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Postharvest Physiology and Storage of Widely Used Root and Tuber Crops

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I. INTRODUCTION

The expression "root and tuber crops" covers a wide cross-section of species with edible subterranean storage organs. Corms, bulbs, and rhizomes, though anatomically distinct, are also considered part of the group. The terms "tuber" and "tuberous root" were first used in the English language in 1668 by Wilkins in *An Essay Towards a Real Character, and a Philosophical Language* (Kays et al. 1992), although "root" was first used 518 years earlier, in 1150 (Napier 1906). Those that are commercially cultivated around the world include 38 root, 23 tuber, 14 rhizome, 11 corm, and 10 bulb crops (Kays and Silva Dias 1996), of which potato, cassava, and sweetpotato are the 4th, 6th, and 7th most important food crops in the world, collectively accounting for over 623 million t of product per year (FAO 2002).

In this review, we cover the storage of the tuber crops potato and Jerusalem artichoke; the root crops sweetpotato, carrot, and cassava; the corm crop taro; and the rhizome crop ginger. The anatomical, physiological, ontological, and biochemical differences among and within the organ types lead to wide differences among these crops in the postharvest alterations they undergo, in their maximum potential longevity, and in their optimum storage conditions (Kays 1997). Plant parts that function as subterranean reproductive organs generally contain considerable stored nutrients, and typically exhibit fairly low metabolic rates and other unique features. For example, during secondary growth, the epidermis is replaced by the periderm, which forms a protective surface tissue. Periderm formation is important, not only in the normal development of these organs, but also postharvest, during which it heals wounds incurred during and after harvest and postharvest handling, and so prevents excessive water loss and microbial infection. Several of these organs contain dormant buds that under appropriate conditions can elongate to form shoots, which may affect product quality.

II. CAUSES OF POSTHARVEST LOSSES

Harvested root and tuber crops are perishable; their high moisture content and metabolic rate lead to losses of both mass and quality. The primary causes of these losses are biotic and abiotic stresses (e.g., thermal, water, atmospheric composition, light, chemical, mechanical, pathological, herbivory), growth responses (e.g., sprouting, rooting), and quality alterations (e.g., diseases, normal metabolic processes).

A. Postharvest Stresses

1. Thermal Stress. After harvest, plant parts are susceptible to injury by exposure to high and low temperatures, and considerable research has been directed toward establishing optimum storage temperatures. Since the resources available vary widely among and within geographical areas, the best storage solution for a given crop may also vary from location to location. In this review, we address the recommended storage conditions, irrespective of possible local resource limitations, and, when appropriate, alternative storage techniques.

2. Water Stress. The rate of moisture loss is modulated by the physical and chemical characteristics of the crop and the conditions to which the product is exposed after harvest. The rate of water vapor loss from the tissue depends on the magnitude of the resistance to the diffusion of water, conferred mainly by the periderm, and the forces driving movement [i.e., primarily the magnitude of the gradient in the chemical potential of water between the product and its surrounding environment, generally expressed as the vapor pressure deficit (VPD)]. The steeper the gradient, the more rapid the water movement. In highly moist root and tuber crops, movement is almost always outward from the product. Thus, minimizing water loss depends on maintaining or increasing the resistance to diffusion and/or reducing the VPD. The diffusion resistance of periderm lenticels, areas of lower resistance, can increase during storage (Banks and Kays 1988). In addition, packaging (e.g., plastic bags) and surface coating can be effective in increasing it. The standard method for retarding postharvest moisture losses is to minimize the water VPD between the product and its storage environment, and while this can be accomplished with a high storage relative humidity, it is critical that free moisture does not form on the surface of the product.

Control of relative humidity (RH) generally involves the addition of water to the air and the precise monitoring of the air moisture content. The devices used to add moisture fall into two general classes: isothermal and adiabatic. Isothermal humidifiers use an external heat source to change water to steam, whereas adiabatic humidifiers use mechanical energy to generate a fog or mist of water droplets (Lefebvre 1989). Spinning disk humidifiers are the most commonly used adiabatic devices in potato storage rooms in the United States, Canada, and Israel (Brook et al. 1995); however, they tend to produce relatively large (i.e., 30–50 μm in diameter) droplets, which, at high RH (i.e., ≥93–94%), may promote condensation on the surface of the product, thus stimulating

pathogen activity (Hide and Lapwood 1992; Afek and Warshavsky 1998). Atomizer humidifiers, whether hydraulic (water - air) or rotary (windmill or motor driven), give smaller droplets (≤10 μm in diameter), allowing higher storage humidities.

3. Gas Stress. The atmosphere within a harvested product can strongly affect its metabolism and potential storage life. Of particular interest are the concentrations of oxygen, carbon dioxide, and ethylene. Ethylene from external sources, for example, can cause bitterness in stored carrots, rendering them unfit for consumption. Controlled atmosphere (CA) storage has been tested on some root and tuber crops, but the benefits are generally not sufficient to warrant commercial use. Of critical importance are the concentrations of the respective gases within the product. The internal concentration depends on the rate of synthesis (ethylene, carbon dioxide) or utilization (oxygen) of the gas by the product, the resistance to its diffusion into or out of the product, and the difference in partial pressure between the interior and exterior. Though CA storage is little used for root and tuber crops, the use of plastic bags or surface coatings can have a pronounced effect.

4. Radiation Stress. Visible, ultraviolet (UV) and ionizing radiations can readily alter the quality of harvested products. Visible light is particularly damaging to potato tubers because it induces the synthesis of toxic glycoalkaloids. Very low levels of diffuse light, however, are recommended during the unrefrigerated storage of certain roots and tubers.

5. Chemical Stress. Chemical stresses during storage are relatively uncommon; when they occur, it is generally because of the inappropriate application of postharvest chemical treatments or inadvertent exposure to pollutants.

6. Mechanical Stress. Root and tuber crops are subjected to a wide range of mechanical stresses during harvest, postharvest handling, and storage. Mechanical stresses that cause a physical injury represent one of the most serious causes of quality loss after harvest. Physical injuries decrease the value of the product, increase its susceptibility to diseases and water loss, and often significantly shorten its life. Such damage tends to increase at each postharvest stage (e.g., harvest, transport, sorting, storage, packaging, marketing, etc.). The three most important types of mechanical stress are friction, impact, and compression, which result in tissue failure via cleavage, slip, bruising, and buckling (Kays 1997). While mechanical stresses are a significant postharvest problem for root and tuber crops, they are beyond the scope of this review.

7. Pathogenic Stress. Large quantities of harvested root and tuber crops are lost because of a wide range of microorganisms (e.g., fungi, bacteria, viruses, mycoplasms, and nematodes), of which fungi and bacteria are the most important. These crops possess a wide array of structural and biochemical barriers to infection; for example, the periderm and surface waxes covering the organ inhibit the invasion of opportunistic pathogens, and harvest and handling procedures that break these barriers greatly increase the risk of pathogen attack during storage. Also, endogenous factors that impede invasion tend to weaken with time, increasing susceptibility to infection. Postharvest treatments or conditions that eliminate the causal organism or impede its development decrease the chances of storage rots.

8. Herbivory Stress. Serious herbivory losses during storage are relatively infrequent in root and tuber crops, except for the sweetpotato, which is attacked by the sweetpotato weevil (*Cylas* spp.) both before and during storage. The relatively high storage temperature for the crop and the pest's short life cycle makes the insect a significant problem in some geographical areas. The larvae tunnel throughout the storage roots, triggering the synthesis of a toxic fruanoterpenoid, thus presenting a food safety problem as well as compromising the quality of the product.

B. Growth Responses

During the storage of root and tuber crops, growth responses, such as the development of shoots, roots, and secondary storage organs (e.g., in cassava and Jerusalem artichoke), are generally undesirable (Hanover 1960; Wickham and Wilson 1988). Many subterranean storage organs are dormant at harvest (e.g., potato, yam, ginger, Jerusalem artichoke), and the duration of dormancy can determine the storage life of certain products. Products that do not have a dormancy mechanism or which have emerged from dormancy must be stored under conditions that impede growth. Sprouting, for example, decreases the dry matter content and also greatly increases the surface area and, consequently, the rate of water loss from the product. As a consequence, a number of treatments have been developed to extend dormancy and/or prevent the resumption of growth. When the storage organ is to be used as a reproductive propagule, however, sprouting is desirable and conditions and/or treatments to facilitate dormancy breaking or growth stimulation are used.

C. Quality Alterations

Most of the quality alterations that root and tuber crops undergo during storage are undesirable. They include disease development and chemical changes associated with normal metabolism (e.g., depletion of stored carbohydrates). Quality alterations are not critiqued specifically in this review but, when relevant, they are addressed under the appropriate topic heading (e.g., disorders, chemical changes).

III. TUBER CROPS

A. Potato

The potato (*Solanum tuberosum* L., Solanaceae) is the fourth most important food crop grown worldwide, with a total yield of over 308 million tonnes (t) in 2001 (FAO 2002). While predominately a temperate crop, potato is also grown in hot climates (Cargill et al. 1989, Afek and Warshavsky 1998). In most production areas, harvested tubers are stored, extending their availability for up to 10 months (Burton et al. 1992). The maximum storage duration is often determined by the propensity to sprout, which varies widely among cultivars. Tuber dormancy, therefore, is a critical component of storage potential. The value of the crop is diminished by losses in fresh weight and desirable quality attributes, losses mainly caused by respiration, disease, evaporation, mechanical damage, sprouting, chemical changes in quality attributes (e.g., color), and damage caused by abiotic stresses (e.g., temperature, gas atmosphere, light) (Es and Hartmans 1987a,b; Meijers 1987a; Rastovski 1987, Burton et al. 1992).

1. Prestorage Treatments. Potato tubers sustain mechanical damage during harvesting and transport, and often during grading and marketing after storage. Injury to the surface periderm not only accelerates moisture loss but exposes the interior of the tubers to opportunistic pathogens that cause dry and soft rots during storage. Curing is a prestorage treatment used for a number of subterranean organs where the harvested product is exposed to temperature and RH conditions that facilitate wound healing (Kays 1997). The precise conditions and their duration vary depending upon the crop in question. Under favorable curing conditions (e.g., 2 weeks at 12–16°C, 90–92% RH), potato tuber tissue forms a protective layer (wound periderm) over the damaged area (Meijers 1987b; Cargill et al. 1989; Burton et al. 1992). A thin layer of suberized

cells is differentiated over the surface of the damaged area, followed by deposition of cork cambium (phellogen) under the sealing layer, giving rise to a densely packed network of new cells with little intercellular space. Curing is essential to limit weight loss and to prevent the penetration of microorganisms, and the temporal relationship between wound healing and microorganism penetration is critical. Tubers are routinely cured on entry into storage, a key feature of successful storage (Meijers 1987b). Sprout inhibitors and treatments to inhibit microbes, also applied prior to storage, are detailed in the Dormancy and the Postharvest Pathology sections, respectively.

2. Recommended Storage Conditions. Recommended conditions vary according to the type of potato (early-crop vs. main-crop), cultivar, intended use, and the storage facilities available (heated, refrigerated, or nonrefrigerated). After curing, potatoes are stored in sacks, pallet boxes, bulk bins or piles, depending upon the facility and the volume being handled. A major part of the potato crop in the U.S. is held in nonrefrigerated, naturally ventilated storage houses, generally in piles or bins 2.5 to 6 m deep (Sparks et al. 1968; Sparks and Summers 1974). Optimum storage conditions are similar to those in refrigerated storage, but the control is less precise. With refrigerated storage, main-crop and seed potatoes are stored at 3.5–4.5°C (4°C is considered optimum) and 90–95% RH; however, certain cultivars are best held at higher temperatures [e.g., 7°C for 'Russett Burbank' (Cunningham, et al. 1971), 10°C for 'Kathahdin' and 'Kennebec' (Sawyer et al. 1965)]. Early-crop potatoes are seldom stored for long, but can be held for up to 4–5 months at 4°C. Main-crop potatoes for chips (crisps) and French fries (chips) are stored at 10–13°C, 90–95% RH. For processing, it is essential that the reducing sugar content be low; if it is too high, the tubers are held at 18–20°C for 1–4 weeks until an appropriate level is reached. Ventilation is critical for heat removal and uniform gas distribution. Generally air flow rates of 0.3–0.4 $m^3 \cdot min^{-1} \cdot t^{-1}$ are recommended; however, in the midwest of the U.S., 0.6–0.7 $m^3 \cdot min^{-1} \cdot t^{-1}$ is recommended for table stock and 0.8–1.0 $m^3 \cdot min^{-1} \cdot t^{-1}$ for seed stock (Smith 1977).

3. Water Loss. Water loss during storage not only reduces the total mass of product and, therefore, its value (Rees et al. 1981; Es and Hartmans 1987a,b; Rastovski 1987; Brook et al. 1995), it is also a primary cause of quality loss (Villa and Bakker-Arkema 1974). Significant moisture loss renders tubers more susceptible to mechanical damage (e.g., bruising) and discoloration, and typically increases peeling losses (Rastovski 1987). The greater the water potential difference or water VPD, the more rapidly

moisture is lost from the tubers (Es and Hartmans 1987c; Kleinkopf 1995). The periderm is the primary barrier to diffusion loss, and localized areas of lower resistance, such as lenticels, facilitate water loss. The diffusion resistance of the lenticels, however, often increases during storage (Banks and Kays 1988). While resistance primarily depends on the anatomical structure of the tubers, moisture loss is reduced by minimizing the water VPD between the tubers and their storage environment by means of a high storage RH (Es and Hartmans 1987c; Burton et al. 1992). The importance of RH is illustrated by the difference in weight loss between tubers ('Desiree') stored for 6 months at 96–98% or at 82–86% RH, i.e., 2 and 7%, respectively (Afek et al. 2000b).

Lowering storage temperature also decreases moisture loss, though temperature fluctuations can exacerbate losses through their effect on the RH and VPD of the storage environment. The recommended storage temperature depends upon the type and/or use of the potatoes (Cargill et al. 1989; Burton et al. 1992).

4. Dormancy and Sprouting Prevention. Dormancy, a period of suspended growth (Emilsson 1949; Burton 1963; Goodwin 1966; Harkett, 1981; Hemberg 1985), often determines the maximum storage time of potato tubers, but postharvest handling and storage conditions may also be important (Espen et al. 1999a,b). In general, potato tubers are dormant at harvest and, in the absence of dormancy-breaking treatments, remain so pending fulfillment of the normal dormancy requirement, when sprouting occurs spontaneously. Sometimes sprouting can be suppressed, as described below. Thus, maintenance or extension of dormancy is an integral feature of storage procedures.

The dormancy duration depends upon the cultivar, tuber maturity, and soil and weather conditions during growth (Krijthe 1962; Burton 1963). Unusually cold, wet weather can extend dormancy by about 4 weeks, whereas extremely dry, warm conditions can reduce it by up to 9 weeks (Burton 1966).

Sprouting impairs the quality of the tuber in that water and other constituents are derived from the tubers for the new cells in the sprouts, which in turn are discarded prior to use. Sprouting also increases the surface area and reduces water diffusion resistance, accelerating weight loss.

The primary methods for inhibiting sprouting during storage are the use of low temperatures (i.e., 2 to 4°C) and sprouting suppressants (Rastovski 1987). However, low temperatures can accelerate the conversion of starch to sugar (Rees et al. 1981; Morrell and Rees 1986b; Es and Hartmans 1987a; Ross and Davies 1992), thus reducing tuber quality

(Rastovski 1987). Sprouting inhibitors found effective in commercial use include isopropyl N-(3-chlorophenyl) carbamate (CIPC), propham (IPC), maleic hydrazide (MH), and tecnazene (TCNB) (Hajslova and Davidek 1986; Buitelaar 1987; Es and Hartmans 1987b; Yada et al. 1991); ethylene, morphactin, nonanol, abscisic acid, and indoleacetic acid, although effective, have not been adopted (Hartmans and van Es 1979; Wang et al. 1980; Beveridge et al. 1981; Rama and Narasimham 1986; Es and Hartmans 1987b). More recently, carvone has proved an efficacious sprouting inhibitor (Oosterhaven et al. 1995; Wiltshire and Cobb 1996; Sorce et al. 1997).

There are problems, however, with some sprout inhibitors. IPC and CIPC suppress suberization and the formation of wound periderm, and therefore should be used only after curing (Edgar 1968; Es and Hartmans 1987b). MH is applied as a foliar spray 4 to 6 weeks before harvest, but too early application reduces yield, while late applications do not inhibit sprouting adequately (Es and Hartmans 1987b; Yada et al. 1991). TCNB is not effective if dormancy has already been broken, the storage room is excessively ventilated, or the storage temperature is above 10°C (Es and Hartmans 1987b). Additionally, there are current or pending restrictions on the use of these compounds in several countries (Lewis et al. 1997; Afek and Warshavsky 1998).

Several other compounds have proved to inhibit sprouting effectively during storage. Hydrogen peroxide (HPP) (stabilized with a mixture of substances whose patent is pending), applied with an atomizing fogger to control postharvest pathogens, was found to inhibit sprouting; after 6 months at 10°C, there were no sprouts longer than 2 mm on potatoes treated with 10% HPP at 5-week intervals (Table 7.1) or with CIPC (Afek et al. 2000a). In untreated controls, 84% of the tubers had sprouted.

The effects of dimethylnaphthalene (DMN) and diisopropylnaphthalene (DIPN) on the sprouting of 'Russet Burbank' tubers were compared with those of CIPC (Lewis et al. 1997). Two applications of DIPN (300 $mg \cdot kg^{-1}$ as a.i. in f.w.) were as effective as one application of CIPC (22 $mg \cdot kg^{-1}$ as a.i. in f.w.) but DMN was less effective than DIPN or CIPC. After 178 days of storage, only tubers treated with CIPC and DIPN were devoid of sprouts, and after 295 days, the average tuber sprout lengths were 8 and 14 mm, respectively. In another study (Kalt et al. 1999), carvone, dimethylnaphthalene (DMN), and ethylene were compared with the commercial standard, CIPC, as sprout inhibitors. After 25 weeks at 9°C, total sprout weight and maximum sprout length were ranked CIPC $\leq$ carvone < ethylene < DMN; neither CIPC- nor carvone-treated tubers had sprouts. Oosterhaven et al. (1995) had previously demonstrated that carvone inhibited sprouting ('Bintje') and that inhibition took effect

Table 7.1. Sprouting of 'Desiree' potato tubers (%) during 6 months of storage at 10°C and 95% RH, following treatment with hydrogen peroxide (HPP) (four applications), isopropyl N-(3-chlorophenyl) carbamate (CIPC), or control. The hydrogen peroxide is stabilized with additional substances (the composition of which is currently patent pending) and applied with an atomizing fogger (Afek et al. 2000a).

	Sprouting (%)					
	Storage period in months					
Treatment	1	2	3	4	5	6
HPP	0 a[z]	0 a	0 a	0 a	0 a	0 a
CIPC	0 a	0 a	0 a	0 a	0 a	0 a
Control	8 b	26 b	48 b	63 b	74 b	84 b

[z]Means separated by Fisher's protected least significant difference test, (5% level).

immediately after treatment. Daniels-Lake et al. (1996) compared the effects of ozone, 1,8-cineole, and CIPC on sprouting ('Russet Burbank') over 25 weeks at 9°C and found that CIPC and 1,8-cineole completely inhibited sprouting, whereas ozone had little effect.

Several essential oils derived from aromatic plants such as *Lavandula angustifolia* Miller (lavender), *Mentha pulegium* L. (mint), *Mentha spicata* L. (spearmint), *Origanum onites* L. (Turkish oregano), *Origanum vulgare* L. ssp. *hirtum* (Greek oregano), *Rosmarinus officinalis* L. (rosemary), and *Salvia fruticosa* Miller (sage) were assessed as sprout inhibitors for 'Spunta' potato (Vokou et al. 1993). Except for oregano oil, all suppressed sprout growth; however, lavender, sage, and rosemary oils were more effective. After 5 weeks of storage, lavender oil was the most effective of these in suppressing sprout emergence and elongation (i.e., ≈90%) while the other oils inhibited elongation by 72–83% (Fig. 7.1).

Rama and Narasimham (1986) tested hot-water-dip and vapor-heat treatments at temperatures ranging from 50–80°C and 60–70°C, respectively, as a means of inhibiting sprouting in stored potatoes. Hot water, at temperatures and durations sufficient to inhibit sprouting, damaged the tubers, whereas vapor treatment (60°C, 95% RH for 60 min) suppressed sprouting for 3 weeks at ambient conditions (22–35°C, 50–80% RH) without any deleterious side effects. If the treatment was repeated after 3 weeks, sprouting was suppressed for an additional 3 weeks.

5. Sprout Induction. Breaking tuber dormancy is often desirable to accelerate and synchronize seed potato stand establishment. Many studies

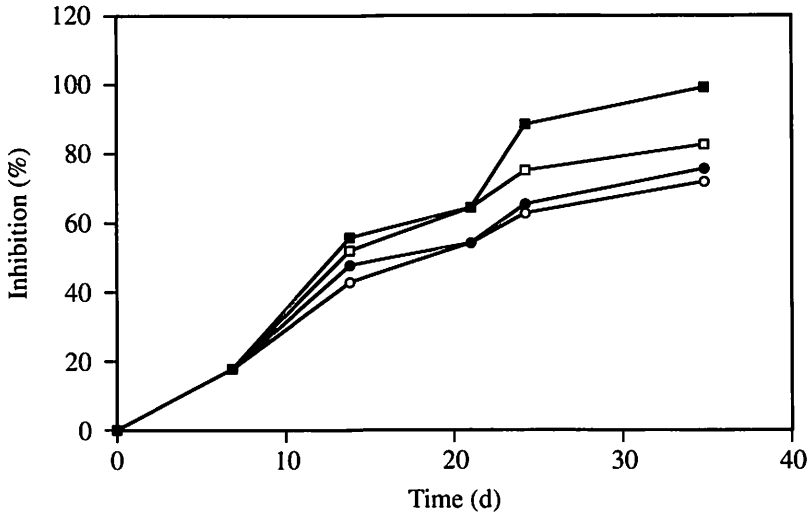

Fig. 7.1. Percentage inhibition in the length of potato tuber sprouts after exposure to the volatile essential oils (1.0 ml · 4 l^{-1} air) of *Lavandula angustifolia* (■), *Salvia fruticosa* (□), *Mentha pulegiuum* (●), and *Mentha spicata* (○) (after Vokou et al. 1993).

have addressed high-CO_2 and/or low-O_2 stress as dormancy breakers (Thornton 1939a,b; Burton 1958, 1968; Goodwin 1966; Burton and Wiggington 1970; Coleman 1987; Esashi 1991; Coleman et al. 1992; Wiltshire and Cobb 1996). For example, exposure of 'Russet Burbank' tubers to CO_2 : O_2 ratios of 20:40% and 60:20% for 7 days facilitated dormancy release and sprout emergence, an effect that was further enhanced by ethylene at 50 µl · l^{-1} (Fig. 7.2) (Coleman and McInerney 1997).

In light of the effects of CO_2 and O_2 on endogenous ethylene synthesis (Esashi 1991), the effect on tuber dormancy release (Rylski et al. 1974) may be related to increased synthesis of aminocyclopropane-l-carboxylic acid and subsequently the phytohormone (Esashi 1991; Mattoo and White 1991; Smith and John 1993). The elapsed time from harvest and the storage temperature were found to be important in dormancy release by CO_2 and O_2 (Coleman 1998). Previous research had also implicated abscisic acid (ABA) in tuber dormancy induction and maintenance (Hemberg 1985; Suttle and Hultstrand 1994; Suttle 1995). Altered CO_2 - O_2 environments also resulted in changes in endogenous ABA levels within the tubers, as did ethylene treatment (Fig. 7.3).

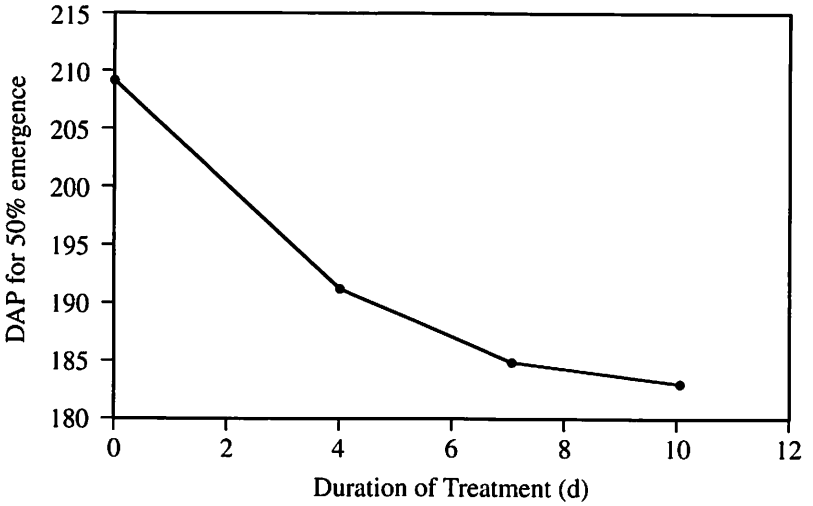

Fig. 7.2. The effect of exposure to 20% CO_2 + 40% O_2 + ethylene at 50 $\mu l \cdot l^{-1}$ on the number of days after planting (DAP) to reach 50% shoot emergence in field-grown 'Kennebec' potato tubers (after Coleman and McInerney 1997).

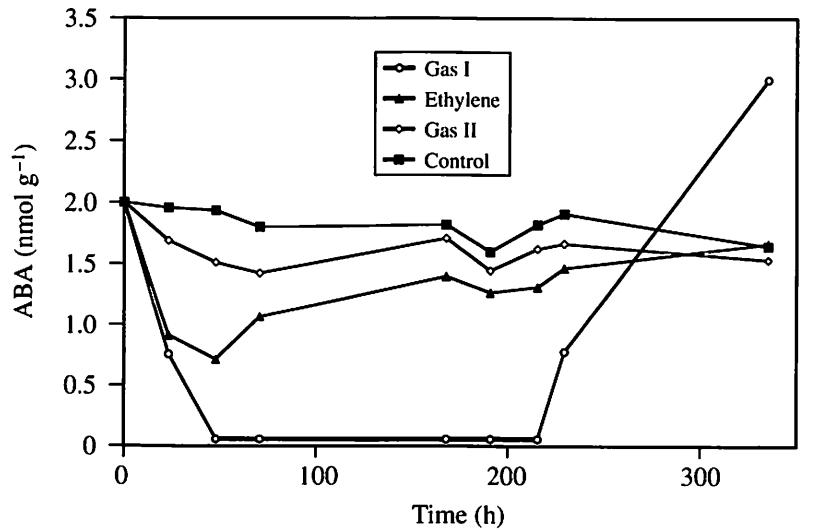

Fig. 7.3. Time course of changes in abscisic acid content on a dry weight basis in the apical eye region of 'Russet Burbank' potato tubers from the late dormancy phase (150–220 DAP), removed from 13°C storage and exposed for 7 d to Gas I (60% CO_2 + 20% O_2), Gas II (20% CO_2 + 40% O_2), C_2H_4 at 1.74 $\mu mol \cdot 1^{-1}$, or air controls. Treatments began 162 days after planting (DAP) and continued for 168 h (after Coleman 1998).

6. Disorders

Greening. Chlorophyll synthesis is triggered when potato tubers are exposed to light after harvest. Light-skinned cultivars tend to be more susceptible than russet types. The green coloration of the skin and underlying flesh is not in itself harmful, but there can be a concurrent synthesis and accumulation of glycoalkaloids (predominately solanine) that are toxic in large quantities (Jadhav and Salunkhe 1975). Greening is prevented by minimizing the exposure to light throughout the postharvest period.

Blackheart. Tubers with blackheart display a blackened center from which cavities may eventually develop. The disorder is caused by lack of oxygen within the tuber, a condition that is exacerbated by elevated temperatures (Stewart and Mix 1917). Control measures include maintenance of adequate ventilation and appropriate storage temperatures (Snowdon 1992).

Mahogany Browning. Exposure of certain cultivars to low temperatures (0°C for 20 weeks or longer) causes an irregular reddish-brown discoloration within the tubers (Hilborn and Bonde 1942).

Stem-End Browning. A dark brown or black discoloration develops at the stem end of the tubers of certain cultivars (Ramsey et al. 1949); it is thought to be related to storage temperature. Tubers can be temperature conditioned (e.g., 0°C for 60 days) as a means of control.

7. Postharvest Pathology. During storage, potatoes may be affected by fungal or bacterial diseases that can spread readily under favorable conditions. Initial infection or inoculation almost always occurs in the field and the organism is then brought into storage. The most common fungal diseases of stored potatoes are early blight—*Alternaria solani* (Ell and Mart.) Sol.; gray mold—*Botrytis cinerea* Fr; black rot—*Colletotrichum coccodes* (Waller.) Hughes; dry rot—*Fusarium* spp.; silver scurf—*Helminthosporium solani* Dur. & Mont.; gangrene—*Phoma* spp.; soft rot—*Phythium* spp.; potato blight—*Phytophthora infestans* (Mont.) De Bary; black scurf—*Rhizoctonia solani* Kuhn; powdery scab—*Spongospora subterranae* (Waller.) Laregh; and common scab—*Streptomyces scabies* (Thaxter) Waksman & Henrici. Bacterial diseases include ring rot—*Corynebacterium sepedonicum*; soft rot—*Erwinia carotovora* subsp. *carotovora* (Jones) Bergey et al.; soft rot—*Erwinia carotovora* subsp. *atroseptica* (van Hall) Dye, Blackleg; and brown rot—*Pseudomonas*

solanacearum (E.F. Sm.) E. F. Sm. (Perombelon et al. 1987a,b; Hide and Lapwood 1992; Tsror et al. 1993, 1999; Afek and Warshavsky 1998). *Erwinia carotovora* (soft rot), *Helminthosporium solani* (silver scurf), and *Fusarium* spp. (dry rot) are particularly serious postharvest pathogens that can readily spread in storage, causing severe losses (Rich 1968; Meijers 1987a; Hide and Lapwood 1992; Wastie et al. 1994). Lack of effective chemical controls exacerbates the problem within cold storage rooms, but control of *E. carotovora* and *H. solani* has been achieved with a fog of hydrogen peroxide containing a stabilizing component (HPP) (Afek et al. 1999a, 2001). Following disinfection, the decay of 'Desiree' tubers caused by *E. carotovora* after 5 months of storage was 4% compared with 26% in control tubers (Table 7.2). Decay caused by *H. solani* after 6 months was reduced to 2% following five monthly treatments with a 10% HPP fog, compared with 38% rot in the non-treated control. A single treatment reduced decay from 38% to 16%; however, multiple treatments were decidedly superior (Table 7.3) (Afek et al. 2001). Hydrogen peroxide is a relatively environmentally safe disinfectant, thought to act through oxidative damage to the fungi and bacteria.

The lack of effective chemicals impedes control of *Fusarium* dry rot in stored tubers. However, several means of biological control have been found effective in laboratory studies. They were based on bacterial antagonists (Kiewnick and Jacobsen 1997; Schisler and Slininger 1994; Schisler et al. 1997), yeasts (Schisler et al. 1995), and arbuscular mycorrhizae (Niemira et al. 1996). In commercial-scale storage experiments, Schisler et al. (2000) found that *Pseudomonas fluorescens* S22:T.04 decreased dry rot in pathogen-coinoculated tubers by 19%, compared

Table 7.2. Percentage of 'Desiree' potato tubers with decay, either with (+) or without (–) *Erwinia carotovora* inoculation, and with (+) or without (–) subsequent treatments with 10% hydrogen peroxide (HPP) during 5 months of storage at 8°C and 95% RH. The hydrogen peroxide is stabilized with additional substances (the composition of which is currently patent pending) and applied with an atomizing fogger (Afek et al. 1999a).

Treatment		Decay (%)				
		Storage period in months				
Inoc	HPP	1	2	3	4	5
+	+	1 b[z]	2 b	2 b	3 b	4 b
+	–	4 c	7 c	12 d	18 d	26 d
–	+	0 a	0 a	0 a	1 a	1 a
–	–	2 b	3 b	6 c	10 c	15 c

[z]Means separated by Fisher's protected least significant difference test, (5% level).

Table 7.3. Percentage decay of 'Desiree' potato tubers caused by *Helminthosporium solani* following 0, 1, or 5 treatments with 10% hydrogen peroxide (HPP) during 6 months of storage at 9°C and 95% RH. The hydrogen peroxide was stabilized with additional substances (the composition of which is currently patent pending) and applied with an atomizing fogger (Afek et al. 2001).

	Decay (%)					
	Storage period in months					
No. treatments	1	2	3	4	5	6
5	0 a[z]	0 a	1 a	1 a	2 a	2 a
1	0 a	3 b	7 b	10 b	14 b	16 b
0	5 b	9 c	16 c	24 c	31 c	38 c

[z]Means separated by Fisher's protected least significant difference test, (5% level).

with controls or the fungicide thiabendazole (TBZ); in subsequent tests, *P. fluorescens* P22.Y:05 and *Enterobacter cloacae* SI I:T:07 reduced the severity of the disease by 25 and 17%, respectively, when the antagonists were applied after the pathogen inoculum.

Prestorage exposure to ultraviolet (UV) irradiation suppressed diseases in potatoes stored for 3 months at 8°C (Ranganna et al. 1997). Dry rot caused by *Fusarium solani* in tubers inoculated with conidia and incubated at 28°C for 24 h was completely suppressed at a UV dose of 15 kJ · m^{-2}. Soft rot caused by *Erwinia carotovora* was suppressed by a UV dose of 15 kJ · m^{-2} after the tubers had been inoculated with the bacterium and incubated at 37°C for 6 h. When inoculated tubers were not incubated before the UV treatment, dry rot was completely suppressed at only 12.5 kJ · m^{-2}, but soft rot was not.

Prestorage steam treatment has also been found effective in reducing the incidence of various pathogens in stored seed tubers (i.e., 2–3% vs. 35–52% in untreated controls) (Afek and Orenstein 2002). In addition, the presence of pathogens in the daughter tubers 120 days post-planting was only 3–4% in steam-treated tubers compared with 26–31% in the untreated controls.

8. Chemical Changes During Storage. Depending upon the cultivar, mature tubers contain 16–23% dry matter, of which starch is the predominant component (~70%) (Burton 1966; Augustin 1975). Potato starch is comprised of 21–25% amylose and 75–79% amylopectin. The rate of starch hydrolysis during storage has a critical impact on tuber quality. Also present are sucrose, glucose, fructose, and substantially smaller amounts of several other sugars. The reducing sugar content is

important in potatoes that are to be fried (Es and Hartmans 1987a; Burton et al. 1992); tubers that are high in reducing sugars (principally glucose and fructose) darken because of the reaction of aldehyde groups with amino groups of amino acids or other sources (Schaaf 1974), and intense discoloration may reduce product marketability. Typically 2.5 to 3 mg of reducing sugar per gram fresh wt is the maximum permissible level for potato chips (crisps) and ~5 mg per gram for French fries.

During storage, starch is progressively hydrolyzed to provide energy and carbon skeletons for maintenance and synthetic reactions. Under certain conditions, there is a pronounced increase in hydrolysis and formation of reducing sugars, the amount of which varies with cultivar, tuber age, injury, decay, moisture status, sprouting, and exposure to temperature extremes (Burton and Wilson 1978; Burton et al. 1992; Espen et al. 1999a,b). Tuber response to low temperatures depends on its initial metabolic status. Storage of potatoes at 0–5°C increases the sugar concentration and may damage membranes (Workman et al. 1976); storage at 25–36°C also results in a significant increase in sugar, probably because of the onset of sprouting (Ludwig 1970; Verma et al. 1974a,b). Storage temperature does not appear to affect the amino acids concentration (Fig. 7.4). Most stress factors that disturb the metabolic

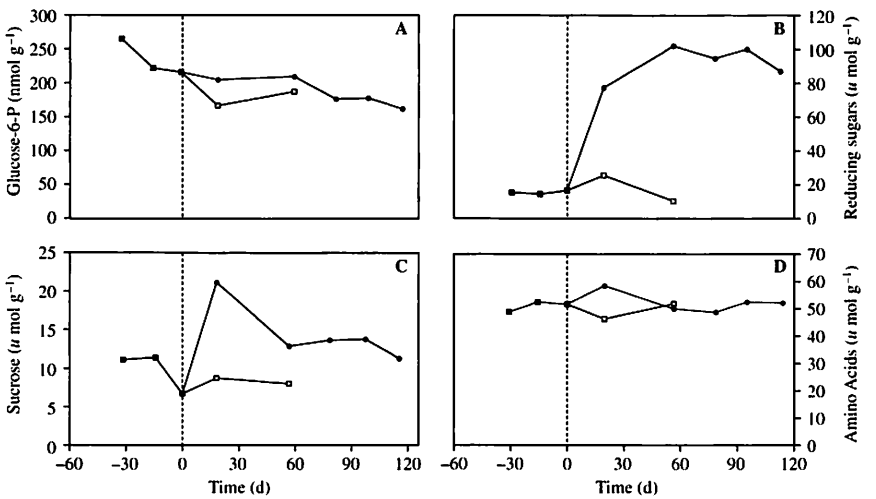

Fig. 7.4. Changes in the concentrations of glucose-6-phosphate (A), reducing sugars (B), sucrose (C), and amino acids (D) on a fresh weight basis in the parenchymatic tissue of potato tubers before normal harvest (■) and in tubers stored at 23°C (□) or 3°C (●) for various periods. Time scale is in days before or after normal harvest (after Espen et al. 1999a,b).

equilibrium in the tubers appear to increase the sugar concentration (Burton 1969; Samotus et al. 1974a,b; Sowokinos 1973; Vliet and Schriemer 1960). Under appropriate storage temperature conditions, the sugar concentration remains relatively constant for an interval after harvest and then increases, at first slowly and finally very sharply. Sugar formation during senescence (i.e., "senescence sweetening") (Burton 1966) is associated with a decline in the condition of the cellular membranes and differs from sugar increases caused by low temperatures; the latter can be remedied by reconditioning (Isherwood 1973, 1976; Ohad et al. 1971), whereas age-induced sugar formation is not reversible and reconditioning is ineffective (Isherwood 1976).

When potato tubers are stored at or below 10°C, they accumulate sugars, primarily glucose, fructose (reducing sugars), and sucrose (Sowokinos 1990) through "cold sweetening." Reducing sugars accumulate because of up-regulation of the gene encoding sucrose phosphate synthase (SPS) (Hill et al. 1996). Along with the activation of sucrose synthesis, de novo amylolytic activity induced in the cold-stored tubers accelerates amylopectin degradation enhancing sweetening (Isherwood 1976).

Starch-degrading enzymes reported in potato tubers include α-amylase, isoamylase, β-amylase, α-glucosidase, and starch phosphorylase (Fan 1975; Gerbrandy et al. 1975; Kennedy and Isherwood 1975; Killilea and Clancy 1978; Schneider et al. 1981; Cochrane et al. 1991a,b; Cottrell et al. 1993). However, it is not clear whether the degradation is primarily amylolytic or phosphorolytic; Morrell and Rees (1986a) concluded that it was phosphorolytic because they detected no amylase activity in the tubers, but several other authors (Sowokinos et al. 1985; Cochrane et al. 1991b; Cottrell et al. 1993) have detected substantial activity.

Low storage temperatures result in increased α-amylase, β-amylase (Cochrane et al. 1991b; Cottrel et al. 1993; Nielsen et al. 1997) and phosphorylase (Claassen et al. 1993) activities, although the latter has not been universally found (Kennedy and Isherwood 1975; Hill et al. 1996). Alpha-glycosidase activity was not influenced (Nielsen et al. 1997), whereas β-amylase activity increased as the temperature decreased from 5 to 3°C; the increase began by day 3 and the activity had increased four- to fivefold within 10 days.

The onset of sugar accumulation coincides with activation of SPS and the appearance of a new form of amylase (Hill et al. 1996; Nielsen et al. 1997). After 10 days, accumulation of sugars was negligible at 11, 9, or 7°C, but considerable at 5 and 3°C (Fig. 7.5). Total starch hydrolysing enzyme activity was unaltered at 7°C, increased at 5°C, and increased further (i.e., up to seven- to eightfold) at 3°C. The increase in amylolytic activity correlated with the appearance of a new amylase,

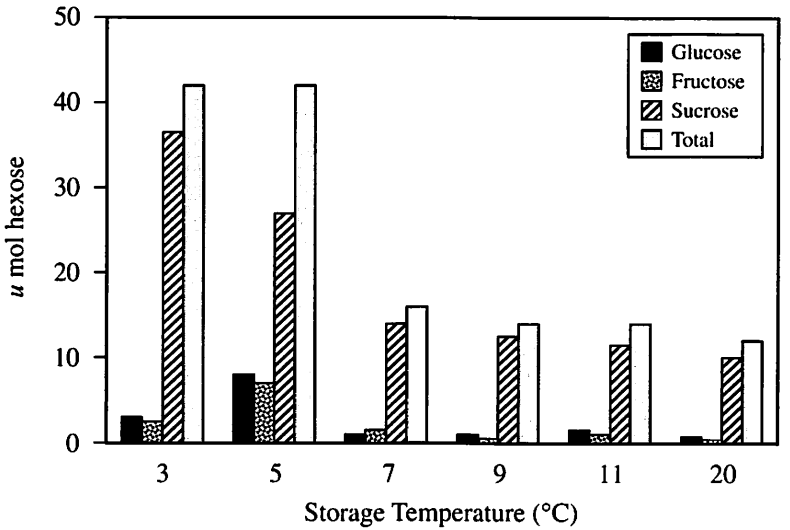

Fig. 7.5. Sugar concentration in potato tubers after storage at various temperatures. Tubers were harvested and stored for 4 wk at 20°C and then held at 11, 9, 7, 5, or 3°C for 10 d (after Deiting et al. 1998).

while increases in acid invertase activity did not correlate with increased sugar concentration.

Cold-induced changes in SPS and amylase are reversed if the tubers are returned to a higher temperature (Deiting et al. 1998). SPS and invertase increased 2.2- and 7.7-fold, respectively, during 28 days at 1°C (Charmara et al. 1998). Sucrose synthase (SS) activity remained constant at 1°C and was similar to that in tubers kept continuously at 10°C. With the transfer of tubers from 1 to 10°C, there was a sharp rise in respiration, which peaked at day 7, and then declined. During reconditioning, sucrose declined rapidly, while glucose and fructose declined more slowly. Sucrose synthase activity increased sharply during 7 days at 10°C and both SPS and invertase decreased. Low O_2 inhibited the decrease in sugar content and suppressed the rise in SS activity, but it did not alter the decreases in SPS and invertase activities.

B. Jerusalem Artichoke

Jerusalem artichoke (*Helianthus tuberosus* L., Asteraceae) is a perennial that is grown as an annual. International production statistics are not collected, but it is a significant crop in some Eastern European countries.

The aerial plant parts die in the early winter, at which time the tubers are harvested. There are many cultivars with selection depending upon latitude and other factors (McCarter and Kays 1984). Increased interest in the crop has stemmed from the fact that carbon is stored in the Jerusalem artichoke as inulin, a straight chain fructan that is minimally digested by humans and can be used as a bulking agent in foods in which sugar is replaced with an artificial sweetener. Thus, the volume previously occupied by sugar is filled with an extremely low-calorie material, greatly reducing the total caloric content of the product. With little reformulation, inulin, though not sweet, functions similarly to sugar (i.e., browning reactions, aroma synthesis, textural properties) in many foods. Also, inulin, whether ingested in Jerusalem artichoke tubers or as a bulking agent, is a dietary fiber and confers a number of health benefits: for example, it lowers blood cholesterol level; promotes Bifido bacteria in the large intestine; reduces the blood levels of sugar, low-density lipoproteins, and triglycerides; and is beneficial to certain heart diseases (Farnworth 1993; Hirayama and Hidaka 1993; Sakun et al. 1996; Varlamova et al. 1996).

Tuber size and shape are critical attributes that are strongly modulated by cultivar and production conditions. Many clones have an irregular and undesirable tuber surface topography due to branching, an objectionable trait. The tubers are harvested in the late fall, generally after the first frost. In production areas where the crop can be harvested throughout the winter, it can be field stored until needed. Elsewhere, harvest is followed by cold storage.

1. Prestorage Treatments. We found no references to prestorage treatments. The tubers are typically not cleaned before storage unless they are to be held under refrigeration and have significant amounts of adhering soil. The surface of the tubers should be devoid of free water to discourage the growth of pathogens.

2. Recommended Storage Conditions. The three primary storage options are refrigerated storage, common storage [e.g., root cellars, clamps or pits (Shoemaker 1927) which utilize natural cooling], and in situ field storage (Sibley 1924; Cormany 1928). Cold storage is highly effective but expensive; nevertheless, it is routinely used for seed and fresh-market tubers, especially where field storage is not practicable. Root cellars, clamps, and pits are used when the tubers must be harvested in the fall, before the ground freezes or other adverse conditions set in, and where refrigeration is not available or is too expensive.

Field storage is a viable option in northern hemisphere locations that are sufficiently far north to ensure cold soil throughout the winter, but not so far that the surface of the soil freezes solid and prevents harvesting. Another requisite is relatively sandy, well-drained soils that allow the use of harvesting machinery throughout the winter. Locations that have a maximum number of potential harvest days are preferable, in that they increase harvest continuity. Locations that do not meet these criteria generally require the use of refrigerated or some other form of indoor storage.

Jerusalem artichoke tubers can be stored for 6–12 months at 0 to 2°C and under a high RH (90–95%). Some cultivars are much more susceptible to storage losses than others (Steinbauer 1932). At low humidities, the tubers shrivel readily and are more likely to decay than if kept in a moist atmosphere. Tubers have relatively low respiratory rates when held at low temperatures (e.g., 10.2 mg $CO_2 \cdot kg^{-1} \cdot h^{-1}$ at 0°C), but they produce heat (vital or respiratory heat) (e.g., 111 $J \cdot kg^{-1} \cdot h^{-1}$ at 0°C) that must be removed if the desired low temperature is to be maintained (Peiris et al. 1997).

The tubers can withstand low temperatures but they freeze below –2.2°C (Whiteman 1957). Freezing inflicts little damage above –10°C, but rapid deterioration occurs at lower temperatures; significant chemical and physical alterations occur in the plasma membrane, most notably losses in sterols and phosphatidylethanolamine (Uemura and Yoshida 1986) with a concurrent loss of membrane function. As with most fleshy plant products, the temperature at which freeze damage occurs and the extent of the damage vary with cultivar, season, preconditioning, rate of freezing, and so on (Kays 1997).

The potential of controlled atmosphere storage has not been adequately assessed. It has been shown to impede inulin depolymerization, apparently through an effect on enzyme activity. Storage of tubers in a 22.5% CO_2/20% O_2 atmosphere significantly retarded inulin degradation (Denny et al. 1944).

Gamma irradiation of Jerusalem artichoke tubers greatly accelerates inulin depolymerization (Salunkhe 1959) and it offers no known advantage to offset this disadvantage.

3. Water Loss. While desiccation losses can be relatively easily prevented with proper storage conditions, they remain a significant storage problem, because the tubers lack a surface layer of corky cells with a high water diffusion resistance (Decaisne 1880). In addition, their surface cells can be readily injured, which facilitates desiccation (Traub et al.

1929a). The thin periderm causes the tubers to lose moisture at a rate depending on the water VPD. Therefore, storage at a high RH (90–95%) is essential (Shoemaker 1927; Traub et al. 1929a; Johnson 1931; Steinbauer 1932).

4. Dormancy and Sprouting. The dormancy period of Jerusalem artichoke tubers varies among cultivars and even among the tubers from a single plant. The dormancy mechanism within the tubers responds to an environmental signal, in this case low temperatures. Sufficient exposure duration to the appropriate temperature (i.e., below a specific maximum and generally near 0°C) fulfills the dormancy requirement, and under appropriate environmental conditions, the cells within the tuber begin to divide and the tuber sprouts.

Tuber dormancy also has significant agricultural implications. Production of the crop in regions where the cold period does not fulfill the dormancy requirement can be greatly impaired by the lack of or uneven sprouting in the spring; also, insufficient cold exposure can lead to spotty sprouting with a significant percentage of tubers remaining dormant. When this occurs, the carry-over of unsprouted weed tubers into a succeeding crop makes their eradication difficult.

The tubers become dormant in the fall, prior to the actual completion of their development. Steinbauer (1939) observed the onset of dormancy (two cultivars) between Aug. 28 and Sept. 7, and found that after this time, freshly dug tubers did not sprout even under suitable environmental conditions. The larger, more mature tubers were the last to enter dormancy. The onset of dormancy appeared to be a gradual process; it was initially established in the stolons and small, younger tubers, well before the completion of the maturation process and the first frost.

The depth of dormancy varies considerably among cultivars and tubers within a cultivar, so that some tubers will sprout as soon as they are exposed to conducive conditions, while others are delayed considerably. In a study of 145 cultivars, Boswell (1932) found the time required for 50% of the tubers to sprout when not subjected to a cold treatment ranged from 54 to 200 days; 150 to 180 days was typical for most cultivars. Also, the degree of dormancy varied among seasons. Cutting the tubers did not alter the depth of the dormancy. Steinbauer (1939) found that treatment with chemicals, e.g., ethylene chlorohydrin, could shorten the dormancy, although some of the chemicals tested also slowed the subsequent sprout growth.

The optimum temperature range for dormancy breaking is 0 to 5°C; higher temperatures, such as 10°C, result in very slow dormancy breaking (Steinbauer 1939) and lead to increased rotting (Steinbauer 1932) and

moisture loss (Traub et al. 1929a). Also, at the higher temperature, the emerging sprouts were less vigorous. Fluctuating low temperatures (–1.1 to 4.4°C) were less effective than a constant 0°C. Typically, 30 to 45 days at 0°C was sufficient to break dormancy in the two cultivars tested ('Chicago' and 'Blanc Ameliore').

Dormancy completion sets off a cascade of reactions that leads to sprouting. Any treatment that breaks dormancy (e.g., 4°C for 12 to 16 weeks) results in modification of the apical bud and its conversion into an elongating shoot (Courduroux 1967; Tort et al. 1985).

With the onset of shoot development, there is an increase in the rate of protein synthesis (Masuda 1965, 1967), a shift in the metabolism of purine nucleotides (Le Floc'h et al. 1982; Le Floc'h and Lafleuriel 1983a,b), and an overall acceleration of the rate of metabolism. With sprouting, the number of ribosomes, present almost exclusively as monosomes, decreased considerably (Bagni et al. 1972). As dormancy is broken, there is an increase in RNA synthesis (Gendraud and Prévôt 1973; Gendraud 1975a,b), enhanced incorporation of amino acids (Cocucci and Bagni 1968), and alterations in free and bound amino acids (Scoccianti 1983). In addition, with the onset of development, there are significant alterations in the activities of certain enzymes, e.g., phosphoenolpyruvate carboxylase activity increased fourfold (Dubost and Gendraud 1987).

Significant changes in water status within the tuber accompany sprouting, with water moving from subtending regions into the growing bud. The reallocation of existing constituents was also indicated by the findings that the parenchyma cells of dormant tubers took up sucrose more readily and that the tetraphenylphosphonium concentration and the intercellular pH was higher than in non-dormant tubers (Gendraud and Lafleuriel 1983). Gendraud and Lafleuriel (1983) interpreted these findings to indicate the possibility that an H^+-sucrose co-transport mechanism was involved in dormancy. In addition, Ottono and Charnay (1986) found that the uptake of abscisic acid differed between dormant and non-dormant tubers.

5. Disorders. Storage losses are due primarily to desiccation, disease, sprouting, freezing, and inulin depolymerization. No physiological disorders have been reported.

6. Postharvest Pathology. Storage rots can be a serious problem (Johnson 1931; McCarter and Kays 1984; Barloy 1988; Cassells et al. 1988) and their development is usually strongly and positively temperature dependent. The disease organisms most frequently isolated from Jerusalem

artichoke tubers were *Botrytis cinerea* Pers. and *Rhizopus stolonifer* (Ehrenb.: Fr) Vuill, though *R. stolonifer* and *Sclerotinia sclerotiorum* (Lib.) de Bary are the most serious rot-causing organisms at low storage temperatures (Johnson 1931). In contrast, *Sclerotium rolfsii* Sacc. and *Erwinia carotovora* spp. (Jones) Bergey et al. are not significant pathogens below 20°C. Control of postharvest rot organisms is facilitated by storage at low temperatures (i.e., 0 to 2°C), removal of diseased tubers prior to storage, minimizing mechanical damage to the tubers, and proper humidity control.

7. Chemical Changes During Storage. Stored tubers undergo significant changes in carbohydrate chemistry which, depending upon the intended use, can have a pronounced effect on their quality. It is important to note that inulin is not one compound but a series of molecules of various chain lengths (Tanret 1893) which begin to depolymerize during storage (Thaysen et al. 1929; Traub et al. 1929b; Bacon and Loxley 1952; Jefford and Edelman 1960; Jefford and Edelman 1963; Rutherford and Weston 1968; Modler et al. 1993a,b; Ben Chekroun et al. 1994; Schorr-Galindo and Guiraud 1997), whether the tubers are harvested or left in situ. The degree of polymerization is critical for uses such as fat replacement or high fructose syrups. With the former, as the chain length decreases, the ability of inulin to mimic a lipid in foods diminishes. Likewise, with progressive depolymerization, the ratio of fructose to glucose decreases and, upon hydrolysis, yields a progressively less pure fructose syrup. For example, during winter storage, the ratio of fructose to glucose decreased from 11 to 3 (Schorr-Galindo and Guiraud 1997); thus, syrups derived from stored tubers would contain substantially more glucose.

IV. ROOT CROPS

A. Sweetpotato

Sweetpotato (*Ipomoea batatas* (L.) Lam., Convolvulaceae) is grown for its fleshy storage roots. Though a perennial, the crop is grown as an annual. The sweetpotato is the seventh most important food crop in the world, with an annual production of 136 million t (FAO 2002), however, in the United States it is used primarily as an occasional vegetable. The sweetpotato confers a wide range of health benefits (Kays and Kays 1998) that have recently enhanced its popularity.

Table 7.4. Weight loss (percentage) of sweetpotato roots stored in the open (control) and in sand, soil, or sawdust, under tropical conditions (adapted from Ray et al. 1994).

	Weight loss (%)				
Storage medium	Storage period in days				
	0	15	30	45	60
Open (control)	0 a[z]	7.7 b	10.4 b	19.9 d	43.9 f
Sand	0 a	6.2 b	8.8 b	20.2 d	28.4 g
Soil	0 a	9.4 b	11.8 b	16.5 e	28.2 g
Saw dust	0 a	10.8 b	15.0 c	17.6 c	29.4 g

[z]Means separation by Duncan's multiple range test, (5% level).

The sweetpotato does not have a developmental stage at which it is mature; the storage roots continue to grow and under favorable field conditions will enlarge until the interior becomes anaerobic and/or rots. Therefore, the crop is harvested when the majority of the roots in a field have reached the desired size.

1. Prestorage Treatments

Curing. Roots should be cured by being kept at 30°C and 90%–97% RH for 4–7 days immediately after harvest (Lutz 1945; Lutz and Simons 1958; Dempsey et al. 1970; Kushman 1975). During curing, ventilation is required to remove CO_2 and replenish O_2. Curing heals wounds inflicted during harvest and handling, helps to reduce moisture loss during storage and decreases the potential for decay. In addition, curing facilitates the synthesis of enzymes that improve the flavor during cooking (Wang et al. 1998; Kays and Wang 2000). The effects of temperature and relative humidity on wound healing have been investigated extensively (Morris and Mann 1955; Strider and McCombs 1958; McClure 1960).

As curing begins, the outermost parenchyma cells at the wound site desiccate. The subtending parenchyma cells then become suberized (Morris and Mann 1955; Wagner et al. 1983; Walter and Schadel 1983) and a lignin-like wound periderm forms beneath the suberized layer. Healing is adequate when the wound periderm is three to seven cells thick, which can be verified with a simple color test (Walter and Schadel 1982). Walter and Schadel (1983) characterized the structure and chemical composition of suberin and lignin in both the epidermis and healed wounds.

Amand and Randle (1989) studied the relationship between ethylene production, wound lignification, and wound periderm formation

following wounding of 'Centennial'. Ethylene production preceded wound lignification and wound periderm formation by 24 to 48 h, respectively. Blocking ethylene action with 2,5-norbornadiene increased ethylene production, blocked wound periderm formation for the 12-day duration of the test, and strongly suppressed and delayed lignification. Blocking ethylene synthesis with aminooxyacetic acid or $CoCl_2$ decreased ethylene production to 10% of that in the control. Lignification and wound periderm formation were also suppressed and their initiation delayed, suggesting the involvement of ethylene in lignification and wound periderm formation

Radiation. Sweetpotato shelf life is not extended by gamma irradiation treatment. Ascorbic acid and starch concentrations decreased after exposure to 1.5–2.0 kGy, while those of sucrose and total sugars increased with increasing radiation dosage (Ajlouni and Hamdy 1988). Gamma irradiation also enhanced softening (Lu et al. 1986).

2. Recommended Storage Conditions. Cured sweetpotatoes should be moved carefully (usually in palletized containers) to a separate storage room and held at 14 ± 1°C and 85–90% RH (Dempsey et al. 1970; Kushman 1975). Long-term storage experiments have shown that roots can be stored successfully under these conditions for up to a year without sprouting (Picha 1986a,b), though sensory quality declines with extended storage. Air flow in the storage room should be about 0.3 $m \cdot s^{-1}$ at optimal temperature and relative humidity (Dempsey et al. 1970). Storage of sweetpotatoes at or above 19°C results in considerable sprouting after several months, and an associated loss in root quality and marketability, but storage below 12°C results in chilling injury.

Storage of sweetpotatoes under controlled atmosphere (CA) reduces the rate of respiratory losses and increases total sugars (Chang and Kays 1981), but additional research on O_2 and CO_2 concentrations, timing, and cultivar requirements is needed. Uncured roots have been shown to decay rapidly when stored in low-O_2 environments, though 2 and 4% O_2 did not appear to be harmful to cured roots (Delate and Brecht 1989). To date, the beneficial effects of CA storage for sweetpotatoes have not been shown to outweigh the cost.

3. Water Loss. Evaporation is a major cause of weight loss during sweetpotato storage; its rate is closely tied to the RH of the surrounding air (Afek and Kays 2002; Afek and Wiseblum 1995). When 'Beauregard' sweetpotatoes were stored for 4 months at 85 or 95% RH and 15°C, the weight losses were 11% and 2.4%, respectively. Thus losses can be sub-

stantially reduced by storing under high RH (Afek and Kays 2002). Ray et al. (1994) in India compared the fresh weight of roots stored in the open (control) with that of those covered with sand, soil, or sawdust, and found that after 2 months the control roots lost approximately 44% of their fresh weight compared with 30% in the other treatments.

4. Sprouting. In temperate growing areas, the storage root is used to produce planting material (slips) for the new crop; in the tropics, vine cuttings are typically used. When slips are required, the potential to sprout and the number of sprouts per root are critical selection traits in breeding programs for acceptable new cultivars. However, precocious sprouting during storage is highly undesirable, since it appreciably shortens the storage life of the roots. Sprouting roots exhibit a distinct proximal dominance (Kays and Stutte 1979); aging in storage increases the sprouting.

Sweetpotato storage roots do not exhibit dormancy; under favorable conditions they sprout readily immediately after harvest. Sprouting can be inhibited by proper temperature management (i.e., 14°C); at 19°C or above, the roots sprout after several months. Sprouting can also be inhibited by a postharvest application of CIPC (Scott et al. 1970), however, the chemical is not currently cleared for use and should not be considered a substitute for proper temperature management. Storage in a medium with sufficient moisture for fibrous root formation encourages sprouting, which utilizes the stored nutrients and moisture in the roots, and thus compromises root quality; it also increases the surface area of the root, accelerating water loss.

5. Disorders

Hardcore. Hardcore is a physiological disorder manifested by the failure of parts of the root to soften during cooking; it is believed to be caused by chilling effects on the cellular membranes (Yamaki and Uritani 1972, 1973, 1974). All cultivars are susceptible to hardcore, but they vary in susceptibility (Buescher et al. 1975a; Daines et al. 1976; Porter et al. 1976; Broadus et al. 1980; Picha 1987) and non-cured roots are more susceptible than cured ones (Lutz 1945; Daines et al. 1976; Picha 1987).

Souring. Roots may be lost during curing and/or storage, following preharvest exposure to anaerobic conditions (Ahn et al. 1980a,b; Chang et al. 1982, 1983) caused by excessive soil moisture. The roots may initially appear sound, but they decompose rapidly in storage, emitting a distinctive sour, fermented odor. The surviving roots undergo greater

shrinkage than sound ones during subsequent storage and typically yield a low-quality baked product (Ton and Hernandez 1978; Ahn et al. 1980a,b; Corey et al. 1982).

Internal Breakdown or Pithiness. Pithiness, found in apparently sound roots, is characterized by a significantly reduced density, and sponginess (Harter et al. 1923; Artschwager 1924; Kushman and Pope 1972); the intercellular air space exceeds 12%. The disorder has been attributed to high respiratory and water losses, and susceptibility varies with cultivar. Curing and storage conditions that are conducive to a rapid metabolism favor pithiness, as storage carbohydrates are depleted rapidly. Sprouting in storage and preharvest exposure to low soil temperatures (5–10°C) also increase pithiness.

Cracking. Storage root cracking may develop during growth or in storage. Cracks formed during storage are typically longitudinal splits near the end of the root and are generally shallower than growth cracks (Clark and Moyer 1988).

Distal End Rot. Distal end rot develops at the root end of the storage organ. Its occurrence is cultivar dependent, and 'Acadian' is highly susceptible (Martin 1958). No pathogenic organism has been isolated and the cause remains undetermined.

Chilling Injury. Sweetpotato storage roots are susceptible to chilling injury when stored below 12°C (Dempsey et al. 1970; Lewis and Morris 1956; Picha 1987; Whiteman and Wright 1946), and they freeze at –1.9°C (Whiteman 1957). Symptoms of chilling injury include root shriveling, surface pitting, abnormal wound periderm formation, fungal decay, internal tissue browning (Whiteman and Wright 1946; Lewis and Morris 1956; Dempsey et al. 1970; Picha 1987), and hardcore formation (Daines et al. 1974, 1976; Buescher et al. 1975a; Broadus et al. 1980). Synthesis of chlorogenic acid and other phenolic compounds has been associated with tissue browning (Dempsey et al. 1970; Lieberman et al. 1958; Porter et al. 1976; Walter and Purcell 1980).

The severity of chilling injury depends on the temperature and the duration of exposure to temperatures below 12°C (Dempsey et al. 1970; Picha 1987). After exposure to chilling, the respiratory rate of roots at 16°C increased linearly with the duration of the preceding exposure to the lower temperature (Dempsey et al. 1970; Lewis and Morris 1956; Picha 1987). The total sugar content of roots stored at 7°C was signifi-

cantly greater than that in those stored at 16°C (Dempsey et al. 1970; Picha 1987), though the effect was highly cultivar dependent.

Effects of Ethylene. Exposure of sweetpotatoes to ethylene should be avoided during storage and handling. Exposure of roots to ethylene at 10 $\mu l \cdot l^{-1}$ reduced their β-amylase activity (Buescher et al. 1975b). When roots of 'Tainung' and 'Chailai' were exposed to ethylene (100 $\mu l \cdot l^{-1}$), their β-amylase activity increased threefold during the first day and then decreased rapidly to the original level (Chang et al. 1996). Ethylene also enhanced the synthesis of phenolic compounds and phenolic oxidizing enzymes that can result in increased discoloration of the roots. The effect, however, requires the roots to be exposed to ambient concentrations of ethylene that would normally not be encountered in properly ventilated storage rooms.

6. Postharvest Pathology. Among the more commonly encountered microorganisms that cause storage rots are *Lasiodiplodia theobromae* (Java black rot) (synonymous with *Botryodiplodia theobromae* and *Diplodia gossypina*), *Ceratocystis fimbriata* (black rot), *Erwinia chrysanthemi* (bacterial soft rot), *Fusarium oxysporum* (surface rot), *Fusarium solani* (root rot), *Macrophomina phaseolina* (charcoal rot), *Monilochaetes infitscans* (scurf), and *Rhizopus stolonifer* (soft rot) (Clark and Moyer 1988; Clark 1992; Clark et al. 1992). Timing of infection varies with the organism and field/harvest/storage conditions (Moyer 1982). Black rot, fusarium root rot, scurf, and bacterial soft rot infections can occur before, during, or after harvest. In contrast, soft rot infections tend to be induced at or after harvest, while charcoal rot, dry rot, surface rot, and root rot occur during harvest. Harvest and postharvest pathogens are typically opportunistic, entering the root via an injury.

Internal cork is a virus-mediated disorder in which the root tissue develops necrotic lesions during storage (Nusbaum 1946, 1950; Martin 1949; Kushman and Pope 1972). The number and size of lesions vary widely and increase with increasing storage duration and storage temperature (Nielsen 1952). The lesions are found primarily in the interior but may also be present on the surface.

Control of postharvest diseases centers on prevention because little can be done once the root is infected. During harvest, care must be taken to minimize damage to the roots and proper sanitation is important. After harvest, the roots should be cured immediately and then stored at the appropriate temperature. Mechanical damage should be carefully prevented during movement from curing to storage rooms, and then

during washing, sorting, and grading prior to marketing. Wash water should be frequently changed to prevent accumulation of inoculum, and calcium hypochlorite should be added to the water. Postharvest pesticides, if used, must be in accordance with the appropriate laws.

In Israel, 'Georgia Jet' is more susceptible to storage pathogens than cultivars with higher dry matter contents, such as 'Jewel' and 'Beauregard' (Picha 1986a,b; Rolston et al. 1987; Stevens et al. 1990). Exposure to low temperatures in the field (e.g., ≤8°C) leads to subsequent rapid deterioration in storage, via *Rhizopus* spp. and *Fusarium* spp. (Afek et al. 1998a; Clark and Moyer 1988; Clark 1992).

In situ treatment of the stored roots by applying a fog of 1% iprodione from an atomizing humidifier substantially reduces rot incidence (e.g., 5% vs. 61% in controls) (Table 7.5) (Afek et al. 1999a). Isolates taken from rotten areas indicated that the decay was caused by *Rhizopus* spp. and *Fusarium* spp.

7. Postharvest Entomology. The sweetpotato weevil (*Cylas formicarius* (F.), Coleoptera: Brentidae) is the most devastating insect pest of the crop worldwide, both in the field and in storage. The adult can find the roots in total darkness by sensing their volatile emissions (Wang and Kays 2002). To date, there are no adequate field or storage control measures available (Nottingham and Kays 2002), therefore, infested roots should not be placed in storage. CA storage may have promise (Delate and Brecht 1989) and some success has been achieved by storing the roots at low temperature or immersing them in hot water (52–62°C for 10 min) (Hahn and Anota 1982). Fruit flies (*Drosophila* spp.) and soldier flies [*Hermetia illucens* (Diptera: Stratiomyidae)] can pose problems where there are diseased, soured, or damaged roots, but both can be controlled with sanitation and/or appropriate insecticide treatment (Boyette et al. 1997).

Table 7.5. Percentage of 'Georgia Jet' sweetpotato roots rotting during 5 months of storage (13°C, 90% RH) with (+) or without (–) a prestorage treatment of 1% iprodione (Afek et al. 1999a).

	Rot (%)				
	Storage period in months				
Iprodione	1	2	3	4	5
+	1 a[z]	2 a	2 a	3 a	5 a
–	6 b	22 b	38 b	50 b	61 b

[z]Mean separation by Fisher's protected least significant difference test, (5% level).

8. Chemical Changes During Storage. Storage roots undergo significant chemical changes during curing and storage. Respiration is highest immediately after harvest and decreases during the first few months of storage (Chang and Kays 1981; Picha 1986b). The concentration of starch, the predominant form of stored carbon in the roots, also decreases during storage (Dempsey et al. 1970; Scott et al. 1970). About 75% of the total starch in the roots at harvest is amylopectin, which increases to about 80% during curing, the remaining 20% being amylose (Scott et al. 1970). Only small amounts of dextrins are present at harvest, but their concentration increases during curing. The primary sugars present, in descending order of abundance, are sucrose, glucose, and fructose (Kays and Horvat 1983; Picha 1986a; Walter and Hoover 1984). There are significant quantitative differences among cultivars in their patterns of carbohydrate change during curing and storage. Typically, reducing (Jenkins and Gieger 1957; Picha 1986a;), nonreducing (Morris and Mann 1955; Jenkins and Gieger 1957; Picha 1986a), and total sugars (Morris and Mann 1955; Picha 1986b) increase during curing and the first few months of storage. However, nonreducing sugars have been reported to decrease in some cultivars (Morris and Mann 1955; Lambou 1958; Picha 1986a), and in some cultivars reducing sugars do not increase (Picha 1986a). Likewise, there is no increase in total sugar concentration in some cultivars (Dempsey et al. 1970; Picha 1986a). Storage temperature also influences the sugar concentration in the roots; low temperatures (e.g., 7°C) stimulate sucrose synthesis (Picha 1987).

The percent dry matter in the roots decreases during long-term storage, as starch is utilized as a respiratory substrate. Alpha-amylase activity increases during storage (Deobald et al. 1971); β-amylase activity increases during storage in some cultivars (Morrison et al. 1988), whereas inconsistent changes in activity were found in others (Walter et al. 1975).

The concentration of pectic substances decreases by as much as 40% by the 6th month in storage (Scott et al. 1970). A decrease in protopectin and an increase in soluble pectin concentrations occur during curing, with the opposite during storage (Heinze and Appleman 1943). In another study, sweetpotatoes stored at 13°C for 3 months displayed minor fluctuations in soluble pectin and protopectin (Daines et al. 1976).

Protein was degraded during storage ('Jewel') and non-protein nitrogen (NPN) concentration decreased during the first 14–15 weeks, followed by an increase (Purcell et al. 1978). As much as 40% of the total N in the roots may be NPN (Purcell et al. 1978), the major components of which, after 107 days in storage, were asparagine (61%), aspartic acid (11%), glutamic acid (4%), serine (4%), and threonine (3%). The

concentration of individual amino acids also changed during storage (Purcell and Walter 1980).

The primary changes in fatty acids after curing and during several months of storage are an increase in tetracosanenoic acid concentration and a decrease in the concentration of short-chain saturated fatty acids (Dempsey et al. 1970). The orange flesh color in sweetpotato is due to carotenoid pigments, primarily β-carotene, a precursor of vitamin A. The concentration of carotenoids increased slightly during curing and short-term storage (Daines et al. 1974; Hammett and Miller 1982). The vitamin C concentration generally decreases during curing and storage (Ezell and Wilcox 1952; Speirs et al. 1953; Hammett and Miller 1982).

B. Carrot

Carrot (*Daucus carota* L., Apiaceae) is a biennial temperate crop grown for its fleshy tap root. Over 20 million t were produced worldwide in 2001 (FAO 2002). Storage techniques range from field storage to refrigerated storage rooms. Harvestable maturity of carrots depends upon their intended use. Most carrots are stored as mature roots, but immature bunched roots, with or without their tops, and small, fresh-cut (i.e., minimally processed) roots are also stored for short periods to facilitate marketing. Optimum handling and storage conditions vary accordingly.

1. Prestorage Treatments

Washing. Prestorage washing is desirable if the carrots have much soil or organic matter adhering to the roots (e.g., when harvested during wet conditions or from sandy soils). Washing can decrease the population of decay-causing organisms and help to facilitate air circulation around the roots in storage.

Precooling. Prompt precooling to or below 5°C after harvest is essential for extended storage because delays increase decay incidence during storage. Hydrocooling is commonly used; loose carrots can be cooled from 25 to 5°C in about 9 min in 1°C water, but it takes about 11 min for those packed in mesh bags (23 kg) (Ryall and Lipton 1979). The precise cooling time varies with root size and temperature, and type of hydrocooler. The half-cooling time should be determined for the specific conditions at each site to maximize the throughput.

2. Recommended Storage Conditions. Mature topped carrots can be stored for 7 to 9 months at 0–1°C, 98–100% RH (Berg and Lentz 1966, 1973; Kirki 1971). Even under optimum conditions, however, 10–20%

of the roots will show some decay after 7 months. At higher storage temperatures (e.g., 5–10°C), sprouting and decay increase appreciably within a few months (Platenius et al. 1934; Kirki 1971; Sherf 1972). Under typical commercial conditions (0–5° C; 90–95% RH), carrots can be successfully stored for only 5–6 months (Ryall and Lipton 1979). The maximum storage duration for immature carrots with their tops attached is about 2 weeks at 0°C, 95–100% RH. Removal of the tops significantly extends the potential storage duration. Contact icing is recommended during transit, especially with loose or bunched carrots with their tops, to retard water loss and maintain freshness.

Temperatures below 0°C should be avoided since carrots can freeze at −1.2° C (Whiteman 1957). Freezing causes cracking and blistering of the surface; after thawing, discoloration, a water soaked appearance, and a flaccid texture develop (Parsons and Day 1970).

Air circulation between crates or pallet boxes in which carrots are stored is desirable to remove respiratory heat, to maintain uniform temperatures, and to help prevent condensation. An air velocity of about 7 to 10 cm · s^{-1} is adequate at low storage temperatures (Berg and Lentz 1966).

Controlled atmosphere storage is rarely used for carrots, but modified atmosphere packages are used for fresh-cut carrots. Storage of intact carrots in atmospheres of 5–10% CO_2 and 2.5–6% O_2 can lead to mold growth and rotting that is greater than that occurring in conventional storage conditions (Berg and Lentz 1966; Weichmann 1977). The oxygen concentration is critical because quality declines rapidly under anaerobic conditions; therefore, selection of packaging materials with appropriate oxygen transfer rates is imperative (Sode and Kuhn 1998). The importance of preventing low-oxygen stress in fresh-cut carrots has stimulated interest in ethanol biosensors (Smyth et al. 1999).

Ozone has been tested as a volatile fungicide, applied during storage to repress postharvest pathogens. Ozone reduced decay but also increased the rates of respiration and electolyte leakage and decreased the color intensity of the roots (Liew and Prange 1994). Gamma irradiation at levels that did not cause significant damage has been shown to alter the respiration rate of grated carrots but not of intact roots (Chervin et al. 1992); it is not used commercially.

3. Water Loss. Carrots lose moisture and shrivel if not stored under high RH (i.e., 98–100%) (Hurschka 1977); lower RH (90–95%) results in greater moisture loss, increased decay, and loss of crispness and other quality attributes (Kirki 1971; Berg and Lentz 1973, 1977; Krahn 1974). Cooling as rapidly as possible after harvest also has a pronounced influence on the root's moisture status. For example, the water potential

decreased by more than 200% within 48 h when freshly harvested roots were stored at 18°C, >98% RH (Herppich et al. 1999). Cultivars vary in their propensity to lose moisture after harvest (Shibairo et al. 1997). Water VPD, air movement, storage temperature, and product surface to volume ratio are the predominant controlling factors.

Immature and partially mature carrots are routinely topped and packed in polyethylene consumer bags or 23-kg mesh bags for marketing. If they were promptly precooled and trimmed of all traces of leaf growth before packaging, they can be held for 4–6 weeks at 0°C, 98–100% RH. The roots should be packed in low-density polyethylene bags, whose thickness depends on the quantity contained (Afek et al. 1998b; Suojala 1999). The bags should be perforated to allow ventilation and to prevent the development of off-flavors; 3–6-mm holes are spaced evenly over the surface (e.g., twelve 3 mm holes for 1-kg bags) (Hardenburg et al. 1953; Afek et al. 1998b, 1999b).

4. Sprouting. The enlarged tap root serves as a storage site for the carbon and nutrients needed to form the flower stalk during the second year. Sprouting, which often occurs during prolonged storage, impairs the quality of the roots and increases water loss and decay. The propensity to sprout increases with storage duration and temperature, and cultivars vary in their susceptibility to sprouting.

5. Disorders

Bitterness. Bitterness, caused by exposure of carrots to ethylene (as little as 0.5 $\mu l \cdot l^{-1}$) during storage, is quite common. The bitter compound is isocoumarin (8-hydroxy-3-methyl-6-methoxy-3,4-dihydroisocoumarin) (Carlton et al. 1961; Sarkar and Phan 1979), which is a phytoalexin synthesized by the tissue (Condon and Kuc 1960). Sources of ethylene range from other stored products and microorganisms to equipment driven by internal combustion engines. Isocoumarin synthesis is modulated by ethylene and oxygen concentrations, temperature, storage duration, and the maturity and physical condition of the roots (Lafuente et al. 1996). Preventive measures include exclusion of ethylene sources, appropriate ventilation, and low storage temperatures (Carlton et al. 1961).

Splitting. Roots frequently split during growth, harvest, and storage, decreasing the value of the product (Millington 1984; Kokooras 1989; Vincent 1990). For example, 30% of the shipments (n = 2,425) of carrots arriving in New York over a 14-year period had significant numbers of

broken or split roots (Cappellini et al. 1987). Splitting is caused by a combination of mechanical weakness and the water status of the tissues, with splitting being inversely proportional to the water content of the cells (McGarry 1993, 1995). A strain is created during water uptake when the expansion potential differs between neighboring cells (Sorensen and Harker 2000); as the root grows, the frequency of splitting increases (Hole et al. 1999). The susceptibility to splitting varies among cultivars, ranging from 3.8 to 73% (McGarry 1993; Hole et al. 1999).

Discoloration. Surface browning may develop during storage, with immature carrots being more susceptible than mature (Chubey and Nylund 1970). Ozone treatment during storage can also decrease the orange coloration of the roots (Liew and Prange 1994).

6. Postharvest Pathology. Postharvest losses due to disease can be a serious problem. For example, eleven parasitic diseases were found in shipments of carrots to New York (Cappellini et al. 1987), with bacterial soft rot being the most prevalent, i.e., present in 33% of the shipments (n = 2,425). Factors that can influence the incidence of pathological losses during storage include the maturity of the roots at harvest (Suojala 1999), timing of harvest (Davies and Lewis 1980; Villeneuve et al. 1993; Suojala 1999), and weather conditions (humidity and precipitation) before and during harvest (Fritz and Weichman 1979; Villeneuve et al. 1993).

The major storage fungi and bacteria of carrot are (a) fungi: black rot—*Alternaria alternata* (Fr.:Fr.) Keissler; black rot—*Alternaria dauci* (Kuhn) Groves & Skolko; black rot—*Alternaria radicina* (Meier); gray mold—*Botrytis cinerea* Pers.; sour rot—*Geothichum candidum* Link; licorice rot—*Mycocentrospora acerina* (Hartig) Deighton; cavity spot—*Pythium* spp.; violet root rot—*Rhizoctonia crocorum* DC. ex. Fr.; Rhizopus rot—*Rhizopus* spp.; watery soft rot—*Sclerotinia sclerotiorum* [(Lib.) De Bary]; Sclerotium rot—*Sclerotium rolfsii* Sacc.; black mold—*Thielaviopsis basicola* (Berk & Br.) Ferraris; and (b) bacteria: soft rot—*Erwinia carotovora* subsp. *carotovora* (Jones) Bergey et al.; soft rot—*Erwinia carotovora* subsp. *atroseptica* (van Hall) Dye, Blackleg; soft rot—*Erwinia chrysanthemi* pv. *chrysanthemi* Burkh., McFadden & Dimock; soft rot—*Pseudomonas marginalis* pv. *marginalis* (Brown) Stevens; soft rot—*Pseudomonas marginalis* pv. *pastinacae* (Burkh.) Young, Dye & Wilkie (Adams and Kropp 1996; Snowdon 1992; Thorne 1972).

The infecting organisms vary with location and production conditions. In Israel, the fungi *A. alternata*, *A. radicina*, and *S. sclerotiorum*, and the bacterium *E. carotovora* subsp. *carotovora* can pose serious problems (Afek et al. 1998b). Several chemical treatments have been

Table 7.6. Decay incidence (percentage) of organically grown carrots after storage (60 days at 0.5°C plus 7 days at 20°C) in retail (1 kg), wholesale (18 kg), and lined (15 kg) packages, following prestorage treatment with steam, fungicide (iprodione + chlorane), or neither, with (+) and without (–) inoculation with *Alternaria alternata*, *A. radicina*, *Sclerotinia sclerotiiorum*, and *Erwinia carotovora* (Afek et al. 1999b).

		Decay incidence (%)		
Prestorage treatment	Inoc	Package type Retail	Wholesale	Lining
Control	+	66 e[z]	61 e	71 e
	–	23 d	25 d	21 d
Steam	+	9 c	7 c	5 bc
	–	6 c	5 bc	8 c
Iprod+Chlor	+	5 b	3 ab	3 ab
	–	3 ab	3 ab	5 bc

[z]Mean separation in columns by Fisher's protected least significant difference test, (5% level).

tested (e.g., thiabendazole, iprodione, sodium-*o*-penylphenate); their use in individual countries depends upon government approval (Wells and Merwarth 1973). Among non-chemical treatments, a prestorage steam treatment has shown benefits (Afek et al. 1999b). After 60 days of storage at 0.5°C and 7 days poststorage at 20°C, steam-treated roots had much less spoilage than untreated controls (Table 7.6).

7. Chemical Changes During Storage. Mono- and disaccharides stored in the vacuoles account for 34–70% of the dry weight of carrot roots (Goris 1969; Ricardo and Sovia 1974; Nilsson 1987a). Sucrose is the dominant form at maturity (Daie 1984), however, its absolute concentration and its ratio to other sugars depend on cultivar and production conditions (Goris 1969; Phan and Hsu 1973; Ricardo and Sovia 1974; Nilsson 1987a; Suojala 2000). During storage, hexoses increase and sucrose declines, especially during the first months (Phan et al. 1973; Nilsson 1987b; Olden and Nilsson 1992; Le Dily et al. 1993). Under appropriate conditions, there are usually only minor changes in the total sugar content, but the sugar composition changes continuously throughout the storage period, irrespective of harvest date (Nilsson 1987b).

C. Cassava

Cassava (*Manihot esculenta* Crantz, Euphorbiaceae) is a primary or secondary food source for more than 500 million people in the tropics

(Bokanga et al. 1994; Hillocks et al. 2002; Padmaja 1995) and is also used as animal feed (Gomez 1979). Worldwide production in 2001 was 189 million t (FAO 2002). Much cassava contains cyanogenic glucosides (principally linamarin and lotaustratin) that degrade to highly toxic hydrogen cyanide (HCN). The concentration of cyanogenic glucosides varies among cultivars, which are separated into *bitter* cassava, which contains high levels of the toxic glucosides throughout the storage roots, and *sweet* cassava, in which the much lower concentrations are confined largely to the peel. The terminology "bitter" vs. "sweet" is misleading, since it has nothing to do with the taste of the roots, and there is a continuum of glycosides concentrations across cultivars, from extremely low to extremely high. The roots, especially those of bitter cultivars, must be processed to remove the HCN, and properly treated cassava is a safe food source. Inadequate processing, however, can yield a toxic product, especially for the very young, elderly, and individuals with compromised health.

A variety of detoxification methods are used, of which drying is the most common in many tropical countries, where cassava is traded as chips or pellets (Padmaja 1995). Sun drying eliminates more cyanide than oven drying because it prolongs the contact between linamarase and the glucosides. The most common technique used for dietary cassava involves mashing, squeezing, fermenting, and then roasting/drying. Soaking followed by boiling removes HCN better than either alone.

The storage root is the most used plant part, though the young leaves are eaten as a green vegetable in many parts of the world. The storage roots are predominately carbohydrate (i.e., starch, 28–33% of fresh weight) and have a total dry matter content of 33–39%. The carbohydrate concentration varies with cultivar, stage of maturity, and growing conditions. The protein concentration of the roots is very low (0.4–0.6% of fresh weight) (Bradbury and Holloway 1988), therefore cassava is mainly a carbohydrate source. When used as a staple food, cassava often provides approximately one-half of the total dietary caloric intake (Rickard and Coursey 1981).

1. Prestorage Treatments

Curing. Cassava roots can be cured by exposure to relatively high temperatures and humidities, a process that heals wounds and impedes the onset of vascular deterioration (Booth and Coursey 1974; Booth 1977). Curing promotes the development of meristematic tissue and suberization beneath the wound, as in other root crops. At 80–85% RH, meristem formation accelerates as the temperature increases from 25 to 40°C, but substantially more deterioration occurs at 40°C than at 35°C. At

35°C a complete meristem is formed around cuts in 4–7 days, whereas at 45°C the roots do not heal and undergo rapid bacterial breakdown. When the roots were held at 95% RH, the wounds rapidly became infected by a wide range of pathogens, whereas at 75% RH they dried out rapidly, especially at wound sites (Booth 1974, 1976).

Waxing. The effect of waxing on the storage life of cassava roots was first studied in the 1940s (Castagnino 1943) and the technique was subsequently refined. Dipping the roots in a fungicidal wax followed by storage at ambient temperatures substantially increased the storage potential (Subramanyam and Mathur 1956). Waxing approximately halved the weight loss during the first 2 weeks of storage and extended the useful storage time period (i.e., <10% loss) from 2 to 10 days. An increase in respiration was also delayed. The principal benefit of waxing seems to be derived from reduced moisture loss rather than the pathogen inhibition. Paraffin wax dips (90–95°C for 45 s) avert serious quality losses for 1–2 months (Anonymous 1972), ensuring sufficient time for export or storage (Burton 1970; de Buckle et al. 1973; Zapata 1978). Water-based carnauba and paraffin waxes are comparable in root quality maintenance (Sargent et al. 1995).

2. Recommended Storage Conditions. Without proper handling, fresh cassava deteriorates rapidly, often within 2–3 days, under tropical conditions, so that losses during storage and shipping can be a serious problem. Several means are available to extend the potential storage period; the choice depends upon the level of technology available and the relative value of the crop. Options include refrigerated, nonrefrigerated (e.g., boxes, pits, clamps), and field storage.

Refrigerated Storage. Cassava can be effectively stored at 0–5°C, 85–90% RH for 1 to 2 months (Normanha and Pereira 1963; Averre 1967; Ingram and Humphries 1972). Storage potential depends significantly on the cultivar, production conditions, mechanical damage sustained during harvest and handling, and prestorage treatments.

Czyhrinciw and Jaffe (1951) monitored changes in several properties of fresh cassava stored for 4 weeks at 3, 12, and 25°C. Storage resulted in a substantial reduction in vitamin C concentration by the 4th week of storage, as well as changes in peroxidase, dehydrogenase, and catalase activity, especially at 12°C. Roots stored for 2 weeks at 0–2°C showed no internal browning, but after 4 weeks a blue mold was observed that increased during subsequent storage (Singh and Mathur 1953). Above 4°C, the roots developed the same symptoms more rapidly

Table 7.7. Effect of temperature and duration on the weight loss of cassava roots in storage (Singh and Mathur 1953).

Storage temp. (°C)	Weight loss (%)							
	Storage period in weeks							
	1	2	3	4	5	6	7	8
0–2	5.6	11.0	15.0	19.2	21.7	34.1	37.5	38.1
2–4	5.1	11.0	15.0	18.0	21.7	34.1	40.3	41.1
4–6	3.7	8.3	—	—	—	—	—	—
6–8	3.4	6.6	—	—	—	—	—	—
8–10	5.0	9.2	—	—	—	—	—	—
11–13	6.7	13.3	—	—	—	—	—	—
19–21	12.1	25.0	—	—	—	—	—	—
22–30[z]	11.7	22.5	—	—	—	—	—	—

[z]ambient

and were unmarketable after only 2 weeks of storage. After 7 to 8 weeks, the weight loss in roots held at 0–2°C was somewhat lower than that at 2–4°C (Table 7.7).

Nonrefrigerated Storage. Storage systems for fresh cassava roots include wooden buildings, bamboo and palm thatch buildings, cellars, pits, clamps, trenches, and boxes (Tracy 1903; Bayday 1922; Booth 1977; Rickard and Coursey 1981). Roots intended for storage should be harvested at the proper maturity, brushed cleaned (not washed), handled so as to minimize injury, and the surface air-dried before storage. Clamps are built by laying a bed of rice or other straw, 1–5 m in diameter, on dry ground and placing 300–500 kg of freshly harvested roots on it in a conical pile. The roots are covered with additional straw and then with soil, taken from around the clamp, so as to leave a drainage ditch. Various thicknesses of soil covering are used and basal or vertical ventilators may be inserted. Properly constructed clamps can preserve cassava roots for 1–2 months, with only modest desiccation and deterioration losses (Booth 1977; Rickard and Coursey 1981). The design of the clamp varies with the season, since it is important to maintain the internal temperature below 40°C. During the hot dry season, extra covering soil and ventilation are used. Even with such modifications results can be unsatisfactory (Booth 1976), so clamps have not been widely adopted for commercial storage (Lozano et al. 1978).

Storage in boxes packed with moist sawdust or other material can be satisfactory. If the sawdust is too dry, the roots desiccate; if too moist,

fungal and bacterial rots develop. Delays between harvesting and packing increase losses. If properly handled, about 90% of the roots, even of susceptible cultivars, are acceptable after 8 weeks of storage (Booth 1977; Sivan 1979; Rickard and Coursey 1981; Cooke et al. 1988). Moist coir dust (coconut husk) can be used in place of sawdust (Marriott et al. 1974) or the roots can be layered between fresh cassava leaves (Aiyer et al. 1978). A plastic box liner can prevent excessive drying of the packing material. The primary limitations to using boxes are the cost of the box and the labor required for packing. Nevertheless, storage boxes are used commercially for fresh cassava (Booth 1977; Rickard and Coursey 1981; Cooke et al. 1988).

Cassava treated with a fungicide (thiabendazole) has been successfully stored in perforated polyethylene bags for 2–3 weeks in Colombia (Wheatley 1989). A similar technique has been less successful in Ghana (Bancroft and Crentsil 1995), where tightly woven fiber bags are used without a fungicide; the roots can be stored for only 7–10 days, which is generally sufficient for marketing in Ghana (Gallat et al. 1998).

Field Storage. In technology-limited locations, such as on farms or in rural villages, cassava is either left unharvested (i.e., "stored" in the field) or processed into a dried product (Lancaster et al. 1982), to minimize losses. Roots stored in the ground continue to increase in size, often become fibrous and woody, and suffer a concurrent decline in starch content. It is difficult to hold freshly harvested cassava for even a few days. The poor storage life of the roots is a major weakness of the crop and forms an economic constraint in developing countries (Rickard and Coursey 1981).

3. Water Loss. In spite of the relatively thick periderm, the loss of moisture from the harvested roots is a serious problem that greatly aggravates losses due to pathogens. Moisture losses are particularly severe if the periderm is broken, which is unavoidable during harvest. Waxes, plastic bags, and film wraps have been used to reduce moisture loss. Although they all impede the outward diffusion of water vapor, they also impede respiration. Wrapping the roots in moist paper before placing them in plastic bags inhibits the development of vascular discoloration, especially below 10° or above 40°C (Averre 1967).

A variety of polyethylene bags and sacks, and polyvinyl chloride cling and shrink films, have been tested on cassava (Oudit 1976; Thompson and Arango 1977; Lozano et al. 1978). Weight losses were considerably reduced by both film wraps and plastic bags, though perforated bags were less effective. Film-wrapped cassava showed signif-

icantly reduced vascular streaking during storage, but the incidence of bacterial infection of the roots was greater than that in unwrapped controls. The use of plastics to impede water loss, particularly when combined with curing, antimicrobial treatments, and low-temperature storage, is effective.

4. Sprouting and Vascular Streaking. Sprouting of cassava storage roots requires the formation of adventitious buds. Due to the relatively short storage potential of cassava, sprouting is not a problem.

Vascular streaking or discoloration is characterized by a dark bluish or blue-black discoloration of the vascular bundles; it usually starts at cut surfaces and progresses along the root, gradually becoming more prominent and turning brown or black over a few days (Castagnino 1943; Pacheco 1952; Drummond 1953; Normanha and Pereira 1963; Averre 1967; Montaldo 1973). The potential for vascular discoloration varies among cassava cultivars (Montaldo 1973). Initially faint blue streaks, caused by a darkening of areas within the xylem vessels and the formation of black occlusions within the vessels, develop near the surface. Discoloration subsequently spreads to neighboring parenchyma cells, where the structure of the starch grains changes (Drummond 1953). The disorder is thought to be enzymatically mediated and has been attributed to several enzymes (e.g., dehydrogenases, peroxidases, phosphorylases, and catalases (Czyhrinciw and Jaffe 1951; Murthy et al. 1956; Nair and Kurup 1963).

The onset of vascular discoloration is closely associated with mechanical damage (Booth 1976) and there is a positive correlation between the degree of damage and the weight loss and deterioration (Rickard and Coursey 1981). The most severe damage typically occurs during harvest, at the point of attachment of the root to the parent plant. The damage and, therefore, discoloration, can be minimized by leaving a short segment of the stem attached to the root, a technique often used by farmers (Drummond 1953; Lozano et al. 1978).

Avoidance of mechanical damage and proper cultivar selection retard development of the disorder and several treatments have been found effective. For example, hot water treatment (53°C for 45 min) or holding the roots under water or in anaerobic conditions inhibits development (Averre 1967). Low storage temperatures (0 or 5°C) also inhibited discoloration (Montaldo 1973).

The particular importance of vascular discoloration is that it appears to predispose the roots to subsequent attack by many fungal and bacterial wound pathogens that cause a variety of wet and dry rots (Plumbley and Rickard 1991).

5. Postharvest Pathology. The poor storage potential of cassava has long been recognized as one of the most critical deficits of the crop. Both fungal and bacterial rots play a major role in postharvest losses (Bayday 1922; Anonymous 1952; Majumder et al. 1956; Subramanyam and Mathur 1956; Normanha and Pereira 1963; Affran 1968; Oudit, 1976). Postharvest fungal pathogens include Botryodiplodia rot—*Botryodiplodia theobrpmae* Pat.; Fusarium rot—*Fusarium solani* (Mart.) Sacc.; Aspergillus rot—*Aspergillus* spp; Mucor rot—*Mucor* spp.; Phytophthora rot—*Phytophthora* spp.; dry rot—*Rhizopus* spp.; Sclerotium rot—*Sclerotium rolfsii* Sacc.; and Trichoderma rot—*Trichoderma harzianum* Rifai. Postharvest bacterial pathogens include soft rot—*Erwinia carotovora* subsp. *carotovora* (Jones) Bergey et al. and soft rot—*Bacillus* sp. (Plumbley and Rickard 1991; Snowdon 1992).

Several chemical treatments for the control of postharvest pathogens have been tested. Fumigation with an ethylene dibromide/ethyl bromide mixture gave a maximum storage life of 19 days (Majumder 1955; Majumder et al. 1956) with bromide residues ranging from 13 to 19 ppm. Some reduction in postharvest pathogens was also achieved by using a 45 s dip in 1% sodium o-phenyl phenate with 900 ppm of 2,6 dichloro-4-nitroaniline followed by rinsing with water (Burton 1970). Manzate and sodium hypochlorite were also tested; manzate at 8×10^4 ppm a.i. inhibited the rotting of roots stored in polyethylene-lined paper bags (Lozano et al. 1978). Benomyl gave positive results in several studies (Booth 1976); it reduced surface fungal growth on roots stored within plastic films. Washing the roots with chlorinated water reduced bacterial soft rot, though neither treatment mitigated vascular streaking (Thompson and Arango 1977). Benomyl was also found to reduce the soft rot complex responsible for the later stages of decay of cassava (Ekundayo and Daniel 1973). Pathogen control measures must be initiated as soon as possible after harvest because their effectiveness decreases if the application is delayed.

6. Postharvest Entomology. Insect damage to stored fresh cassava is relatively uncommon, partly because of the short storage life. In dried cassava, however, losses to the larger grain borer (*Prostephanus truncatus* Horn.) have been reported in South America and Africa (Golob 1988; Hodges 1986).

7. Chemical Changes During Storage. The quality changes of cassava roots stored in moist sawdust and in clamps have been studied by Booth et al. (1976). Starch was rapidly converted to glucose, fructose, and sucrose, raising the original free sugar levels two- or threefold, during

the first 2 weeks of storage. At the same time, the root tissue, especially the central portion, softened considerably.

V. CORM AND RHIZOME CROPS

A. Taro

Taro (*Colocasia esculenta* (L.) Schott, Araceae) is produced mainly in Nigeria, China, Ghana, Japan, the Ivory Coast, Papua New Guinea, Burundi, the Philippines, Egypt, and Madagascar (Wang and Higa 1983; Pardales 1987). World production is about 9 million t (FAO 2002).

Two types of taro are usually recognized, distinguished by the way in which the corms are formed. In dasheen (*Colocasia esculenta* var. *esculenta*) there is a large edible main corm (e.g., cylindrical, about 30 × 15 cm) to which are attached a few lateral "cormels." In eddoe (*C. esculenta* var. *antiquorum*), numerous edible cormels surround a small, bitter main corm. The names "eddoe" and "dasheen" are sometimes used interchangeably, which causes confusion since the two types differ in their postharvest characteristics, partly because of their differing structures. The harvested corm of dasheen carries several wounds after removal of the cormels and it sustains further injury if the basal portion is removed to provide propagation material (Snowdon 1992). The extensive wounding predisposes the corms to pathogen invasion. Eddoe cormels, in contrast, have only a single small wound, following separation from the mother corm. Furthermore, the cormels of certain cultivars of eddoe possess some degree of dormancy that confers additional storage life. Thus, eddoe may be stored for up to 3 months at ambient temperature, but dasheen is more perishable; in warm moist storage environments, dasheen corms cannot usually be held for longer than about a month without sprouting or decaying (Snowdon 1992). Passam (1982) reports a very much shorter postharvest life for both dasheen and eddoe.

The corms and cormels, as well as the petioles and leaves, are consumed after cooking, a process that eliminates the acridity caused by calcium oxalate crystals (raphides) (Paull et al. 1999).

1. Prestorage Treatments. The corms/cormels may be exposed to warm (20–30°C) moist conditions to facilitate the curing of wounds incurred during harvest. High RH favors curing (Been et al. 1975), which minimizes the entry of disease-causing microorganisms. After curing, the product is room cooled to the appropriate storage temperature.

2. Recommended Storage Conditions. Storage between 11 and 13°C, 80–95% RH can extend the longevity of the product significantly (i.e., up to 8 weeks) compared with that under ambient tropical conditions. At higher temperatures the corms perish rapidly; for example, in the Philippines corms are no longer fit for human consumption after 7 to 14 days in ambient conditions (27–32°C) because of microbial rotting (Quevedo and Ramos 1992). Taro is sometimes stored/shipped at temperatures lower than 11–13°C (e.g., 7–10°C, 80–95% RH for up to 18 weeks) but it must be consumed within a day or two after removal from refrigeration because the subsequent shelf life is significantly impaired. The corms are susceptible to chilling injury, the symptoms of which are surface pitting and an increased incidence of disease.

3. Weight Loss. Corms stored in clamps lost less weight than those stored in huts, because there was less evaporation and respiration. After 2 weeks of storage under ambient conditions, the weight losses of corms with intact roots and petiole, stored in a hut and in a clamp were 25 and 12%, respectively (Quevedo and Ramos 1992).

4. Postharvest Pathology. Decay of the corms is the most serious problem of taro during storage (Gollifer and Booth 1973; Passam 1982; Quevedo et al. 1992; Quevedo and Ramos 1992; Snowdon 1992). The following postharvest pathogens have been identified: (a) fungal pathogens include Botryodiplodia rot—*Botryodiplodia theobromae* Pat.; gray mold—*Botrytis* spp.; dry rot—*Fusarium* spp.; sour rot—*Geothichum candidum* Link; Phytophthora rot—*Phytophthora colocasiae* Racib.; Pythium rot—*Pythium* spp.; Rhizoctonia rot—*Rhizoctonia solani* Kuhn; dry rot—*Rhizopus* spp.; Rosellinia rot—*Rosellinia bunodes* (Berk & Br.) Sacc.; Sclerotium rot—*Sclerotium rolfsii* Sacc.; black rot—*Thielaviopsis* spp.; pink mold rot—*Trichothecium roseum* Link; and (b) bacterial: soft rot caused by *Erwinia* sp. (Gollifer and Booth 1973; Passam 1982; Quevedo and Ramos 1992; Quevedo et al. 1992; Snowdon 1992).

Several prestorage treatments for the control of storage pathogens have been tested. For example, dipping the roots in hot water (50°C) containing 200 ppm (a.i.) benomyl for 5 min reduced storage decay. After 2 weeks under ambient conditions, only 12% of the treated corms were lost compared with 65% for the controls (Quevedo et al. 1992). Corms from which the shoot apices and petiole bases had been removed had a shorter storage life than untreated ones (Wilson 1983); decay incidence was lower in the latter corms and was less still when they were stored in clamps (lower temperature and higher relative humidity). After 2

weeks, corms stored in clamps suffered only 6% losses vs. 22% among those in huts (Quevedo and Ramos 1992).

B. Ginger

Ginger (*Zingiber officinale* Rosc., Zingiberaceae) is grown for its underground rhizomes that are used raw (green) or processed as an aromatic condiment. A significant portion of the crop is processed, generally preserved in a brine solution or sugar syrup or as a dried product (Vasala 2001). The world production in 2001 was 835,000 t (FAO 2002), with India and China the leading producers. The unique aroma of ginger is due to a complex mixture of monoterpenes and sesquiterpenes (Govindarajan 1982a,b), with the dominant odorant being 6-gingerol (Connell and Sutherland 1969). The best rhizomes are those that are harvested immediately after they mature, i.e., when the above-ground plant parts die (Akamine 1962). Premature or delayed harvest increases postharvest losses. Factors that decrease the quality of the ginger during storage include surface shriveling and weight loss due to desiccation, decay, physiological breakdown, sprouting, and discoloration.

1. Prestorage Treatments. After harvest, the rhizomes are washed, air dried, and cured at 20°C, 70% RH for 7 days, after which they are cooled to the appropriate storage temperature using forced-air or room precooling.

2. Recommended Storage Conditions. Mature ginger rhizomes can be stored for 2–3 months at 12–14°C, 85–90% RH with only minor losses. Losses due to chilling injury and sprouting are avoided at this temperature range. Lower RH (e.g., 65%) results in excessive dehydration and a diminished appearance (Akamine 1962). Storage in polyethylene bags is not recommended because the high humidity increases sprouting, the development of fibrous roots, rotting, and discoloration. When refrigerated storage is not available, the rhizomes can be stored in pits covered with sand or dry grass in a shaded barn (Oti et al. 1988), but their maximum longevity is substantially less than under refrigeration.

3. Water Loss. Storage at a high relative humidity (i.e., 85–90%) is essential for preventing desiccation; rhizomes held at 65% RH lost approximately 16% of their water content in 6 months (Akamine 1962). Low RHs result in excessive moisture loss, while very high RHs promote pathogen attack. Moisture is readily lost from cut surfaces, so proper

curing is essential. Waxing does not appear to impede water loss, and the beneficial effects of polyethylene bags in decreasing desiccation are negated by increased decay (Akamine 1962).

4. Sprouting. At 21°C, 65% RH, sprouting limits the maximum storage potential of the rhizomes to about 1 month. Preharvest application of maleic hydrazide and postharvest application of CIPC reduced sprouting (Paull et al. 1988b), as did exposure to ionizing radiation (gamma or X-rays) (Mukherjee et al. 1995; Paull et al. 1988b). Excessively high exposures increase pathogen development (Akamine 1962).

5. Disorders

Chilling Injury. Exposure of the rhizomes to temperatures below 10°C results in chilling injury. Rhizomes that have been chilled soften and shrivel more readily. The degree of damage is a function of the temperature to which the rhizomes were exposed and the duration of exposure. For example, 2–3 wks storage at or below 7°C seriously damages the rhizomes (Akamine 1962). Physiological breakdown of the rhizomes is enhanced by exposure to chilling. Rhizomes that have been frozen readily degrade upon thawing.

Discoloration. Stored rhizomes may develop a purple discoloration on the cut surfaces, and possibly also, to a lesser degree, on the intact surfaces (Akamine 1962). Pigment development appears to be related to the presence of free moisture on the surface of the rhizomes, and storage in plastic bags accentuates the problem.

6. Postharvest Pathology. Many storage disease organisms attack ginger. They include Fusarium rot—*Fusarium* spp., Pythium rot—*Pythium* spp., Armillaria rot—*Armillaria mellea* (Vahl:Fr) Kummer, bacterial soft rot—*Erwinia carotovora* ssp. *carotovora* (Jones) Bergey et al., bacterial wilt—*Pseudomonas solanacearum* (E. F. Sm.) E. F. Sm., black rot—*Memnoniella echinata* (Rev.) Gall., blue mold rot—*Penicillium* spp., Botryodiplodia rot—*Botryodiplodia theobromae* Pat., charcoal rot—*Macrophomina phaseolina* (Tassi) Goid, gray rot—*Trichurus spiralis* Hasselbr., red rot—*Nectria inventa* Pethybr., Rosellina rot—*Rosellinia bunodes* (Berk and Br.) Sacc., and Sclerotium rot—*Sclerotium rolfsii* Sacc. (Snowdon 1992). Benomyl has been shown to decrease rotting (Okwuowulu and Nnodu 1988) and an antagonistic microorganism (an isolate of *Trichoderma*) was found to suppress the growth of *Sclerotium rolfsii* (Mukherjee et al. 1995). Treatment of the rhizomes with hot water,

sodium hypochlorite, and low-oxygen storage atmospheres each failed to control storage pathogens (Akamine 1962). Postharvest pathogens are best controlled by storing the rhizomes at the recommended temperature and RH.

7. Chemical Changes During Storage. The chemical composition of rhizomes stored at 12.5°C for 32 weeks remained remarkedly stable, with the exception of their gingerol concentration, which increased fivefold (Paull et al. 1988a). Fiber, oil, total phenolics, and protein concentrations did not change significantly, though the color of the rhizomes darkened from a very light yellow at harvest to a grayish yellow after 2 months. The concentration of total sugars increased on a fresh weight basis, but only after extended storage (28 weeks).

LITERATURE CITED

Adams, G. C., and B. R. Kropp. 1996. *Athelia arachnoidea*, the sexual state of *Rhizoctonia carotae*, a pathogen of carrot in cold storage. Mycologia 88:459–472.

Afek, U., and S. J. Kays. 2002. Minimizing sweetpotato weight loss during storage by using an atomizing humidification system. HortScience 37:755.

Afek, U., and J. Orenstein. 2002. Disinfecting potato tuber seeds by steam treatment. Can. J. Plant Pathol. 24:36–39.

Afek, U., J. Orenstein, and E. Nuriel. 1998a. Increased quality and prolonged storage of sweet potatoes in Israel. Phytoparasitica 26:307–312.

Afek, U., J. Orenstein, and E. Nuriel. 1998b. Prolonged storage and increased quality of stored carrots in Israel. (in Hebrew) Gan Sadeh Vameshek 9:62–70.

Afek, U., J. Orenstein, and E. Nuriel. 1999a. Fogging disinfectants inside storage rooms against pathogens of potatoes and sweet potatoes. Crop Prot. 18:111–114.

Afek, U., J. Orenstein, and E. Nuriel. 1999b. Steam treatment to prevent carrot decay during storage. Crop Prot. 18:639–642.

Afek, U., J. Orenstein, and E. Nuriel. 2000a. Inhibition of potato sprouting during storage by HPP (hydrogen peroxide plus). Am. J. Potato Res. 77:63–65.

Afek, U., J. Orenstein, and E. Nuriel. 2000b. Using the Tabor Atomizer System to maintain weight and firmness in stored potatoes. Am. J. Potato Res. 77: 203–205.

Afek, U., J. Orenstein, and J. J. Kim. 2001. Control of silver scurf disease in stored potato by using hydrogen peroxide plus (HPP). Crop Prot. 20:69–71.

Afek, U., and S. Warshavsky. 1998. Problems in storage of potatoes in Israel. (in Hebrew) p. 97–114. In: D. Levy (ed.), Potato in hot climate. Israel Agresearch, J. Agr. Res. Org., Israel.

Afek, U., and A. Wiseblum. 1995. Storage of sweet potatoes in Israel. (in Hebrew) Hassadeh 75:42–55.

Affran, D. K. 1968. Cassava and its economic importance. Ghana Farmer 12:172–178.

Ahn, J. K., W. W. Collins, and D. M. Pharr. 1980a. Influence of preharvest temperature and flooding on sweet potato roots in storage. HortScience 15:261–263.

Ahn, J. K., W. W. Collins, and D. M. Pharr. 1980b. Gas atmosphere in submerged sweet potato roots. HortScience 15:795–796.

Aiyer, R. S., P. G. Nair, and L. Prema. 1978. No-cost method for preserving fresh cassava roots. Cassava Newslett. 4:8–9.

Ajlouni, S., and M. K. Hamdy. 1988. Effect of combined gamma-irradiation and storage on biochemical changes in sweet potatoes. J. Food Sci. 53:477–481.

Akamine, E. K. 1962. Storage of fresh ginger rhizomes. Hawaii Agr. Expt. Sta. Bul. 130.

Amand, P. C., and W. M. Randle. 1989. Ethylene production and wound healing in sweet potato roots. HortScience 24:805–807.

Anonymous. 1952. Tapioca Enquiry Committee, Govt. Travancore-Cochin, India. Final Report. Trivandrum, India.

Anonymous. 1972. La yuca parafinada. Rev. Inst. Invest. Tecnol. Bogota 14:47–51.

Artschwager, E. 1924. On the anatomy of the sweet potato root with notes on internal breakdown. J. Agr. Res. 27:157–166.

Augustin, J. 1975. Variations in the nutritional composition of fresh potatoes. J. Food Sci. 40:1295–1299.

Averre, C. W. 1967. Vascular streaking of stored cassava roots. Proc. 1st Int. Symp. Trop. Root Crops, Trinidad 2:31–35.

Bacon, J. S. D., and R. Loxley. 1952. Seasonal changes in the carbohydrates of the Jerusalem artichoke tuber. Biochem. J. 51:208–213.

Bagni, N., A. Donini, and D. Serafini Fracassini. 1972. Content and aggregation of ribosomes during formation, dormancy, and sprouting of tubers of *Helianthus tuberosus*. Plant Physiol. 27:370–375.

Bancroft, R. D., and R. D. Crentsil. 1995. Application of a low-cost storage technique for fresh cassava (*Manihot esculenta*) roots in Ghana. p. 547–555. In: T. Agbor Egbe, A. Brauman, D. Griffon, and S. Treche (eds.), Transformation alimentaire du manioc. ORSTOM, Paris.

Banks, N. H., and S. J. Kays. 1988. Measuring internal gases and lentical resistance to gas diffusion in potato tubers. J. Am. Soc. Hort. Sci. 113:577–580.

Barloy, J. 1988. Jerusalem artichoke tuber diseases during storage. p. 145–149. In: G. Grassi and G. Gosse (eds.), Topinambour (Jerusalem artichoke). Proc. Workshop, Comm. European Communities, Madrid.

Bayday, D. S. 1922. Storage of some root crops and other perishable farm products. Philippine Agr. 10:423–440.

Been B. O., J. Marriott, and C. Perkins. 1975. Wound periderm formation in dasheens and its effects on storage. Caribbean Food Crops Society Meeting, Trinidad. p. 1–5.

Ben Chekroun, M., J. Amzile, M. el Yachioui, N. E. el Haloui, and J. Prevost. 1994. Qualitative and quantitative development of carbohydrate reserves during the biological cycle of Jerusalem artichoke (*Helianthus tuberosus* L.) tubers. New Zealand J. Crop Hort. Sci. 22:31–37.

Berg, L. van den, and C. P. Lentz. 1966. Effect of temperature, relative humidity, and atmospheric composition on changes in quality of carrots during storage. Food Technol. 20:954–979.

Berg, L. van den, and C. P. Lentz. 1973. High humidity storage of carrots, parsnips, rutabagas, and cabbage. J. Am. Soc. Hort. Sci. 98:129–132.

Berg, L. van den, and C. P. Lentz. 1977. Effect of relative humidity on storage life of vegetables. Acta Hort. 62:197–208.

Beveridge, J. L., J. Dalziel, and H. J. Duncan. 1981. The assessment of some volatile organic compounds as sprout suppressants for ware and seed potatoes. Potato Res. 24:61–76.

Bokanga, M. A., J. A. Essers, N. Poulter, H. Rosling, and O. Tewe (eds.). 1994. Int. workshop on cassava safety. Acta Hort. 375.

Booth, R. H. 1974. Post harvest deterioration of tropical root crops: losses and their control. Trop. Sci. 16:49–63.

Booth, R. H. 1976. Storage of fresh cassava (*Manihot esculenta*) I. Post harvest deterioration and its control. Exptl. Agr. 12:103–111.

Booth, R. H. 1977. Storage of fresh cassava (*Manihot esculenta*) II. Simple storage techniques. Exptl. Agr. 13:119–128.

Booth, R. H., and D. G. Coursey. 1974. Storage of cassava and related post harvest problems. p. 43–49. In: Cassava processing and storage. Ottawa, Canada, Int. Dev. Res. Centre Monogr. IDRC-031e

Booth, R. H., T. S. de Buckle, O. S. Cardenas, G. Gomez, and E. Hervas. 1976. Changes in quality of cassava roots during storage. J. Food Technol. 11:245–264.

Boswell, V. R. 1932. Length of rest period of the tuber of Jerusalem artichoke (*Helianthus tuberosus* L.). Proc. Am. Soc. Hort. Sci. 28:297–300.

Boyette, M. D., E. A. Estes, A. R. Rubin, and K. A. Sorensen. 1997. The postharvest handling of sweetpotatoes. North Carolina State Univ. Ext. Bul. AG-413-10.

Bradbury, J. H., and W. D. Holloway. 1988. Chemistry of tropical root crops: Significance for nutrition and agriculture in the Pacific. Austral. Cent. Intl. Agr. Res., Canberra.

Broadus, E. W., W. W. Collins, and D. M. Pharr. 1980. Incidence and severity of hardcore in sweet potatoes as affected by genetic line, curing and lengths of 1°C and 21°C storage. Scientia Hort. 13:105–113.

Brook, R. C., R. J. Fick, and T. D. Forbush. 1995. Potato storage design and management. Am. Potato J. 72:463–480.

Buescher, R. W., W. A. Sistrunk, J. L. Bowers, and C. F. Boettinger. 1975a. Susceptibility of sweet potato lines to development of hardcore, a chilling disorder. Ark. Farm Res. 24:2.

Buescher, R. W., W. A. Sistrunk, and P. L. Brady. 1975b. Effects of ethylene on metabolic and quality attributes in sweet potato roots. J. Food Sci. 40:1018–1020.

Buitelaar, N. 1987. Sprout inhibition in ware potato storage. p. 331–341. In: A. Rastovski and A. van Es (eds.), Storage of potatoes: Post-harvest behavior, store design, storage practice, handling. Pudoc, Wageningen, The Netherlands.

Burton, C. L. 1970. Diseases of tropical vegetables in the Chicago market. Trop. Agr. Trin. 47:303–313.

Burton, W. C., and M. J. Wiggington. 1970. The effect of a film of water upon the oxygen status of a potato tuber. Potato Res. 13:180–186.

Burton, W. G. 1958. The effect of the concentrations of carbon dioxide and oxygen in the storage atmosphere upon the sprouting of potatoes at 10°C. Eur. Potato J. 1:47–57.

Burton, W. G. 1963. Concepts and mechanism of dormancy. p. 17–41. In: J. D. Ivins and F. L. Milthorpe (eds.), The growth of the potato. Butterworths, London.

Burton, W. G. 1966. The potato. Second edition. Veenman and Zonen, Wageningen.

Burton, W. G. 1968. The effect of oxygen concentration upon sprout growth on the potato tuber. Eur. Potato J. 11:249–265.

Burton, W. G. 1969. The sugar balance in some British potato varieties during storage. II. The effect of tuber age, previous storage temperature and intermittent refrigeration upon low-temperature sweetening. Eur. Potato J. 12:81–95.

Burton, W. G., and A. R. Wilson. 1978. The sugar content and sprout growth of tubers of potato cultivar Record, grown in different localities, when stored at 10, 2 and 20°C. Potato Res. 21:145–162.

Burton, W. G., A. van Es, and K. J. Hartmans. 1992. The physics and physiology of storage. p. 608–727. In: P. M. Harris (ed.), The potato crop. Chapman and Hall, London.

Cappellini, R. A., M. J. Ceponis, and G. W. Lightner. 1987. Disorders of celery and carrot shipments to the New York market, 1972–1985. Plant Dis. 71:1054–1057.

Cargill, B. F., R. C. Brook, and T. D. Forbush. 1989. Potato storage technology and practice. Am. Soc. Agr. Eng., St. Joseph, Michigan.

Carlton, B. C., C. E. Peterson, and N. E. Tolbert. 1961. Effect of ethylene and oxygen on production of a bitter compound by carrot roots. Plant Physiol. 36:550–552.

Cassells, A. C., M. L. Deadman, and N. M. Kearney. 1988. Tuber diseases of Jerusalem artichoke (*Helianthus tuberosus* L.): production of bacterial-free material via meristem culture. p. 1–8. In EEC Workshop on Jerusalem Artichoke, Rennes, France.

Castagnino, G. A. 1943. Conservacion de la raiz de mandioca. El Campo (BuenosAires). 27:23.

Chang, C. T., H. Y. Liou, H. L. Tang, and H. Y. Sung. 1996. Activation, purification and properties of β-amylase from sweet potatoes (*Ipomoea batatas*). Biotechnol. Appi. Biochem. 24:13–18.

Chang, L. A., and S. J. Kays. 1981. Effect of low oxygen on sweet potato roots. J. Am. Soc. Hort. Sci. 106:481–483.

Chang, L. A., L. K. Hammett, and D. M. Pharr. 1982. Ethanol, alcohol dehydrogenase and pyruvate decarboxylase in storage roots of four sweet potato cultivars during simulated flood damage and storage. J. Am. Soc. Hort. Sci. 107:674–677.

Chang, L. A., L. K. Hammett, and D. M. Pharr. 1983. Carbon dioxide effects on ethanol production, pyruvate decarboxylase, alcohol dehydrogenase activities in anaerobic sweet potato roots. Plant Physiol. 71:59–62.

Charmara, I., D. Schlimme, and T. Solomos. 1998. Changes in sugars and activities of sucrose phosphate synthase, sucrose synthase, and invertase during potato tuber (Russet Burbank) reconditioning at 10°C in air and 2.53 kPa oxygen after storage for 28 days at 1°C. J. Am. Soc. Hort. Sci. 123:311–316.

Chervin, C., C. Triantaphylides, M. F. Libert, R. Siadous, and P. Boisseau. 1992. Reduction of wound-induced respiration and ethylene production in carrot root tissues by gamma irradiation. Posthar. Biol. Biotech. 2:7–17.

Chubey, B. B., and R. E. Nyland. 1970. The effect of maturity and environment on phenolic compounds and oxidative browning in carrots. J. Am. Soc. Hort. Sci. 95:393–395.

Claassen, P. A. M., M. A. W. Budde, and M. H. van Calker. 1993. Increase in phosphorylase activity during cold-induced sugar accumulation in potato tubers. Potato Res. 36:205–217.

Clark, C. A. 1992. Postharvest diseases of sweet potatoes and their control. Postharvest News Inform. 3:75–79.

Clark, C. A., and J. W. Moyer. 1988. Compendium of sweet potato diseases. p. 1–74. APS Press, St. Paul, MN.

Clark, C. A., P. D. Dukes, and J. W. Moyer. 1992. Diseases. p. 88–105. In: A. Jones and J. C. Bouwkamp (eds.), Fifty years of cooperative sweetpotato research 1939–1988. Southern Coop. Ser. Bul. 369.

Cochrane, M. P, C. M. Duffus, M. J. Allison, and G. R. Mackay. 1991a. Amylolytic activity in stored potato tubers. 1. Estimation using *p*-nitriphenyloligosaccharides. Potato Res. 34:325–332 .

Cochrane, M. P., C. M. Duffus, M. J. Allison, and G. R. Mackay. 1991b. Amylolytic activity in stored potato tubers. 2. The effect of low temperature storage on the activities of α- and β-amylase and α-glucosidase in potato tubers. Potato Res. 34:333–341.

Cocucci, S., and N. Bagni. 1968. Polyamine-induced activation of protein synthesis in ribosomal preparation from *Helianthus tuberosus* tissue. Life Sci. 7:113–120.

Coleman, W. K. 1987. Dormancy release in potato tubers: A review. Am. Potato J. 64:57–68.

Coleman, W. K. 1998. Carbon dioxide, oxygen and ethylene effects on potato tuber dormancy release and sprout growth. Ann. Bot. 82:21–27.

Coleman, W. K., and S. E. Coleman. 1986. The effects of bromoethane and ethanol on potato (*Solanum tuberosum*) tuber sprouting and subsequent yield responses. Am. Potato J. 63:373–377.

Coleman W. K, and J. McInerney. 1997. Enhanced dormancy release and emergence from potato tubers after exposure to a controlled atmosphere. Am. Potato J. 74:173–182.

Coleman, W. K., G. Hawkins, J. McInerney, and M. Goddard. 1992. Development of a dormancy release technology: A review. Am. Potato J. 69:437–445.

Condon, P., and J. Kuc. 1960. Isolation of a fungitoxic compound from carrot root tissue inoculated with *Ceratocytis fimbriata*. Phytopathology 50:267–270.

Connell, D. W., and M. D. Sutherland. 1969. A re-examination of gingerol, shogaol, and zingerone, the pungent principles of ginger (*Zingiber officinale* Roscoe). Austral. J. Chem. 22:1033–1043.

Cooke, R. D., J. E. Rickard, and A. K. Thompson. 1988. The storage of tropical root and root crops—cassava, yam and edible aroids. Exptl. Agric. 24:457–470.

Corey, K. A., W. W. Collins, and D. M. Pharr. 1982. Effect of duration of soil saturation on ethanol concentration and storage loss of sweet potato roots. J. Am. Soc. Hort. Sci. 107:195–198.

Cormany, C. E. 1928. Yields of Jerusalem artichoke tested. Mich. Agr. Expt. Sta. Quart. Bul. 10:156–158.

Cottrell, J. E., C. M. Duffus, L. Paterson, G. R. Mackay, M. J. Allison, and H. Bain. 1993. The effect of storage temperature on reducing sugar concentration and the activities of three amylolytic enzymes in tubers of the cultivated potato, *Solanum tuberosum* L. Potato Res. 36:107–117.

Courduroux, J. C. 1967. Étude du mécanisme physiologique de la tubérisation chez le Topinambour (*Helianthus tuberosus* L.). Ann. Sci. Nat. Bot. Biol. Vég. 8:215–356.

Coursey, D. G. 1968. The edible aroids. World Crops 20:25–30.

Cunningham, H. H., M. V. Zaehringer, and W. C. Sparks. 1971. Storage temperature for maintenance of internal quality in Idaho Russet Burbank potatoes. Am. Pot. J. 48:320–328.

Czyhrinciw, N., and W. Jaffe. 1951. Modificaciones quimicas durante la conservacion de raices y tuberculos. Arch. Venez. Nutr. 2:49–67.

Daie, J. 1984. Characterization of sugar transport in storage tissue of carrot. J. Am. Soc. Hort. Sci. 109:718–722.

Daines, R. H., M. J. Ceponis, and D. F. Hammond. 1974. Relationship of chilling to development of hardcore in sweet potatoes. Phytopathology 64:1459–1462.

Daines, R. H., D. F. Hammond, N. F. Haard, and M. J. Ceponis. 1976. Hardcore development in sweet potatoes. A response to chilling and its remission as influenced by cultivar, curing temperatures and time and duration of chilling. Phytopathology 66:582–587.

Daniels-Lake, B. J., P. K. Prange, W. Kait, C. L. Liew, J. Walsh, P. Dean, and R. Coffin. 1996. The effects of ozone and 1,8-cineole on sprouting, fry color and sugar of stored russet Burbank potatoes. Am. Potato J. 73:469–481

D'Antonio, V. L., and N. A. McHale. 1988. Effect of storage temperature and extraction methods on dormancy and germination of true potato seed. Am. Potato J. 65:573–581.

Davies, W. P., and B. G. Lewis. 1980. The inter-relationship between the age of carrot roots at harvest and infection by *Mycocentrospora acerina* in storage. Ann. Appl. Biol. 95:11–17.

de Buckle, S. Teresa, H. Castelblanco, L. E. Zapata, M. F. Bocanegra, L. E. Rodriguez, and D. Rocha. 1973. Preservation of fresh cassava by the method of waxing. Rev. Inst. Invest. Tecnol, Bogota 15:33–47.

Decaisne, J. 1880. *Helianthus tuberosus* (Topinambour, Poire de terre). Flore des Serres 23:112–119.

Deiting, U., R. Zrenner, and M. Stitt. 1998. Similar temperature requirement for sugar accumulation and for the induction of new forms of sucrose phosphate synthase and amylase in cold-stored potato tubers. Plant Cell Environ. 21:127–138.

Delate, K. M., and J. K. Brecht. 1989. Quality of tropical sweetpotatoes exposed to controlled-atmosphere treatment for postharvest insect control. J. Am. Soc. Hort. Sci. 114:963–968.

Dempsey, A. H., L. J. Kushman, and J. E. Love. 1970. Storage. p. 36–38. In: Thirty years of cooperative sweet potato research, 1939–1969. Southern Coop. Series Bul. 159.

Denny, F. E., N. C. Thornton, and E. M. Schroeder. 1944. The effect of carbon dioxide upon the changes in the sugar content of certain vegetables in cold storage. Cont. Boyce Thompson Inst. 13:295–311.

Deobald, H. J., V. C. Hasling, and E. A. Catalano. 1971. Variability of increases in α-amylase and sugars during storage of Goldrush and Centennial sweet potatoes. J. Food Sci. 36:413–415.

Drummond, O. A. 1953. Da etiologia de rajas pretas das raizes de manidoca. Anais de Congreso Nacional da Sociedade Botanica do Brasil, 4, Recife. Anais. Recife Brasil, 57-M.

Dubost, G., and M. Gendraud. 1987. Activité phosphoenolpyruvate carboxylase dans les parenchymes de tubercules de Topinambour dormants ou non dormants: relation avec le pH intracellulaire. C.R. Acad. Sci. Paris 305:619–622.

Edgar, A. D. 1968. Storage of potatoes. p. 344–358. In: O. Smith (ed.), Potatoes: Production, storing, processing. AVI, Westport, CT.

Ekundayo, J. A., and T. M. Daniel. 1973. Cassava rot and its control. Trans. Brit. Mycol. Soc. 61:27–32.

Emilsson, B. 1949. Studies on the rest period and dormant period in the potato tuber. Acta Agr. 3:189–284.

Es, A. van, and K. J. Hartmans. 1987a. Starch and sugar during tuberization, storage and sprouting. p. 79–113. In: A. Rastovski and A. van Es (eds.), Storage of potatoes: Post-harvest behavior, store design, storage practice, handling. Pudoc, Wageningen, The Netherlands.

Es, A. van, and K. J. Hartmans. 1987b. Dormancy, sprouting and sprout inhibition. p. 114–132. In: A. Rastovski and A. van Es (eds.), Storage of potatoes: Post-harvest behavior, store design, storage practice. Pudoc, Wageningen, The Netherlands.

Es, A. van, and K. J. Hartmans. 1987c. Water balance of potato tuber. p. 141–147. In: A. Rastovski and A. van Es (eds.), Storage of potatoes: Post-harvest behavior, store design, storage practice. Pudoc, Wageningen, The Netherlands.

Esashi, Y. 1991. Ethylene and seed germination. p. 133–157. In: A. K. Mattoo and J. C. Suttle (eds.), The plant hormone ethylene. CRC Press, Boca Raton, FL.

Espen, L., S. Morgutti, A. Abruzzese, N. Negrint, A. Rivetta, M. M. Quattrini, M. Cocuccu, and S. M. Cocucci. 1999a. Changes in the potato (*Solanum tuberosum* L.) tuber at the onset of dormancy and during storage at 23°C and 3°C. 1. Biochemical and physiological parameters. Potato Res. 42:189–201.

Espen, L., S. Morgutti, and S. M. Cocucci. 1999b. Changes in the potato tuber (*Solanum tuberosum* L.) in the onset of dormancy and during storage at 23°C and 3°C. 2. Evaluation of protein patterns. Potato Res. 42:202–214.

Ezell, B. D., and M. S. Wilcox. 1952. Influence of storage temperature on carotene, total carotenoids and ascorbic acid content of sweet potatoes. Plant Physiol. 27:81–94.

Fan, M. L. 1975. Partial purification and properties of potato beta-amylase. Taiwania 20:132–138.

FAO. 2002. (www.apps.fao.org/lim500/agri_db.pl)

Farnworth, E. R. 1993. Fructans in human and animal diets. p. 257–272. In: M. Suzuki and N. J. Chatterton (eds.), Science and technology of fructans. CRC Press, Boca Raton, FL.

Fritz, D., and J. Weichmann. 1979. Influence of the harvesting date of carrots on quality and quality preservation. Acta Hort. 93:91–100.

Gallat, S., D. Crentsil, and R. D. Bancroft. 1998. Development of a low cost cassava fresh root storage technology for the Ghanaian market. p. 77–84. In: R. S. B. Ferris (ed.), Postharvest technology and commodity marketing. Proc. Postharvest Conf., IITA, Ibadan, Nigeria.

Gendraud, M. 1975a. Quelques aspets du métabolisme des acides ribonecléotides libres en relation avec i'édification des tubercules de Topinambour (*Helianthus tuberosus* L.). Thèse de Doctorat. Univ. de Clermont-Ferrand, France.

Gendraud, M. 1975b. Tuberisation; dormance et synthèse in situ des RNA chez le Topinambour (*Helianthus tuberosus* L. cv. D-19). Biol. Plant. 17:17–22.

Gendraud, M., and J. Lefleuriel. 1983. Charactéristiques de l'absorption du saccharose et du tétraphénylphosphonium par les parenchymes de tubercules de Topinambour, dormants et non dormants, cultivés in vitro. Physiol. Vég. 21:1125–1133.

Gendraud, M., and J. C. Prévôt. 1973. Mesure in situ de l'incorporation d'un précurseur dndogène dans les ARN des tubercules de Topinambour cultivés in vitro en fonction de leur état de dormance. Etude critique et exemple de résultats. Physiol. Vég. 11:417–427.

Gerbrandy, S. J., V. Shankar, K. M. Shivaram, and H. Stegemann. 1975. Conversion of potato phosphorylase isozymes. Phytochemistry 14: 2331–2333.

Goillifer, D. E., and R. H. Booth. 1973. Storage loss of taro in British Solomon Islands Protectorate. Ann. Appl. Biol. 75:349–356.

Golob, P. 1988. Current status of the Larger Grain Borer (*Prostephanus truncatus* Horn) in Africa. Insect Sci. Applic. 9:737–745.

Gomez, G. 1979. Cassava as a swine feed. World Animal Rev. 29:13–20.

Goodwin, P. B. 1966. The effect of water on dormancy in the potato. Eur. Potato J. 9:53–63.

Goris, A. 1969. Les sucres de la racine de carotte cultivee (variete Nantaise demi-longue): variations climatiques et saisonnieres, repartition dans les tissus, modification au cours du stockage. Qual. Plant. Mater. Veg. 18:283–306.

Govindarajan, V. S. 1982a. Ginger—Chemistry, technology and quality evaluation: Part 1. Crit. Rev. Food Sci. Nutr. 17:1–96.

Govindarajan, V. S. 1982b. Ginger—Chemistry, technology and quality evaluation: Part 2. Crit. Rev. Food Sci. Nutr. 17:189–258.

Hahn, S. K., and T. Anota. 1982. l. The effect of temperature on the development of the sweet potato weevil. IITA Annu. Rept. 1981, Ibadan, Nigeria.

Hajslova, J., and J. Davidek. 1986. Sprout inhibitors IPC and CIPC in treated potatoes. Nahrung Food 30:75–79.

Hammett, L. K., and C. H. Miller. 1982. Influence of mineral nutrition and storage on quality factors of 'Jewel' sweet potatoes. J. Am. Soc. Hort. Sci. 107:972–975.

Hanover, P. 1960. Formation et débourrement de nouveaux tubercules axillaires sur les tubercules mères de Topinambour et phénomènes accompagnant les degrés de levée de dormance de ces plantes. C.R. Acad. Sci. 251:2267–2769.

Hardenburg, R. E., M. Lieberman, and H. A. Schomer. 1953. Prepackaging carrots in different types of consumer bags. Proc. Am. Soc. Hort. Sci. 61:404–412.
Harkett, P. J. 1981. External factors affecting length of dormant period in potato. J. Sci. Food Agr. 32:102–103.
Harter, L. L., J. I. Lauritzen, and J. L. Weimer. 1923. Internal breakdown of sweet potatoes. Phytopathology 13:146–147.
Hartmans, K., and A. van Es. 1979. The influence of growth regulators GA_3, ABA, kinetin and IAA on sprout and root growth and plant development using excised potato buds. Potato Res. 22:319–332.
Heinze, P. H., and C. O. Appleman. 1943. A biochemical study of the curing processes in sweet potatoes. Plant Physiol. 18:548–555.
Hemberg, T. 1985. Potato rest. p. 353–388. In: P. H. Li (ed.), Potato physiology. Academic Press, Orlando, FL.
Herppich, W. B., H. Mempel, and M. Geyer. 1999. Effects of postharvest mechanical and climatic stress on carrot tissue water relations. Postharv. Biol. Biotech. 16:43–49.
Hide, G. A., and D. H. Lapwood. 1992. Disease aspects of potato production. p. 403–437. In: P. M. Harris (ed.), The potato crop. Chapman and Hall, London.
Hilborn, M. T., and R. Bonde. 1942. A new form of low temperature injury in potatoes. Am. Pot. J. 19:24–29.
Hill, L., R. Reimholz, R. Schroder, T. H. Nielsen, and M. Stitt. 1996. The onset of sucrose accumulation in cold stored potato tubers is caused by an increased rate of sucrose synthesis and coincides with low levels of hexose phosphates, an activation of sucrose phosphate synthase and the appearance of a new form of amylase. Plant Cell Environ. 19:1223–1237.
Hillocks, R. J., J. M. Thresh, and A. C. Bellotti (eds.). 2002. Cassava biology, production and utilization. CABI, Wallingford, England.
Hirayama, H., and H. Hidaka. 1993. Production and characteristics of fructo-oligosaccharieds. p. 347–353. In: Suzuki, M. and N. J. Chatterton (eds.), Science and technology of fructans, CRC Press, Boca Raton, FL.
Hodges, R. J. 1986. The biology and control of *Prostephanus truncatus*—a destructive pest with an increasing range. J. Stored Prod. Res. 221–214.
Hole, C. C., R. L. K. Drew, B. M. Smith, and D. Gray. 1999. Tissue properties and propensity for damage in carrot (*Daucus carota* L.) storage roots. J. Hort. Sci. Biotech. 74:651–657.
Hurschka, H. W. 1977. Post-harvest moisture loss and shrivel in five fruits and vegetables. U.S.D.A. Agr. Res. Ser., Mkt. Res. Rept. 1059.
Ingram, J., and J. R. O. Humphries. 1972. Cassava storage—A review. Trop. Sci. 14:131–148.
Iritani, W. M., and L. Weller. 1978. Factors influencing reconditioning of Russet burbank potatoes. Am. Potato J. 55:425–430.
Isherwood, F. A. 1973. Effects of changes in storage temperature on the metabolism of the potato tuber. Proc. 5th Triennial Conf. EAPR., Norwich. 1972:156–157.
Isherwood, F. A. 1976. Mechanism of starch-sugar interconversion in *Solanum tuberosum*. Phytochemistry 15:33–41.
Jadhav, S. J., and D. K. Salunkhe. 1975. Formation and control of chlorophyll and glycoalkaloids in tubers of *Solanum tuberosum*, L. and evaluation of glycoalkaloid toxicity. Adv. Food Res. 21:307–354.
Jefford, T. G., and J. Edelman. 1960. Changes in content and composition of the fructose polymers of *Helianthus tuberosus* L. during growth of daughter plants. J. Expt. Bot. 12:177–187.

Jefford, T. G., and J. Edelman. 1963. The metabolism of fructose polymers in plants. II. Effect of temperature on the carbohydrate changes and morphology of stored tubers of *Helianthus tuberosus* L. J. Expt. Bot. 14:56–62.

Jenkins, W. F., and M. Gieger. 1957. Quality in baked sweet potatoes affected by varieties and post-harvest treatments. Food Res. 22:32–36.

Johnson, H. W. 1931. Storage rots of the Jerusalem artichoke. J. Ag. Res. 43:337–352.

Kalt, W., R. K. Prange, and B. J. Daniels-Lake. 1999. Alternative compounds for the maintenance of processing quality of stored potatoes (*Solanum tuberosum*). J. Food Proc. Preserv. 23:71–81.

Kays, S. J. 1997. Postharvest physiology of perishable plant products. Exon Press, Athens, GA.

Kays, S. J., W. W. Collins, and J. C. Bouwkamp. 1992. A response: The sweetpotato storage organ is a root, not a tuber. p. 307–313. In: W. A. Hill, C. K. Bonsi, and P. A. Loretan (eds.), Sweetpotato technology for the 21st century. Tuskegee Univ., Tuskegee, AL.

Kays, S. J., and R. J. Horvat. 1983. A comparison of the volatile constituents and sugars of representative Asian, Central American and North American sweet potatoes. 6th Intern. Symp. Trop. Root Crops, p. 577–587.

Kays, S. J., and S. E. Kays. 1998. Sweetpotato chemistry in relation to health. p. 231–272. In: Sweetpotato production systems toward the 21st century. Kyushu National Agri. Expt. Sta., Miyakonojo, Miyazaki, Japan.

Kays, S. J., and J. C. Silva Dias. 1996. Cultivated vegetables of the world: Latin binominal, common name in 15 languages, edible part, and method of preparation. Exon Press, Athens, GA.

Kays, S. J., and G. W. Stutte. 1979. Proximal dominance and sprout formation in sweet potato [*Ipomoea batatas* (L.) Lam.] root pieces. 5th Intern. Symp. Trop. Root Crops, Manila, p. 41–47.

Kays, S. J., and Y. Wang. 2000. Thermally induced flavor compounds. HortScience 35:1002–1012.

Kennedy, M. G. H., and F. A. Isherwood. 1975. Activity of phosphorylase in *Solanum tuberosum* during low temperature storage. Phytochemistry 14:667–670.

Kiewnick, S., and B. J. Jacobsen. 1997. Control of *Rhizoctonia* black scurf and *Fusarium* dry rot in potatoes with fungicides and antagonistic bacteria. Phytopathology 87:51–52.

Killilea, S. D., and M. J. Clancy. 1978. Properties of potato α-glucosidase. Phytochemistry 17:1429–1431.

Kirki, L. 1971. Moisture in vegetable storage. Acta Hort. 20:146–151.

Kleinkopf, G. F. 1995. Early season storage. Am. Potato J. 72:449–462.

Kokooras, I. F. 1989. The effect of temperature and water status on the susceptibility of carrot roots to damage. Thesis, Cranfield Inst. Tech., Silsoe, England.

Krahn, R. 1974. Long term storage of perishable vegetables. Acta Hort. 38:443–450.

Krijthe, N. 1962. Observations on the sprouting of seed potatoes. Eur. Potato J. 5:316–333.

Kushman, L. J. 1975. Effect of injury and relative humidity during curing on weight and volume loss of sweet potatoes during curing and storage. HortScience 10:275–277.

Kushman, L. J., and D. T. Pope. 1972. Causes of pithiness in sweet potatoes. N.C. Agr. Expt. Sta. Tech. Bul. 207.

Lafuente, M. T., G. Lopez-Galvez, M. Cantwell, and S. F. Yang. 1996. Factors influencing ethylene-induced isocoumarin formation and increased respiration in carrots. J. Am. Soc. Hort. Sci. 121:537–542.

Lambou, M. G. 1958. Effects of curing, storage and dehydration on the mono and disaccharides of the sweet potato. Food Technol. 12:150–155.

Lancaster, P. A., J. S. Ingram, M. Y. Lim, and D. G. Coursey. 1982. Traditional cassava based foods: a survey of processing techniques. Econ. Bot. 36:12–45.

Le Dily, F., F. Villeneuve, and J. Boucaud. 1993. Qualite et maturite de la racine de carotte: influence de la conservation au champ et au froid humide sur la composition biochirnique. Acta Hort. 354:187–199.

Lefebvre, A. H. 1989. Atomization and sprays. Hemisphere Pub., New York.

Le Floc'h, F., and J. Lafleuriel. 1983a. The AMP aminohydrolase of Jerusalem artichoke tubers: partial purification and properties. Physiol. Vég. 21:15–27.

Le Floc'h, F., and J. Lafleuriel. 1983b. The role of mitochondria in the recycling of adenine into purine nucleotides in the Jerusalem artichoke (*Helianthus tuberosus* L.) Z. Pflanzenphysiol. 13:61–71.

Le Floc'h, F., J. Lafleuriel, and A. Guillot. 1982. Interconversion of purine nucleotides in Jerusalem artichoke shoots. Plant Sci. Lett. 27:309–316.

Lewis, D. A., and L. L. Morris. 1956. Effects of chilling storage on respiration and deterioration of several sweet potato varieties. Proc. Am. Soc. Hort. Sci. 68:421–428.

Lewis, M. D., G. E. Kleinkopf, and K. K. Shetty. 1997. Dimethylnaphthalene and diisopropylnaphthalene for potato sprout control in storage. Application methodology and efficacy. Am. Potato J. 74:183–197.

Lieberman, M., C. C. Craft, W. V. Audia, and M. S. Wilcox. 1958. Biochemical studies of chilling injury in sweet potatoes. Plant Physiol. 33:307–311.

Liew, C. L., and R. K. Prange. 1994. Effect of ozone and storage temperature on postharvest diseases and physiology of carrots (*Daucus carota* L.). J. Am. Soc. Hort. Sci. 119:563–567.

Lozano, J. C., J. M. Cock, and J. Castano. 1978. New developments in cassava storage. p. 38–45. In: Proc. cassava protection workshop, CIAT, Cali, Colombia, Series CE-14.

Lu, J. Y., S. White, P. Yakuku, and P. A. Loretan. 1986. Effects of gamma radiation on nutritive and sensory qualities of sweetpotato storage roots. J. Food Quality 9:425–435.

Ludwig, J. W. 1970. De Aardappel en aardappelprodukten. Syllabus van de studiest of voor de MO-akte N 8, IBVL-Wageningen.

Lutz, J. M. 1945. Chilling injury of cured and non-cured Puerto Rico sweet potatoes. USDA Circ. 729.

Lutz, J. M., and J. W. Simons. 1958. Storage of sweet potatoes. USDA Farmers Bull. 1442.

Majumder, S. K. 1955. Some studies on the microbial rot of tapioca. Bull. Cent. Food Technol. Res. Inst. Mysore 4:164.

Majumder, S. K., S. Y. Pingale, M. Swaminathan, and V. Subrahmanyam. 1956. Control of spoilage in fresh tapioca roots. Bul. Cent. Food Technol. Res. Inst. Mysore 5:108–109.

Marriott, J., B. O. Been, and C. Perkins. 1974. Storage of fresh cassava roots in moist coir dust. Proc. Carib. Food Crop Soc. 12:79–81.

Martin, W. J. 1949. Internal cork of sweet potatoes. Annu. Rept. Louisiana Agr. Expt. Sta. 1947–48:98.

Martin, W. J. 1958. Distal-end rot of sweetpotatoes of the Acadian variety. Plant Dis. 42:983.

Masuda, Y. 1965. Auxin-induced growth of tuber tissue of Jerusalem artichoke. I. Cell physiological studies on the expansion growth. Bot. Mag. 78:417–423.

Masuda, Y. 1967. Auxin-induced expansion growth of Jerusalem artichoke tuber tissue in relation to nucleic acid and protein metabolism. Ann. N.Y. Acad. Sci. 144:68–80.

Mattoo, A. K., and W. B. White. 1991. Regulation of ethylene biosynthesis. p. 21–42. In: A. K. Mattoo and J. C. Suttle (eds.), The plant hormone ethylene. CRC Press, Boca Raton, FL.

McCarter, S. M., and S. J. Kays. 1984. Diseases limiting production of Jerusalem artichokes in Georgia. Plant Dis. 64:299–302.

McClure, T. T. 1960. Chlorogenic acid accumulation and wound healing in sweet potato roots. Am. J. Bot. 47:277–280.

McGarry, A. 1993. Influence of water status on carrot (*Daucus carota* L.) fracture properties. J. Hort. Sci. 68:431–437.

McGarry, A. 1995. Cellular basis of tissue toughness in carrot (*Daucus carota* L.) storage roots. Ann. Bot. 75:157–163.

Meijers, C. P. 1987a. Diseases and defects liable to affect potatoes during storage. p. 48–174. In: A. Rastovski and A. van Es (eds.), Storage of potatoes: Post-harvest behavior, store design, storage practice, handling. Pudoc, Wageningen, The Netherlands.

Meijers, C. P. 1987b. Wound healing. p 328–330. In: A. Rastovski and A. van Es (eds.), Storage of potatoes: Post-harvest behavior, store design, storage practice, handling. Pudoc, Wageningen, The Netherlands.

Millington, S. 1984. Quality of carrots in the United Kingdom with respect to harvester and packinghouse damage. Acta Hort. 163:127–136.

Modler, H. W., J. D. Jones, and G. Mazza. 1993a. Observations of long-term storage and processing of Jerusalem artichoke tubers (*Helianthus tuberosus*). Food Chem. 48:279–284.

Modler, H. W., J. D. Jones, and G. Mazza. 1993b. The effect of long-term storage of the fructo-oligosaccharide profile of Jerusalem artichoke tubers and some observations on processing. p. 57–64. In: A. Fuch (ed.), Inulin and inulin-containing crops. Elsevier Science, Amsterdam.

Montaldo, A. 1973. Vascular streaking of cassava roots. Trop. Sci. 15:39–46.

Morrell, S., and T. Rees. 1986a. Control of the hexose content of potato tubers. Phytochemistry 25:1073–1076.

Morrell, S., and T. Rees. 1986b. Sugar metabolism in developing tubers of *Solanum tuberosum*. Phytochemistry 25:1579–1585.

Morris, L. L., and L. K. Mann. 1955. Wound healing, keeping quality and compositional changes during curing and storage of sweet potatoes. Hilgardia 24:143–183.

Morrison, T. A., R. Pressey, and S. J. Kays. 1988. Amylase activity in stored staple-type sweet potatoes. HortScience 23:767 (Abstr.).

Moyer, J. W. 1982. Postharvest disease management for sweet potatoes. p. 177–184. In: Sweet potato. Proc. First Intl. Symp., AVRDC, Taiwan.

Mukherjee, P. K., P. Thomas, and K. Raghu. 1995. Shelf-life enhancement of fresh ginger rhizomes at ambient temperatures by combination of gamma-irradiation, biocontrol and closed polyethylene bag storage. Appl. Biol. 127:375–384.

Murthy, H. B. N., G. R. Rao, and M. Swaminathen. 1956. Studies on the starch synthesizing enzymes in tapioca (*Manihot utilissima*) root. Enzymologia 18:63–75.

Nair, C. N., and P. A. Kurup. 1963. Phosphorylase inhibitor in the rind of tapioca root. Naturwissenschaften 50:667.

Napier, A. S. 1906. Contributions to old English lexicography. Trans. Philological Soc., London 10:265–385.

Nielsen, L. W. 1952. Effect of temperature on the development of internal cork lesions in sweet potato roots. Phytopathology 42:625–627.

Nielsen, T. H., U. Deiting, and M. Stitt. 1997. β-Amylase in potato tubers is induced by storage at low temperature. Plant Physiol. 113:503–510.

Niemira, B. A., R. Hammerschmidt, and G. R. Safir. 1996. Postharvest suppression of potato dry rot (*Fusarium sambucinum*) in prenuclear minitubers by arbuscular mycorrhizal fungal inoculum. Am. Potato J. 73:509–515.

Nilsson, T. 1987a. Growth and chemical composition of carrots as influenced by the time of sowing and harvest. J. Agr. Sci. (Camb.) 108:459–468.

Nilsson, T. 1987b. Carbohydrate composition during long-term storage of carrots as influenced by the time of harvest. J. Hort. Sci. 62:191–203.

Normanha, E. S., and R. S. Pereira. 1963. Cultura de mandioca. O Agronomico, Sao Paulo 15:9–35.
Nottingham, S. F., and S. J. Kays. 2002. Strategies for sweetpotato weevil control. Acta Hort. 583:155–161.
Nusbaum, C. J. 1946. Internal cork, a new disease of sweet potato of unidentified cause. Phytopathology 36:18–23.
Nusbaum, C. J. 1950. Internal cork of sweet potato. South Carolina Agr. Expt. Bul. 381.
Ohad, I., I. Friedberg, Z. Ne'eman, and M. Schramm. 1971. Biogenesis and degradation of starch. The fate of the amyloplast membranes during maturation and storage of potato tubers. Plant Physiol. 47:465–477.
Okwuowulu, P. A., and E. C. Nnodu. 1988. Some effects of pre-storage chemical treatments and age at harvest on the storability of fresh ginger rhizomes (*Zingiber officinale* Roscoe). Trop. Sci. 28:123–125.
Olden, B., and T. Nilsson. 1992. Acid and alkaline invertase activities in carrot during root development and storage. Swed. J. Agr. Res. 22:43–47.
Oosterhaven, K., B. Poolman, and E. J. Smid. 1995. S-carvone, as a natural potato sprout inhibition, fungistatic and bacteristatic compound. Ind. Crops Prod. 4:23–31.
Oti, E., P. A. Okwuowulu, and G. O. Chijioke. 1988. Biochemical changes in ginger (*Zingiber officinale* Roscoe) rhizomes stored under river sand and under dry grass in pits in the humid tropics. Trop. Sci. 28:87–94.
Ottono, P., and D. Charnay. 1986. Absorption de l'acide abscissique dans les parenchymes de tubercules de Topinambour dormants et non dormants: mise en évidence d'un effet de charge des parois. C.R. Acad. Sci. Paris 303:415–418.
Oudit, D. D. 1976. Polyethylene bags keep cassava roots fresh for several weeks at ambient temperature. J. Agr. Soc. Trin. Tobago. 76:63–66.
Pacheco, J. A. 1952. Alteracoes de qualidade da fecula durante o armazenenamento das raizes de mandioca. Bragantia 12:297–298.
Padmaja, G. 1995. Cyanide detoxification in cassava for food and feed used. Crit. Rev. Food Nutr. 35:299–399.
Pardales, J. R. 1987. State of the art for Taro. Root Crop Anniversary Symp. PRCRTC, ViSCA, Baybay, Leyte, Philippines.
Parsons, C. S., and R. H. Day. 1970. Freezing injury of root crops: beets, carrots, parsnips, radishes, and turnips. U.S. Dept. Agr. Mkt. Res. Rpt. 866.
Passam, H. C. 1982. Experiments on the storage of eddoes and tannias (*Colocasia* and *Xanthosoma* spp.) under tropical ambient conditions. Trop. Sci. 24:39–46.
Paull, R. E., N. J. Chen, and T. T. C. Goo. 1988a. Compositional changes in ginger rhizomes during storage. J. Am. Soc. Hort. Sci. 113:584–588.
Paull, R. E., N. J. Chen, and T. T. C. Goo. 1988b. Control of weight loss and sprouting of ginger rhizome in storage. J. Am. Soc. Hort. Sci. 113:734–736.
Paull, R. E., C. S. Tang, K. Gross, and G. Uruu. 1999. The nature of the taro acridity factor. Postharv. Biol. Technol. 16:71–78.
Peiris, K. J. S., J. L. Mallon, and S. J. Kays. 1997. Respiratory rate and vital heat of some specialty vegetables at various storage temperatures. HortTechnology 7:46–48.
Perombelon, M. C. M., V. M. Lumb, and L. J. Hyman. 1987a. A rapid method to identify and quantify soft rot Erwinia on seed potato tubers. EPPO Bul. 17:25–35.
Perombelon, M. C. M., V. M. Lumb, and D. Zutra. 1987b. Pathogenicity of soft rot Erwinia to potato plants in Scotland and Israel. J. Appl. Bacterial. 63:73–84.
Phan, C., and H. Hsu. 1973. Physical and chemical changes occurring in the carrot root during growth. Can. J. Plant Sci. 53:629–634.
Phan, C., H. Hsu, and S. K. Sarkar. 1973. Physical and chemical changes occurring in the carrot root during storage. Can. J. Plant Sci. 53:635–641.

Picha, D. H. 1986a. Carbohydrate changes in sweet potatoes during curing and storage. J. Am. Soc. Hort. Sci. 111:89–92.

Picha, D. H. 1986b. Weight loss in sweet potatoes during curing and storage: Contribution of transpiration and respiration. J. Am. Soc. Hort. Sci. 111:889–892.

Picha, D. H. 1987. Chilling injury, respiration and sugar changes in sweet potatoes stored at low temperature. J. Am. Soc. Hort. Sci. 112:497–502.

Platenius, H., F. S. Jamison, and H. C. Thompson. 1934. Studies on cold storage of vegetables. N.Y. (Cornell) Agr. Expt. Sta. Bul. 602.

Plumbley, R. A., and J. E. Rickard. 1991. Post-harvest deterioration of cassava. Trop. Sci. 31:295–303.

Porter, W. C., D. M. Pharr, L. J. Kushman, and D. T. Pope. 1976. Discoloration of chilled sweet potato [*Ipomoea batatas* (L.) Lam] roots: Factors related to cultivar differences. J. Food Sci. 41:938–941.

Purcell, A. E., and W. M. Walter, Jr. 1980. Changes in composition of the non-protein nitrogen fraction of 'Jewel' sweet potatoes [*Ipomoea batatas* (Lam.)] during storage. J. Agr. Food Chem. 28:842–844.

Purcell, A. E., W. M. Walter, Jr., and F. G. Giesbrecht. 1978. Changes in dry matter, protein and non-protein nitrogen during storage of sweet potatoes. J. Am. Soc. Hort. Sci. 103:190–192.

Quevedo, M. A., and A. D. Ramos. 1992. Method of corm preparation for storage of taro. Trop. Sci. 32:121–127.

Quevedo, M. A., R. T. Sanico, and M. E. Baliad. 1992. The control of post-harvest diseases of taro corms. Trop. Sci. 31:359–364.

Rama, M. V., and P. Narasimham. 1986. Heat treatments for the control of sprouting of potatoes during storage. Ann. Appl. Biol. 108:597–603.

Ramsey, G. B., J. S. Wiant, and M. A. Smith. 1949. Market diseases of fruits and vegetables; potatoes. U.S. Dept. Agr. Misc. Publ. 98.

Ranganna, B., A. C. Kushalappa, and G. S. V. Raghavan. 1997. Ultraviolet irradiance to control dry rot and soft rot of potato in storage. Can. J. Plant Pathol. 19:30–35.

Rastovski, A. 1987. Storage losses. p. 177–180. In: A. Rastovski and A. van Es (eds.), Storage of potatoes: Post-harvest behavior, store design, storage practice, handling. Pudoc, Wageningen, The Netherlands.

Ray, R. C., S. R. Chowdhury, and C. Balagopalan. 1994. Minimizing weight loss and microbial rotting of sweet potatoes (*Ipomoea batatas* L.) in storage under tropical ambient conditions. Adv. Hort. Sci. 8:159–163.

Rees, T., W. L. Dixon, C. J. Pollock, and F. Franks. 1981. Low temperature sweetening of higher plants. p. 41–61. In: J. Friend and M. J. C. Rhodes (eds.), Recent advances in the biochemistry of fruits and vegetables. Academic Press, London.

Ricardo, C. P. P., and D. Sovia. 1974. Development of tuberous roots and sugar accumulation as related to invertase activity and mineral nutrition. Planta 118:43–55.

Rich, A. E. 1968. Potato diseases. p. 397–437. In: O. Smith (ed.), Potatoes: Production, storing, processing. AVI, Westport, CT.

Rolston, L. H., C. A. Clark, J. M. Cannon, W. M. Randler, E. G. Riley, P. W. Wilson, and M. L. Robbins. 1987. 'Beauregard' sweet potato. HortScience 22:1338–1339.

Ross, H. A., and H. V. Davies. 1992. Sucrose metabolism in tubers of potato (*Solanum tuberosum* L.): Effect of sink removal and sucrose flux on sucrose-degrading enzymes. Plant Physiol. 98:287–293.

Rutherford, P. P., and E. W. Weston. 1968. Carbohydrate changes during cold storage of some inulin-containing roots and tubers. Phytochemistry 7:175–180.

Ryall, A. L. and W. J. Lipton. 1979. Handling, transportation and storage of fruits and vegetables. Vol. 1. Vegetables and melons. 2nd ed. AVI, Westport, CT.

Rylski, I., L. Rappaport, and H. K. Pratt. 1974. Dual effects of ethylene on potato dormancy and sprout growth. Plant Physiol. 53:658–662.

Sakun, U. M., V. F. Sviridor, V. V. Grebenuk, I. G. Grinenko, R. I. Groushetsky, I. S. Guliy, and L. D. Bobrovnik. 1996. p. 21. Inulin in prophylactics of heart disease. Abst. Sixth Seminar on Inulin, Braunschweig, Germany.

Salunkhe, D. K. 1959. Physiological and biochemical effects of gamma radiation on tubers of Jerusalem artichoke. Bot. Gaz. 120:180–183.

Samotus, B., M. Niediwiedz, Z. Kolodziej, M. Leja, and B. Czajkowska. 1974a. Storage and reconditioning of tubers of Polish potato varieties and strains. Influence of storage temperature on sugar level in potato tubers of different varieties and strains. Potato Res. 17:64–81.

Samotus, B., M. Niediwiedz, Z. Kolodziej, M. Leja, and B. Czajkowska. 1974b. Storage and reconditioning of tubers of Polish potato varieties and strains. 2. Changes in sugar level in potato tubers of different varieties and strains during reconditioning of cold-stored potatoes. Potato Res. 17:82–96.

Sarkar, S. K., and C. T. Phan. 1979. Naturally occurring and ethylene-induced phenolic compounds in carrot root. J. Food Prot. 42:526–534.

Sargent, S. A., T. B. S. Corrêa and A. G. Soares. 1995. Application of postharvest coatings to fresh cassava roots (*Manihot esculenta*, Crantz) for reduction of vascular streaking. p. 331–338. In: L. Kushwaha, R. Serwatowski, and R. Brook (eds.), Proc. Harv. Postharv. Technol. Fresh Fruits and Veg., ASAE Pub. 1–95.

Sawyer, R. L., L. L. Boyd, R. C. Cetas, and A. H. Bennett. 1965. Potato storage research on Long Island with forced-air ventilation systems. Cornell Univ. Agr. Expt. Sta. Bul. 1002.

Schaaf, D. van der. 1974. De Maillard reaktie bij het drogen van land- bouwprodukten in het algemeen en bij het verwerken van aardappelen in het bijzonder (Een literaturstudie). IBVL-Rapport 38. IBVL, Wageningen, The Netherlands.

Schisler, D. A., and P. J. Slininger. 1994. Selection and performance of bacterial strains for biologically controlling *Fusarium* dry rot of potatoes incited by *Gibberella pulicaris*. Plant Dis. 78:251–255.

Schisler, D. A., C. P. Kurtzman, R. J. Bodiast, and P. J. Slininger. 1995. Evaluation of yeasts for biological control of *Fusarium* dry rot of potatoes. Am. Potato J. 72:339–353.

Schisler, D. A, P. J. Slininger, and R. J. Bothast. 1997. Effects of antagonist cell concentration and two-strain mixtures of biological control of Fusarium dry rot of potatoes. Phytopathology 87:177–183.

Schisler, D. A., P. J. Slininger, G. Kleinkopf, R. J. Bothast, and R. C. Ostrowski. 2000. Biological control of *Fusarium* dry rot of potato tubers under commercial storage conditions. Am. J. Potato Res. 77:29–40.

Schneider, E. M., J. U. Becker, and D. Volkmann. 1981. Biochemical properties of potato phosphorylase change with its intracellular localization as revealed by immunological methods. Planta 151:124–134.

Schorr-Galindo, S., and J. P. Guiraud. 1997. Sugar potential of different Jerusalem artichoke cultivars according to harvest. Biores. Tech. 60:15–20.

Scoccianti, V. 1983. Variations in the content of free and bound amino acids during dormancy and the first tuber cycle in *Helianthus tuberosus* tuber explants. Giorn. Bot. Ital. 117:237–245.

Scott, L. E., H. Harris, R. J. Constantin, T. S. Boggess, and M. W. Hoover. 1970. Processing. p. 39–45. In: Thirty years of cooperative sweet potato research, 1939–1969. Southern Coop. Ser. Bul. 159.

Sherf, A. F. 1972. Storage diseases of cabbage and carrots. N.Y. (Cornell) Agr. Inform. Bul. 35.

Shibairo, S. I., M. K. Upadhyaya, and P. M. A. Toivonen. 1997. Postharvest moisture loss characteristics of carrot (*Daucus carota* L.) cultivars during short-term storage. Scientia Hort. 71:1–12.

Shoemaker, D. N. 1927. The Jerusalem artichoke as a crop plant. USDA Tech. Bul. 33.

Sibley, J. C. 1924. Observations and experiments with the Mammoth French White Jerusalem artichoke to date of April 1, 1924. Franklin, PA.

Singh, K. K., and P. B. Mathur. 1953. Cold storage of tapioca roots. Bul. Cent. Food Technol. Res. Inst., Mysore. 2:181–182.

Sivan, P. 1979. Post harvest durability of fresh roots of cassava varieties in Fiji and storage of roots in moist sawdust. Fiji Agr. J. 41:95–102.

Smith, J. J., and P. John. 1993. Activation of 1-aminocyclopropane-1- carboxylate oxidase by bicarbonate-carbon dioxide. Phytochemistry 12:1381–1386.

Smith, O. 1977. Potatoes: Production, storing, processing. AVI, Westport, CT.

Smyth, A. B., P. C. Talasila, and A. C. Cameron. 1999. An ethanol biosensor can detect low-oxygen injury in modified atmosphere packages of fresh-cut produce. Postharv. Biol. Biotech. 15:127–134.

Snowdon, A. L. 1992. Color atlas of post-harvest diseases and disorders of fruits and vegetables. Vol. 2: Vegetables. CRC Press, Boca Raton, FL.

Sode, F., and B. F. Kuhn. 1998. Respiration in MA-packed, cut carrots. J. Food Eng. 37:223–232.

Sorce, C., R. Lorenzi, and P. Ranalli. 1997. The effects of (s)-(+)-carvone treatments on seed potato tuber dormancy and sprouting. Potato Res. 40:155–161.

Sorensen, L., and F. R. Harker. 2000. Rheological basis of splitting in carrot storage roots. J. Am. Soc. Hort. Sci. 125:212–216.

Sowokinos, J. R. 1973. Maturation of *Solanum tuberosum*. Comparative sucrose and sucrose synthetase levels between several good and poor processing varieties. Am. Potato J. 50:234–247.

Sowokinos, J. R. 1990. Stress-induced alterations in carbohydrate metabolism. p. 137–158. In: M. E. Vayda and W. D. Park (eds.), The molecular and cellular biology of the potato. CAB Intl., Wallingford, UK.

Sowokinos, J. R., E. C. Lulai, and J. A. Knoper. 1985. Translucent tissue defects in *Solanum tuberosum* L. 1. Alterations in amyloplast membrane integrity, enzyme activities, sugars, and starch content. Plant Physiol. 78:489–494.

Sparks, W. C., and L. V. Summers. 1974. Potato weight losses, quality changes and cost relationships during storage. Idaho Agr. Expt. Sta. Bul. 535.

Sparks, W. C., V. T. Smith, and J. G. Garner. 1968. Ventilation Idaho potato storages. Idaho Agr. Expt. Sta. Bul. 500.

Speirs, M., A. H. Dempsey, J. Miller, W. J. Peterson, J. T. Wakeley, F. D. Cochran, R. Reder, H. B. Cordner, E. A. Fieger, M. Hollinger, W. H. James, M. Lewis, J. F. Eheart, M. J. Eheart, F. S. Andrews, R. W. Young, J. H. Mitchell, D. B. Garrison, and F. T. McLean. 1953. Carotene, ascorbic acid and moisture content of sweet potatoes. Southern Coop. Ser. Bul. 30.

Statham, O. J. H. 1989. A review of potato storage practices trends and development in Great Britain. p. 9–37. In: B. F. Cargill, R. C. Brook, and T. D. Forbush (eds.), Potato storage technology and practice. Am. Soc. Agr. Eng., St. Joseph, MI.

Steinbauer, C. E. 1932. Effects of temperature and humidity upon length of rest period of tubers of Jerusalem artichoke (*Helianthus tuberosus*). Proc. Am. Soc. Hort. Sci. 29:403–408.

Steinbauer, C. E. 1939. Physiological studies of Jerusalem artichoke tubers, with special reference to the rest period. USDA Tech. Bul. 657.

Stevens, C., V. A. Khan, A. Y. Tang, and J. Y. Lu. 1990. The effect of ultraviolet radiation on mold rots and nutrients of stored sweet potatoes. J. Food Proc. 53:223–226.
Stewart, F. C., and A. J. Mix. 1917. Blackheart and the aeration of potatoes in storage. New York Agr. Expt. Tech. Bul. 436:321–362.
Strider, D. L., and C. L. McCombs. 1958. Rate of wound phellem formation in the sweet potato. Proc. Am. Soc. Hort. Sci. 72:435–442.
Subramanyam, H., and P. B. Mathur. 1956. Effect of a fungicidal wax coating on the storage behaviour of tapioca roots. Bull. Cent. Food Technol. Res. Inst., Mysore 5:110–111.
Suojala, T. 1999. Effect of harvest time on the storage performance of carrot. J. Hort. Sci. Biotech. 74:484–492.
Suojala, T. 2000. Variation in sugar content and composition of carrot roots at harvest and during storage. Scientia Hort. 85:1–19.
Suttle, J. C. 1995. Postharvest changes in endogenous ABA levels and ABA metabolism in relation to dormancy in potato tubers. Physiol. Plant. 95:233–240.
Suttle, J. C., and J. F. Hultstrand. 1994. Role of endogenous abscisic acid in potato microtuber dormancy. Plant Physiol. 105:891–896.
Tanret, C. 1893. Sur les hydrates de carbone du topinambur. Compt. Rend. Acad. Sci. (Paris) 117:50–53.
Thaysen, A. C., W. E. Blakes, and B. M. Green. 1929. On the nature of the carbohydrates found in the Jerusalem artichoke. Biochem. J. 23:444–455.
Thompson, A. K., and M. Arango. 1977. Storage and marketing of cassava in plastic films. Proc. Trop. Reg., Am. Soc. Hort. Sci. 21:30–33.
Thorne, S. M. 1972. Studies on the behavior of stored carrots with respect to their invasion by *Rhizopus stolonifer* Lind. J. Food Technol. 7:139–151.
Thornton, N. C. 1939a. Carbon dioxide storage. XIII. Relationship of oxygen to carbon dioxide in breaking dormancy of potato tubers. Contrib. Boyce Thompson Inst. 10:201–204.
Thornton, N. C. 1939b. Oxygen regulates the dormancy of the potato. Contrib. Boyce Thompson Inst 10:339–361.
Ton, C. S., and T. P. Hernandez. 1978. Wet soil stress effects on sweet potatoes. J. Am. Soc. Hort. Sci. 103:600–603.
Tort, M., M. Gendraud, and J. C. Courduroux. 1985. Mechanisms of storage in dormant tubers: correlative aspects, biochemical and ultrastructural approaches. Physiol. Veg. 23:289–299.
Tracy, S. M. 1903. Cassava. Farmers Bul., U.S. Dep. Agr., 167, Washington, DC.
Traub, H. P., C. J. Thor, J. J. Willaman, and R. Oliver. 1929a. Storage of truck crops: the girasole, *Helianthus tuberosus*. Plant Physiol. 4:123–134.
Traub, H. P., C. J. Thor, L. Zeleny, and J. J. Willaman. 1929b. The chemical composition of girasole and chicory grown in Minnesota. J. Agr. Res. 39:551–555.
Tsror (Lahkim), L., M. Aharon, and O. Erlich. 1999. Survey of bacterial and fungal seedborne diseases in imported and domestic potato seed tubers. Phytoparasitica 27:15–226.
Tsror (Lahkim), L., A. Nachmias, O. Erlich, M. Aharon, and M. C. M. Perombelon. 1993. A 9-year monitoring study of diseases on potato seed tubers imported to Israel. Phytoparasitica 21:321–328.
Uemura, M., and S. Yoshida. 1986. Studies on freezing injury in plant cells. Plant Physiol. 80:187–195.
Varlamova, C., E. Partskhaladze, V. Oldhamovsky, and E. Danilova. 1996. Potential uses of Jerusalem artichoke tuber concentrates as food additives and prophylactics. p. 141–144. In: Sixth Symp. Inulin, Carbohydrate Res. Found., The Hague, Netherlands.

Vasala, P. A. 2001. Ginger. p. 319. In: K. V. Peter (ed.), Handbook of Herbs and Spices. Woodhead Pub., Cambridge, England.

Verma, S. C., T. R. Sharma, and S. M. Verma. 1974a. Sucrose accumulation during high temperature storage of potato tubers. Potato Res. 17:224–226.

Verma, S. C., T. R. Sharma, and S. M. Verma. 1974b. Effects of extended high temperature storage on weight losses and sugar content of potato tubers. Indian J. Agr. Sci. 44:702–706.

Villa, L. G., and F. W. Bakker-Arkema. 1974. Moisture losses from potatoes during storage. ASAF Paper 74-6510.

Villeneuve, F., J. P. Bosc, and C. Luneau. 1993. La conservation en chamber froide: étude de quelques facteurs. Acta Hort. 354:221–232.

Vincent, J. F. V. 1990. Fructure properties of plants. Adv. Bot. Res. 17:235–287.

Vliet, W. F. van, and W. H. Schriemer. 1960. The sugar accumulation in potatoes kept at a low temperature, as studied in a small selection of samples of Dutch varieties. Eur. Potato J. 3:263–271.

Vokou, D., S. Vareltzidou, and P. Katinakis. 1993. Effects of aromatic plants on potato storage: sprout suppression and antimicrobial activity. Agr. Ecosyst. Environ. 47:223–235.

Wagner, A. B., E. E. Burns, and D. R. Paterson. 1983. The effects of storage systems on sweet potato quality. HortScience 18:336–338.

Walter, W. M., Jr., and M. W. Hoover. 1984. Effect of pre-processing storage conditions on the composition, microstructure and acceptance of sweet potato patties. J. Food Sci. 49:1258–1261.

Walter, W. M., Jr., and A. E. Purcell. 1980. Effect of substrate levels and polyphenol oxidase activity on darkening in sweet potato cultivars. J. Agr. Food Chem. 28:941–944.

Walter, W. M., Jr., A. E. Purcell, and A. M. Nelson. 1975. Effect of amytolytic enzymes on "moisture" and carbohydrate changes of baked sweet potato cultivars. J. Food Sci. 40:793–796.

Walter, W. M., and W. E. Schadel. 1982. A rapid method for evaluating curing progress in sweet potatoes. J. Am. Soc. Hort. Sci. 107:1129–1133.

Walter, W. M., and W. E. Schadel. 1983. Structure and composition of normal sidn (periderm) and wound tissue from cured sweet potatoes. J. Am. Soc. Hort. Sci. 108:909–914.

Wang, C. Y., J. G. Buta, H. E. Moline, and H. W. Hruschka. 1980. Potato sprout inhibition by camptothecin, a naturally occurring plant growth regulator. J. Am. Soc. Hort. Sci. 105:120–124.

Wang, J. K., and S. Higa. 1983. Taro: a review of *Colocasia esculenta* and its potentials. Univ. Hawaii Press, Honolulu, HI.

Wang, Y., R. J. Horvat, R. A. White, and S. J. Kays. 1998. Influence of postharvest curing treatment on the synthesis of the volatile flavor components in sweetpotato. Acta Hort. 464:207–212.

Wang, Y., and S. J. Kays. 2002. Sweetpotato volatile chemistry in relation to weevil (*Cylas formicarius* Summers) behavior. J. Am. Soc. Hort. Sci. 127:656–662.

Wastie, R. L., G. R. Mackay, and A. Nachmias. 1994. Effect of *Erwinia carotovora* or *Alternaria solani* or *Verticillium dahliae* on disease development in Israel. Potato Res. 37:113–120.

Weichmann, J. 1977. Physiological response of root crops to controlled atmospheres. In: D. H. Dewey (ed.), Proc. 2nd Natl. Controlled Atmosphere Res. Conf., Mich. State Univ. Hort. Rep. 28:122–136.

Wells, J. M., and F. L. Merwarth. 1973. Fungicide dips for controlling decay in carrots in storage for processing. Plant Dis. Rptr. 57:697–700.

Wheatley, C. C. 1989. Conservation of cassava in polyethylene bags. CIAT Study Guide 04SC-07,06, CIAT, Cali, Colombia.
Whiteman, T. M. 1957. Freezing points of fruits, vegetables and florist stocks. USDA Mkt. Res. Rep. 196.
Whiteman, T. M., and R. C. Wright. 1946. The effects of temperature on losses in sweet potatoes. Proc. Am. Soc. Hort. Sci. 48:437–442.
Wickham, L. D., and L. A. Wilson. 1988. Quality changes during long-term storage of cassava roots in moist media. Trop. Sci. 28:79–86.
Wilkins, J. 1668. An essay towards a real character, and a physiological language. Printed by J. M. for S. Gellibrand and John Martin, London.
Wilson, J. E. 1983. Storage of taro corms and leaves. Alafua Agr. Bul. 8:35–37.
Wiltshire, J. J. J., and A. H. Cobb. 1996. A review of the physiology of potato tuber dormancy. Ann. Appl. Biol. 129:553–569.
Workman, M., E. Kerschner, and M. Harrison. 1976. The effect of storage factors on membrane permeability and sugar content of potatoes and decay by *Erwinia carotovora* var. *atroseptica* and *Fusarium roseum* var. *sambucinum*. Am. Potato J. 53:191–207.
Yada, R. Y., R. H. Coffin, M. K. Keenan, M. Fitts, C. Dufault, and G. C. Tai. 1991. The effect of maleic hydrazide (potassium salt) on potato yield, sugar content and chip color of Kennebec and Norchip cultivars. Am. Potato J. 68:705–709.
Yamaki, S., and I. Uritani. 1972. The mechanism of chilling injury in sweet potato. VI. Changes of lipid components in the mitochondrial membrane during chilling storage. Plant Cell Physiol. 13:67–69.
Yamaki, S., and I. Uritani. 1973. The mechanism of chilling injury in sweet potato. X. Change in lipid protein interaction in mitochondria for cold-storage tissue. Plant Physiol. 51:883–888.
Yamaki, S., and I. Uritani. 1974. The mechanism of chilling injury in sweet potato. XI. Irreversibility of physiological deterioration. Plant Cell Physiol. 15:385–388.
Zapata, L. E. 1978. Preservacion de yuca fresca. Anales del tercer seminario avanzado de tecnologia de alimentos Bogota, D.E. Colombia.