NITROGEN FORMS IN THREE KENYAN SOILS NITISOLS, LUVISOLS AND FERRALSOLS

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ABSTRACT

The nitrogen cycle in soil is an integral part of the overall cycle of N in nature. The primary source of N is the atmosphere where the strongly bonded gas molecule N is predominately gas (78.08%). Total N content in soils ranges from 0.02% in the subsoil and more than 2.5% in peats; ploughed layers of most cultivated soils contain between 0.06 and 0.5%. The amount present in each case is, however determined by climate, type of vegetation, topography, parent material and activities of man. Over 95% of TN in surface soils is organically bound while the portion of non-exchangeable N is high in subsurface soil. Knowledge concerning the nature of organic N in soils is based on studies involving identification and estimation of N forms released by treatment with hot acids. Organic N forms were determined in three soils by acid hydrolysis. The total hydrolysable organic N for the 0-15 and 15-30cm layers were 57.2 and 59.3% for Gituamba andosols; 56.9 and 61.9 for Kitale ferralsols; 39.0 and 42.1% for Katumani luvisols, respectively. Amide N ranged from 11.6 to 21.4% of total N; Hexosamine from 5.2 to 10.1% and Amino acid N from 26.2 to 37.1%. Amino acid N therefore formed the highest portion followed by Amide N of the hydrolysable organic N.

Key words: Total nitrogen forms; Hydrolysable nitrogen forms; Organic nitrogen forms

INTRODUCTION

Nitrogen (N) is the one of the most limiting factor to the growth of almost all crops in terrestrial ecosystems. The dynamics of inorganic nitrogen (NH_4^+, NO_3^-) has been intensively studied in soils, and increasingly research has been focused on the dynamics of soil organic nitrogen in agricultural soils. Hydrolysable organic N is the organic N present in different soil extracts either extracted with water or potassium sulphate (K_2SO_4) or that can be hydrolyzed by acid hydrolysis or solubilized (Ros et al., 2009). Hydrolysable contents of soils can be less than 5% of the total N that can be extracted by mild salt solutions, or may be more than 50% of the total extracted by acid hydrolysis methods (Stevenson, 1994; Matsumoto and Ae, 2004; Ros et al., 2009). Plant roots can absorb not only inorganic N but also hydrolysable organic N in the form of amino acids (AAs) from soil and solutions (Wu et al., 2005). Hydrolyzed soil organic matter contains high organic N, of which 30%–50% is present in the form of AAs.

The rhizosphere is the soil zone in which microbial activity is influenced by plant roots, distinguishing it from 'bulk' soil (Russell, 1982; Herman et al., 2006). Active interaction occurs among plant roots, soil, and microbes in rhizosphere soils (Herman et al., 2006). The interaction results in increase in soil nitrogen mineralization, which correspondingly increases net plant nitrogen assimilation (Bregliani et al., 2010).

Several studies have attempted to measure the size and characteristics of Hydrolysable N pools in soils (Ros et al., 2009). In contrast, very few have attempted to determine the distribution of Hydrolysable N either in the rhizosphere or in bulk soil of traditional agricultural soils. While it seems reasonable to assume that organic N pools would be larger in the rhizosphere than in bulk soil (DeAngelis et al., 2008; Näsholm et al., 2009), there are also compelling reasons why this may not be the case under fertilizer application or may depend on the plant species grown (monocotyledonous or dicotyledonous). Soil processes may also influence the size of N pools in the soils. For example nitrogen fixation which involves reduction of elementary N_2 to the -3 oxidation state in NH_3. This is a biological process catalyzed by nitrogenise, a largely anaerobic metalloenzyme. The NH_3 produced is retained by N-fixing cells and reacts with glutamate to form glutamine. Newly fixed NH_3 is only rarely released by healthy nitrogen fixing cells but must first pass through organic form before entering the N-cycle. Mineralization of organic N is mainly by microorganisms where organically bound N is released as NH_3. Whether N is mineralized or immobilized depends on the magnitude of C:N ratio of substrate compared to that of decomposer organism. When the C:N ratio is greater than that of decomposer organisms, immobilization occurs (Keinan and Schechter, 2016).

Organic residues which have high C:N ratios are very difficult to decompose because of their high carbon content. These include materials such as hay straw, pine needles, corn stalks, dry leaves, saw dust, etc. Some of these materials may have C:N ratios in excess of 100:1. The more difficult to decompose organic N compounds include cellulose, lignin, oils, fats, and resins. Organic residues which have low C:N ratios decompose relatively easy. These include alfalfa, clover, manures, sludge, immature grasses, etc. Generally, the more immature the plant material, the lower C:N ratio. These materials can decompose rapidly and in many cases actually contribute to soil N levels. Easily decomposed organic N compounds include sugars, starches, proteins, and hemicellulose (Keinan and Schechter, 2016).

Immobilization occurs either through biotic or abiotic process. For example clay minerals can efficiently immobilize NH_4^+ in the soil. Exchangeable NH_4^+ is available for biological immobilization. In the past, it had been assumed that non-exchangeable NH_4^+ fixed in clay mineral lattices had low biological availability; but more recent data has shown that 30-60% of fixed Exchangeable NH_4^+ is available for biological uptake (Keinan and Schechter, 2016). In some situations, N in exchangeable NH_3 is oxidized through nitrification process by autotrophic bacteria (*Nitrosomonas* spp. and *Nitrobacter* spp.) to form nitrites (NO_2^-) and nitrates (NO_3^-) (Schaechter, 2009, Ward, 1996 and Mancinelli, 1996). The NO_2^- and / NO_3^- are more mobile and prone to leaching losses (Lin et al., 2001). The NO_2^- can further be reduced to nitrous oxide (N_2O) and dinitrogen (N_2) through denitrifying bacteria (Schulze and Harold, 2012) hence

sealing the fate of N in soils. Reduction of NO_3^- through NO_2^- and NH_4^+ normally occurs in low rates in the soils (3). If it were possible to enhance this process where NO_3^- is reduced to NH_3 and its subsequent incorporation into soil organic matter (SOM), large losses resulting from denitrification and leaching could be prevented hence improving the economy of N in soils.

MATERIALS AND METHODS

Three soil types were selected on the basis of groups, agro-ecological zone, organic matter content and land use. These were the Gituamba andosols, Kitale feralsols and Katumani luvisols (WRB, 2006). Gituamba is centered on coordinate $0^045'S-36^051'E$ where the Geology is mainly Basalts and Basaltic Conglomerates of Simberian Series. The land is under tea and pyrethrum cultivation under ecological zone II; P/E 82%. The soils are acidic, well drained dark to dark-reddish clay. The Kitale ferralsols were sampled on center coordinates $1^001'N-34^039'E$ and the Geology consists of Basement system of Gneisses, Schists rich in Feldspars, Biotite, Horneblende and Garnet with minor exposure of Granite and Pegmatitic dykes. The land is mainly used for maize cultivation and pasture research. It is in agro-ecozone III, P/E 66%. The soils on one side are well drained deep to moderately deep, reddish brown to yellowish red, friable clay on upper valley slopes. The other is poorly drained dark grayish brown in valley bottoms. Main clay mineralogy is kaolin. There are significant quantities of Illite and Montimorrillonite. The Katumani luvisols are on coordinates 01^035 $'S-37^014'E$. The Geology of the area is mainly Quartzo-feldspartic gneiss of the Precambrian basement system. The land was originally under Acacia bush which has been cleared to pave way for Cereals such as maize, Sorghum and also beans and pastures. It is in Eco-zone IV. The soils are well drained sandy clay.

Soil Sampling

The soil samples were taken from the 0-15 and 15-30cm depth. A profile pit 40cm deep was dug and the 15-30cm depth sampled first to avoid contamination from above layer. The samples were placed in special sampling bags, sealed and placed in cool boxes before transportation to the laboratory for processing. Undisturbed samples were also taken using core rings for physical determinations of bulk density and hydraulic conductivity.

SOIL ANALYSIS

Inorganic Nitrogen fractions

Total Nitrogen and available N were determined in accordance with micro-Kjeldhal method described by Bremner (1996). For Non-exchangeable ammonium N that is fixed, 1gram of air dried soil (<2mm) was weighed into a 400ml beaker. To it was added 20mls of potassium hypobromite solution and shaken. The beaker was then covered with a watch glass and allowed to stand for 2hours (to get rid of exchangeable NH_4^+ ions and labile organic compounds). After 2hours, 60mls of distilled water was added and the strongly boiled for 5-6minutes. The mixture was allowed to stand overnight and the supernatant decanted. The residue was transferred to a 250ml polythene centrifuge tubes with 0.5N KCl from plastic watch

bottles. The centrifuge tubes were filled to the 80ml mark and centrifuged for 20minutes by 1000 x G rev/min and again the supernatant discarded. The remaining residue was then transferred to a 250ml digestion flask with distilled water and 7mls Conc. H_2SO_4 and 0.5g K_2SO_4 added, and digestion commenced. The digestion started at low temperatures until the mixture cleared (30 minutes) and the temperature was then raised up to the end off digestion (90minutes). The mixture was allowed to cool, before cooling completely, small amounts of distilled water was added to prevent solidification of digest. The digest was transferred into a distillation flask and 40mls of 10N NaOH added quickly to avoid escape of ammonia gas before actual distillation commenced. The NH_3 gas released during distillation was collected in 20mls of 1% boric acid with a mixed indicator. Titration was done with 0.01N H_2SO_4 and the indicator turned from green to pink.   Calculation done as per Equ Ia & b.

$$0.01N\ H_2SO_4 = 140\mu gN; \quad \text{hence \%N} = \left(\frac{(Titre - Blank).140}{10,0000} \right) \qquad \text{Ia}$$

$$kgN/ha = \frac{Soil\ depth\ x\ Bulk\ density\ Conc.\mu gN\ x\ area\ (cm^2)}{Weight\ of\ Soil\ x\ 10^9} \qquad \text{Ib}$$

Organic Nitrogen fractions

Hydrolysate preparation

Was done in accordance with Bremner (1965) where 10grams of air dry soil finely ground <2mm   was weighed into a 250ml flask and 3 drops of octyl alcohol and 50mls of 6N HCl were added and the mixture swirled till it was thoroughly mixed with soil. The flask was then placed on a hot electric plate with a heat control device and connected to Liebig condenser. Cold water was passed through the water junket of the condenser and the soil acid mixture heated to boil under a reflux for 12hours. The mixture was allowed to cool before being filtered through Buchner funnel with suction apparatus using whatman filter paper No.42. The filtrate was then neutralized with Sodium hydroxide (5N NaOH and 0.01N NaOH, respectively) up to pH 6.5±0.1 and then made to the mark in a 250ml volumetric flask. The alkali was added slowly with constant stirring to prevent Hydrolysate from becoming alkali at any stage of neutralization (first with and 5N NaOH to bring the hydrolysate to pH 5 and then with 0.01N NaOH to raise pH further to 6.5). The Hydrolysate was stopperd and kept in a fridge as stock for analysis of various forms of organic nitrogen.

Total Hydrolysable Nitrogen

Done in accordance with Bremner 1965 where 25mls of neutralized Hydrolysate was pipetted into a distillation flask. To it was added 0.5g K_2SO_4; 0.5g selenium mixture as catalyst and 7mls Concentrated H_2SO_4. The mixture was heated continuously in a Kjedhal digestion flask until frothing ceased after water was damped off. This was done at low temperatures for 30minutes after which it temperature was increased until the mixture cleared and digestion was completed by gently boiling for a further one hour. The flask was allowed to cool and 10mls of distilled water was added slowly while shaking the mixture. The flask was again allowed to cool under water in a beaker containing crashed ice. This digest was

transferred into a 250ml distillation flask and 40mls of 10N NaOH added. This was then distilled and ammonia gas released was collected in 1% boric acid and titration done using 0.01N H_2SO_4. Calculation done as per Equ II.

0.01N H_2SO_4 = 140µgN;   hence %N = $\left(\dfrac{(Titre - Blank).140}{10,0000} \right)$ II

Amide-N

25mls of the neutralized Hydrolysate was pipetted into a 250ml distillation flask and 0.07g of MgO added. The mixture was distilled as in total hydrolysable nitrogen above. The distillate was again collected in 1% boric acid and titration done using 0.001N H_2SO_4. Calculation done as per Equ III.

0.001N H_2SO_4 = 14µgN, hence %N = $\dfrac{(Titre - Blank).14}{10,000}$ III

(Amide + Hexosaamine)-N

25mls of the neutralized Hydrolysate was pipetted into a 250mls distillation flask. To it was added 10mls of Phosphate-Borate (PB-buffer at pH 11.2). The mixture was steam distilled and NH3 gas collected in1% boric acid. Titration was done with 0.001N H_2SO_4. The amount of Hexosamine was obtained by subtraction method from the Amide results above. Calculation of % N followed same Equ. III above.
% Hexosamine-N = % (amide+Hexosamine)-N-(% Amide-N).

(Serine + Threonine)-N

After removal of Amid + Hexosaamine Nitrogen above by steam distillation with PB-buffer at pH 11.2; the flask was detached from distillation apparatus and the steam jet rinsed with 5mls of distilled water and rinse collected in a distillation flask. The flask containing the rinse and the mixture was cooled thoroughly under cold tap water. 2mls of periodic acid solution was added to the mixture and the flask swirled for 30seconds. Then 2mls of Sodium meta-Arsenate solution was added to reduce excess periodic acid. Distillation was done and calculation continued as in Amide-N above.

A-Amino acid-N

25mls of the neutralized Hydrolysate was pipetted into a 250mls distillation flask. To the mixture was added 1 ml 0.5N NaOH and the flask heated in boiling water for approximately 20minutes. The flask was allowed to cool before 0.5g Citric acid and 0.1g Ninhydrin powder were added. The flask was again immersed in a boiling water bath for about 1 minute, swirled for a few seconds without removing from the bath. The flask was allowed to remain in the bath for 9minutes, and then cooled under a water tap and 10mls of PB-buffer and 1 ml 5N NaOH added. Distillation and calculations were done as in Amide-N above Equ. IV.

0.001N H_2SO_4 = 14µgN, hence %N = $\dfrac{(Titre - Blank).14}{10,000}$

IV

(Ammonium+Hexosamine+Amino acid)-N

25mls of the neutralized Hydrolysate was pipetted into a 250mls distillation flask. To it was added Citrate buffer (pH 2.5) and the mixture heated vigorously in a boiling water bath for 1minute. The mixture was then swirled for a few seconds without removing it from the water bath and then left in the bath for a further 9minutes, cooled and 10mls of PB-buffer added. This was then connected to distillation apparatus and steam distilled as above. Liberated NH_3 was collected in 1% boric acid and titrated with 0.01N H_2SO_4. Mixed indicator was used and the end point was pinkish colour. Calculation was as in total hydrolysable N above.

RESULTS AND DISCUSION

Soil characterization

The behavior of nitrogen in soils is controlled by the physical, chemical and biological properties of the soil. This being the case, it was therefore necessary to know some of the salient soil properties of the three soils used. Table 1 shows the physical and chemical characteristics of the three soils namely; Gituamba andosols, Kitale ferralsols and Katumani luvisols. The soil pH varied within the soil depths and across the soil groups. The pH ranged from 4.0 in Gituamba andosols (0-15cm) to 7.0 in Katumani luvisols (15-30cm) depths. The pH is markedly influenced by the parent material and climatic conditions of the site.

Table 1. Some salient soils characteristics of the study

Soil sampling site and groups	Gituamba andosols		Kitale ferralsols		Katumani luvisols	
Soil Properties/ Depth (cm)	0 - 15	15 - 30	0 - 15	15 - 30	0 - 15	15 - 30
pH-water	4.0	4.1	5.6	5.6	6.6	7.0
pH-KCl	3.9	4.0	4.4	4.5	4.8	5.6
CEC (me/100g soil)	28.6	26.7	15.3	13.4	13.4	12.1
ECEC(me/100g soil)	11.6	10.2	11.4	9.1	9.5	10.7
Ca(me/100g soil)	0.7	0.3	4.7	2.9	5.7	6.3
Mg(me/100g soil)	0.5	0.1	2.4	2.0	1.3	1.9
Na(me/100g soil)	0.5	0.4	1.0	0.5	0.6	0.4
K(me/100g soil)	4.3[*]	3.3[*]	1.5	1.2	1.5	0.9
% Base Saturation	21.0	19.5	62.7	55.2	61.4	78.5
Exch Al^{3+} (me/100g soil)	4.6	3.3	1.0	0.8	1.1	0.9
Exch H^+(me/100g soil)	1.0	0.7	0.9	0.9	0.4	0.3
Available P (ppm)	12.5	10.0	2.5	1.5	46.0	29.0
% Total N	0.6	0.5	0.2	0.1	0.2	0.1
% Organic C	7.9	4.8	4.5	1.8	1.0	0.5
C:N	12.8	9.2	22.5	13.9	5.7	5.3
Bulk Density (g/cm^{-3})	0.6	0.8	1.2	1.1	1.4	1.3
% Sand	40.3	38.3	41.9	37.3	68.6	74.2

%Clay	19.9	27.9	52.9	55.0	23.9	22.4
%Silt	39.8	33.8	5.2	7.7	7.5	3.4
Textural Class	Loam	Loam	Clay	Clay	Sandy Clay Loam	Sandy Clay Loam

Gituamba area is relatively humid and soils derived from volcanic activity hence low pH. Low pH also has a marked influence on Exchangeable Aluminium (Al) as clearly seen in Gitumaba andosols with highest Al content of 4.6me in the 0-15cm and 3.3me/100g soil in the 15-30cm depth, respectively. Analyzed Al was found to be highly negatively correlated to soil pH (r = -0.88, P≤0.05), meaning that as pH decreased, the Al content increased and vice versa as in the case of Gituamba soils. The pH was also found to be significantly and positively correlated with percent base saturation (r = -0.86, P≤0.05).

The soils also gave different though expected pattern of organic carbon (OC) and total nitrogen (TN) distribution in soil profiles. Both the %OC and TN decreased with depth within soil profile, a phenomenon undoubtedly due to the addition of organic matter mainly at the top. Nitrogen is an integral part of organic carbon. Gituamba andosols had the highest of both 7.9 and 0.6% of OC and TN, respectively.

C: N ratio differed in the two depths in all soil groups with the ratio lower in the 15-30cm depth. Low C: N ratio ranged from 5.3 in Katumani luvisols, to 22.5 in Kitale ferralsols 0-15cm depths. C: N rations are controlled by conditions such as moisture, temperatures and presence of substrate to be mineralized. The C: N ratios observed were within the range which favors net N-mineralization (Kaleeem et al., 2015; Jansen, 1992; Karuku, 1989).

Exchangeable potassium (Exch. K) was very low in Kitale ferralsols followed by Katumani luvisols. In Gituamba andosols, Exch. K was high at 3me/100g soil. These Andosols are derived from volcanic ash hence high K content. Keter (1974) suggested that East African rocks are often rich in this element especially when derived from volcanic rocks. Generally, Calcium (Ca), Magnesium (Mg), Sodium (Na) and K were higher in top than sub-soil with exception in Katumani luvisols which could partly be due to leaching from above or simply reflect supply of cations from parent rock .

Cation exchange capacity (CEC) is a measure of soil fertility and was observed to be higher in the top than in the sub soils in the soil groups with exception of Katumani luvisols. The Clay content was highest in Kitale ferralsols at 52.9 and 55.0% and lowest in Katumani luvisols at 23.9 and 22.4% in the 0-15 and 15-30cm depths, respectively for each soil group. Katumani soils had highest sand content at 68.6 and 74.3% for the 0-15 and 15-30cm depths, respectively. The texture of the three soil groups varied greatly and could have been influenced by such factors as the vegetation of the location, climate as well as the parent material in which the soils were derived. Gituamba soils are loamy; Kitale clayey and Katumani are sandy clay loam.

The bulk density (ρb) was highest in Katumani luvisols at 1.4 and 1.3g/cm^3 and lowest in Gituamba andosols at 0.6 and 0.8g/cm^3 for the 0-15 and 15-30cm depths, respectively and seems to reflect the texture of respective soils. Soils low in clay content and are high in sand content like Katumani luvisols tend to exhibit higher ρb and vice versa (Chaudhari et al. 2013, Morisada et al. 2004; Sakin, 2012; Catherine and Ouimet, 2007;   Aşkın and Özdemir, 2003 and Sakin et al., 2011)**.** However, influence of organic matter content on ρb in these soils cannot be ruled out.

Nitrogen forms in the three soil groups

Table 2 shows various forms of nitrogen of the three soils groups in kgN/ha. The TN ranged from 1,038.5 kg in Kitale ferralsols (15-30cm) to 6,558.6kg/ha in Gituamba andosols (0-15cm) depths. The TN decrease with depth in all the three soil groups: Gituamba from 6,558.6 to 5,832.7kg; Kitale from 1,627.0 to 1,038.5kg and Katumani from 3,582.2 to 1,663.2kg/ha in 0-15 to 15-30cm depths. Gituamba andosols had the highest amount of TN in the 0-30cm layer of 12,391.3kg/ha and Kitale had the lowest at 2,665.5kg/ha. Again the total amounts reflect climate and vegetation of the area (http://www.zo.utexas.edu/courses/bio301/ /Chapter4.html, 2016). The same trends was observed in exchangeable ammonium Nitrogen (NH_4^+) and nitrate Nitrogen (NO_3^-) and were higher in the 0-15 cm than in 15-30 cm depth in all three soil groups. Gituamba had highest available N at 319.6kg/ha in the 0-30 cm layer followed by Kitale at 129.3kg/ha then Katumani at 28.6kg/ha.

The %TN in all three soil groups ranged from 0.6 in Gituamba andosols (0-15cm) to 0.1% in Katumani luvisols (15-30cm) depths, a range that has been reported in most cultivated soils (Ehud and Israel, 20016). Observed exchangeable NH_4^+-N was very low and ranged from 00.2% in Katumani (0-15cm) to 3.2% in Kitale (15-30cm) of the TN in each depth. NO_3^- - N on the other hand ranged from 0.2% in Katumani (0-15cm) to 1.6% in Kitale (0-15cm) of the TN in each depth. NO_3^- -N were lower than exchangeable NH_4^+-N in all three soil groups.

Organic nitrogen showed same trend as TN where Gituamba andosols had the highest amount at 9337.2kgN/ha followed by Katumani luvisols with 4939.0kg/ha and Kitale ferralsols the lowest at 2455.0kg/ha in the 0-30 cm depths. Organic N formed the highest portion of the various forms of nitrogen. Organic N formed the highest portion of various N forms and ranging from 75.3 to 95.3% of total N. Gituamba andosols had 75.4 and 75.3%, Kitale ferralsols 92.3 and 91.8% and Katumani luvisols with 95.3 and 91.7% in the `0-15 and 15-30 cm depths, respectively.

Table 2. Nitrogen distribution in the three soil groups; Andosols, Ferralsols and Luvisols (kgN/ha)

Soil group	Depth (cm)	Total-N	Organic-N	Hydrolysable organic-N	Hydrolysable-N as % of organic-N	Available-N	Fixed-N (NH4-f)

Andosol	0-15	6558.6	4945.2	3742.2	75.7	167.1	1305.2
	15-30	5832.7	4392.0	3499.2	81.5	152.5	1403.9
	Total	12391.3	9337.2	7241.4	77.6	319.6	2709.1
	% of TN	-	75.4	58.4	-	2.7	21.9
Ferralsol	0-15	1627.0	1501.7	826.5	55.0	72.5	54.0
	15-30	1038.5	953.3	618.0	64.8	56.8	40.3
	Total	2665.5	2455.0	1444.5	58.8	129.2	94.3
	% of Total	-	92.0	54.2	-	4.7	3.3
Luvisol	0-15	3582.2	3413.8	1476.3	43.2	15.1	152.5
	15-30	1663.2	1525.2	768.0	50.4	13.5	116.9
	Total	5245.4	4939.0	2244.3	45.4	28.6	269.4
	% of Total	-	94.2	42.8	-	0.6	5.2

Fixed N (NH_4^+-f) ranged from 40.4kgN in Kitale ferralsols to 1403.9 kgN/ha in Gituamba and sols both in 15-30 cm depth and was relatively higher in the topsoil compared to the sub-soil. The NH_4^+-f in percent were 4.3 and 7.5%; 22.1 and 22.05% and 3.3 and 3.8% for Katumani luvisols, Gituamba andosols and Kitale ferralsols 0-15 and 15-30 cm depths, respectively. The reason for the low NH_4^+-f in Kitale ferralsols is not clear but could be due to the inherent clay minerals. The high amounts of all forms of N in Gituamba andosols seem to reflect on the vegetative cover present in the area. Much of the litter fall is decomposed by microorganisms as it falls giving rise to organic matter (OM). This is clearly seen in Table 1.0 where the Gituamba andosols had the highest %OC and in Table 2 where all N forms were highest. The decline in organic matter down the soil profiles had similar effect on all forms of N. As the OM accumulates at the top, more N is released but as it declines down the profile, all N forms decreases. This fortifies the fact that soil N unless with inorganic additions comes from organic matter decomposition (Ryals and Wendler, 20013).

Katumani luvisols with a scarce vegetation, there was marked reduction in OM which gave rise to low N forms. For Katumani, the area sampled was bare and had been under cultivation for many years. On the other hand, Kitale ferralsols were sampled under grass cover and this could be the reason for the high OM accumulation on the surface. It can therefore be concluded that the high accumulation of N forms observed in Gituamba andosols was as a result of high vegetation cover and probably due also to low pH (Table 1) values which limit the work of microorganism in decomposing OM quickly and completely. Organic N accumulates in acid soils due to slow mineralization but mineralization becomes rapid when the soils are limed (Harmense and Van Schreven, 1955; Karuku, 1989).

Organic forms of nitrogen in three soils groups

Table 3 shows the organic N fractions for Gituamba andosols, Kitale ferralsols and Katumani luvisols. The total hydrolysable-N ranged between 39.0-43.9% and 41.7-42.8% for Katumani luvisols, 54.6-9.4% and 61.3-63.0% for Kitale ferralsols and then 55.8-58.4 and 57.4-60.9% for Gituamba Andosols in the 0-15 and 15-30 cm depths, respectively.

Table 3. Mean hydrolysable soil organic nitrogen forms as percent of total nitrogen

Soil group	Gituamba andosols		Kitale ferralsols		Katumani luvisols	
Organic N forms/Depth (cm)	0 - 15	15 - 30	0 - 15	15 - 30	0 - 15	15 - 30
Total Hydrolysable N						
Range	55.8-58.4	57.4-60.9	54.6-59.4	61.3-63.0	39.0-43.9	41.7-42.8
Mean	57.2	59.3	56.9	61.9	40.0	42.1
Amide-N						
Range	16.1-18.7	17.7-19.8	18.1-20.8	20.3-22.5	10.9-12.6	17.0-18.1
Mean	17.2	18.4	19.2	21.4	11.6	17.6
Hexosamine N						
Range	7.5-8.6	8.0-8.2	5.6-6.5	9.9-10.6	4.9-5.4	8.8-10.4
Mean	8.0	8.1	6.0	10.1	5.2	9.4
Amino acid-N						
Range	34.8-35.4	32.7-33.8	37.0-37.6	35.2-38.9	29.1-30.8	23.1-30.8
Mean	35.1	33.3	37.3	37.1	30.0	26.2

Amide-N ranged between 10.9-12.6% and 17.0-18.1% for Katumani luvisols, 18.1-20.8% and 20.3-22.5% for Kitale ferralsols and then 16.1-18.7 and 16.7-18.8 for Gituamba andosols in the 0-15 and 15-30 cm depths, respectively. Hexosamine-N was between 4.9-5.4% and 8.8-10.4% for Katumani luvisols, 5.6-6.5% and 9.9-10.63% for Kitale ferralsols and then 4.9-5.4% and 8.0-8.2% for Gituamba and sols in the 0-15 and 15-30cm depths, respectively. Following the same trend, Amino acid-N as percent of TN was 34.8-35.4% and 32.7-33.8% for Gituamba andosols, 37.0-37.6% and 35.2-38.9% for Kitale ferralsols and 29.1-30.8 and 23.1-30.8% for Katumani luvisols in the 0-15 and 15-30cm depths, respectively. The 0-15cm depth had slightly lower hydrolysable N amounts compared to the 15-30cm depth in all the three soil groups and this same trend was observed for Amide-N and Hexosamine-N. Amino acid-N was higher in the top compared to sub-soils.

The total hydrolysable-N together with other N-fractions in the soils could have been influenced by the amount and extent of decomposition and humification of OM as well as the microbial population and their activities in the two depths (Kelley and Stevenson, 1985; Kai et al., 1973). Results obtained for total hydrolysable-N (42-62%) fall within the range already obtained by Shigeyoshi and co-workers in 1986. These workers observed total hydrolysable-N of between 40 and 80%. Amount of Amide-N (17-21%) also agrees with those obtained by Bremner (1949a); Keeney and Bremner (1964), who observed values ranging from 15 to 25%.

Values obtained for Hexosamine-N (5-10%) were in range of those obtained by Bremner and Shaw (1954), Keeney and Bremner (1964) of between 5 and 10% and Shigeyoshi et al. (1986) of 2 and 5%.

All these organic N-fractions tend to vary from one type of soil to another. This seems to be influenced by soil characteristics such as OM content in soils, decomposition level of organic materials and management systems of the soil. The high amount of Hexosamine-N observed in Kitale Ferralsols (15-30cm) could be due to the grass cover which extends its roots down the soil profile. Stevenson (1957) observed maximum accumulation of Hexxosamine-N in the B-horizon of a silt loam (Planosol under grass cultivation) where 24% of TN was in form of Hexosamine-N.

Types of OM added in these soils are mostly of diverse nature and hence influences the various fractions observed. Also the soils have been under different agricultural land use and this tends to influence the N turnover differently. Some forms such as Amino acid could be hydrolyzed to available forms of N such as NH_4^+ and NO_3^- for microorganism and plant uptake.

CONCLUSIONS AND RECOMMENDATIONS

The nitrogen content in the three soils varied considerably. This variation seemed to reflect on the amount of organic matter present in each soil. Gituamba andosols with the highest amount of organic matter also had the highest quantity of all forms of N. On the other hand, Katumani luvisols with lowest amount of OM had the least quantity of various N forms. The soil pH also seemed to influence the rate and extent of OM decomposition thereby in turn influencing the various N forms present in each soil. Organic N forms formed the highest portion of various N forms while nitrates formed the least percentage. Non-exchangeable NH_4^+-N was highest in Gituamba and sols and was attributed to the high amount of N present in this soil and probably also to the pH and type of clay mineral of this soil. For organic N fractions, hydrolysable total N ranged from 40.0 to 61.9%. Amide-N fraction from 11.6 to 21.4%; Hexosamine-N from 5.2 to 10.1% and Amino acid-N from 26.2 to 37.3 % (on average) of the total N in the soils. Amino acid-N formed the highest portion of the organic N fractions. Those fractions that were easily hydrolysable, for example Amino acid-N were lower in Katumani luvisols which had been cultivated for a long time than in Kitale ferralsols which were under grass vegetation.

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