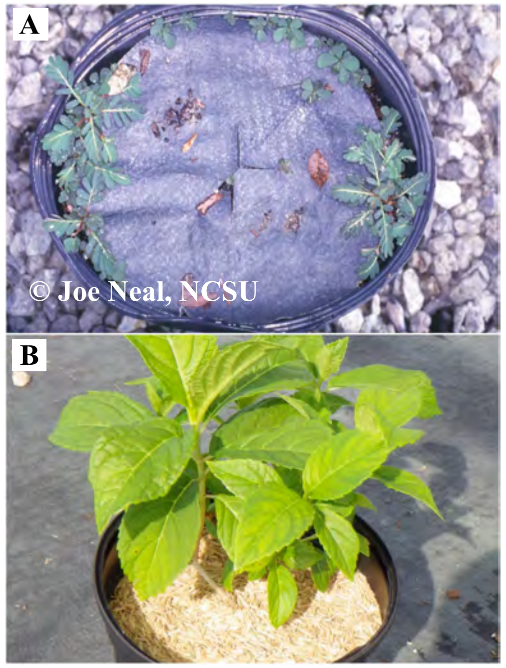


Figure 6.16 Fabric disk impregnated with copper to inhibit weed growth (A) and rice hull mulch (B). Such mulches can be about as effective as preemergence herbicides but weeds can emerge along the edges or through “slits” in the fabric or through organic mulches. Emerged weeds need to be removed when small.

When using mulches for weed control, several cultural practices may need to be adjusted. Mulches help retain moisture in the containers, thus irrigation frequency will need to be adjusted. Having mulched crops in the same irrigation zone with other crops may result in excess irrigation on some and inadequate irrigation on others. Top-dressed fertilizer may be less effective when placed atop mulches. Incorporated slow release fertilizer or dibbled fertilizer will be more effective when mulches are used.

Preemergence herbicide use in container production

After transplanting, apply irrigation to settle the substrate. A preemergence herbicide can then be applied to limit new weed establishment. Certain herbicide labels suggest waiting a week or two to allow root development of the nursery crop prior to herbicide application. Although this is a valid concern since a number of commonly-used nursery herbicides are root inhibitors, waiting two weeks before application will allow for considerable weed germination (Figure 6.17). Since most preemergence herbicides do not control emerged weeds, one is therefore already behind on weed management. It is easier to keep containers free of weeds than it is to clean-up a crop after weeds have become established.

Preemergence herbicides will need to be reapplied to maintain control throughout the growing season. Depending on the situation, three to six applications may be required per year. The reapplication timing will depend on the herbicide used, the application rate, the watering frequency/daily amount, and local environmental conditions. Perhaps more importantly, the need for reapplication will depend on weed populations. Sites with a history of heavy weed infestations will need more frequent herbicide applications than will sites with a history of few weeds.

Figure 6.17 Spotted spurge seedings two weeks after potting. Many weed species can emerge and establish very rapidly if preemergence herbicide applications are delayed.

When selecting herbicides, crop safety is paramount. Many preemergence herbicides are labeled for use in nursery crops (Table 6.1). Herbicide choice will depend on the nursery crop species, its growth stage at application, available formulations, herbicide cost, and major weed species present at the nursery. See Table 6.2 for a summary of herbicides labeled (at the time of publication) for use on azalea, hydrangea, holly, loropetalum, Indian hawthorn and rhododendron.

There are a number of granular combination products on the market that combine a grass herbicide and a broadleaf herbicide. Common products include oxyfluorfen or isoxaben for broadleaf weeds, plus a dinitroaniline herbicide (oryzalin, prodiamine, pendimethalin, trifluralin) for control of annual grasses. These combination products control a wider spectrum of weeds than single active ingredient products. Accurate and uniform application is very important for granular herbicides as it is easy to over- or under-apply the product.

After a granular herbicide application, either run the irrigation or use a leaf blower to remove granules from the foliage of nursery crops. Granular herbicides containing oxyfluorfen, oxadiazon or flumioxazin, (such as Biathlon, BroadStar, Rout, Ronstar, and OH2) can burn tender foliage when granules adhere to foliage. Symptoms of injury range from leaf spotting, to tip necrosis when granules are caught in the growing points. Research has shown that this is especially important in hydrangea production as all granular herbicides tested have the potential to cause tip necrosis if the granules are not removed from the foliage. Also, removing preemergence herbicides from the foliage gets the product to the surface of the substrate where weed seeds germinate.

Sprayable herbicides can be less expensive to purchase and apply compared to granular products. There are a limited number of herbicides available for broadleaf control in a sprayable form, with isoxaben being the primary one. Because isoxaben is ineffective for controlling large crabgrass and other grassy weeds, it is typically combined with a dinitroaniline herbicide or other grass control product for broader spectrum weed control. Spray applications during a flush of new growth can result in damage to the developing buds. When applied at this time, up to 20% reduction in overall growth has been reported on many broadleaf crop species (Figure 6.18). To avoid this injury, make spray applications

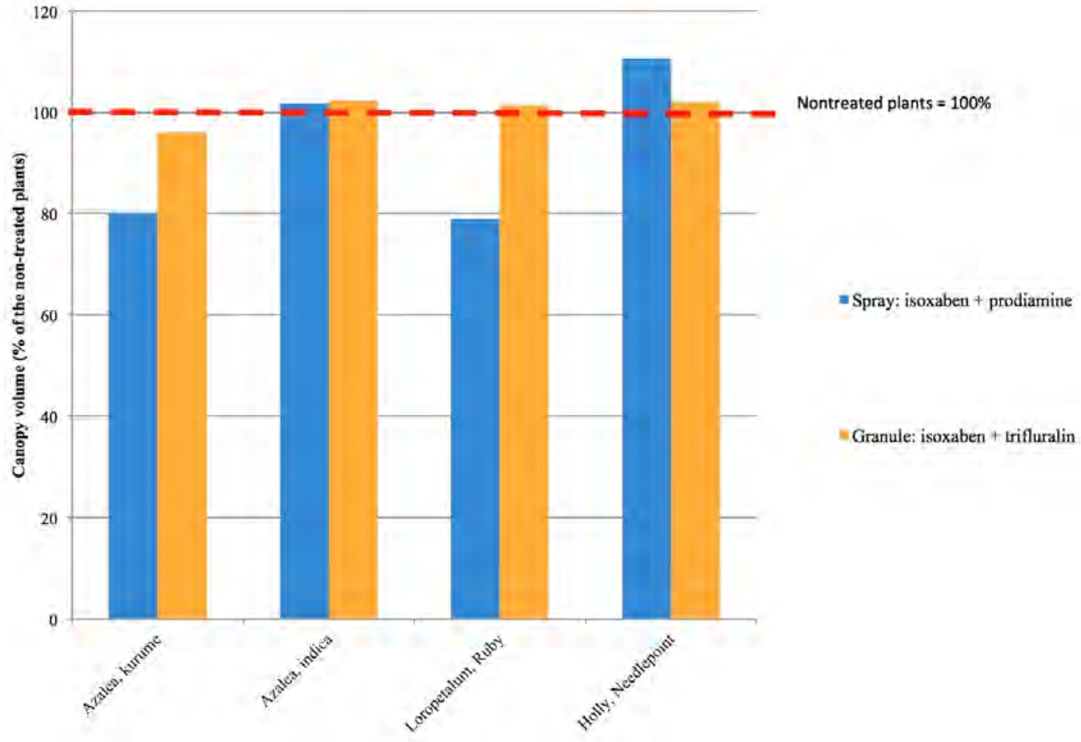


Figure 6.18 Spray applications of preemergence herbicides can reduce overall growth of some species when treatments are made during flushes of growth. This chart shows that spray applications of isoxaben + prodiamine resulted in a decreased canopy volume (estimated by plant height and width measurements for a Kurume azalea and Ruby loropetalum but not an Indica azalea or needlepoint holly. Granular applications of isoxaben + trifluralin did not reduce growth. Non-treated plants would be 100% and values for all columns are relative to the non-treated. Courtesy of J. Neal, NCSU.

after new growth has fully expanded or before bud break. Irrigating immediately after treatment, before the herbicide spray has dried can reduce, but not eliminate, damage.

Table 6.1 Preemergence herbicides labeled for use in container nurseries. See Table 6.2 for a summary of preemergence herbicides labeled for use on *Hydrangea, Ilex, Raphiolepis*, *Loropetalum*, and *Rhododendron* spp.

Other single active ingredient products:

Common name	Trade names	Available forms
isoxaben	Gallery	DF or SC
dimethenamid-p	Tower	EC
dithiopyr	Dimension	EC or EW
indaziflam	Marengo	SC or GR
flumioxazin	BroadStar	SC, DG or GR
napropamide	Devrinol	DF
s-metolachlor	Pennant Magnum	EC
sulfentrazone	Dismiss	SC
oxadiazon	Ronstar	GR or FL

Table 6.1 (*continued*) Preemergence herbicides labeled for use in container nurseries. See Table 6.2 for a summary of preemergence herbicides labeled for use on *Hydrangea, Ilex, Raphiolepis*, *Loropetalum*, and *Rhododendron* spp..

Dinitroaniline herbicides:

Common name	Trade names	Available forms
prodiamine	Barricade, Regalkade	Sprayable, granular
pendimethalin	Pendulum, Corral	Sprayable, granular
oryzalin	Surflan, Oryzalin	Sprayable
trifluralin	Treflan, Preen Garden Weed Preventer, others	Granular
oryzalin + benefin	XL	Granular

Granular Herbicide Combination Products

Common name	Trade names
benefin + oryzalin	XL
dimethenamid-p + pendimethalin	Freehand
isoxaben +_ trifluralin	Snapshot TG, DG
isoxaben + prodiamine	Gemini
oxadiazon + prodiamine	RegalStar
oxyfluorfen + oryzalin	Rout
oxyfluorfen + pendimethalin	OH2
oxyfluorfen + prodiamine	Biathlon
oxyfluorfen + trifluralin	HGH
oxyfluorfen + oxadiazon	Regal OO, others

Table 6.2 Preemergence Herbicides Registered for Use on *Hydrangea*, *Ilex*, *Loropetalum*, *Raphiolepis*, and *Rhododendron* spp. Products are listed in alphabetic order from left to right, with this table including Barricade - Marengo / Specticle SC. Remaining products are listed on the final two pages of this table.

Genus and species	Common Name	Barricade®	Biathlon®	Broadstar®	Casoron®	Dacthal®	Devrinol®	Dimension®	Freehand®	Gallery®	Gemini®	Goal®	HGH 75®	Image®	Kerb®	Marengo®/ Specticle® G	Marengo®/ Specticle® SC
Hydrangea spp.	**hydrangea species**		Ø*	Ø		F				Ø	Ø						
Hydrangea arborescens	hydrangea, smoot			Ø		F				Ø							
Hydrangea macrophylla	hydrangea, mophead, bigleaf	f/c		Ø		F			f/c	Ø						Ø*	Ø*
Hydrangea paniculata	hydrangea,			Ø		F				Ø							
Hydrangea quercifolia	hydrangea, oak-leaf			Ø		F				Ø							
Ilex spp.	**holly species**				F*	F	f/c		f/c*				f/c		F		
Ilex aquifolium	holly, English				F	F	f/c		f/c	F*	f/c*		f/c		F		
Ilex cassine	holly, dahoon				F	F	f/c	f/c	f/c	f/c	f/c		f/c		F		
Ilex cornuta	holly, Chinese	F	f/c*	f/c	F*	F	f/c	F*	f/c	f/c*	f/c*		f/c	F*	F	f/c	d
Ilex crenata	holly, Japanese	f/c		f/c	Ø	F	f/c	f/c*	f/c	f/c*	f/c*		f/c	F*	F	f/c	d
Ilex glabra	holly (inkberry)			f/c	F	F	f/c		f/c	f/c*	f/c*		f/c		F	f/c	d
Ilex latifolia	holly, lusterleaf				F	F	f/c		f/c				f/c		F		
Ilex opaca	holly, American	f/c			F	F	f/c		f/c	f/c			f/c		F		d
Ilex pernyi	holly, perny	f/c			F	F	f/c		f/c		f/c		f/c		F		
Ilex verticillata	holly, winterberry			Ø	F	F	f/c		f/c				f/c		F		d
Ilex vomitoria	holly, yaupon	f/c			Ø	F	f/c	F*	f/c	f/c*	f/c*		f/c	F*	F		
Ilex xaquipernyi	holly, San Jose				F	F	f/c		f/c	f/c	f/c		f/c		F		d
Ilex xattenuata	holly, Foster's hybrid		f/c*		F	F	f/c	f/c*	f/c	f/c	f/c*		f/c		F		d
Ilex xmeserveae	holly, meserve hybrids		f/c*	f/c	F	F	f/c	F*	f/c	f/c*	f/c*		f/c		F	f/c	d
Ilex xNellie R. Stevens	holly, Nellie R. Stevens			f/c	F	F	f/c	f/c	f/c	f/c	f/c		f/c		F		d
Loropetalum chinense	**fringe flower, Chinese**	f/c*	f/c*						f/c	f/c*	f/c*					f/c	d
Raphiolepis spp.	**hawthorne species**			f/c*			f/c			f/c*							
Raphiolepis indica	hawthorne, Indian	f/c		f/c			f/c	f/c*	f/c	f/c*	f/c*		f/c*	F		f/c	d
Raphiolepis ovata	hawthorne, roundleaf						f/c			f/c	f/c		f/c				
Raphiolepis umbellata	hawthorne, Yedda	f/c					f/c										

Table 6.2 (*continued*) Preemergence Herbicides Registered for Use on *Hydrangea*, *Ilex*, *Loropetalum*, *Raphiolepis*, and *Rhododendron* spp. Products are listed in alphabetic order from left to right, with this table including Barricade - Marengo / Specticle SC. Remaining products are listed on the final two pages of this table.

Genus and species	Common Name	Barricade®	Biathlon®	Broadstar®	Casoron®	Dacthal®	Devrinol®	Dimension®	Freehand®	Gallery®	Gemini®	Goal®	HGH 75®	Image®	Kerb®	Marengo®/ Specticle® G	Marengo®/ Specticle® SC
Rhododendron spp. (azalea)	**azalea, species**			f/c*		F	f/c		f/c	f/c*	f/c*		f/c	Ø	F	f/c*	d*
Rhododendron spp. (hardy deciduous azaleas)	**azalea, hardy decidous**				F		f/c		f/c				f/c	Ø	F		
Rhododendron (az) calendulaceum	azalea, flame			f/c*		F	f/c	F	f/c	f/c*			f/c	Ø	F		
Rhododendron (az) exbury	azalea, Exbury Hybrid Group			f/c		F	f/c	F	f/c	f/c*			f/c	Ø	F		
Rhododendron (az) indica	azalea, Southern Indica Type	f/c*	f/c*	f/c*		F	f/c		f/c	f/c*			f/c	Ø	F	f/c	d
Rhododendron (az) Girard Hybrids	azalea, Girard Hybrids	f/c*		f/c		F	f/c		f/c	f/c*			f/c	Ø	F		
Rhododendron (az) Glen Dale Hybrids	azalea, Glen Dale Hybrids					F	f/c		f/c				f/c	Ø	F		
Rhododendron (az) obtusum	azalea, Kurume Group	f/c*	f/c*	Ø	F	F	f/c	F	f/c	f/c*	f/c		f/c	Ø	F	f/c	d
Rhododendron (az) Satzuki Hybrids	azalea, Satzuki Hybrids	f/c*		f/c		F	f/c		f/c	f/c*			f/c	Ø	F		
Rhododendron spp. (rhododendron)	**rhododendron species**	f/c*		f/c*	F*	F	f/c	F*	f/c	f/c*	Ø*		f/c		F		d

Key: F = registered for use in the field (or landscape)
C = registered for use in containers
f/c= registered for both field and container use
d = registered for directed applications only
Ø = label prohibits use on this species
F*, C*, or f/c* = registered for some species or cultivars; consult label for details
Ø* = prohibited for some cultivars within a species; consult label for details.

Note: The information in this table is intended as a guide only. Information was extracted from listed herbicide labels available at the time of publication. Label changes occur and despite the authors' best efforts errors do occur. The user is encouraged to consult the herbicide label before application. The use of brand names does not imply an endorsement of these products nor discrimination against similar products.

Some injury has been reported on certain cultivars / hybrids of *R. carolinianum* and *catawbiense* from Gallery, OH2, Rout and Snapshot TG. Check labels for details and precautions.

Table 6.2 (*continued*) Preemergence Herbicides Registered for Use on *Hydrangea*, *Ilex*, *Loropetalum*, *Raphiolepis*, and *Rhododendron* spp. Products are listed in alphabetic order from left to right, with this table including OH2 - XL.

Genus and species	Common Name	OH2®	Pendulum® (EC)	Pendulum® (GR)	Pennant Magnum	Princep®	Regalkade®	Regal OO®	Regal Star II®	Ronstar® (GR)	Rout®	Snapshot TG®	Surflan®	Sureguard®	Tower®	Treflan®	XL®
Hydrangea spp.	**hydrangea species**				F			f/c				Ø			f/c		
Hydrangea arborescens	hydrangea, smoot	Ø*			F			f/c			Ø*	Ø					
Hydrangea macrophylla	hydrangea, mophead, bigleaf	f/c*		f/c	F		f/c	f/c				Ø	f/c				
Hydrangea paniculata	hydrangea,				F			f/c			Ø*	Ø					
Hydrangea quercifolia	hydrangea, oak-leaf				F			f/c				Ø	f/c				
Ilex spp.	**holly species**				F	F		f/c *	f/c*	f/c					f/c*	f/c	
Ilex aquifolium	holly, English				F	F		f/c	f/c*	f/c		F*	F*			f/c	F*
Ilex cassine	holly, dahoon				F	F		f/c	f/c*	f/c						f/c	
Ilex cornuta	holly, Chinese	f/c*	f/c	f/c	f/c	F	f/c	f/c	f/c*	f/c	f/c*	f/c*	f/c*		f/c	f/c	f/c*
Ilex crenata	holly, Japanese	f/c*	f/c	f/c	f/c	F	f/c	f/c	f/c*	f/c	f/c*	f/c*	f/c*		f/c	f/c	f/c*
Ilex glabra	holly (inkberry)				F	F		f/c	f/c*	f/c		f/c*	F			f/c	F
Ilex latifolia	holly, lusterleaf				F	F		f/c	f/c*	f/c						f/c	
Ilex opaca	holly, American	f/c	f/c	f/c	F	F	f/c	f/c	f/c*	f/c	f/c				f/c	f/c	
Ilex pernyi	holly, perny				F	F	f/c	f/c	f/c*	f/c						f/c	
Ilex verticillata	holly, winterberry				F	F		f/c	f/c*	f/c					F	f/c	
Ilex vomitoria	holly, yaupon	f/c*	f/c	f/c	F	F	f/c	f/c	f/c*	f/c	f/c*	f/c	f/c			f/c	f/c
Ilex x*aquipernyi*	holly, San Jose				F	F		f/c	f/c*	f/c	f/c*	f/c	f/c		F*	f/c	f/c
Ilex x*attenuata*	holly, Foster's hybrid		f/c*	f/c*	f/c	F		f/c	f/c*	f/c		f/c*				f/c	
Ilex x*meserveae*	holly, meserve hybrids	f/c*			F	F		f/c	f/c*	f/c	f/c*	f/c*	F*			f/c	F*
Ilex xNellie R. Stevens	holly, Nellie R. Stevens	f/c			F	F		f/c	f/c*	f/c						f/c	
Loropetalum chinense	**fringe flower, Chinese**	f/c*					f/c				f/c*	f/c	f/c		f/c		
Raphiolepis spp.	**hawthorne species**				F			f/c									
Raphiolepis indica	hawthorne, Indian	f/c*	f/c	f/c	F		f/c	f/c	f/c	f/c*	f/c*	f/c*	f/c*		f/c		f/c*
Raphiolepis ovata	hawthorne, roundleaf				F			f/c				f/c	F			f/c	F
Raphiolepis umbellata	hawthorne, Yedda				F		f/c	f/c		f/c							

Table 6.2 (*continued*) Preemergence Herbicides Registered for Use on *Hydrangea*, *Ilex*, *Loropetalum*, *Raphiolepis*, and *Rhododendron* spp. Products are listed in alphabetic order from left to right, with this table including OH2 - XL.

Genus and species	Common Name	OH2®	Pendulum® (EC)	Pendulum® (GR)	Pennant Magnum	Princep®	Regalkade®	Regal OO®	Regal Star II®	Ronstar® (GR)	Rout®	Snapshot TG®	Surflan®	Sureguard®	Tower®	Treflan®	XL®
Rhododendron spp. (azalea)	**azalea, species**	f/c*	f/c	f/c	F			f/c *		f/c*	f/c*		f/c		f/c	f/c	f/c
Rhododendron spp. (hardy deciduous azaleas)	**azalea, hardy deciduous**			f/c	F			f/c *					f/c		f/c	f/c	f/c
Rhododendron (az) calendulaceum	azalea, flame	f/c*	f/c	f/c	F			f/c *		f/c	f/c*	f/c*	F*			f/c	f/c
Rhododendron (az) exbury	azalea, Exbury Hybrid Group		f/c	f/c	F			f/c *		f/c	f/c	f/c*	f/c*			f/c	f/c
Rhododendron (az) indica	azalea, Southern Indica Type		f/c	f/c	f/c		f/c*	f/c *	f/c	f/c	f/c*	f/c*	f/c*			f/c	f/c
Rhododendron (az) Girard Hybrids	azalea, Girard Hybrids		f/c	f/c	F			f/c *		f/c*		f/c*	f/c			f/c	f/c
Rhododendron (az) Glen Dale Hybrids	azalea, Glen Dale Hybrids							f/c *		f/c		f/c*				f/c	f/c
Rhododendron (az) obtusum	azalea, Kurume Group	f/c*	f/c	f/c	f/c		f/c*	f/c *	f/c	f/c*	f/c*	f/c*	f/c*			f/c	f/c
Rhododendron (az) Satzuki Hybrids	azalea, Satzuki Hybrids	f/c	f/c	f/c	F			f/c *		f/c		f/c*	F*			f/c	f/c
Rhododendron spp. (rhododendron)	**rhododendron species**	f/c*	f/c	f/c	f/c*		f/c*	f/c *	f/c	f/c*	Ø*	f/c*	f/c*		f/c	f/c	f/c

Key: F = registered for use in the field (or landscape)
C = registered for use in containers
f/c= registered for both field and container use
d = registered for directed applications only
Ø = label prohibits use on this species
F*, C*, or f/c* = registered for some species or cultivars; consult label for details
Ø* = prohibited for some cultivars within a species; consult label for details.

Note: The information in this table is intended as a guide only. Information was extracted from listed herbicide labels available at the time of publication. Label changes occur and despite the authors' best efforts errors do occur. The user is encouraged to consult the herbicide label before application. The use of brand names does not imply an endorsement of these products nor discrimination against similar products.

Some injury has been reported on certain cultivars / hybrids of *R. carolinianum* and *catawbiense* from Gallery, OH2, Rout and Snapshot TG. Check labels for details and precautions.

The treatment prior to covering houses in fall is important to reduce the number of weeds that germinate in overwintering structures during the winter and early spring. No preemergence herbicides are registered for use on crops in covered houses due to the potential for trapping of herbicide vapors around nursery foliage. These vapors can injure nursery crops, especially those that have broken bud in the overwintering house. Thus preemergence herbicide application must wait until after houses are completely uncovered in spring before resuming preemergence herbicide applications. That is why it is important to have a preemergence application in fall. Generally, there is a two week waiting period between application and covering of the house. This ensures that herbicide vapors have dissipated before plastic is placed over the structure.

Postemergence herbicide use in container production

In container production, emerged weeds are typically removed by hand. Hand weeding is the most costly component of nursery weed control. While all other aspects of a nursery weed management program should strive to reduce the time spent hand weeding, it should be done on a regular schedule to reduce weed competition and prevent spread of weed seeds. Under certain circumstances emerged weeds may be controlled with herbicides. Postemergence herbicides labeled for use on woody shrubs are summarized in Table 6.3. There are selective options for grass control in broadleaf shrubs and trees. These include clethodim (Envoy Plus), fenoxaprop (Aclaim Extra), fluazifop-p-butyl (Fusilade II, Ornamec Over-The-Top), and sethoxydim (Segment). These selective grass-control herbicides may be applied over-the-top of some species but must be applied as a directed spray around others. Fenoxaprop is generally less effective for controlling perennial grasses than the other three herbicides. Check the label as some of these products require addition of an adjuvant. Nonionic surfactants are generally preferred over crop oils, especially under high temperature/high humidity conditions when spraying overtop nursery crops.

Fewer options are available for broadleaf weed and sedge control. Herbicides that control broadleaf weeds and sedges have a greater risk of crop injury and are typically used only as directed sprays – avoiding contact with the foliage of the crop plants (Table 6.3). Isoxaben (Gallery) can be applied to control small bittercress seedlings, and the granular products containing oxyfluorfen or flumioxazin can control cotyledon-stage broadleaf weeds. Otherwise, there are essentially no selective herbicides for removing most emerged broadleaf weeds in container production. Non-selective herbicides containing glyphosate are not labeled for use around container-grown plants. Spot treatment of weeds in containers is possible with non-selective, contact-type herbicides such as diquat (Reward) or pelargonic acid (Scythe) but any such treatments must avoid contact with foliage or tender stems of the crop plants. Sulfentrazone (Dismiss) controls seedling broadleaf weeds and sedges such as kyllinga but must also be used only as a directed application, avoiding contact with foliage and green stems. Directed applications of these herbicides have limited utility in small containers; consequently, producers generally must rely on hand weeding and preemergence herbicide applications for weed management in container production. In larger containers, directed applications of contact postemergence herbicides can be a very cost-effective way to remove emerged weeds.

Weed management in Pot-in-Pot production

Since the substrate used in pot-in-pot production is soil-less, producers can generally follow the guidelines listed above for above-ground container production. For areas between rows (Figure 6.19), one could utilize combinations of registered preemergence and postemergence herbicides, such as glyphosate plus flumioxazin or indaziflam, for weed control. But, do not use glyphosate to control weeds in the containers. Emerged weeds in containers can be controlled with spot applications of diquat (Reward), or pelargonic acid (Scythe).

Weed and Ground Cover Management in Field Nursery Crop Production

Weeds are defined as plants growing where they are not desired. A plant species growing in the area between the crop rows may not be considered a weed, but may be considered one if growing in the row. Ideally one would prefer a ground cover that suppresses weeds both within and between rows and requires little mowing but not compete with tree or shrubs or harbor insect or disease pests. Finding such a living mulch has been difficult. Therefore, a nursery manager needs to manage vegetation both in the row and between the rows to minimize adverse effects on nursery production.

Weeds compete with nursery crops for water, nutrients, and light. Growers cannot simply overcome this competition by adding fertilizer because adding fertilizer also increases weed growth. Competition in the soil seems to be especially important for nitrogen, explaining why a nursery crop can have a pale color when growing alongside weeds. Competition for water in the root zone is especially important in dry summers. Tall weeds shade crop leaves, reducing photosynthesis and growth. Vine-type weeds shade the crop and cause distorted crop growth (Figure 6.20).

Figure 6.19 Before "replanting" pot-in-pot production areas, take care to control weeds, especially perennial weeds in and around socket pots.

Weeds can also interfere with nursery practices. One example is poison ivy, which contains a toxin to which most people are sensitive. Weeds with thorns, such as horsenettle and wild blackberry, are a nuisance to individuals working in the nursery.

Table 6.3 Postemergence Herbicides Registered for Use on *Hydrangea*, *Ilex*, *Loropetalum*, *Raphiolepis*, and *Rhododendron* spp.

Genus and species	Common Name	Acclaim® Extra	Envoy®	Fusilade® II	Segment®	Basagran® TO	Casoron®*	Certainty®	Dismiss®	Lontrel®*	Sedgehammer®**
Hydrangea spp.	hydrangea	OT		O*	O*	D		D			D
Hydrangea macrophylla	hydrangea, big leaf	OT		O*	O*	D		D			D
Hydrangea paniculata	hydrangea, panicle	OT		OT		D		D			D
Hydrangea quercifolia	hydrangea, oakleaf	OT		OT		D		D			D
Ilex spp.	holly species	O*	OT	O*	O*	D	O*	D			D
Ilex aquifolium	holly, English		OT			D	OT	D			D
Ilex aquifolium x cornuta	holly, English x Chinese		OT			D	OT	D			D
Ilex cassine	holly, dahoon		OT			D	OT	D			D
Ilex cornuta	holly, Chinese		OT	OT	O*	O*		OT	D		D
Ilex crenata	holly, Japanese	OT	OT	O*	O*	OT		D			D
Ilex glabra	inkberry		OT	OT		D		D			D
Ilex latifolia	holly, lusterleaf		OT			D	OT	D			D
Ilex meserveae	holly, blue boy/girl	OT	OT	OT		D	OT	OT	D		D
Ilex opaca	holly, American	OT	OT	OT		D	OT	D			D
Ilex vomitoria	holly, yaupon		OT	OT	OT	D	OT	D			D
Ilex xattenuata	holly, Foster's hybrid		OT	OT		D	OT	D			D
Loropetalum chinense	fringe flower, Chinese					D		D			D
Raphiolepis indica	hawthorn, Indian		OT	OT	OT	D		D			D
Raphiolepis umbellata	hawthorn, Yedda			OT		D		D			D
Rhododendron (spp.) (azalea)	azalea spp.	OT	OT	D*	O*		O*	D	D		D
Rhododendron indicum	azalea, Indica	OT	OT	D*	O*			D			D
Rhododendron molle	azalea, Chinese	OT	OT		O*		O*	D			D
Rhododendron obtusum	azalea, Kurume hybrids	OT	OT	D*	O*		OT	D		OT*	D
Rhododendron x (azalea) Gable hybrids	azalea, Gable hybrids	OT	OT	O*			OT	D			D
Rhododendron x (azalea) Girard hybrids	azalea, Girard hybrids	OT	OT	O*			OT	D			D
Rhododendron x (azalea) Glend Dale hybrids	azalea, Glend Dale hybrids	OT	OT	OT			OT	D			D

Table 6.3 (*continued*) Postemergence Herbicides Registered for Use on *Hydrangea*, *Ilex*, *Loropetalum*, *Raphiolepis*, and *Rhododendron* spp.

Genus and species	Common Name	Acclaim® Extra	Envoy®	Fusilade® II	Segment®	Basagran® TO	Casoron®*	Certainty®	Dismiss®	Lontrel®*	Sedgehammer®**
Rhododendron x (azalea) Northern Lights hybrid	azalea, Northern Lights hybrid	OT	OT		O*		OT	D			D
Rhododendron x (azalea) Pericat hybrids	azalea, Pericat hybrids	OT	OT	O*				D			D
Rhododendron x (azalea) Satsuki hybrids	azalea, Satsuki hybrids	OT	OT	O*				D			D
Rhododendron spp. (rhododendron	rhododendron species	OT	OT	D*	O*		OT	D	D		D*
Rhododendron carolinianum	rhododendron, Carolina	OT	OT		O*		OT	D			D*
Rhododendron catawbiense	rhododendron, Catawba	OT	OT	O*	O*		OT	D		OT*	D*
Rhododendron maximum	rhododendron, rosebay	OT	OT		O*		OT	D			D*
Rhododendron PJM group	rhododendron, PJM	OT	OT	D*			OT	D			D

Key:
OT = registered for over-the-top use.
D = registered for directed applications.
O* = registered for over-the-top use on certain species within the genus - consult label for details.
D* = registered for directed applications on certain species within the genus; consult label for details.

*Casoron and Lontrel may be used in field nurseries and landscape plantings, not in container nurseries. Crop should be established before using Casoron. See label for details.
**Sedgehammer is labeled for use in landscape plantings only, not for use in field or container nurseries.

Weeds harbor insect pests, disease pathogens, and rodents that can damage nursery crops. One way to help manage disease and insect pests is through control of weed species that serve as alternative hosts. Weed growth can reduce air flow through the nursery, increasing humidity which could lead to increased disease severity. Weeds can intercept fungicide or insecticide sprays meant for the crop, reducing their effectiveness. Weeds such as white clover may attract bees to a site, which is of particular concern if insecticides are being applied. Weeds provide cover and a food supply for mice, voles, and other rodents.

A wide range of weed species can grow in a field nursery. Weeds can be divided into taxonomic groups or can be grouped by life cycle. Understanding taxonomic plant groups will assist in understanding selectivity of herbicide action. Understanding the life cycle of a weed species will guide the cultural and chemical control options needed for management. Growers must maintain a year-round weed management plan because both summer and winter weeds infest field production sites.

Figure 6.20 Weedy vines, such as morningglory, compete for light and can disfigure crops.

Ground covers
A ground cover within a field nursery is essential to minimize soil losses due to water and wind erosion, to maintain long-term soil productivity, and to prevent the encroachment and spread of weeds. In addition, a well-established sod makes it easier to drive through fields when they are muddy or covered in snow or ice. A well-established sod is frequently the only economical way to support equipment such as tractors, sprayers, or mechanical harvesters when the soil is wet. Furthermore, research has demonstrated the need for a vegetative barrier around the edges of a field nursery. Grassed contour strips slow down and direct flow of water across a slope and serve as a buffer and final biological filter to remove any excess nutrients before runoff leaves the nursery. Kentucky bluegrass, perennial ryegrass, tall fescue, orchardgrass, and fine-leafed fescues (hard, creeping) are potential ground covers in cooler environments (zones 5 to 7). In warmer zones, tall fescue and bermudagrass are effective in providing workable sod because they are vigorous and provide a great deal of biomass. Bermudagrass is invasive, though, and would be a management problem in the crop row.

If not properly managed, however, ground covers can compete with nursery crops for fertilizer, light, and water, thereby reducing crop growth. A common way to minimize competition while enjoying the advantages of a ground cover is to keep the rows clean or mostly weed free with preemergence and/or postemergence herbicides, while maintaining a ground cover in the row middles.

Pre-planting weed management planning
The most important weed management tasks are done before planting. Good site preparation includes scouting for perennial weeds and controlling the difficult species such as trumpet creeper, multiflora rose, mugwort, Florida betony, kudzu, and hedge bindweed before planting. Manual or mechanical control of perennial weeds can be difficult and costly once the field is planted. In most cases, there are no selective herbicides for controlling perennial broadleaf weeds after planting.

Controlling perennial weeds requires killing the root system, because most perennial weeds will regrow if only the top is destroyed. There are three options for controlling perennial weeds: cultivation, fumigation, or a systemic postemergence herbicide (glyphosate). While cultivation can be effective against certain perennial weeds, multiple cultivations over a period of several months are often required to control the root systems. Furthermore, some perennial weeds such as mugwort and yellow nutsedge are spread by cultivation. Fumigation is an option of last resort to eradicate infestations of weeds that cannot be controlled after planting. Systemic postemergence herbicides such as glyphosate (Roundup ProMax and many other trade names) will control many perennial weeds, but timing of the application is critical to ensure satisfactory perennial weed control. Late summer or early fall is an effective time to apply glyphosate for many perennial weeds.

Planting temporary cover crops such as sorghum-sudangrass, plowing in fall, and allowing the land to remain fallow can help to reduce some weed and insect problems (Figure 6.21). The intense shading, mowing, and competition created in a green manure or cover crop program will greatly reduce, if not eliminate, certain weed problems. Fall plowing exposes roots and tubers to the freeze-thaw cycles of a Southeastern U.S. winter but will also expose the soil to erosion and is therefore <u>not</u> encouraged.

A permanent cover crop can be planted in fall prior to planting. The cover crop can be killed in the row using a nonselective herbicide such as glyphosate prior to planting trees or shrubs.

Weed scouting
The first step in any pest management program is to identify the pest(s). Scouting the fields for weeds will enable the grower to determine which weeds are present and to plan appropriate management strategies. Field nurseries should be scouted at least twice a year – in late summer or early fall, and again in early summer. In late summer

Figure 6.21 Temporary cover crop established to suppress weeds and improve soil.

most summer annual, perennial, and biennial weeds are easily identified. In early summer, winter annuals, perennials, and summer annuals that escaped control procedures can be identified. Weed scouting involves assembling an inventory of the weeds in each block. This is done by simply walking each field and recording the species encountered. Then, highlight the most important species – those that are most prevalent, perennial, new to the field, on noxious weed lists, or should have been controlled by the weed management program in place. With this information, the grower can better plan a weed management program that matches the needs of each crop and field.

In-row vegetation management

Weeds growing within or in close proximity to the crop plants are most competitive and must be controlled. This is typically done by establishing a two to three-foot wide weed free band. We typically refer to this practice as the "in-row" treatment (Figure 6.22). The two basic strategies for in-row vegetation management are (1) *residual*, relying primarily on preemergence herbicides, and (2) *non-residual*, relying almost exclusively on non-selective postemergence herbicides. Each strategy has advantages and disadvantages. The residual approach will utilize more expensive herbicides but will require fewer trips through the fields. The non-residual approach is simple and inexpensive but will require many trips across the fields each season to control emerged weeds.

In the *residual strategy* preemergence herbicides are applied in late winter to control summer annual weeds, and late summer to control winter annual weeds. Depending upon the local conditions, weeds present, and the particular herbicides used, an additional treatment in late spring or early summer may be required. Perennial weeds are spot treated with appropriate postemergence herbicides. Most preemergence herbicides will not control emerged weeds so any weeds present at application can be controlled by adding a postemergence herbicide.

The *non-residual strategy* involves frequent applications of non-selective, postemergence herbicides, on an as-needed basis. Treat weeds when they are 6 to 8 iches in height; this will enable the applicator to contact weeds and avoid the crop. It is common during the growing season to treat fields every 4 to 6 weeks but the interval between applications will vary with weed spectrum present, the mode of action of the herbicide used, and the prevailing weather conditions. Frequent field scouting should be conducted to identify the appropriate times for re-treatment.

Figure 6.22 In-row vegetation control prevents weed competition, facilitates maintenance of drip irrigation lines and reduces vertebrate pest damage to crops.

Herbicides commonly used in the non-residual management strategy include glyphosate (Roundup ProMax and many other trade names), glufosinate (Finale), and diquat (Reward). Each has its advantages and disadvantages. To decide upon the most appropriate one, follow the guidelines for using postemergence herbicides (below), and consult with your county Cooperative Extension Service and an appropriate reference. However, complete reliance on any one herbicide or mode of action will lead to weed population shifts and herbicide resistance. This is particularly important for glyphosate-reliant vegetation management programs. Widespread adoption of the glyphosate-resistant crops technology has led to relatively rapid development and spread of glyphosate-resistant weeds such as horseweed. Fields should be scouted for "escaped" weeds. When resistance is suspected, remedial action should be taken immediately to prevent the reproduction and spread of resistant weed populations.

Between-row vegetation management

Vegetation between rows (Figure 6.23) is managed to prevent encroachment of the vegetation into the crop rows, eliminate habitat for vertebrate pests (such as deer), to provide improved access to the field, and to prevent the spread of weeds into the crop row. The vegetation between crop rows is managed by mowing, cultivation or sub-lethal rates of herbicides (referred to as chemical mowing), and/or growth regulators can be used to slow growth of grass but not kill it. The most common vegetation management practices are cultivation and mowing.

Relying solely on cultivation is discouraged because it leads to erosion and may actually spread perennial weeds. Mowing is generally conducted three to four times each year. The interval between mowing events can be extended by careful selection and use of plant growth regulators. For example, spraying tall fescue or orchardgrass in early spring, when there are four to five new leaves or seven to ten days after mowing, with 1 pint/acre of sethoxydim (Segment), a selective grass herbicide, will suppress the grass for 8 to 10 weeks. Sethoxydim may also be used for chemical mowing. Another alternative is glyphosate (Roundup-Pro, others), which can provide 6 to 10 weeks of suppression at a cost of a few dollars per acre, much less than regular mowing. Glyphosate needs to be applied as a directed spray between the nursery stock rows. Use no more than 25 gallons of the final spray mix per broadcast acre. Chemical mowing will result in chlorotic

Figure 6.23 Maintaining vegetation between crop rows can prevent erosion and improve field access in wet weather. But between-row vegetation must be managed to prevent encroachment into crop rows and to discourage vertebrate pests.

(yellow) grass for up to 30 days and will result in a gradual transition of the vegetation from grass to broadleaf cover. Such a transition may not be desirable as broadleaf cover generally provides less traction for vehicle tires and poorer wear tolerance compared to perennial grasses.

Preemergence and Postemergence Herbicide Use

Guidelines for using preemergence herbicides

Most preemergence herbicides will not control emerged weeds, so they should be applied before weeds germinate. In field production, preemergence herbicides should be applied after transplanting to weed-free soil and then irrigated for effectiveness. Most preemergence herbicides can be applied after the soil is settled around the transplants, but before weeds emerge. This prevents weed seeds from germinating for several weeks to months. Frequency of herbicide application will depend upon the residual activity of the herbicide. Residual weed control will increase with increasing herbicide application rate; control decreases with increasing amounts of rainfall or irrigation, higher temperatures and higher soil organic matter levels. The proper herbicide for each situation will be dictated by the crop species, weed species, and future use of the field. As with any other tool, each herbicide has unique characteristics that should be considered when planning a weed management program:

Managing herbicide resistance:
- Do not rely on a single herbicide mode of action
- Rotate herbicides
- Utilize both residual and postemergence herbicides
- Control "escaped" weeds before they go to seed

- ✶ Crop Safety – First and foremost, the herbicides must be safe on your crops.
- ✶ Weed control spectrum – Which weeds the herbicide will and will not control.
- ✶ Rate of application – The correct rate will vary with weed pressure, organic matter content of the soil, and ornamental species.
- ✶ Residual – The length of time the herbicide will provide effective weed control.
- ✶ Activation – For maximum effectiveness, each herbicide needs to be watered (about 1/2-inch irrigation or rain) into the soil surface within a specified number of days.
- ✶ Mechanism or mode of action – Prevent herbicide resistance by using different modes of action in combinations or rotation.
- ✶ Potential losses – Avoid leaching, runoff, spray drift and volatility through proper product selection and use.

The most common practice in field nursery weed control is to combine a "grass" herbicide with a "broadleaf" herbicide in order to control a broad spectrum of weeds. Products commonly used in field production of woody shrubs are listed below (products for field production of trees and conifers may be different).

Each herbicide differs in the weeds controlled, longevity of residual control, ornamental crop safety and cost. Among the "grass" herbicides, only dimethenamid-p and s-metolachlor control yellow nutsedge (*Cyperus esculentus*), yet they can have a shorter residual life in the soil and are generally less effective on many common broadleaf weeds than the other herbicides listed above.

Dichlobenil (Casoron) is a winter-applied herbicide that can be used in some established shrub beds. It controls emerged winter annuals, provides residual control of annual weeds, and will suppress or control many perennial weeds such as tall fescue, Florida betony (*Stachys floridana*), field horsetail (*Equisetum arvensis*) and mugwort (*Artemesia vulgaris*), A typical residual weed control program in field shrub production will likely use some variation of the following scenario:

- After planting: band-apply isoxaben combined with pendimethalin, prodiamine or oryzalin. If yellow nutsedge is a major problem, instead combine isoxaben with dimethenamid or metolachlor.
- At ten to twelve weeks later, repeat the application, especially if it is a wet spring.
- In late summer, apply a “broadleaf and grass” herbicide combination for winter weed control.
- In late winter – apply a combination treatment to control spring germinating species.
- Escaped weeds may be controlled with careful spot applications of non-selective postemergence herbicides.

Before deciding which herbicides are right for each crop, consider these guidelines and consult with your county Cooperative Extension Agent and an appropriate reference.

Landscape beds

The preemergence herbicides listed above generally can also be used in landscape beds. The formulation of indaziflam registered for this use is Specticle and is applied as a directed spray. The sprayable formulation of flumioxazin (SureGuard) can also be used for premeergence as well as early postemergence weed control. SureGuard must be applied as a directed spray, ideally when ornamentals are dormant, as it causes contact injury to tender foliage.

Common “broadleaf” preemergence herbicides	Common “grass” preemergence herbicides
flumioxazin	dimethenamid-p
isoxaben	indaziflam
oxyfluorfen	s-metolachlor
simazine	napropamide
	oryzalin
	oxadiazon
	pendimethalin
	prodiamine

Note: Refer to Table 6.2 and confirm each year with product labels for specific product registrations for your crop(s). Other “broadleaf” herbicides are registered for use around other species of field-grown nursery crops.

Guidelines for using postemergence herbicides

Postemergence herbicides are applied to weeds after they have emerged. Characteristics of postemergence herbicides that should be considered before selection and use are:

- Crop Safety – Even non-selective herbicides differ in their levels of phytotoxicity to crops.
- Weed control spectrum – which weeds the herbicide will and will not control.
- Selective versus nonselective – Selective herbicides will provide greater crop safety but limited spectrum of weed control.
- Systemic versus contact – Contact herbicides are less effective on perennial weeds but are potentially less injurious to actively growing crop plants.
- Application timing (or season) for optimum control – Seedling weeds are easier to control, and season of application will influence the efficacy of glyphosate on many perennial weeds.
- Drying time – Foliar applications must get into the plant before rain or irrigation washes the herbicide off.
- Mechanism or mode of action – Prevent herbicide resistance by using different modes of action in combinations or rotation.
- Persistence – A few postemergence herbicides have soil residual that can affect subsequent crops.
- Potential losses – Avoid leaching, runoff, spray drift and volatility through proper product selection and use.

Postemergence herbicides can be classified as systemic or contact, and as selective or nonselective. Systemic herbicides are absorbed and move through the plant. These are useful for controlling perennial weeds. For best control, the weeds must be actively growing so the herbicides can move throughout the plant. Contact herbicides kill only the portion of the plant that is actually contacted by the herbicide. Nonselective herbicides have the potential to kill or injure any plant they contact; whereas, selective herbicides kill some types of plants and not others.

The majority of postemergence herbicides used in field nursery crops are non-selective. Diquat dibromide (Reward) and pelargonic acid (Scythe) are nonselective, contact herbicides. They control small annual weeds but only burn-back perennial or large annual weeds. Good spray coverage is especially important when applying contact herbicides. Re-application is necessary to control larger weeds. Glyphosate (Roundup ProMax and many other trade names) and glufosinate (Finale) are systemic, nonselective herbicides, and are more effective than contact herbicides on perennial weeds or larger annual weeds. Glufosinate is considered to be a contact herbicide by many practitioners, but limited translocation does occur within treated leaves or branches. Glyphosate is better translocated in plants, and is consequently more effective at controlling perennial weeds and grasses compared to glufosinate.

Grasses can be selectively controlled in many broadleaf and non-grass monocot nursery crops. Clethodim (Envoy Plus), fluazifop-P-butyl (Fusilade II), and sethoxydim (Segment) are selective, systemic, postemergence herbicides which only kill grasses while leaving broadleaf weeds and crops unharmed. Conversely, very limited options are available for selective postemergence broadleaf weed control in nursery crops. Consequently, broadleaf weeds are generally controlled preemergence or with non-selective herbicides. Clopyralid (Lontrel) can be used in certain field-grown tree and shrub species for control of weeds in the legume, nightshade, and aster families. Desired crops in these plant families cannot be treated, though, with clopyralid. Major weeds controlled by clopyralid include Canada thistle, groundsel, horseweed, vetch, dogfennel, and white clover.

All postemergence herbicides have a specified drying time for maximum effectiveness ranging from 30 minutes to 6 hours. This is the length of time that needs to pass after herbicide application before irrigation or rain to ensure that the herbicide has had adequate time to be absorbed and therefore affect the weed (termed “rainfast” on product labels). Although some postemergence herbicides labeled for field production remain in the soil for a short length of time after application, most commonly used postemergence herbicides have little or no soil activity; therefore, multiple applications are needed for perennial weeds. The majority of herbicides used for postemergence weed control in field production are used either for grass control or for nonselective weed control. Products that provide nonselective weed control should not be applied to the foliage or green stems of ornamental plants as severe injury or plant death may occur.

Figure 6.24 Attempting to treat the entire crop row with spray from one side can result in too much contact with the stem of the crop leading to bark damage.

Postemergence herbicides may be applied in many different manners. Where weed populations are sparse, spot spraying with hand held spray nozzles is common. If weed populations are consistent, tractor-mounted (or ATV mounted) sprayers may be used. Tractor-mounted sprayers should be adjusted and calibrated to spray a band on either side of the crop row. Do not attempt to spray the entire band width from one side of the row! This will result in too much spray contact with the crop stem and bark damage (Figure 6.24). Controlled droplet applicators have also been used successfully to apply postemergence herbicides. However, bark damage to the crop has resulted when the applicators are operated in such a manner that crop stems are contacted by the concentrated spray. Regardless of the application method utilized, do not treat with systemic herbicides when suckers, water sprouts, or fresh pruning scars are present, or else crop injury will likely result. Remove suckers at least 48 hours before applying systemic herbicides such as glyphosate. Spray shields or curtains can be added to the spray rig to reduce crop exposure, but such systems do not eliminate the potential for crop exposure through spray drift or rubbing from wetted spray shields or curtains.

When using postemergence herbicides:

- Apply at correct rate.
- Select the most appropriate herbicide for the task.
- Remember that multiple applications are usually required to control perennial weeds.
- Use the type and amount of surfactant specified on the label.
- Apply when the air temperature is above 50^{o}F and the comfort index (temperature in ^{o}F plus humidity) is below 140.
- Treat weeds at proper growth stage.
- Allow plenty of drying time (each herbicide is different, consult the label for specific guidelines).

Yellow nutsedge control

Yellow nutsedge is one of the most common perennial weeds encountered in field nursery crop production. It can be controlled with non-selective herbicide but some selective herbicides are available. Directed sprays of bentazon (Basagran), sulfentrazone (Dismiss), or sulfosulfuron (Certainty) can be used for controlling emerged yellow nutsedge. Check the label to see if an adjuvant is needed. Limited research has been conducted with sulfentrazone and sulfosulfuron in nursery crops so only treat small areas first and monitor the crop for two months to check for any injury. These herbicides can also be used for yellow nutsedge control in landscapes. Halosulfuron (SedgeHammer) can be used for postemergence nutsedge control in landscape beds but is not registered for use in field nursery production.

Alternatives to Herbicides in Field Production

Herbicides cannot always be used, nor are they effective in controlling all weeds. In these situations, cultivation and hand pulling may be the only available options.

Cultivation can be a valuable tool for weed management (Figure 6.25). It effectively controls small annual weeds and many later successional weeds such as tree saplings, and can be used to improve cover crop establishment. However, many creeping perennials are spread by cultivation. Also, cultivation can stimulate successive flushes of germinating weeds by bringing new weed seeds to the soil surface. You will need to check for emerging weeds on a two- to three-week cycle if you are routinely cultivating.

Figure 6.25 Cultivation is a common method of controlling seedling annual weeds but can lead to excessive erosion and damage to shallow crop roots.

If preemergence herbicides have been applied and activated, they form an herbicide barrier that must be left undisturbed to be effective. Cultivation disrupts this barrier and lessens the effectiveness of the herbicide. Therefore, cultivate sparingly if you use preemergence herbicides.

Cultivation has several other drawbacks. Cultivated soil is very susceptible to erosion since there is little or no vegetation to hold the soil in place. In addition, implements such as in-row weeders, which cut off weeds below the soil surface, can build up ridges, which are detrimental to growth of nursery crops. Ridged soil around the stem collar of newly set liners tends to suffocate them just as if they had been planted too deeply. Cultivating nursery stock in subsequent years causes considerable root pruning and delays growth, and can lead to a soil buildup around the collar of nursery stock. Many landscape contractors have had problems with trees grown with soil thrown over the surface of the roots. The surface of the root ball may have several inches of soil with few roots above what was the original soil line. Landscapers think they are planting at the correct height. In reality, they are planting several inches too deep. Thus, cultivation has received considerable attention in landscape magazines and has been identified as a poor practice for growing nursery stock.

It is important to develop a weed management strategy that encompasses all 12 months of the year and uses all available options. These include preventative measures, such as preemergence herbicides and sanitary practices that prevent the introduction and spread of weed seeds and vegetative parts, and remedial measures such as postemergence herbicides, cultivation and supplemental hand weeding. A comprehensive weed management program will incorporate the most appropriate options for each cropping situation. Below are crop-specific weed management guidelines for *Ilex, Loropetalum, Raphiolepis*, and *Rhododendron* (including azalea). Weed management in hydrangea production can be quite different from these crops and is presented in Chapter 1 of this manual.

Weed Management in Loropetalum (*Loropetalum* spp.) Production

LOROPETALUM

Background

Loropetalum, also known as Chinese fringe-flower or Chinese witch-hazel, is most commonly produced in containers but may also be grown in the field. Loropetalum is one of the most commonly planted and popular landscape plants in the South, and as such, many different cultivars have been released in recent years. It is typically grown and sold in shrub-form; however, in recent years growers have begun to prune many varieties into standards or small trees. The production method (container vs. field) and pruning practices will affect which herbicides can be used for weed control.

In comparison with many other container nursery crops, loropetalum is more sensitive to certain preemergence herbicides, especially in container production and before new growth has hardened off. Thus it is very important to utilize only those herbicides labeled for loropetalum production and to follow all product label restrictions and precautions.

Weed Control in Containers

Preemergence weed control in containers

In container production, growers rely heavily on regular applications of broad-spectrum preemergence herbicides. Young loropetalum plants tend to be sensitive to herbicide injury. Start with well-rooted, healthy, weed-free liners. Newly potted plants should be irrigated well to settle the substrate before preemergence herbicides are applied. Granular herbicides labeled for use in loropetalum production include oxyfluorfen + prodiamine, pendimethalin + dimethenamid-p, indaziflam, oxyfluorfen + pendimethalin, prodiamine, oxyfluorfen + oryzalin, and trifluralin + isoxaben. Sprayable herbicides labeled for use on loropetalum include prodiamine and isoxaben (generally applied as a tank mix), oryzalin and dimethanimid-p. Spray applications are convenient and more cost-effective, compared to granular treatments, but recent research has shown that spray-applied herbicides may carry a greater risk of crop injury than granular treatments. For example: isoxaben + prodiamine reduced loropetalum growth by 18% compared to non-treated plants; whereas a granular application of the same active ingredients resulted in no injury (Figure 6.26).

To prevent damage to foliage and growing points, granular herbicides should be applied when the foliage is dry. If granules adhere to the foliage, they should be removed by shaking the plants, blowing granules off the foliage (some growers use leaf blowers for this purpose), or irrigating after treatment. When using spray applications of

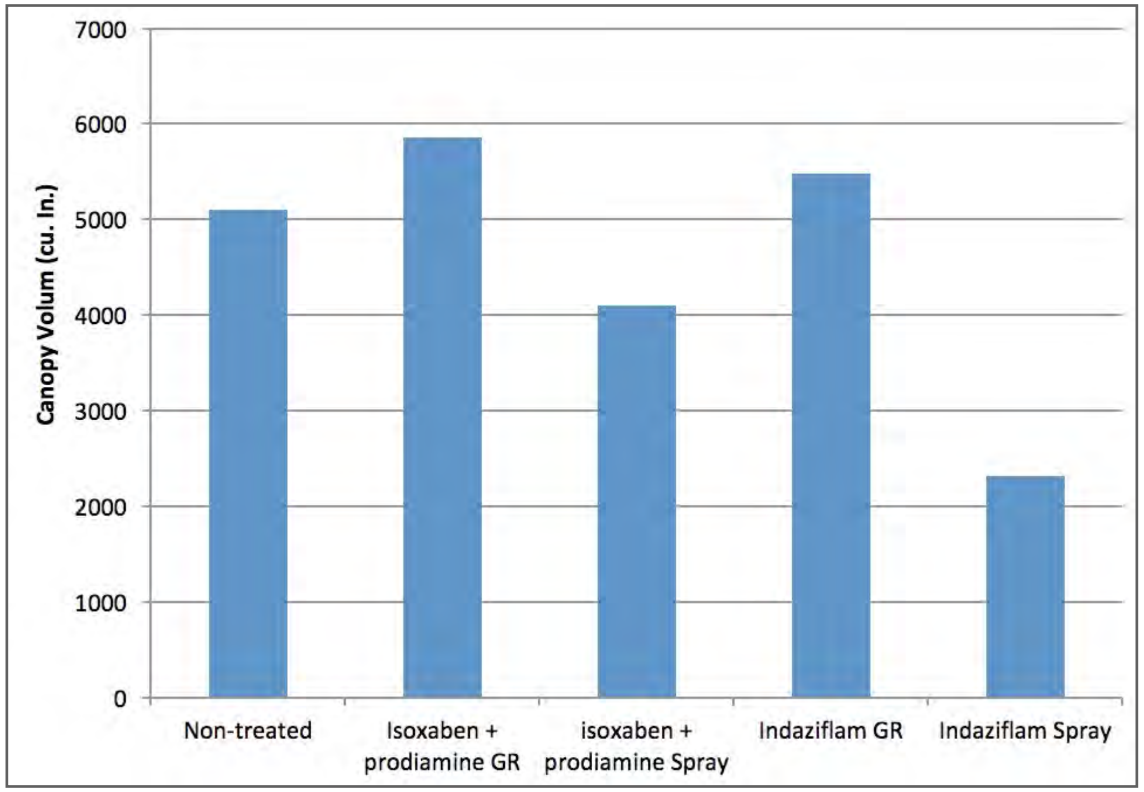


Figure 6.26 Loropetalum canopy volume following two applications of granular or spray-applied preemergence herbicides. Courtesy J. Neal, NCSU.

preemergence herbicides, avoid applications at or shortly after bud-break, and irrigate immediately after treatment to wash herbicides off the treated foliage.

Herbicides will need to be re-applied on a regular schedule. In the Southeastern U.S., preemergence herbicides are typically applied every eight to ten weeks during the growing season. However, several preemergence herbicide labels include specific restrictions for treating loropetalum and limit the total number of applications allowed each season. Herbicide rotations will be required and one must consider general restrictions on the total number of applications allowed per year. For example: pendimethalin + dimethenamid-p and indaziflam are each limited to about two applications per season; oxyfluorfen + pendimethalin and oxyfluorfen + oryzalin are limited to a combined total of three applications; and trifluralin + isoxaben applications may not exceed a total of 600 lbs. product per acre per year. Additionally, growers should not make “back to back” applications of dimethenamid-p + pendimethalin or sprayable formulations of dimethenamid-p. These product labels state that sequential applications should be separated by 16 weeks or more. Applying these products at an 8-week interval can result in significant reductions in growth (Figure 6.27).

To minimize the potential for herbicide injury growers must:

- Utilize only labeled herbicides;
- Remove the herbicide from the crop foliage;
- Rotate herbicide active ingredients; and
- Observe label restrictions for maximum doses allowed.

Figure 6.27 To avoid crop injury growers must follow herbicide label precautions. The Freehand label specifically states that Freehand applications should be separated by at least 16 weeks. In this photo loropetalum plants treated (Right) with Freehand twice at an 8-week interval are about 50% smaller than the non-treated plant (Left).

Postemergence weed control in containers

There are very limited options for postemergence weed control in this species. Isoxaben can be used for control of small bittercress plants. Otherwise, growers are limited to hand weeding.

Weed Control in Loropetalum Field Production and Landscape Maintenance

Preemergence weed control

The herbicides mentioned for container production could also be used in field production and/or landscape maintenance. The liquid formulation of indaziflam can be used in loropetalum production under the name Marengo and in landscape maintenance under the name Specticle, but must be applied as a directed application as it can injure foliage. The sprayable formulation of flumioxazin (SureGuard) is another herbicide that can be used only as a directed application to established shrubs in landscapes. The sprayable formulation of flumioxazin provides contact postemergence weed control, burning leaf tissue. It can cause significant plant injury if any of the foliage or green bark is contacted, but can be applied for weed control in most deciduous tree species and shrubs if label precautions are followed. If possible, apply products like sprayable formulations of flumioxazin and indaziflam when nursery crops are dormant as young tender growth will be more susceptible to injury. The postemergence injury from these two herbicides prevents use of sprayable formulations in container production.

Postemergence Herbicide Use in Field Production and Landscape Maintenance
Currently, there are no postemergence herbicides labeled for over the top application in loropetalum production or in landscapes. However, several herbicides can be applied as a directed application to established woody ornamentals in nursery production and landscape maintenance, including bentazon, diquat, glufosinate, glyphosate, and pelargonic acid. Halosulfuron can be used as a directed spray around established woody plants to control sedges and certain broadleaf weeds in landscapes, but not in nursery production. As loropetalum is most commonly grown in compact shrub form, making directed herbicide applications difficult without contacting some of the foliage. Production of loropetalum in tree form allows for easier herbicide application since there is less exposure of foliage to the spray compared to the shrub form.

Halosulfuron and glyphosate are systemic and move throughout the plant, meaning that even if only a small portion of the plant is contacted, the whole plant can be affected. Therefore extreme caution should be used when applying these chemicals to avoid contact with plant foliage. When treating tree-form plants, remove basal sprouts at least 24 hours before applying systemic herbicides. Diquat and pelargonic acid are both contact herbicides, and bentazon is primarily contact in action, so only the portion of the plant that is contacted with these products will be injured, and the plant has a better chance of recovery. Glufosinate has limited systemic action. However, it should be noted that better weed control will likely be achieved with systemic herbicides, especially for perennial weeds. In landscape plantings it is important to avoid the use of glyphosate formulations that contain an ALS-inhibiting residual herbicide such as imazapic or imazapyr. Loropetalum is particularly sensitive to this class of herbicides and may be severely injured by directed applications within the root zone (Figure 6.28).

While there are several options in terms of herbicides labeled for use on loropetalum, it is impossible to include all cultivars or varieties on product labels. Remember that although a genus may be listed on a herbicide label, some cultivars or species may still be sensitive to certain herbicides. Chances are that you will likely grow several varieties of loropetalum which are not included specifically on any herbicide label. In these cases, if the label allows, treat a small group of plants and monitor them for several weeks noting any discoloration, stunting, or other crop injury before treating an entire block. A complete list of preemergence and postemergence herbicides for use in and around loropetalum is included in Tables 6.2 and 6.3.

Figure 6.28 Loropetalum is quite sensitive to injury from root uptake of several ALS-inhibitor herbicides. The mulched area around this loropetalum was treated with a dose of glyphosate + imazapic labeled for use in landscape beds. Injury developed slowly over a 6 month time.

Weed Management in Holly (*Ilex spp.*) Production

ILEX

1. Background
2. Weed Control in Container Production
3. Weed Control in Field Production and Landscape Maintenance

Background

Ilex is a diverse genus including large tree-forms, shrubs and ground covers as well as evergreen and deciduous species. Most *Ilex* species are produced in containers, but several are also field grown. In general, *Ilex* species are not particularly sensitive to injury from preemergence herbicides. Thus, weed management programs in *Ilex* production can be tailored to the spectrum of weeds present.

Figure 6.29 Spray applications of residual herbicides can stunt plants when applied during or shortly after budbreak. These Carissa holly plants were stunted by over the top applications of preemergence herbicides – Nontreated (A) vs treated with labeled doses of isoxaben + oryzalin (B) or dithiopyr (C). Herbicides were applied at labeled doses.

Weed Control in Container Production

Preemergence weed control in containers

In comparison with many other container nursery crops, *Ilex* is quite tolerant of most preemergence herbicides labeled for use in nursery crop production. Granular herbicides labeled for use on many newly potted or planted hollies include oxyflurorfen + prodiamine, flumioxazin, pendimethalin + dimethenamid-p, indaziflam G, oxyfluorfen + pendimethalin, oxyfluorfen + oryzalin , trifluralin + isoxaben , oxyfluorfen + isoxaben + triflurlalin, and others. Follow label restrictions and precautions.

Injury may occur when granular treatments are misapplied at higher-than-labeled doses. For example: about 20% mortality was observed from indaziflam G applied at twice the labeled rate to newly potted *Ilex cornuta* hybrids, but applications at the labeled dose were not injurious.

Spray-applied preemergence herbicides may also be used over the top of many species of hollies. Standard treatments will include isoxaben combined with one of the following: prodiamine, dimethenamid-p, oryzalin , pendimethalin, or others. Spray treatments should be avoided during flushes of new growth as the tender new growth can be sensitive to herbicide damage. The amount of injury varies among herbicides and species. For example: Carissa holly was severely damaged by dithiopyr applied over the top during active growth and stunted by oryzalin applications (Figures 6.29 and 6.30), yet, needlepoint hollies receiving the same treatments were unaffected. When applying any preemergence herbicide, ensure that all label precautions are followed and there is no tender growth or bud swelling, and that foliage is dry when making granular herbicides. Chances of injury can be reduced by applying irrigation as soon as possible after herbicides are applied or mechanically shaking the granules from the foliage. Some growers have used leaf blowers to remove granules from plant foliage.

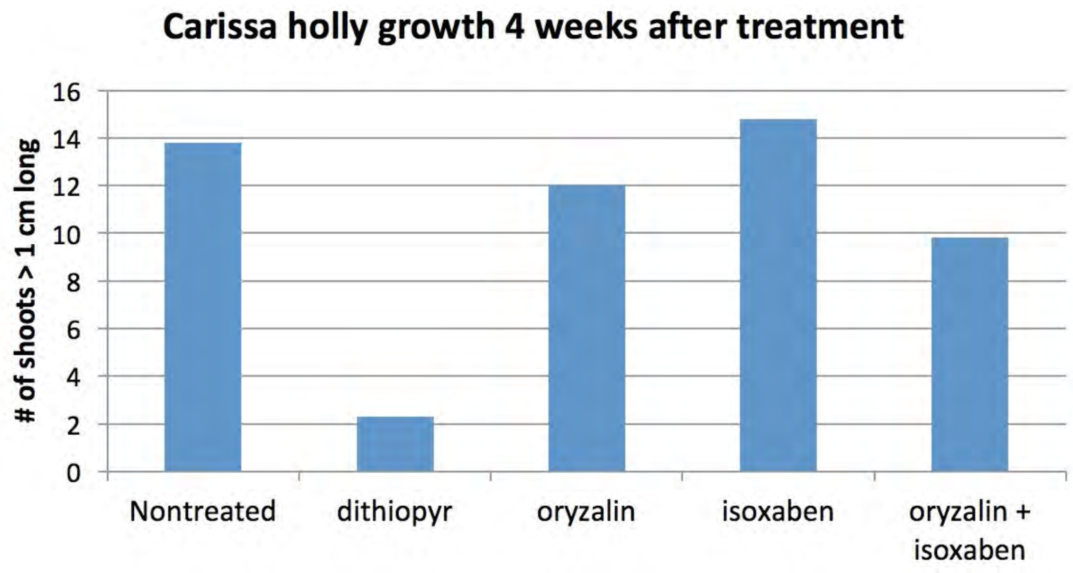


Figure 6.30 Plant responses to spray herbicide treatments may vary between species and herbicides. In this research, new growth of Carissa holly was reduced 13% and 29% by oryzalin and oryzalin + isoxaben, respectively. Plants were severely stunted by dithiopyr EW. Isoxaben applied alone did not reduce growth. This data measured the number of new shoots that were greater than 1 cm long. Overall shoot length and overall plant growth

Postemergence weed control
Emerged weeds not controlled by preemergence herbicides should be removed by hand before they are well established in the containers. In large containers, where herbicides may be applied in a manner that avoids contact with the foliage, emerged weeds may be controlled with non-selective, postemergence herbicides such as diquat or pelargonic acid. These herbicides are non-selective and will damage the foliage and green stems of holly plants if contacted by the spray.

Emerged grasses may be controlled with selective grass herbicides, fluazifop-p-butyl, sethoxydim, or clethodim. These selective grass herbicides may be applied over the top of holly plants to control annual or perennial grass weeds.

Weed Control in Field Production and Landscape Maintenance
Preemergence weed control
Newly planted hollies may be treated over the top with some of the same preemergence herbicides labeled for use in container production. Spray-applied herbicides are generally preferred for field production. If plants have new growth present at the time of treatment, directed applications that avoid the growing points are recommended. When directed applications are made, it is also possible to use sprayable formulations of indaziflam. Flumioxazin may be used around established hollies in landscape plantings but not nurseries.

In established plantings, directed applications of preemergence herbicides are recommended. Directed applications of preemergence herbicides may contact the lower branches but should be made before bud-break to prevent injury to the new growth. Applications during the growing season should be timed to avoid treating plants during a flush of new growth (Figure 6.31).

Figure 6.31 Directed applications of most preemergence herbicides may contact lower foliage but should be made be made before flushes of new growth.

A “typical” preemergence weed management program for field grown hollies will include at least two applications per year – late winter and late summer. A third application in late spring may be necessary if difficult to control summer annual weeds such as morningglory (*Ipomoea* spp.) are present. Each application usually includes two preemergence herbicides – one for broadleaf weeds and one for grass control. Examples include: isoxaben + oryzalin, or flumioxazin + prodiamine. Simazine is an option for preemergence broadleaf control in holly but high application rates should be avoided, especially on sandy soils. If chlorosis occurs in holly following a simazine application, and the yellowing is not due to a nutrient deficiency or other cause, then either the simazine rate needs to be decreased or an alternative broadleaf herbicide used. Dichlobenil may be applied to established field plantings of some holly species to control perennial broadleaf weeds. Check the herbicide label for species restrictions as certain holly species are sensitive.

Because many preemergence herbicide options are available, the selection of products can be tailored to the site. For example: if the site is infested with yellow nutsedge (*Cyperus esculentus*) then spring and early summer treatments should include dimethenamid-p or s-metolachlor. Similarly, if morningglory is present, the summer application should include simazine or flumioxazin.

Emerged weeds may be controlled by directed applications of non-selective herbicides such as glyphosate or glufosinate. Care should be taken to avoid contact with the foliage of the crop. Research has shown that injury to *Ilex* from glyphosate spray drift is greatest when the exposure occurs in the spring. Applications in late summer, fall, or before bud-break resulted in little or no injury to evergreen hollies. However, expect severe injury to deciduous hollies from spray drift (or misapplications) in late summer or fall.

Some selective herbicides are labeled for directed applications around field-grown hollies. Directed applications of bentazon, sulfosulfuron, or sulfentrazone may be used to control yellow nutsedge around some hollies. Check product labels for specifics. These herbicides may also control some seedling broadleaf weeds. For example: bentazon controls seedling ragweed; and sulfentrazone controls seedling morningglory. Little research data is available on sulfosulfuron safety around ornamental plants. Therefore, before widespread use, test this product around a small number of plants and observe plant growth for several months.

In established landscapes, a spray formulation of flumioxazin can be used as a directed application around established shrubs to kill small, seedling broadleaf weeds while also providing broad spectrum preemergence control. However, directed spray applications of flumioxazin can damage hollies and other ornamentals if foliage or green bark is contacted by the spray (Figure 6.32); thus such treatments are not labeled for hollies in container or field nurseries. Directed applications of halosulfuron may be used for nutsedge control in landscape plantings but not in nurseries.

Annual and perennial grasses may be controlled with the selective grass herbicides, clethodim, fluazifop-p, or sethoxydim. These herbicides may be applied over the top of holly plants. For best results, begin treatments early in the season when weedy grasses are less than 10 inches tall. Multiple applications are usually required.

In field grown hollies it is also possible to manage weeds with an entirely postemergence herbicide program. Directed applications of non-selective herbicides may be applied on a regular basis, about every six weeks. However, because the foliage of hollies is often close to the ground, it is often not possible to avoid foliage contact. For this reason, we recommend utilizing a residual preemergence herbicide program for most field grown hollies. Where plants are limbed-up for special markets, an all-postemergence program is more feasible. However, relying on a single postemergence herbicide, such as glyphosate, can increase the chances of herbicide resistance development. A number of weed species, such as horseweed and pigweed, have developed resistance to glyphosate. Rotate herbicides with different modes of action and utilize preemergence and nonchemical control options to reduce the potential for herbicide resistance.

Figure 6.32 Flumioxazin (SureGuard) causes injury to new holly growth when foliage is contacted by the spray; whole plant exposed to spray (A), and a close up of injury to the growing point and new leaves (B).

Weed Management in Indian Hawthorn (*Rhaphiolepis* spp.) Production

RHAPHIOLEPIS

Background

Indian hawthorn are primarily grown in containers, although certain varieties may be field grown in tree-form for special landscape uses. Most preemergence herbicides commonly used in container nurseries are labeled for use in *Rhaphiolepis*, so herbicide choice could be based more on specific weeds of concern at a particular nursery, rather than choosing merely to avoid potential crop damage.

Weed Control in Container Production

Preemergence weed control in containers

A complete list of preemergence herbicides for use in *Rhaphiolepis* production is included in Table 2. Granular herbicides used in Indian hawthorn production include flumioxazin, dimethenamid + pendimethalin, oxyfluorfen + trifluralin, oxyfluorfen + pendimethalin, pendimethalin, prodiamine, oxyfluorfen + oxadiazon, oxyfluorfen + oryzalin, isoxaben + trifluralin, trifluralin, and benefin + oryzalin. Apply granular herbicides when the foliage is dry then irrigate or use a leaf blower to move granules from the crop foliage.

Granular formulations are still the most widely used preemergence herbicides in container nursery production, but more growers are beginning to transition into spray applied herbicides which are often more economical both in terms of chemical costs and application costs. Sprayable herbicides for over the top application to Indian hawthorn include prodiamine, napropamide, dithiopyr, isoxaben, isoxaben + prodiamine, pendimethalin, s-metolachlor, oryzalin, and dimethenamid-p. As isoxaben is primarily effective on broadleaf weeds, it should be combined with a grass herbicide, such as prodiamine, for broader-spectrum weed control. The liquid formulation of indaziflam may also be used in *Rhaphiolepis* production but only as a directed application in pot-in-pot or field systems. Most *Rhaphiolepis* are grown in compact shrub form with foliage extending down to the surface of the container substrate and often over the edges of the container. Directed applications will be difficult to make, so select preemergence herbicides that can be applied over the top.

While most preemergence herbicides can be used in Indian hawthorn without harming the crop, chances of potential injury are greater immediately after potting, during periods of active growth, or under environmental stresses. For example, in some cases flumioxazin G has caused slight tip burn or speckling on leaves when applied to newly potted Indian hawthorn liners, but the formulation of flumioxazin G released in 2009 (and the current formulation) reduced the potential for such injury. Avoid over-the-top application of emulsifiable concentrate or oil-based herbicide formulations, such as

Pendulum EC, Tower or Pennant Magnum, during active growth or under hot, humid growing conditions, as injury to new growth may occur. When a particular cultivar or species of *Rhaphiolepis* is not included on a particular herbicide label, apply the herbicide to a small group of plants to check for injury before applying to an entire block if the label allows.

Weed Control in Field Production and Landscape Maintenance

Preemergence weed control

The preemergence herbicides registered for use in container production of *Raphiolepsis* can generally be used in field production as well. Sprayable applications are generally used in field production due to lower cost and easier application compared to granular forms. Dormant applications are preferred to minimize the potential for crop injury.

Postemergence weed control

For postemergence control of grassy weeds, several graminicides are labeled for over-the-top use in Indian hawthorn production including clethodim, fluazifop-p-butyl, and sethoxydim. While all the graminicides provide postemergence control of grassy weeds, product selection should be based on which grass species is problematic at a particular nursery. Check specific product labels for a complete list of recommendations and precautions for each product.

In container-grown plants, emerged broadleaf and sedge weeds should be removed by hand. In field plantings and landscapes, the nonselective postemergence herbicides diquat, glyphosate and pelargonic acid can be applied as directed applications, keeping the spray off Indian hawthorn foliage. Bentazon can be applied as a directed spray for control of yellow nutsedge and certain broadleaf weeds. Sulfosulfuron may be applied as a directed spray to control yellow or purple nutsedge. In landscape plantings, sedge control can also be achieved by making directed applications of halosulfuron. As making directed applications in Indian hawthorn will likely be difficult, control of broadleaf and sedge weeds should be accomplished through proper sanitation practices, hand weeding, and other nonchemical means, as well as through use of preemergence herbicides.

Weed Management in Rhododendron and Azalea

RHODODENDRON AND AZALEA

1. Background
2. Weed Control in Azalea and Rhododendron Liner Production
3. Weed Control in Container Production
4. Weed Control in Field Production and Landscape Maintenance

Background
Rhododendron species, including azalea, are popular landscape shrubs. Azaleas are almost exclusively produced in containers but rhododendrons are field and container grown. Producers growing these species in containers are primarily faced with controlling annual grass and broadleaf weeds. Field growers are faced with a wider diversity of weeds, including annual, biennial, and perennial species. Weed control in azaleas is critical to eliminate weed competition and as a component of an integrated pest management program. Weed species can be symptomless hosts of diseases such as *Phytophthora* root rot and therefore impact disease severity of azalea. The presence of weeds increased the percentage recovery of *Phytophthora cinnamomi* from container-grown azaleas; the pathogen was isolated from azalea roots in all weedy treatments, as well as from 3 of 8 non-inoculated, weedy controls (Kalmowitz et al. 1991). *P. cinnamomi* was not isolated from any other non-inoculated controls. The authors concluded that weed control is essential to control *Phytophthora* root rot of container-grown azaleas.

Weed Control in Azalea and Rhododendron Liner Production
Starting with clean liner stock is the first step in minimizing weed infestations for the life of the crop. Rooted cuttings of azaleas typically grow rapidly enough to be transplanted directly from propagation trays to outdoor production. However, many varieties of rhododendron have slower growth rates and will be placed in “liner” production for one growing season before transplanting to larger containers or field sites. If plants are kept in a covered structure, such as a greenhouse, cold frame, or plastic-covered hoop house, no residual herbicides may be used on the crop. If liners are produced in shade houses or other open production areas, some residual herbicides may be used. Granular herbicides containing oxadiazon or a combination of oxadiazon plus oxyfluorfen are commonly used in azalea and rhododendron liner production. Other herbicides, such as pendimethalin, prodiamine, oryzalin, trifluralin, indaziflam, flumioxazin or dimethenamid-p, are likely to injure young liners and should be avoided.

Weed Control in Container Production
Preemergence weed control in containers
Weed management in container production is typically based on preemergence herbicide applications supplemented with hand weeding. Azaleas appear to be somewhat more sensitive to certain preemergence herbicides compared to other woody nursery species. One concern is adverse effects on root development from the dinitroaniline herbicides (oryzalin, pendimethalin, prodiamine, trifluralin) (Figure 6.33). Pendimethalin, for example, reduced root growth in treated ‘Tradition’ azalea, likely due to movement into the upper inch of the growing mix (Derr and Simmons 2006). Ronstar G (oxadiazon) is not a root inhibitor and it had no adverse effect on azalea root weight after one, two, or three applications during a growing season (Derr and Salihu 1996). Azaleas treated with sprayable formulations of oryzalin, isoxaben, and prodiamine all had lower root weight than plants treated with oxadiazon. Azaleas treated with Rout or OH2 had an intermediate level of root growth between oxadiazon and oryzalin, probably due to the lower dose of a dinitroaniline herbicide in these combination granular herbicides. Stunting of shoot and root growth, though, appeared to be temporary and, plant size at

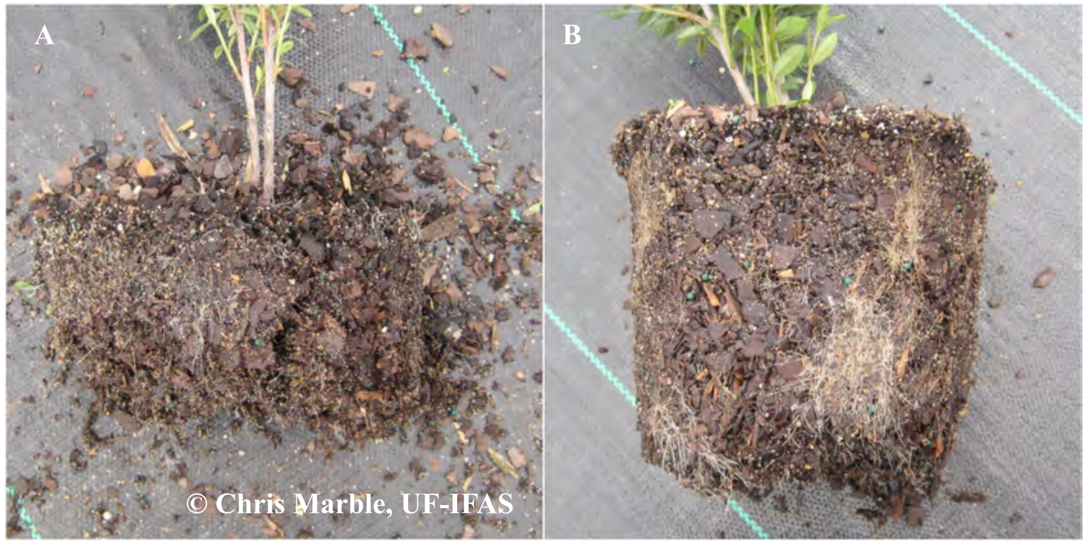


Figure 6.33 'Fashion' azalea root injury from dinitroaniline herbicide overdose. Stunted root development (A) following an over application of pendimethalin vs. (B) non-treated azalea (B).

treatment may be a factor in the extent of stunting caused by preemergence herbicides, with larger plants being more tolerant.

Observe root growth prior to making sequential applications of root-inhibiting herbicides. When diagnosing injury from root-inhibiting herbicides like the dinitroaniline class of chemicals, keep in mind that these herbicides do not kill roots; they inhibit the development of new lateral roots, as well as stunting growth of existing root tips. Root diseases, such as Phytophthora, can kill roots.

Another concern following application of dinitroaniline herbicides to azalea is effects on stems near the soil line. Swelling or constriction of azalea stems has been observed following applications of certain dinitroaniline herbicides, leading to stem breakage. For example, spray applications of oryzalin decreased stem diameter and strength of 'Hino Crimson' and 'Snow' (Skroch and Catanzaro 1994). Such injury is most commonly associated with excessive doses applied to young plants. Therefore, it is important to apply the correct dose and to follow the manufacturer's re-application interval guidelines and restrictions.

Herbicides are typically applied as dry granules but spray applications are possible. Common granular herbicides used in azalea and rhododendron production often combine active ingredients to control a broader spectrum of weeds and include Biathlon (oxyfluorfen + prodiamine), Rout (oxyfluorfen plus oryzalin), OH2 (oxyfluorfen + pendimethalin), Snapshot (isoxaben + trifluralin), FreeHand (dimethenamid-p + pendimethalin), and others. Broadstar, a granular formulation of flumioxazin, and Marengo G, a granular formulation of indaziflam, also provide broad spectrum weed control. Care is needed when azaleas are flushing in the spring, as tip burn can occur from products containing oxyfluorfen or flumioxazin as granules may be caught in the growing points, causing injury. To avoid injury to the foliage, granules should be applied when the foliage is dry and any granules adhering should be removed by irrigation, leaf blowers, rakes or manually. Snapshot and FreeHand do not cause burning of foliage so they may be a better option during periods of active azalea growth.

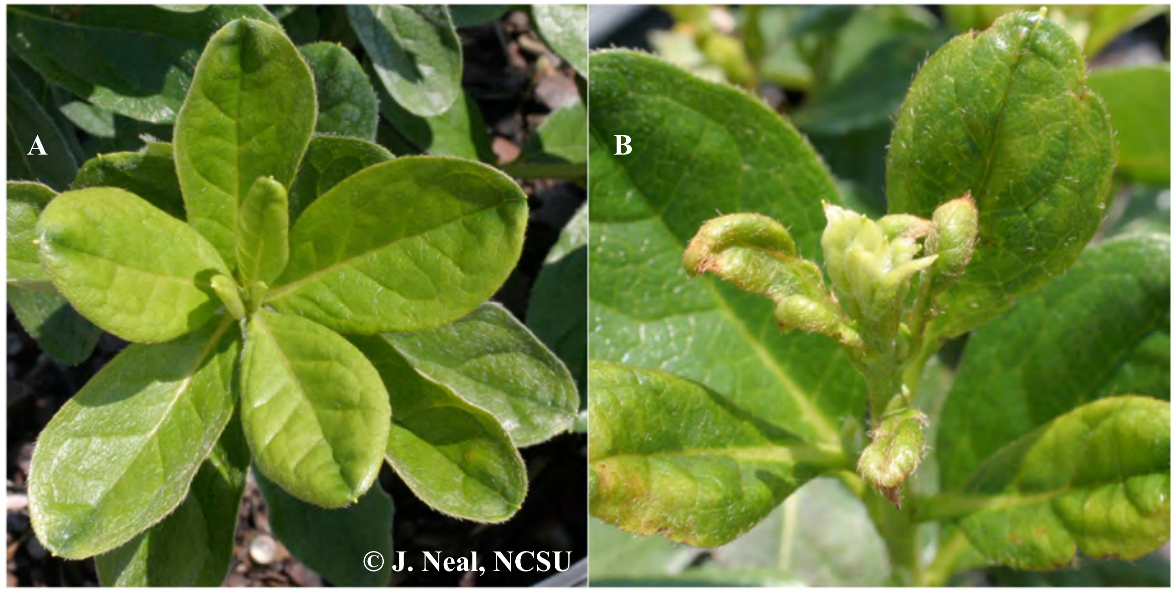


Figure 6.34 Spray applications during active growth can injure azalea growing points. Isoxaben + prodiamine applied during flush of new growth caused bud damage and stunted growth. Non-treated, normal foliage (A); Treated plant (B).

Spray applications of preemergence herbicides are gaining in popularity but there are fewer herbicide options available and these can carry a greater risk of crop injury compared to granular treatments. For example: spray applications of flumioxazin (SureGuard), oxadiazon (Ronstar FLO or 50 WSP), or oxyfluorfen (Goal) can cause severe injury to container-grown azaleas but granular formulations of these herbicides cause little or no injury. Sprayable formulations of herbicides labeled for use on azaleas include prodiamine, isoxaben, pendimethalin, s-metolachlor and dimethenamid-p. A common strategy is to combine isoxaben with one of the other herbicides for broader spectrum weed control. Applications over the top of actively growing azaleas can cause bud damage and reduced growth. (Figure 6.34). To avoid crop injury, treat plants before bud break or after new growth has matured. Irrigation immediately after treatment can reduce the severity of the damage, but does not eliminate the injury. In addition to bud injury, spray applications may affect both the root and shoot systems of azaleas resulting in reduced canopy growth (Figure 6.35).

Tipburn has also been seen with a sprayed combination of isoxaben plus s-metolachlor, perhaps due to the organic solvent in the s-metolachlor formulation increasing herbicide

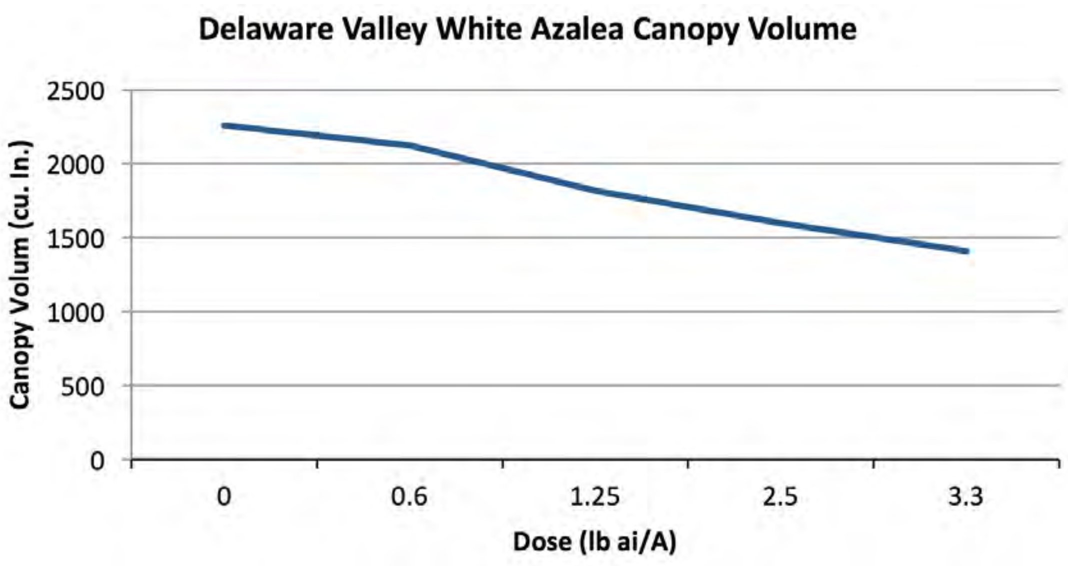


Figure 6.35 Effects of increasing isoxaben + prodiamine doses on Delaware Valley White azalea canopy volume. Spray applications reduced growth. Canopy volume was calculated as one-half the volume of a sphere. Courtesy J. Neal, NSCU.

uptake. Sprayed applications of pendimethalin can stunt azalea growth from strictly shoot exposure, possibly due to an impact on the growing point. Therefore, granular formulations of these herbicides should be considered instead of sprayable forms when azaleas are actively growing. Plants should only be treated after they have an established root system. Any stunting of growth following overtop application of a sprayable pendimethalin formulation likely results from adverse impacts on both the root and shoot system of azaleas. Emulsifiable concentrate (EC) formulations of the dinitroanilines should be avoided when spraying overtop nursery crops.

Postemergence herbicide use

The postemergence grass herbicides clethodim, fenoxaprop, fluazifop-p, and sethoxydim can be used to selectively control emerged annual and perennial grasses. Clethodim is the most effective on annual bluegrass, while fenoxaprop generally provides lower control of perennial grasses than the other three herbicides. Certain of these herbicides require adjuvant addition. Use of a nonionic surfactant instead of a crop oil is preferred, especially under high temperature/high humidity conditions since oil additives may increase potential for spotting. However, check product labels for variety and species restrictions.

Few options are available for selective control of broadleaf weeds. Sprayed applications of isoxaben can be used for postemergence control of small bittercress seedlings. Isoxaben will not provide postemergence control of other common broadleaf weeds in nursery production. Emerged broadleaf weeds are generally controlled by hand removal.

Differences in response to herbicides among azalea cultivars

Over 10,000 varieties of azalea have been named, although most are not in production (Azalea Society of America 2015). Azalea varieties may differ in their susceptibility to injury from pre- and postemergence herbicides (Skroch and Catanzaro 1994). Trifluralin and oryzalin reduced shoot and root growth of certain cultivars. Oryzalin decreased stem diameter and strength of 'Hino Crimson' and 'Snow'. Damage caused by other herbicides was cultivar dependent. In one study, bentazon applied twice at 1 lb ai/A plus crop oil caused 45% injury to 'Pleasant White', approximately 25% injury to 'Delaware Valley White' and Elsie Lee' and no injury to 'Rosebud', 'Tradition', or 'Hershey Red' azalea (Derr and Appleton 1989). Fluazifop-p at 0.25 lb ai/A plus nonionic surfactant caused 15 to 19% injury to the azalea cultivars 'Poukhanense' and 'Hinocrimson' but no injury to 'Coral Bell', 'Hershey Red', or 'Tradition' (Derr 1987). Frank and Beste (1983) reported injury to 'Hinocrimson' azalea from fluazifop-p but no injury to 'Hershey Red' azalea. In that same test, sethoxydim caused no injury to either cultivar. Sethoxydim is therefore preferred over fluazifop for overtop applications to control emerged grasses in azalea.

Weed Control in Field Production and Landscape Maintenance

Nonchemical control

Rhododendron species tend to be shallow rooted. Cultivation, therefore, must be done with caution to avoid excessive damage to root systems. Mulching is a way to conserve soil moisture as well as suppress weed growth. Excessive mulch depth (greater than 4 inches) can lead to wet conditions that can favor disease development. Certain mulches, such as pine needles or pine bark, will decrease soil pH, beneficial for growth of *Rhododendron* species.

Preemergence herbicide use

Most of the preemergence herbicides labeled for container-grown rhododendrons may also be used in field production. In addition, dichlobenil can be applied during winter for control of emerged winter weeds as well as several important perennial weeds, including perennial grasses and mugwort (wild chrysanthemum). Lower rates of dichlobenil, however, need to be used in azalea compared to certain other woody nursery crops. In landscape beds, flumioxazin (SureGuard) can be applied as a directed spray around established azalea. Avoid contact to azalea foliage or damage to foliage could occur.

Also, be careful concerning herbicide use on stock plants used for cuttings. Some research suggests certain herbicides may adversely impact rooting of cuttings. Trifluralin, EPTC, simazine, and dichlobenil were applied to stock plants of the azalea cultivars 'Coral Bells' and 'President Clay' (Fretz 1973). Softwood tip cuttings were taken 80 days after treatment and plant root development was assessed 60 days after being stuck. Rooting was unaffected by most herbicides but was reduced by dichlobenil applied at twice the labeled rate.

Postemergence herbicide use

The postemergence grass herbicides already mentioned can be used for controlling annual and perennial grass weeds. Nonselective herbicides, such as glyphosate, pelargonic acid, and diquat, need to be used with caution to avoid rhododendron injury. Use of shields will assist in keeping spray droplets off rhododendron foliage.

Yellow nutsedge control

Yellow nutsedge (*Cyperus esculentus* L.) is a troublesome weed in container and field nursery production as well as in landscape maintenance. Most preemergence herbicides do not control yellow nutsedge, but s-metolachlor and dimethenamid-p do suppress this weed and can be used in nurseries and landscapes. Metolachlor was evaluated in the field for four years to determine phytotoxicity to 11 varieties of azaleas and *Rhododendron* 'PJM' (Frank and Beste 1990). Fall or spring applications of metolachlor caused no injury to the established azaleas 'Delaware Valley White', 'Hershey Red' and 'Hinocrimson'. Some temporary injury occurred to newly planted 'Hinocrimson' azaleas treated with metolachlor at rates above the recommended one. A July application at this same rate caused some temporary injury to established 'Delaware Valley White', 'Double Pink', 'Hershey Red', 'Hot Shot', 'Karen', 'Lady Robin', 'Rosebud' and 'Tradition' azaleas and 'PJM' rhododendron. No reduction in crop quality or size was observed with any species at time of crop harvest.

No selective postemergence herbicide is available for over-the-top application to azaleas. Bentazon can be used as a directed spray in nursery production and landscape maintenance. The Basagran TO label, however, warns about potential for injury to rhododendron with directed sprays of this herbicide. Directed applications of sulfosulfuron and sulfentrazone may be used to control emerged nutsedge and some seedling broadleaf weeds. However, only treat small areas initially as limited research has been conducted with these herbicides, especially for sulfosulfuron. While halosulfuron can be used as a directed spray in landscape beds for nutsedge control, it is not labeled for nursery use.

References

Azalea Society of America 2015. Azaleas. Accessed January 21, 2017 <http://azaleas.org/index.pl/azaleas.html>.

Derr, J.F. 1987. Response of Azalea (*Rhododendron obtusum*) cultivars to sethoxydim and fluazifop-P. Weed Technology 1:226-230.

Derr, J.F. and B.L. Appleton. 1989. Phytotoxicity and yellow nutsedge control in azalea and liriope with Basagran (bentazon) J. Environ. Hort. 7:91-94.

Derr, J.F. and S. Salihu. 1996. Preemergence herbicide effects on nursery crop root and shoot growth. J. Environ. Hort. 14:210-213.

Derr, J.F. and L.D. Simmons. 2006. Pendimethalin Influence on Azalea Shoot and Root Growth. J. Environ. Hort. 24:221–225.

Frank, J.R. and C.E. Beste. 1983. Sethoxydim and fluazifop-butyl for weed control in field-grown azaleas. Proc. Northeastern Weed Sci. Soc. 37:331.

Frank, J.R. and C.E. Beste. 1990. Growth inhibition of ericaceous plants from metolachlor. J. Environ. Hort. 8:173-176.

Fretz, T.A. 1973. Effects of herbicides on the rooting response of container-grown azaleas. Research Summary, Ohio Agricultural Research and Development Center 1973 Vol. 71 pp. 23-25.

Kalmowitz, K., T. Whitwell., E.I. Zehr, and J.E. Toler. 1991. Pesticides and weeds influence *Phytophthora cinnamomi* presence and growth in container-grown azaleas. HortScience 26:1428.

Skroch, W.A. and C.J. Catanzaro. 1994. Variation in sensitivity of azaleas to herbicides. Proc. International Plant Propagator's Soc. 43:395-396.

Abiotic

Non-living agents that can induce injury to plants. These include environmental, physiological, and other non-biological factors.

Any factor or stressor that is non-living (e.g., salinity, drought, temperature, etc.)

Related Glossary Terms

Biotic, Salinity, Stressor

Abscission

Process in which leaves are shed, in some cases as result of injury (e.g., exposure to excess chlorine in irrigation water).

Related Glossary Terms

Drag related terms here

Index Find Term

Alkalinity

Capacity of water to neutralize acids, expressed as the concentration of basic ions (e.g. carbonate or bicarbonate equivalents); alkaline substrates have a pH greater than 7.

Related Glossary Terms

pH

Index Find Term

Ascospores

Sexual spores contained or produced in the asci (sexual spore-bearing cells produced in ascomycete fungi, plural of ascus) of certain fungi that can serve as a source of infection for certain fungal diseases.

Related Glossary Terms

Drag related terms here

Index Find Term

Bark Cracking

Damage (i.e., cracks) in the bark of a tree due to disease, freeze damage, or some other kind of physical injury.

Related Glossary Terms

Drag related terms here

Index Find Term

Beneficial insects

Insects that help to control harmful pest populations (i.e., biological control), but can also be harmed by excessive use of insecticides.

Related Glossary Terms

Drag related terms here

Index Find Term

Biological control

Introduction of organisms such as insect predators, fungi, and nematodes, as a control method to naturally reduce harmful pest populations.

Related Glossary Terms

Drag related terms here

Index

Find Term

Biotic

Living organisms that can induce injury on plants. These include insects, pathogens, nematodes, parasitic plants, and viruses.

Related Glossary Terms

Abiotic, Stressor

Index Find Term

Blight

Sudden, conspicuous wilting and/or dying of affected plant parts, especially young, growing tissues; results from bacterial, fungal, viral, or abiotic factors.

Related Glossary Terms

Wilting

Index Find Term

Budding

Grafting technique that uses a bud as the scion wood.

Related Glossary Terms

Scion

Index Find Term

Buffer strip

Vegetated area along the edges of a field that serves as a ‘buffer’ zone to filter contaminants from runoff and to reduce erosion and sedimentation.

Related Glossary Terms

Runoff

Cambium

Meristematic tissues in the trunk/stem of plants that actively divide to increase the girth of stems by generating bark cells, wood, and vascular transport tissues (xylem and phloem).

Related Glossary Terms

Xylem vessels

Index Find Term

Canker

Necrotic, sunken disease lesion on a stem or branch.

Related Glossary Terms

Drag related terms here

Index Find Term

Chlorosis

Yellowing or bleaching of normally green plant tissue, often brought on by the loss of chlorophyll due to nutrient deficiency or disease.

Related Glossary Terms

Drag related terms here

Cleistothecia

Type of fungal fruiting body in which meiosis occurs during sexual reproduction.

Related Glossary Terms

Drag related terms here

Index Find Term

Contact pesticide

Insecticide, fungicide, or herbicide that is active primarily through direct interaction (via ingestion or dermal uptake) with a living organism.

Related Glossary Terms

Drag related terms here

Index Find Term

Controlled release fertilizer

Fertilizers that release macro- and micronutrients gradually over a specified period of time. Proprietary coatings degrade and release nutrients over time in response to irrigation frequency and temperature.

Related Glossary Terms

Drag related terms here

Index Find Term

Crawler

Juvenile stage (often restricted to the first instar) of scale insects that is mobile on the host plant.

Related Glossary Terms

Drag related terms here

Index Find Term

Cyclic irrigation

Irrigation method in which a plant's daily water allotment is divided and applied in a series of irrigation and rest intervals throughout a day.

Related Glossary Terms

Drag related terms here

Index Find Term

Cytokinin

A phytohormone that acts as a growth regulator in plants; commonly associated with cell division and differentiation.

Related Glossary Terms

Drag related terms here

Index Find Term

Degree day

Accumulated developmental units that accrue after a minimum temperature threshold (often standardized at 50 °F) is met; insect development generally does not occur below this threshold.

Related Glossary Terms

Drag related terms here

Index Find Term

Dicots

Class of flowering plants characterized by two seed leaves in the embryo.

Related Glossary Terms

Drag related terms here

Index Find Term

Directed applications

A pesticide application that is concentrated on a more specific area. Directed applications help to preserve natural enemies and use less product, as opposed to non-directed applications, which use more pesticide and can harm natural enemy populations.

Related Glossary Terms

Natural enemies

Index Find Term

Dormant oil

Oil applied during the dormant season that serves as a pesticide by suffocating insects (scales, leaf-curling aphids, etc.); oils do not require an IRAC code.

Related Glossary Terms

Drag related terms here

Index Find Term

Drought stress

Stress brought on by lack of water caused by drought which can adversely affect some plant species.

Related Glossary Terms

Drag related terms here

Index Find Term

Electrical conductivity

Measure of the salt content of water based on the flow of an electrical current; higher salt contents mean higher electrical conductivity (EC is measured in mmhos/cm or deciSiemens/m).

Related Glossary Terms

Drag related terms here

Entomopathogenic fungi

A fungal species that acts as a parasite of insects, either killing or disabling insect pests. Can be used as a biological control measure in some cases.

Related Glossary Terms

Drag related terms here

Index Find Term

Evapotranspiration

Combined water loss due to evaporation from the substrate surface and transpiration from the plant itself.

Related Glossary Terms

Drag related terms here

Index Find Term

Fertigation

Process in which water-soluble fertilizers are applied via drip irrigation.

Related Glossary Terms

Drag related terms here

Index Find Term

Flagging

Die-back on branches that is a result of cankers. Flagging can occur on one or more branches while adjacent branches appear healthy.

Related Glossary Terms

Drag related terms here

FRAC

The Fungicide Resistance Action Committee gives guidance to prolong the effectiveness of fungicides liable to encounter resistance problems and to limit crop losses should resistance appear. FRAC refers to a code that indicates the mode of action of a particular fungicide.

Related Glossary Terms

Drag related terms here

Index

Find Term

Frass

Sawdust, pelletized, and “tooth-pick” extrusions of woody tissues that are generated in response to insect tunneling or feeding activity in host plants.

Related Glossary Terms

Drag related terms here

Gall

Abnormal growth or swelling (outward) of plant stems, flowers, or roots caused by certain insects and pathogens.

Related Glossary Terms

Drag related terms here

Index Find Term

Gallery

Long passages or galleries excavated within twigs and branches in which insect pests lay eggs.

Related Glossary Terms

Drag related terms here

Index

Find Term

Genus

Delineation of the taxonomic order between ‘family’ and ‘species’; singular of genera.

Related Glossary Terms

Drag related terms here

Index Find Term

Hardy

Ability to survive the minimum average low temperature in a given area.

Related Glossary Terms

Drag related terms here

Index

Find Term

Herbicide damage

Damage resulting from the unintentional exposure to herbicides, often as a result of drift or excessive use.

Related Glossary Terms

Drag related terms here

Index Find Term

Honeydew

Carbohydrate and water solution excreted by some insects (e.g., aphids and soft scales) during feeding activity; honeydew may either drop (via gravity) or be forcefully expelled by the insect away from the host plant resource.

Related Glossary Terms

Drag related terms here

Hyphae

Branching filamentous strands of a fungus' vegetative mycelium; plural of hypha.

Related Glossary Terms

Drag related terms here

Index Find Term

Inoculum

Portions (e.g. hyphae, spores, mycelia, etc.) of bacteria, viruses, and fungi that perpetuate or propagate new infections or colonies.

Related Glossary Terms

Drag related terms here

Index

Find Term

Invasive

Any species that are not native to a particular ecosystem that can cause economic and/or environmental harm.

Related Glossary Terms

Drag related terms here

Index Find Term

IRAC

Insecticide Resistance Action Committee formed to advise on the prevention and management of insecticide resistance; IRAC refers to a code that indicates the mode of action of a particular insecticide.

Related Glossary Terms

Drag related terms here

Index

Find Term

Irrigation efficiency

Ratio of the volume of water applied to the volume of water retained by the plant. Some water losses are acceptable in proper irrigation management. The more water absorbed or retained in the container during an irrigation event, without leaching, the more efficient the irrigation system.

Related Glossary Terms

Drag related terms here

Index Find Term

Key pests

Pests that occur in damaging levels year after year at a particular site, usually consisting of a small number of insects, diseases, weeds, and nematodes.

Related Glossary Terms

Drag related terms here

Index Find Term

Larvae

Immature, wingless, and often wormlike insect form that emerges (hatches) from eggs.

Related Glossary Terms

Drag related terms here

Index Find Term

Leaching fraction

The proportion of applied water that leaches from the container after an irrigation event.

Related Glossary Terms

Drag related terms here

Index Find Term

Lenticels

Raised pores on stems, roots, or fruit in which cells are loosely arranged and appear as dots on stems; thought to function in the exchange of gases

Related Glossary Terms

Drag related terms here

Index

Find Term

Lesions

Defined areas of diseased plant tissue.

Related Glossary Terms

Drag related terms here

Index Find Term

Liners

Young transplants produced in field nurseries.

Related Glossary Terms

Drag related terms here

Index Find Term

Mature

State in which an organism (such as a plant, arthropod) has reached the end of its juvenile stage(s) and can reproduce sexually.

Related Glossary Terms

Drag related terms here

Index Find Term

Mode of action

Refers to the mechanism by which a pesticide kills or controls a particular pest organism; denoted by either FRAC or IRAC codes.

Related Glossary Terms

Drag related terms here

Index Find Term

Monocots

Class of flowering plants characterized by one seed leaf in the embryo.

Related Glossary Terms

Drag related terms here

Index Find Term

Mottling

Appearance of uneven coloration, often on leaves, which can be an indication of injury caused by malnutrition, disease, or chemical injury.

Related Glossary Terms

Drag related terms here

Index Find Term

Mycelia

Mass of vegetative fungal tissue; thread-like body of a fungus.

Related Glossary Terms

Drag related terms here

Index Find Term

Natural enemies

Refers to any naturally occurring organism (e.g., parasitic, predatory, etc.) that can help control a pest and that should be considered when implementing an IPM program.

Related Glossary Terms

Directed applications

Index Find Term

Necrosis

Death of plant tissue accompanied by dark brown discoloration.

Related Glossary Terms

Drag related terms here

Index Find Term

Nematicides

Pesticide that targets nematodes.

Related Glossary Terms

Drag related terms here

Neonicotinoids

Group of systemic insecticides that includes acetamiprid, clothianidin, dinotefuran, imidacloprid, thiacloprid, and thiamethoxam; often best used as a preventative measure against certain pests.

Related Glossary Terms

Drag related terms here

Index Find Term

Non-selective

Pesticides with a wide spectrum of control; can be damaging to non-target species.

Related Glossary Terms

Selective

Index Find Term

Nymphs

Immature form of an insect that undergoes gradual metamorphosis, which consists of a three-stage lifecycle, i.e. egg, nymph, and adult.

Related Glossary Terms

Drag related terms here

Index Find Term

Ovisacs

Egg-containing capsules.

Related Glossary Terms

Drag related terms here

Index Find Term

Parasitoids

Insect that parasitizes another organism (sometimes pests).

Related Glossary Terms

Drag related terms here

Index Find Term

Parthenogenesis

Type of asexual reproduction in which viable embryos occur without fertilization.

Related Glossary Terms

Drag related terms here

Index Find Term

Perennials

Plant with a lifecycle that lasts more than two years usually producing seed each year.

Related Glossary Terms

Drag related terms here

Index Find Term

Perithecia

Type of fungal fruiting body in which meiosis occurs during sexual reproduction.

Related Glossary Terms

Drag related terms here

Index Find Term

Pesticide resistance

Insensitivity of a pest population to a pesticide; occurs through natural selection when pest populations develop that are genetically able to detoxify a particular pesticide as a result of exposure to a single pesticide mode of action.

Related Glossary Terms

Drag related terms here

Index

Find Term

pH

Measurement, which ranges from 0 to 14, of the concentration of hydrogen ions (H+) in a solution; pH of 7 is considered neutral.

Related Glossary Terms

Alkalinity

Pheromone lures

Bait used in traps to monitor pest populations; often the synthetic version of a naturally produced chemical that attract the opposite sex.

Related Glossary Terms

Drag related terms here

Index

Find Term

Photosynthetic capacity

Capacity of a plant to convert energy from sunlight into carbohydrates.

Related Glossary Terms

Drag related terms here

Index Find Term

Phytotoxicity

Plant damage, often in reference to the extent to which a pesticide can damage a plant.

Related Glossary Terms

Drag related terms here

Index Find Term

Polyphagous

Insects that feed on multiple types of plants.

Related Glossary Terms

Drag related terms here

Index

Find Term

Postemergence

Herbicides used to control weeds after germination.

Related Glossary Terms

Drag related terms here

Index Find Term

Preemergence

Herbicides used to control weeds before germination.

Related Glossary Terms

Drag related terms here

Index Find Term

Provenance

Geographic origin of seed.

Related Glossary Terms

Drag related terms here

Index Find Term

Pupae

Structures in which insects undergo metamorphosis between the immature and mature stages of their development.

Related Glossary Terms

Drag related terms here

Index Find Term

Pycnidia

Round or flask-shaped structure containing asexual fungal spores.

Related Glossary Terms

Drag related terms here

Index Find Term

Pyrethroids

Group of insecticides that includes permethrin and bifenthrin.

Related Glossary Terms

Drag related terms here

Index Find Term

Reclaimed water

Tertiary-treated wastewater released from wastewater treatment plants. Used only for non-potable purposes, such as irrigation.

Related Glossary Terms

Drag related terms here

Index

Find Term

Resistant cultivars

Plant cultivars that are able to completely or partially exclude or overcome the effect of ra pathogen or arthropod pest.

Related Glossary Terms

Rootstock

Index

Find Term

Rootstock

Lower portion of a graft that develops into the root system.

Related Glossary Terms

Resistant cultivars

Runoff

Water lost due to irrigation or rain events that may contain nutrients, pesticide residues, and fertilizer residues.

Related Glossary Terms

Buffer strip

Index Find Term

Salinity

The presence of salt or level of salt dissolved in water; excessive salinity can cause damage to plants.

Related Glossary Terms

Abiotic, Salinity tolerance

Index Find Term

Salinity tolerance

A plant’s tolerance to the presence of salt in irrigation water or soil.

Related Glossary Terms

Salinity

Index Find Term

Scion

Shoot or bud that is grafted onto a rootstock.

Related Glossary Terms

Budding

Index Find Term

Scorch

Burned appearance on leaf margins as a result of disease, water stress, or other environmental conditions.

Related Glossary Terms

Drag related terms here

Index

Find Term

Scouting

Regular, repeated inspections of plants in the field throughout the year which serve as a way to determine which pests are present in sufficient numbers to cause economic loss and warrant pesticide application.

Related Glossary Terms

Drag related terms here

Index

Find Term

Secondary outbreak

Population growth of a pest that often develops following pesticide applications made to control a different pest.

Related Glossary Terms

Drag related terms here

Selective

Pesticides with a limited spectrum of control that target specific pests; their use implies less chance of unintentional damage of crops or natural enemies.

Related Glossary Terms

Non-selective

Index Find Term

Soilless substrate

Medium used to grow plants that does not contain mineral soil and is mostly comprised of organic materials such as pine bark and wood, as well as any number of substrate amendments such as coconut coir, compost, cotton gin waste, or vermicompost.

Related Glossary Terms

Substrate

Index Find Term

Sooty mold

Dark growth of fungal hyphae that may be found on surfaces covered with carbohydrate-rich honeydew.

Related Glossary Terms

Drag related terms here

Index Find Term

Sporulate

To form spores.

Related Glossary Terms

Drag related terms here

Index Find Term

Stippling

Patterns of small spots that occur on leaves as a symptom of insect feeding or pollution damage.

Related Glossary Terms

Drag related terms here

Index Find Term

Stomata

Pores found in the leaf epidermis through which gas exchange occurs.

Related Glossary Terms

Drag related terms here

Index Find Term

Stressor

For our purposes, biotic pests or abiotic factors that induce or cause plant stress.

Related Glossary Terms

Abiotic, Biotic

Index Find Term

Substrate

Organic and inorganic materials such as bark, peat, and sand that are used as components of planting media in containers in order to support the plant and contain its root system.

Related Glossary Terms

Soilless substrate

Index Find Term

Suckers

Vigorous shoots that originate below the soil line or graft union.

Related Glossary Terms

Union, Vigorous

Index Find Term

Sunscald

Injury to bark or cambium resulting from low temperatures, particularly a sharp or sudden decrease in temperature.

Related Glossary Terms

Drag related terms here

Index Find Term

Systemic

Insecticide, fungicide, or herbicide that is active within the plant tissue, either by root uptake or translaminar (i.e., through the leaf) absorption; moves most commonly within the xylem.

Related Glossary Terms

Translaminar, Xylem vessels

Index Find Term

Tolerant cultivars

Plant cultivars which are susceptible to disease infection, but their yield and reproduction are unaffected by disease.

Related Glossary Terms

Drag related terms here

Translaminar

Fungicides that redistribute from the sprayed surface of the leaf to the unsprayed surface, not truly systemic.

Related Glossary Terms

Systemic

Union

Point at which a scion and rootstock are joined during grafting.

Related Glossary Terms

Suckers

Vector

Any agent (person, animal, or microorganism) that carries and transmits a pathogen into plants.

Related Glossary Terms

Drag related terms here

Index Find Term

Vigorous

Healthy, actively-growing (as in the case of plants) and showing no signs of disease or weakness.

Related Glossary Terms

Suckers

Water holding capacity

Ability of container substrate to retain water.

Related Glossary Terms

Drag related terms here

Water-soaked lesions

Sunken and/or wet-looking areas on leaf or bark; often caused by bacteria.

Related Glossary Terms

Drag related terms here

Index Find Term

Wilting

Loss of turgidity of plants due to water loss; disease agents or physical injuries can block xylem vessels and impede flow of water through plants.

Related Glossary Terms

Blight, Xylem vessels

Index Find Term

Xylem vessels

Water transport system within plants.

Related Glossary Terms

Cambium, Systemic, Wilting

Index Find Term