NITROGEN FORMS IN SOILS, AND EFFECT OF LIME, NITROGEN AND PHOSPHORUS SALTS ON NITROGEN MINERALIZATION //

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DECLARATION

This thesis is my original work and has not been presented for a degree in any other University.

Signature..... 21/7/89

GEORGE N. KARUKU

This thesis has been submitted for examination with my approval as a University supervisor.

Signature.....

DR. B.O. MOCHOGE

DEDICATION

TO MY BELOVED PARENTS WHO TIRELESSLY PAID MY SCHOOL FEES

AND ALL THOSE PEOPLE WHO VALUE EDUCATION

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LIST OF ABBREVIATIONS

ha	=	hectare
kg	=	kilogram
g	=	gram
mm	=	millimetre
ml	=	millilitre
cm^3	=	cubic centimetre
me	=	milliequivalent
N	=	Nitrogen
N_o	=	Nitrogen mineralization potential
µg	=	Microgram
CEC	=	Cation exchange capacity
ECEC	=	Effective cation exchange capacity
EDTA	=	Ethylenediamine tetracetic acid
DAP	=	Diammonium phosphate
AS	=	Ammonium sulphate
TSP	=	Tripple superphosphate
$CaC0_3$	=	Calcium carbonate (Lime)
FAO	=	Food and Agricultural Organisation
UNESCO	=	United Nations Education, Scientific and Cultural Organisation
C:N	=	Carbon: Nitrogen Ratio
Log	=	Logarithm
^oC	=	Degrees centrigrade
Ca	=	Calcium
Mg	=	Magnesium
Na	=	Sodium
K	=	Potassium
H	=	Hydrogen
%	=	Per cent
P	=	Phosphorus
Al	=	Aluminium
NH_4	=	Ammonium
$N0_3$	=	Nitrate
$N0_2$	=	Nitrite
N_20	=	Nitrous oxide
Mins	=	Minutes
r	=	Correlation coefficient
rev	=	Revolutions
Calc.	=	Calculation
Con.	=	Concentration

NITROGEN FORMS IN SOIL AND EFFECT OF LIME, NITROGEN AND PHOSPHORUS SALTS ON NITROGEN MINERALIZATION

ABSTRACT

Incubation experiment studies were done to determine the effect of lime ($CaCO_3$) at a rate of 10 tons/ha, Diamonium Phosphate (DAP) and Ammonium Sulphate (AS) at 200 Kg N/ha and Tripple Super Phosphate (TSP) at 100 Kg P/ha on N mineralization. The three soils used were from Gituamba (Andosols), Kitale (Ferralsols) and Katumani (Luvisols). A secondary objective was to determine the values for the mineralization potential (N_0), rate constant (k) and the rate of mineralization using Stanford formula for untreated soils only; $N_0=9.77N_t$, where N_t is the nitrogen mineralized in two weeks.

The soils were incubated aerobically in polythene bags for a period of 120 days in the laboratory at room temperature and available nitrogen (NH_4^++NO_3^-) was determined at specified periods during the experiment. The mineralized N was higher where treatments were applied than in the control. Where lime was applied in Gituamba soils, mineralized N was significantly higher ($P \leq 0.05$) than the control. However, for Kitale soils, although liming increased N mineralization, it was not significant ($P \geq 0.05$) as compared with the control. Where DAP and TSP were applied, a slight effect on nitrogen mineralization was observed. This was attributed to P which affects microbial biomass steadily. The rates of mineralization were higher in the 0-15 cm than 15-30 cm sampling depths with Gituamba indicating the highest. The mineralization potential for the 0-15 cm and 15-30 cm layers were 392.3 and 162.5 µgN/g for Gituamba; 195.6 and 178.7 µgN/g for Kitale; and 198.0 and 75.8 µgN/g soil for Katumani soils. The time required for 50% of N_0 to be mineralised indicated that less than half the potential value would become available in a normal growing season with the possible exception of Gituamba (15-30 cm) and Katumani soils. The trends of the calculated values compared well with the observed ones except for Katumani soils. The Stanford formula appears therefore not to work well with soils low in organic matter.

Organic N forms were also determined in the three soils. The hydrolysable organic N for the 0-15 and 15-30 cm layers was 57.2 and 59.3% for Gituamba; 56.9 and 61.9% for Kitale; 39.0 and 42.1% for Katumani soils, respectively. Amide N ranged from 11.6 to 21.4% of total N; Hexosamines from 5.2 to 10.1% and Amino acid N from 26.2 to 37.1%. Amino acid therefore formed the highest portion followed by Amide N of the hydrolysable organic N.

CHAPTER ONE

1.0 INTRODUCTION

Nitrogen is a very important nutrient in crop production, it promotes vegetative growth of plants and it is a constituent of proteins and other tissues. Plants deficient in nitrogen have stunted growth with yellowish leaves and their yields are low. Unfortunately, this element is extensively deficient in most cultivated soils and therefore one of the most limiting factors in plant production.

The atmosphere is the primary source of nitrogen where it is strongly bonded as nitrogen (N_2) gas. In its elemental form, nitrogen is useless to higher plants unless converted to usable forms. This is accomplished for example through fixation by Rhizobia and other micro organisms which live symbiotically in the roots of legumes and certain non leguminous plants.

Nitrogen found in the soil can be classified as inorganic or organic. By far the greater portion occurs as soil organic complex which accounts for nearly 98% of total nitrogen in top soil. The inorganic forms of soil nitrogen include NH_4^+, NO_3^- and NO_2^- whereby NH_4^+ and NO_3^- are of greatest importance in crop uptake. This inorganic nitrogen rarely exceeds 2 to 3% of total soil nitrogen (Bremner, 1951). Various factors affect transformation of nitrogen fractions in the soil. These include climatic factors, soil conditions, and the type and condition of organic matter found in soils. Transformation is brought about by soil microorganisms and so their activity in terms of rate and amount of nitrogen turnover will depend on the environment in which they will be found. The microorganisms decompose organic matter into simpler forms and hence mineralization is accomplished.

The amount of nitrogen released to plants each season is largely a function of the rate at which this nitrogen in the organic matter is mineralized by microorganisms. Bremner (1955) noted that organic forms of soil nitrogen occur in various stages of humification and decomposition, and are closely related to microbial activity. This in turn can lead to net release of N from the organic reserve as mineral nitrogen (Kai et al., 1973).

In natural ecosystems, the amount of mineral nitrogen (released from organic matter after mineralization) appears to be adequate because nitrogen in form of fertilizers is rarely applied.

However, for agricultural ecosystems, this amount is not enough to warrant adequate crop performance, hence application of nitrogen fertilizer (Mochoge and Beese, 1985). Addition of fertilizer salts have been shown to increase nitrogen mineralization. Munevar and Wollum (1977) observed increased N mineralization when phosphorus salts were added. Similar results were obtained by Stotzky and Norman (1961). Singh and Beauchamp (1986) also observed increased nitrogen mineralization with addition of nitrogen salts. However, some other workers have found contrasting results. Jackman (1960) showed that addition of N salts depressed microbial respiration hence low N mineralization.

Knowledge on N transformations in temperate regions is wide but low in the tropical regions. In Kenya, very little is known on the dynamics, and rates of change of nitrogen in soils. Understanding the extent and rate of change of organic nitrogen in soils is important from the standpoint of N management and rates of N application in form of mineral fertilizers. For example, any addition of nitrogen fertilizers to the soil should take into consideration the amount of residual nitrogen, the rate and amount of mineralized nitrogen and the needs of an individual crop (Mochoge and Beese, 1985). The purpose of this study therefore was (i) to identify and quantify the inorganic forms of nitrogen in the soils and their distribution (ii) to identify and quantify the fractions of organic nitrogen in soils (iii) to establish the effect of lime, N and P salts on N turnover in soils and (iv) to determine the rates of N mineralization in soil.

CHAPTER TWO

2.0 LITERATURE REVIEW

2.1 Total nitrogen in soils

The nitrogen cycle in soil is an integral part of the overall cycle of nitrogen in nature. The primary source of soil nitrogen is the atmosphere, where the strongly bonded gaseous molecule nitrogen is the predominant gas (79.08%) (Bartholomew and Clark 1965).

Total nitrogen content in the soil ranges from 0.02% in subsoils to more than 2.5% in peats; ploughed layers of most cultivated soils contain between 0.06% and 0.5% nitrogen (Bremner, 1960). The amount present in each case is, however, determined by climate, type of vegetation, topography, parent material and activities of man.

Climate is a very important factor as it determines the rate and magnitude of nitrogen transformation in the soil. It should first of all be understood that climate determines plant species available at any location, quantity of plant materials produced and the intensity of microbial activity in the soil. This in turn determines the nitrogen and organic matter levels in the soil. Tropical soils are extremely poor in nitrogen nutrient. Das (1949) with Indian soils found that total N amounted to about 0.05% while most of European soils amounted to 0.15%. This situation is caused by a number of factors. The virgin lands of the tropics have been cultivated for several years and thus the removal of nutrients by crops has been more than the addition into the soils. This therefore means that the soils have been very much impoverished. However, in high regions of the tropics, the amount of N-content is said to be higher than 0.05%. Harradine and Jenny (1958) in California found that the amounts of nitrogen content increased with decreasing temperature and with increasing precipitation.

Nitrogen in soils is largely bound to organic matter and mineral materials. Under natural conditions, this nitrogen is gained in soils through fixation of elemental nitrogen by microorganisms and from the accession of ammonia and

nitrate in rain water. Losses occur through crop removal, leaching and volatalization (Bartholomew and Clark, 1965). Although nitrogen uptake by crops and microorganisms is considered a loss of nitrogen from the soil, it is not completely a loss but a temporary one. After death, crops and microorganisms decompose and release nitrogen to the soil. The tropical crops also do not make proper use of available nitrogen in the soil. Plant recovery of available soil nitrogen lies between 50 and 60% (Rogers, 1961). The other 40 to 50% somehow escapes from the soil system. Denitrification and volatalization processes account for 5 to 15% of the available nitrogen (NO_3^-) (Allison; 1966).

Soil management also influences the magnitude of total nitrogen in the soils. Juo and Lal (1977) reported a significant increase in organic carbon and nitrogen content in Nigeria (Alfisol) in the presence of residue mulch as compared to soils without residue mulch. Harding (1955) in southern California observed that constant cropping diminished organic matter and total nitrogen in the soil unless careful use was made of green manure and barnyard manure. He also showed that high nitrogen contents were associated with high organic matter content. These in turn were associated with soil texture. The lowest values of both organic matter and total nitrogen were characteristic of sandy loams, whereas the highest values were characteristic of clay soils. The surface (0-15 cm) had higher organic matter and total nitrogen than did the subsoils. Baum (1975) found that in forest soils, the nitrogen content differed from one humus form to another.

2.2 Available nitrogen (NH_4^+ +NO_3^-N)

This is nitrogen found in the soil solution is in a chemical form that can be readily absorbed by plant roots. Rarely do soils contain more than 1% of total nitrogen in an available form at any one time. Most nitrogen is present as organically bound form which is unavailable to higher plants. Available nitrogen in the soil is supplied by symbiotic fixation, non-symbiotic fixation, organic matter through mineralization, fertilizer additions and rainfall. The most important

available N are ammonium (NH_4^+), Nitrate (NO_3^-) ions and some certain simple organic compounds, principally those containing free amide or amino groups. Nitrites (NO_2^-) are minor source of available nitrogen but more than a few parts per million are toxic to most plants (Tisdale and Nelson, 1966). Although plants can utilize any of these nitrogen forms, there are numerous exceptions (Wallace, 1954).

One form may be preferentially absorbed depending on the environment, the species and the age of the plants (Gosh and Burris, 1950). Ammonium ions in soils are found in soil solution, as part of the exchange complex, and in clay lattices restricting its exchangeability. The amount in soil solution is extremely small but it is in dynamic equilibrium with exchangeable ammonium. Ammonium ion is an important source of available nitrogen especially in grassland and to a lesser extent in forests (Vlassak, 1970). In grasslands a large part of soil organic matter consists of the remains of old roots where the organic matter is distributed throughout the root zone (Bartholomew and Clark, 1965). The ammonium ions are released by ammonification process in an area permeated by plant roots that immediately absorb any ammonium not utilised by microorganisms (Bartholomew and Clark, 1965).

Available nitrogen in forest soils is fairly low. According to Runge (1971), the concentration ranges from 0.5 mg in mineral soil to nearly 17 mg/100 g soil in humus layer . Mineralization of nitrogen in the forest floor and turnover of small roots are now generally recognized as the main pools of available nitrogen in forest ecosystems (Wollum and Davy, 1975). Tillage also affects the concentration of available nitrogen in the soil. Gallaher and Ferrer (1987), in the University of Florida reported that where there was no tillage, the Kjeldahl nitrogen resulted in 23 to 24% N more than conventional tillage in the 0 to 5 cm soil depth after 3 to 6 years of cultivation. Also after 6 years, the 0-5 cm depth had 36% more organic matter in no-tillage than in conventional tillage. The same trend has been observed by Lal (1976) and Blevins et.al; (1977). Agboola (1981) reported that conventional tillage and application of fertilizer enhanced organic matter

breakdown, leading to an increase in soil erosion and hence a greater loss of organic matter. This in turn led to a decrease in available nitrogen.

Soil texture also influences nitrogen availability in soils. Gallaher and Ferrer (1987) suggested that coarse- textured soils subjected to high amount of rainfall are more prone to losses of nitrogen through leaching. They also suggested that high temperature seems to be more correlated to denitrification.

Aeration, especially in paddy fields, affects nitrogen availability. Hanif et al; (1986), working in an irrigated rice system, showed that ammonium, deeply incorporated in a submerged irrigation system, diffused upwards from anaerobic to aerobic layer where biochemical oxidation nitrified it to nitrites and nitrates. These oxidised nitrogen species diffused downwards to the anaerobic layer where most or at least part, was lost as gaseous end products. Most nitrogen losses in alternately flooded and dried conditions occur due to nitrification and denitrification processes that are affected by the environmental changes and microbial population (Ponnamperuma, 1972; Reddy et al., 1976; Reddy and Patrick, 1975). On continuously submerged soils, the subsurface soil layer may be reduced with an overlain oxidised surface layer. This consequently leads to simultaneous nitrification - denitrification processes hence nitrogen losses occur (Hanif et. al., 1986; Reddy et al. 1976).

Soil pH also influences nitrogen availability. Ishaque and Cornfield (1972) showed that other factors being equal, production of inorganic nitrogen was greater in neutral than in acid soils although some soils show little influence of pH on N-transformation.

2.3 Fixed ammonium nitrogen in clay mineral

Some soils have ability to bind added ammonium (NH_4^+) and also potassium (K^+) in such a manner that it will not be readily replaced by other cations. MacBeth (1917) demonstrated that in some soils, the added ammonium nitrogen could not be completely recovered by extraction with 10% HCl solution or by Alkaline

distillation. This was later checked and confirmed by Chinamade and Druoineau (1936), and Chinamade (1940). The concept of the mechanism of ammonium ion fixation in soils is derived from the studies of potassium fixation which is considerably more extensive than ammonium. The concept is based on the inclusion of NH_4^+ ions between the lattices of clay minerals which have been impoverished with K^+ ions (Nommik, 1957).

On the basis of available information, it seems evident that the ability of soils to fix NH_4^+ may be ascribed to occurrence of micaceous minerals. However, the vermiculites have the greatest capacity to fix K^+ and NH_4^+ ions (Bajwa, 1982). An equilibrium is assumed to exist between non-fixed (i.e. NH_4^+ ions in solution and in exchangeable form) and fixed ammonium in soils (Nommik, 1957) as shown below.

$$(NH_4^+ + K^+)s \leftrightarrows (NH^+_4 + K^+)e \leftrightarrows (NH_4^+ + K^+)f$$

Where "s", "e" and "f" denote solution, exchange and fixed phases, respectively. However Nommik (1957) working with some Swedish soils concluded that a more realistic interpretation of the equilibrium could be as follows.

$$(NH^+_4 + K+)s \rightleftharpoons (NH^+_4 + K^+)\, e \rightleftharpoons (NH^+ + K^+)f$$

The rate of fixation is much greater than the rate of release. In this respect, the fixed ammonium can be considered as being unavailable for microbes and plants.In general, the fixing capacity of soils does not exceed 10 meq/100 g dry soil, but in some, a capacity of more than 15 meq/100 g dry soil has been observed (Legg and Allison, 1959).

Some research workers have suggested that the portion of non-exchangeable ammonium is rarely more than 8% in surface soils but can exceed 40% in subsoils. Bremner (1959) found that fixing capacity of surface soils is less than in subsoils. This could be due to the fact that the content of native fixed ammonium related to the total amount of nitrogen in subsurface soils is generally higher than in surface soils (Stevenson and Dhariwal, 1959).

The rate and magnitude of ammonium fixation depends on such factors as type of clay minerals present, the amount of potassium and to a lesser extent on the other cations present, soil pH, temperature and moisture content of the soil. Vermiculite has the greatest fixing capacity of K^+ and NH_4^+ ions (Allison, et. al 1953a). Other clay minerals include illite (Allison, 1953b, 1953c) and montimorillonite (Allison, 1953c, Hinman, 1964). The capacity of clay minerals to fix ammonium depends on moisture conditions. The fixing capacity of illite increases greatly both in rate and magnitude after drying by heating the soils (Allison, et. al., 1951). That of montimorillonite is much less than illite and negligible under moist conditions (Nommik, 1967), while vermiculite is capable of fixing to the same extent under moist as well as dry conditions.

Cations such as Rubidium, Caesium and many others have shown inhibitory effect on fixation and release of NH_4^+ ions (Wiklander, 1950). However,high concentration of NH_4^+ ions in soil can increase ammonium fixation (Nommik, 1957, Allison, et. al., 1951). Though it is stated that increased temperature increases fixation (Harada and Kutsuma, 1954), it is, however, not clear how far the magnitude of fixation is. Jansson (1958) showed that addition of potassium (2 hours before addition of NH_4^+) inhibited fixation of ammonium by about 33%; whereas simultaneous addition of K^+ and NH_4^+ ions did not affect it.

2.4 Organic nitrogen in soil

Present evidence indicates that over 95% of total nitrogen in most surface soils is organically combined. Bremner (1959) suggested that the portion of non-exchangeable nitrogen is high in subsurface soils. Until 1954, it was generally assumed that only 2% nitrogen in soils is in inorganic forms and that organic nitrogen could be estimated accurately through the determination of total nitrogen in the soils. Now it has been established that some forms of nitrogen are fixed hence non-exchangeable (Bremner, 1959; Walsh and Murdock, 1960; Bajwa, 1982). Research on vertical distribution of nitrogen in soils shows that the organic nitrogen

decreases with depth except in soils derived from recent sediments where irregularities may occur (Brown and Thorpe, 1942; Stevenson, 1957c, 1959). In most soils, the bulk of organic nitrogen occurs in the surface 60 cm of the profile where there is decomposition of organic residues.

Within the equatorial type of soils, it has been found that ochrosols which are less acidified and more aerated have less organic matter accumulation when compared to oxisols, whose organic matter is concentrated markedly on the top few centimeters. Concerning the decomposition of organic matter, Birch and Friend (1956b) found that in East Africa, there was in general an apparent inverse relationship between humus content and increase of rainfall with altitude. Increasing temperature no doubt favoured high rate of decomposition of organic material, but it also favoured its rates of production. Gallaher and Ferrer (1987) in Florida showed that with no-tillage, Kjeldahl nitrogen increased with increase in organic matter content as compared to tilled plots in the 0 to 5 cm depth of the soil profile. Juo and Lal (1977), working with Alfisols in Nigeria found significant increase in nitrogen content of soil in the presence of residual mulch. Lal (1976) and Blevins et al. (1977) had similar observations. These results agree with Shigeyoshi et al. (1986) who found that application of organic matter increased organic nitrogen (all forms) and this decreased with depth. They also support the fact that organic forms of nitrogen are affected by soil management such as cultivation, cropping and fertilizer application (Keeney and Brenner, 1964; Porter et al., 1964).

2.4.1 Organic forms of nitrogen in soils (Fractionation)

Knowledge concerning the nature of organic nitrogen in soils is based largely on studies involving identification and estimation of forms of nitrogen released by treatment with hot acids. These hydrolysis studies have shown that 20 to 40% of total nitrogen in most surface soils is in form of bound amino acids (Stevenson, 1954; 1956b; Young and Mortensen, 1958; Keeney and Bremner, 1964) and 5 to

10% in form of combined hexosamines (Keeney and Bremner, 1964). The proportion of amide nitrogen is between 4 and 28%, that of amino sugar nitrogen between 2 and 5% and unidentified nitrogen make up 16 to 24%) (Shigeyoshi et. al., 1986). All these forms decrease with depth whereas ammonium nitrogen remains fairly constant throughout the profile. However, non-hydrolysable nitrogen increases with depth.

Shigeyoshi et. al. (1986) found that application of organic matter such as rice composite, rice straw and Italian rye grass led to a marked increase in all forms of soil nitrogen whose content was highest in surface soil and decreased down the profile. Bremner (1955) noted that organic forms of nitrogen occur in various stages of humification and decomposition and are closely related to microbial activity. This was later observed by other workers (Chu and Knowles, 1966; Nommik, 1967; Wagner and Mutatkar, 1968; Kelley and Stevenson, 1985).

It has also been shown that added organic residues decompose rapidly and disappear. It is therefore assumed that much of the organic nitrogen which remains in the soil is of microbial origin (Wagner and Mutatkar, 1968; Kai et al., 1973; Maramoto et al., 1982; Kelley and Stevenson, 1985). Accordingly, there is possible diverse organic matter products which could be added to the soil.This, therefore, could mean that different sources of organic matter added to the soil do not exhibit a similar quantity and distribution of organic forms of nitrogen (Cheshire et al., 1982; Gupta and Reuszer, 1967).

Amino acids and hexosamines liberated by acid hydrolysis have been identified by paper Chromatography. However, their modes of linkages have not yet been established. Purine and pyrimidine derivatives have been detected, but current evidence indicates that they do not account for more than 1% of total nitrogen in surface soils (Adams et al., 1954). Others such as Choline, Creatine and Allantoin have been isolated from the soils, but quantities detected have been very small. There is no evidence to suggest that these compounds account for a significant amount of organic nitrogen in the soils.

Theories have been advanced that this nitrogen is in form of lignin-ammonia, quinone-ammonia, quinone-amino acids or carbohydrate-amino acid condensation product (Tisdale and Nelson, 1966). Sowden (1958) suggested that formation of nitrogenous components of humus is more complex than simple lignin-protein interaction. This is so in view of the fact that, the highly decomposed humus materials are lower in amino nitrogen and higher in ammonium nitrogen than the less decomposed materials. Nevertheless there is very little evidence to support the above theories. The chemical nature of about half of the organic nitrogen in the soils remains obscure. Moreover relatively little is known with respect to the characteristics of organic nitrogen in different soil horizons of soil profile (Stevenson, 1982) especially in paddy fields.

There are several limitations associated with hydrolysis of soils with 6N Hcl. The most obvious is that it does not characterise a considerable proportion of nitrogen released by hydrolysis. It also does not provide information concerning the nature of non-hydrolysable (acid insoluble) nitrogen which constitutes 20 to 40% of total nitrogen in surface soils. Nevertheless, they provide useful information concerning the nitrogen compounds in soils and have proved useful in studies of the distribution of various forms of nitrogen in soil profiles (Stevenson, 1957c; Sowden, 1958; Bremner, 1958b; 1959). These studies have also provided information of the effects of cultivation and other treatments on nitrogen compounds in soils (Rendig, 1951; Keeney and Bremner, 1964).

One interesting result of acid hydrolysis is that most of the nitrogen is released as ammonium nitrogen which increases markedly with time (12 hours of refluxing), representing 15 to 25% of total nitrogen (Keeney and Bremner, 1964). Evidence has it that some of this ammonium nitrogen is formed by hydrolysis of amide (Glutamine and Asparagine) residues in soil organic matter (Sowden, 1958). However, calculations based on the assumption that glutamic acid and aspartic acid in soils occur entirely in form of glutamine and asparagine, show that much of the ammonium liberated by acid hydrolysis of soil cannot be derived from these

amides. Present evidence indicates that some of this ammonium is formed by decomposition of hydroxyamino acids (Serine and Threonine) (Bremner, 1949a; Bremner and Shaw, 1954). The proportion of soil nitrogen present as amino acids decreases with depth in the soil profiles (Sowden, 1956, 1958; Bremner, 1958b; 1959) and is affected by cropping, cultivation and fertilization (Porter et al., 1964; Keeney and Bremner, 1964).

2.5 Transformations of organic forms of nitrogen in soils

2.5.1 Nitrogen mineralization

Nitrogen in surface soil horizons is predominantly (=98%) in organic form (Bremner, 1951). In this form, nitrogen is unavailable for plant and microbial use in the soil. It means therefore that it has to be converted to forms available for plant use, that is, ammonium (NH_4^+) and nitrates (NO_3^-).

The conversion of organic nitrogen to more mobile, inorganic state is known as "Nitrogen Mineralization" and is accomplished in two steps; ammonification (production of ammonium from organic matter) and nitrification (Conversion of ammonium to nitrate). The reverse, i.e. incorporation of inorganic to organic forms, is known as "nitrogen immobilization". The opposing processes, immobilization and mineralization occur simultaneously in most ecosystems, for example in the soil, where organic material is undergoing microbial decomposition.

Mineralization of nitrogen is the result of the metabolism of a multitude of microbial strains, mostly chemoheterotroph (Buckman and Brady, 1969). Because the ultimate liberation of ammonium from organic matter is associated with many physiologically dissimilar microorganisms, nitrogen is mineralized in most extreme conditions (Alexander, 1977). The amount of ammonium that accumulates, however, varies with the nature of organisms, the substrate, the soil type and environmental conditions.

The break down of organic compounds containing nitrogen is through proteolytic enzymes synthesized extracellular by microorganisms. This process is

known as proteolysis, whereby proteins are hydrolysed into simpler units, i.e. peptides and amino acids. The release of ammonium is then accomplished from amino acids through ammonification process. The rate of mineralization-immobilization is influenced by a number of factors. These include C:N ratio, type of organic substrate, pH, soil moisture content, temperature and aeration of the soil.

The C:N ratio of organic matter added to the soil has great influence on net mineralization and immobilization of nitrogen in the soil. It is often said that organic matter with a C:N ratio wider than 30:1 favours net immobilization while materials with a ratio narrower than 20:1 favour net mineralization (Jansson, 1929). Nevertheless, this is not always the case because in forest soils with litter fall having C:N ratio wider than 20:1 i.e. ranging from 40 to 60:1 (Roberge and Knowles, 1966; Sowden and Ivarson, 1974; Ohta and Kumada, 1977; 1978), a considerable net mineralization has been observed (Richards, 1978).

During initial decomposition, immobilization dominates over mineralization, and the total nitrogen concentration of the litter increases while the C:N ratio decreases. Gosz et al; (1973), using the Nylon Mesh - bag technique, found that during 12 months on forest floor, the percent nitrogen of yellow Birch leaves (Betulla allegheniensis Brith) increased from 0.85 to 2.31% and C:N ratio decreased from 62:1 to 23:1. Ohta and Kumada (1978 a, b) determined elemental composition and laboratory nitrogen mineralization rates (20 weeks at 30°C) in 29 forest soils. The C:N ratio ranged from 35 to 67:1. By the end of incubation period, all samples showed increased organic nitrogen (net mineralization). The rate of mineralization was markedly less in samples with C:N ratio over 40:1.

Significance of some major soil variables to nitrogen mineralization has been established under laboratory conditions. These include texture with which the emphasis has been on varying aggregate sizes (Craswell and Waring, 1972), soil moisture, with which mineralization has been found to vary directly within the

available range (Reichmann et al., 1966) and organic matter, particularly that which accumulates under pasture (Huntjens, 1972).

Lindemann and Cardenas (1984), using a clayey soil (Glendale, a typic Torrifluvent) and a sandy soil (Latene, a typic Calciorthid), found that mineralization rates decreased rapidly in the first two weeks followed by slower rates with time. The total mineralization rate was found to be higher in clayey soil than sandy soils which had been amended with sewerage sludge while net mineralization decreased. Nitrogen mineralization also decreases with depth. This reflects the amount of organic matter present and the microbial population. This has been observed before by Mochoge and Beese (1985) dealing with a Calcic Luvisol and dystric cambisol in central Europe.

Other factors being equal, production of inorganic nitrogen is greater in neutral than in acid soils (Ishaque and Cornified, 1972; Mochoge, 1981), although some soils show little influence of pH on N transformation. Organic nitrogen accumulates in acid soil due to the slow mineralization. A rapid release of mineral nitrogen is seen, however, when these soils are limed (Harmsen and van Schreven, 1955). This also applies to immobilization which increases with increase in pH (Broadbent and Tyler, 1965). Various workers have shown that nitrogen mineralization increases with increase in temperature (Biederbeck and Campbell, 1972; Mochoge and Beese, 1985; Shen et al., 1984). Mochoge and Beese (1985) reported that mineralization is influenced by physical and biological properties of the soil and that soils with maximum disturbance have a higher microbial activity at the soil surface which decreases with depth. Vlassak (1970) observed that there is rapid mineralization in cultivated soils and pastures while soils under natural vegetative covers show low but steady mineralization and mostly are rich in ammonium nitrogen.

Mochoge and Beese (1985) showed that in the arable luvisols, the net mineralization increased cumulatively. In forest soils the rate of nitrogen mineralization decreased with depth (Mochoge and Beese, 1985). This was thought

to reflect the quantity of mineralizable organic matter with depth. The reduction in net nitrogen mineralization with depth reflects the decreasing humus content with depth (Runge, 1971). Agarwal et. al; (1972) in Hawaii observed that mineralization of native nitrogen was curtailed by addition of carbon sources. Addition of readily available energy sources also showed that mineralization of native nitrogen was apparently related to total nitrogen and organic carbon contents. Soils with high total nitrogen and carbon had higher mineralization than those with low contents of the two elements. One soil with highest total nitrogen showed least mineralization. This nitrogen was thought to be tied up in a highly resistant organo-inorganic complex.

Stanford et al., (1974) developed a concept of potentially mineralizable soil nitrogen. This is the fractional quantity of soil organic nitrogen that is susceptible to mineralization according to first order kinetics. This reaction is expressed by equation $dn/dt = kn$. By integration, it gives: $\log (N_O - N_t) = \log N_O - k_t/2.303$, where N_O is nitrogen mineralization potential and k is the mineralization rate constant. They found that, the rate constant, k, was reasonably equal for a large number of soils and that a period of two weeks incubation following a short pre-incubation (1 or 2 weeks) is sufficient for estimating the mineralization potential (N_O); Stanford et. al. (1974) proposed a simplified equation for calculating the nitrogen mineralization potential (N_O): $N_O = 9.77\ N_t$, where N_t is nitrogen mineralized in two weeks. Stanford and Smith (1972), dealing with five types of soils (Entisols, Alfisols, Ultisols, Aridisols and Mollisols) reported that the rate of nitrogen mineralization was proportional to the quantity of nitrogen comprising the mineralizable substrate.

Some workers have reported cumulative inhibitory effects arising during incubation. Allison and Sterling (1949) observed a drop in pH during 23 weeks of continuous incubation. The drop in pH on incubating the soil could possibly be due to H^+ ions released during the process of nitrification. In some cases, accumulation

of unspecified toxins have been suspected (Harmsen and Schreven, 1955) especially in grassland soils.

Enzymatic conversion of organic nitrogen substances yielding ammonium ions is considered as the rate limiting step in nitrogen mineralization. This reaction is not regarded as being diffusion controlled. The low nitrogen mineralization during the initial stages of incubation is considered to reflect the lag phase (Mochoge and Beese, 1985; Singh and Beauchamp, 1986; Stanford and Smith, 1972). It could also be due to assimilation of nitrogen by organisms, owing to decomposition of small amounts of low nitrogen plant materials (Stanford and Smith, 1972). This could also be due to the lower population of micro-organisms present at the time (Zake, 1989; personnal communication).

The correlation of nitrogen mineralization in two weeks and nitrogen mineralization potentials are usually low but improve with time. This is also the case for cumulative net nitrogen mineralization. The estimates of potentially mineralizeable soil nitrogen are based on short-term incubations (Stanford et. al., 1974). These research workers noted that the estimates derived from short-term incubations were similar to those derived after extensive periods of incubation. A greenhouse study (Stanford et. al., 1974) indicated that nitrogen mineralization potential (N_O) has intrinsic values in predicting amount of soil nitrogen mineralized under specified environmental conditions.

2.5.2 Nitrification

This is a biochemical process which results in the formation of nitrites and nitrates from compounds containing reduced nitrogen forms. The organisms (autotrophs) obtain their carbon from carbon dioxide and energy from ammonium and nitrite oxidation. Under this condition, nitrates formed are available to plants and are also easily lost from soil through denitrification and leaching. Though other genera of the nitrifying organisms have been isolated (Meiklejohn, 1954; Delwiche, 1956), it is generally assumed that only two autotrophic genera are dominant in

nitrifying in most soils. These are nitrosomonas and nitrobacter (Alexander and Clark, 1965). These two genera are strict chemo-autotrophs and accomplish nitrification in two steps.

Nitrosomonas; $NH_4^+ + 1\frac{1}{2}O_2 \rightarrow NO_2^- + 2H^+ + H_2O$ = -84.0 Kcal

Nitrobacter; $NO_2 + 1/2O_2 \rightarrow NO_3$ = -17.8 Kcal.

The rate of this conversion depends on environmental conditions affecting these organisms such as soil pH, aeration, temperature, soil moisture content, nutrient status of the soil and the organic matter content of the soil.

The optimum pH is between 6.5 and 8.0 while extreme acidity and alkalinity retard their activities (Sarathchandra, 1978). However reasonable nitrification has been observed up to pH 4.0 (Boswell, 1955; Weber and Gainey, 1962). This is attributed to possibly acid adapted strains or chemical differences in various habitats. This means that strains derived from acid soils are more tolerant to high hydrogen ion concentration and also to aluminium than those from areas of alkaline pH (Brar and Giddens, 1968). Tyler and Broadbent (1960) reported that nitrate transformation in acid soil were highly sensitive to nitrite concentration in the soil and were therefore inhibited even at very low levels. They reported that nitrite had less effect in alkaline soils and suggested that losses here were in gaseous form. Nitrites inhibited the respiration rate of soil microbes as a whole and not only the nitrifying group (Tyler and Broadbent, 1960). In warm soils, the rate of nitrification increases with increase in temperature, reaching the maximum at between 25°C and 35°C. As conversion of nitrate is an oxidative process, its rate of conversion therefore depends on oxygen supply to the bacteria concerned. Where oxygen supply is inadequate for microorganisms there is little ammonium oxidation and the reaction ceases in the total absence of oxygen. Fathi and Bartholomew (1951) observed a considerable reduction in nitrification due to reduction in partial pressure of oxygen below that found in the air. Moisture affects aeration regime of the soil. The water status of microbial habitats therefore, has a marked influence upon nitrate production. Waterlogging limits oxygen diffusion

hence suppressing nitrification while at the other extreme, in arid conditions, bacterial proliferation is retarded by insufficiency of water rather than oxygen supply (Bartholomew and Clark, 1965). Optimum moisture level varies considerably with different soils and appears to lie between 50% and 60% of the water holding capacity of the soil.

Purchase (1974), in Rhodesia (Zimbabwe), observed that in tropical grassland soils deficient in some nutrients such as phosphorus, nitrification was restricted, causing nitrite oxidizers to show delayed or negligible response to applied ammonium salts.

2.5.3 Denitrification and volatilization processes

Though much work has been done to account for nitrogen fertilizer applied in the soil, i.e. through leaching losses, plant uptake and amount remaining in the soil, there is always a deficit which is unaccounted for (Allison, 1955). Soulides and Clark (1958), working with closely adjacent grassland and intertilled sites of Codorus silt loam, Bladden clay loam, Cecil sandy loam, Hixton fine loam, Fayette silt loam, Hanecye silt loam and Pachappa sandy loam soils encountered losses up to 50% of applied nitrogen. Similar observations were made by Wagner and Smith (1958). These losses were attributed to denitrification. This is the process whereby nitrogen is lost as a gas from the soil. It is a biological process whereby nitrates and nitrites are reduced to volatile gases, usually nitrous oxides and molecular nitrogen.

In this process, certain bacteria use oxygen in nitrate and nitrites hence reducing it to Volatilizable gas (N_2O or N_2). This process occurs where there is poor drainage and lack of oxygen. It is especially favoured in poorly-drained upland soils or waterlogged soils containing nitrates (Ulysses, 1982).

Recent research work has shown that there are other pathways of N losses in soil which involve a combination of chemical and biological reactions. Allison (1963) in Maryland, U.S.A., showed that nitrous oxide (N_2O) is evolved during microbial oxidation of ammonium nitrate. Bollag et al., (1973), Yoshinda and

Alexander (1970) had similar observations. Thus, increased nitrification following liming of soils could also favour increased losses of N_2O. This loss is not greatly affected by pH (Focht and Verstraete, 1977). When nitrogen is applied as Urea or as ammonium salts, ammonia volatilization (non-microbial process) is likely to increase with increased pH (Terman, 1979). Lindemann and Cardenas (1984) observed increased denitrification in soils where sewerage sludge was added. It was also higher in Clayey than in sandy soils.

2.6 Effect of liming on nitrogen mineralization in soils

Liming acid soils has been shown to increase both soil pH and nitrogen mineralization. Similar findings were reported by Simard, et. al. (1988); Sarathchandra (1978); Nyborg and Hoyt (1978); Sarathchandra and Upsdell (1981). Singh and Beauchamp (1986) in Ontario, Canada, observed that liming increased organic nitrogen mineralization and that mineralization rate was lowest at lower levels of liming.

Nyborg and Hoyt (1978) in a Canadian agricultural station noted that in surface acid soils (half of them luvisols while the rest consisted of gleysols, Chernozems, Podzols and Solonetz), increase in nitrogen mineralization correlated with an increase of soil pH. Carter (1986) observed a reduction in N mineralization as a result of gypsum addition which led to a fall in soil pH.

Nommik (1978) in central Sweden and using a podzolic soil evaluated the effect of lime (added calcium oxide at pH 6.0) in humus under pine, spruce, alder or beach forests. The C:N ratio of the humus ranged from 17.5 to 35.4:1. In all cases, nitrogen mineralization occurred over 115 days of incubation period (incubation at 19 to 22°C), but often times, liming either depressed the mineralization rate or led to net immobilization.

Singh and Beauchamp (1986) suggested that apart from nitrification being low due to low nitrifier activity, it could also be due to low clay content. These two workers reported one acid soil (pH 4.7) where liming reduced the nitrifiers.

Manrique (1987) showed that shortly after liming a clayey, kaolinitic typic Tropudult in Panama, both soil pH in 1M KCl and exchangeable calcium increased with 2 mg of $CaCO_3$ or more of lime. However, the ameliorating effect of liming was short- lived under the frequent rainstorms during the rainy season of 1984. Liming failed to neutralise all exchangeable acidity and recurrent soil acidity and this was evident after 60 days.

2.7 Effect of nitrogen salts on nitrogen mineralization

Addition of N salts increases mineralization of soil nitrogen. This has been suggested by various workers (Broadbent, 1965; Broadbent and Nakashima, 1971). Stocky and Mortensen (1958) reported stimulation of N mineralization in cultured Riffle peat when nitrogen fertilizers were added. Similar finding have been reported by Chu and Knowles (1966) using N^{15} enriched pseudomonas cells added to chicot sandy loam and a black spruce raw humus. However, Janssen (1958) reported a negative effect. Perez and Gollardo (1987) found that incubation of calcareous soils with vegetative water (waste from olive industry), increased availability of inorganic nitrogen forms from the 6th week of incubation. These two workers also showed that nitrogen content in soils below 0.15% led to immobilization. Peterson and Smith (1982)made similar observations and that total nitrogen decreased during incubation. They suggested that it was due to nitrite formation which escaped detection by Kjeldahl method used.

Budimir (1977) showed that intensive application of nitrogen fertilizer in temperate soils changed the physical and chemical properties of soil humus(e.g. pH , C:N ratio, etc). He observed that the content of inorganic nitrogen increased after incubation in samples from fertilized plots especially where nitrogen was applied as Urea. Budimir (1977) also showed that application of ammonium nitrate fertilizer did not result in nitrification except in plots with highest application rate. Nakos (1976) found no nitrification after urea treatment.

Supply of soil inorganic nitrogen may depress or stimulate nitrogen mineralization (Megasur, 1968). Very high concentration of soluble inorganic nitrogen added to the soil may reduce the process of nitrogen mineralization. Inorganic nitrogen added to volcanic ash soils has been shown to have either a depressing effect on microbial respiration (Jackman, 1960) or cause no effect in the process. Agarwal et. al. (1972) in Hawaii observed a stimulatory effect on nitrogen mineralization after addition of inorganic nitrogen.

Williams (1972) evaluated humus stands of Scot pine (Pinus. sylvestris L) that had received various fertilizer treatments. He observed that net nitrogen mineralization for plots treated with urea or ammonium salts was about twice the control. This indicated that the immobilized fertilizer nitrogen was in a more active pool than native organic nitrogen. Application of high nitrogen rates lowers the soil pH and also the phosphorus and potassium content of the soil (Reneau et al., 1968; Link, 1979; Sims and Wells, 1985). High nitrogen rates also increase the osmotic potential of the soil (Sims et al., 1984). This in turn could lead to nutrient toxicities and deficiencies.

Francis et. al., (1960) found that the recovery rate of applied N fertilizer was less than 75% in poorly buffered soils which accumulated NO_2 during incubation or could acquire acidity due to nitrogen fertilizer application or were initially acid during the cause of nitrification of added urea. Neeteson et al., (1986) in Netherlands demonstrated that there was an apparent disappearance of at least some of the added mineral nitrogen followed by its reappearance later on (about 85% disappeared and reappeared after 5 weeks). This was similar in all soils despite those differing in organic matter, pH and texture.

2.8 Effect of phosphorus salts on nitrogen mineralization

Munevar and Wallum (1977) in Columbia reported that addition of phosphorus to Andepts increased nitrogen mineralization. It also increased CO_2 evolved when compared to soils not treated with phosphorus. These two workers

observed significant mineralization of carbon and nitrogen where there was phosphorus-nitrogen interaction. They therefore concluded that available phosphorus was a rate limiting factor for organic matter mineralization in Andepts.

Other workers observed that the level of soil phosphorus was a limiting factor to microbial growth (Stotzky and Norman, 1961; Munevar and Wallum, 1977). These workers suggested that in severely phosphorus deficient soils, available phosphorus limited microbial growth and that its application significantly increased the quantity of nitrogen mineralized.

Purchase (1974) in Zimbabwe showed that phosphorus deficiency in grassland soils restricted nitrification causing nitrite oxidizers to show delayed or negligible response to applied ammonium salts. Ross and Barbara (1978) in New Zealand found the same stimulatory effect on nitrogen mineralization with some grassland soils where phosphorus was applied as a fertilizer, while in some (Tima and Cluden soils), there was no stimulatory effect. Simard et al; (1988) observed that addition of increasing rates of phosphorus to surface soils from a Humoferric podzol in Canada reduced the total concentration of nitrates and ammonium ions in solution but did not affect the pH or CEC of the soils appreciably.

CHAPTER THREE

3.0 MATERIALS AND METHODS

3.1 Research methodology

The need for satisfactory laboratory methods of obtaining an estimate of the amount of nitrogen likely to be made available for crop growth by mineralization of soil organic matter during the growing season of a crop has been a matter of great concern. Numerous studies of biological and chemical approach have been conducted (Keeney and Bremner, 1966; Gianello and Bremner, 1986; Stanford and Smith, 1972; Stanford, 1982). It is generally accepted that the most reliable methods currently available are those involving determination of inorganic N produced by incubation of soil samples under aerobic and anaerobic conditions (Runge, 1974; Mochoge, 1981; Stanford, 1982, Gianello and Bremner, 1986).

Different incubation methods have been used and these include incubating soils in situ (i.e. in bags), use of column studies and incubating of soil samples in polythene bags in the laboratory.

Incubating of soil samples in situ involves burying of soil samples in polythene or mesh bags under soils at different depths from their sites of sampling and monitoring frequently the inorganic N release (Runge, 1974). This method exhibits some field conditions but use of disturbed soil samples and sandwitching of bags in between soil layers puts it slightly off from actual situations in the field in terms of water and air regimes. Moreover, bags used have been often attacked by insects and therefore the results have not been very reliable.

Use of column studies have many advantages especially when undisturbed soils are used. This is because they exhibit transient flow of water and aeration in soil as field situations. However, the method has also some short-comings in that it is usually done in the laboratory or in greenhouse where it is easier to control than in the field.

Incubating of soil samples in polythene bags in the laboratories, though rather unnatural, has however been proved to give reliable inorganic N availed in a given

season and this has correlated well with crop performances (Stanford and Smith, 1972; Stanford, 1982). This method is also easy to carry out hence its use in this study.

The soil samples for incubation studies were weighed (2kg per bag) and then set for incubation experiment. They were placed in polythene bags after moisture content was brought to field capacity, sealed and then incubated for 120 days. The polythene bags have an advantage in that they allow air exchange and minimize water loss (Mochoge and Beese, 1984). Gordon et al. (1987) in Ontario, Canada and working with forest floor material showed that the thickness of polythene bags did not affect mineralization of nitrogen. They also showed that all thickness remained impermeable to waterloss. This ensured constant moisture content over the duration of the experiment. Since there was no leaching possibility in this experiment only the available nitrogen was analysed. That is the part of total mineralized nitrogen that exceeds microbial demand.

3.2 Soils used

Three types of soils were chosen and these differed on the basis of their pH and organic matter content. Their physical and chemical properties are shown in Table (1-3). These soils include the Gituamba andosols, Katumani luvisols and Kitale ferralsols.

3.2.1 Gituamba Andosols

Gituamba is located in Murang'a district, Central Province, with centre coordinates 0°45'S - 36°51'E. It is on the dissected foot ridges (Aberdares) of the rolling volcanic uplands and minor hills. The altitude is 2130 m. The geology of the area is mainly basalts and balsaltic conglomerates of Simbara series. The land is used for tea and pyrethrum cultivation. The ecological zone is II, P/E 82%. The soils are well drained, deep, dark reddish brown, firm, clay with a humic A

horizon (Oswago, 1975). The soils are classified as andosols (FAO/UNESCO, 1974) according to Oswago (1975).

3.2.2 Kitale Ferralsols

Kitale research station is located 3 km south west of Kitale town,Trans Nzoia district, Rift Valley province,with centre coordinates 1°01'N -34°39'E. The physiography is slightly undulating upland with an average altitude of 1860 m. The geology consists of basement system of gneisses and schists, rich in feldspar, biotite, hornblende and garnet with minor exposures of granite and pegmatitic dykes. The vegetation was originally moist combretum woodland to bushland.

In rainfed areas, cultivation consists of maize and pasture research. Soil moisture and soil temperature regimes are Ustic and Isothermic, respectively. Its ecological zone is III, P/E 66%. There are two main soils which can be distinguished here. One is well drained, deep to moderately deep, reddish brown to yellowish red, friable clay on the upper valley slopes. The other one is poorly drained, (dark) greyish brown, firm clays, encountered at bottom lands (Anon, 1971; 1976).

The organic matter content is fairly high. Major clay mineralogy is Kaolin. Others include montimorillonite and illite. Significant quantities of illite may impart to the soil characteristic non -response to potassium and ammonium ions (Anon, 1971). Less water is stored in these soils due to high Kaolin content. These soils are classified as ferralsols (FAO/UNESCO, 1974) (Anon, 1971).

3.2.3 Katumani Luvisols

Katumani is 10 km South of Machakos town, Machakos district, Eastern Province with centre coordinates 01°35 'S - 37°14'E. The physiography is undulating upland. It lies at an altitude of 1575 m. The geology is of quartzo-felds paruc gneisses of precambrian basement system. The land was originally of Acacia

bushland which has now been cleared for cultivation and research on various cereals; in particular maize varieties, but also sorghum, beans and pasture research.

It is in ecological zone IV, P/E 40%. The dominant soils are well drained, deep reddish brown and friable sand clay (Mbuvi and Van de Weg, 1975). The soils are classified as Luvisols (FAO/UNESCO, 1974) according to Mbuvi and Van de weg (1975).

3.3 Soil Sampling

The soil samples were taken from the top 30 cm layer, i.e. 0 to 15 cm and 15 to 30 cm. A profile pit 40 cm deep and 40 cm long was dug. The 15 to 30 cm depth was sampled first and then 0 to 15 cm. This was necessary in order to avoid mixing of the two depths. The samples were then placed in nylon bags to avoid much moisture loss.Undisturbed samples were collected using core rings for bulk density determination.

3.4 Preparation of Soil Samples for Incubation

The soils were weighed in 2 kg for each treatment. For Gituamba and Kitale soils, there were five treatments.These included the control, Lime addition at 10 ton/ha; Ammonium sulphate at 200 kg/ha; Diammonium phosphate at 200 kg/ha and Tripple superphosphate at 100 kg/ha. For Katumani soils, all the above treatments except that of lime were applied. This soil required no liming because of its high pH (Table 3). The soils were then spread in a thin layer for treatments. 1 gram each of ammonium sulphate and diammonium phosphate, 0.5 grams of tripple superphoshate and 10 grams of calcium carbonate ($CaCO_3$) were weighed, put in distilled water and then sprayed on the thin layer of soil to achieve some uniformity for the treatment. Distilled water was added to the soil up to field capacity. The soils were then put in polythene bags and sealed to prevent moisture loss. Incubation was done in the laboratory at room temperature and lasted 120 days.

3.5 Soil Preparation for Extraction

Mineralization and nitrification processes were followed by the changes of ammonium nitrogen and nitrate nitrogen forms in the soils. These ions were measured after 0, 60 and 120 days of incubation. Moisture content and pH water were also determined. Wet soils (of 10 grams) were used for extraction. Extraction was done by 2N KCl. The mixture was filtered and the filtrate was used for the analysis of mineral nitrogen (NH_4^+ - N and NO_3^- - N).

3.6 Analysis of Soil Samples

3.6.1 Total Nitrogen (Kjeldahl Method)

This was done in accordance with Kjeldahl method described by Bremner (1960). The soil samples were air- dried and then passed through a 2 mm sieve. 1 g of the sieved soil was weighed and put into a 250 ml digestion flask. To it was added 3.5 mls of phenol-sulphuric acid. 15 minutes later, 0.5 g sodium thiosulphate was added. After another 15 minutes 0.5 g selenium mixture, 0.5 g K_2SO_4 and 3.5 mls of concentrated sulphuric acid was added. Digestion started at low temperature until the mixture cleared (30 minutes). The temperature was then raised up to the end of digestion (90 mins). The mixture was allowed to cool, but before cooling completely, a small amount of distilled water was added. This was to prevent solidification of the digest. The digest was then transferred into a distillation flask and 40 mls of 10 N NaOH added. This was done quickly to avoid escape of NH_3 gas before actual distillation started. The NH_3 released during distillation was collected in 1% boric acid (20 mls) with a mixed indicator. Titration was done with 0.01 N H_2SO_4 and the indicator turned from green to pink.

Calculations: 1 ml of 0.01 N H_2SO_4 = 140 µg N.

$$\text{KgN/ha} = \frac{\text{Depth (cm)} \times \text{Bulk density (g/cm}^3) \times \text{Concentration µN/g soil} \times \text{area (cm}^2)}{\text{weigth of soil} \times 10^9}$$

3.6.2 Available Nitrogen ($NH_4^+ + NO_3^-$)-N

Ten grams of moist soil was weighed into a flask. To it was added 100 mls. of 2N KCl and shaken vigorously in a mechanical shaker for one hour. The mixture was then filtered using Whatman filter paper No. 42. This filtrate was transferred to a distillation flask and magnesium oxide added. The mixture was then steam distilled and ammonia gas released was collected in 1% boric acid. This distillate was then titrated with 0.001 N H_2SO_4 giving amount of available ammonium nitrogen (NH_4+ - N). To the same sample containing MgO, Devarda's alloy was added and distillation continued. NH_3 gas released was collected in fresh boric acid. Titration with 0.001N H_2SO_4 was done giving amount of available nitrate nitrogen (NO_3^- - N).

Calculation: 1 ml of 0.001N H_2SO_4 = 14 µgN

3.6.3 Non-Exchangeable Ammonium Nitrogen (fixed)

One gram of air-dried soil (< 2 mm) was weighed into a 400 ml beaker. To it was added 20 mls of potassium hypobromite solution and shaken. The beaker was then covered with a watch glass and allowed to stand for 2 hours (to get rid of exchangeable NH_4^+ ions and labile organic compounds). 60 mls of distilled water was added after 2 hours and the mixture boiled strongly for 5 - 6 minutes. The mixture was allowed to stand overnight and then decanted. The supernant was discarded and the residue transferred to a 250 ml polythene centrifuge tubes with 0.5N KCl from plastic watch bottles. The centrifuge tubes were then filled to the 80 ml mark and centrifuged for 20 minutes by 1000 x G rev/min. Again the supernant was discarded and KCl added to the residue up to the 80 ml mark. This was centrifuged again and the supernant discarded. The residue was then transferred completely into a 250 ml Kjeldahl digestion flask with distilled water. To it was added 7 mls. Conc. H_2SO_4 and 0.5 g K_2SO_4 and digestion started. Low temperature was employed for the first 30 minutes to damp off water. The rest was done as in total nitrogen (section 3.61).

3.7 Organic nitrogen fractionation

3.7.1 Preparation of Hydrolysate

This was done in accordance with Bremner (1965). 10 grams of air dry, finely ground soil (< 2 mm) was weighed into a 250 ml flask. 3 drops of octyl alcohol and 50 mls of 6N HCl were added to it and the mixture was swirled until it was thoroughly mixed with the soil. The flask was then placed on a hot electric plate with a heat control device and connected to a liebig condenser. Cold water was passed through the water Jacket of the condenser and the soil acid mixture heated to boil under reflux for 12 hours. This was allowed to cool and then 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 before being made to the mark in a 250 ml volumetric flask. The alkali was added slowly with constant stirring to prevent hydrolysate from becoming alkali at any stage of neutralization (first 5 N NaOH was to bring hydrolysate to pH 5 and then 0.01 NaOH to bring it to pH 6.5). Hydrolysate was stoppered and then kept in the fridge as stock for analysis of various forms of organic nitrogen.

3.7.2 Total Hydrolysable Nitrogen

This was done in accordance with Bremner (1965). Twenty five mls of neutralized hydrolysate was pipetted into a 250 ml distillation flask. To it was added 0.5 g K_2SO_4, 0.5 g Selenium mixture (catalyst) and 7 mls. conc. H_2SO_4. The mixture was continuously heated in a Kjeldahl digestion flask until frothing ceased after which water was damped off. This was done at low temperature (30 mins). The heat was increased until the mixture cleared and digestion was completed by gently boiling for another one hour. The flask was allowed to cool and 10 mls of distilled water was added slowly while shaking. The flask was again cooled under water in a beaker containing crushed ice. This digest was transferred into a 250 ml distillation flask and 40 mls of 10 N NaOH added. This was distilled

and the ammonia gas released was collected in 1% boric acid. Titration was done using 0.01N H_2SO_4.

Calculation: 0.01N H_2SO_4 = 140 µgN

$$\text{Therefore \% N} = \frac{(\text{Titre - Blank}) \times 140}{10{,}000}$$

3.7.3 Amide-N

Twenty five mls of hydrolysate was pipetted into a 250 ml distillation flask. 0.07 g of magnesium oxide was added and the mixture distilled as in section 3.72. Again the distillate was collected in 1% boric acid and titration was done using 0.001N H_2SO_4.

$$\text{Calc. 0.001N } H_2SO_4 = 14\mu gN\text{: \%N} = \frac{(\text{Titre - Blank}) \times 14}{10{,}000}$$

3.7.4 (Amide + Hexosamine) - N

25 mls of hydrolysate was pipetted into a 250 ml distillation flask. 10 mls of phosphate - borate buffer (pb), pH 11.2 was added. The mixture was steam distilled and NH_3 gas released was collected in 1% boric acid. Titration was done with 0.001 N H_2SO_4. The amount of hexosamine was obtained by subtraction method, i.e. from the results in section 3.73.

Calculation as section 3.73. % Hexosamine - N = % (amide + Hexosamine) N - (% Amide - N).

3.7.5 (Serine and Threonine)-N

After the removal of ammonium + Hexosamine nitrogen by steam distillation with PB - buffer (section 3.74), the flask was detached from the distillation apparatus. The steam inlet was then rinsed with 5 mls. of distilled water and the rinse collected in the distillation flask. The flask containing the rinse and the mixture was cooled thoroughly under a cold water tap 2 mls of periodic acid

solution was added and the flask swirled for 30 seconds. Then 2 mls of sodium meta-Arsenite solution was added to reduce excess periodic acid. Distillation and calculation continued as above (section 3.74).

3.7.6 (α-Amino acid) - N

Twenty five mls of neutralized hydrolysate was pipetted into a 250 mls distillation flask. To it was added 1 ml of 0.5 N NaOH and the flask was heated in boiling water for approximately 20 minutes. The flask was then allowed to cool and 0.5 g citric acid; 0.1 g Ninhydrin powder were added. The flask was again immersed in a boiling water bath for about one minute and then swirled for a few seconds without removing it from the bath. It was allowed to remain in the bath for another 9 minutes. The flask was cooled under tap water and 10 mls of PB - buffer; 1 ml of 5 N NaOH added. Distillation was done as above section (3.73, 3.74).

Calc. 0.001N H_2SO_4 ≡ 14 µg N: %N = $\frac{\text{(Titre-Blank) x 14}}{10{,}000}$

3.7.7 (Ammonium + hexosamine + amino acid) - N

Twenty five mls of neutralized hydrolysate filtrate was pipetted into a 250 ml distillation flask. Citrate buffer (pH 2.5) was added and the mixture heated vigorously by immersing the flask in a boiling water bath for 1 minute. The mixture was then swirled for a few seconds without removing it from the bath. It was left in the bath for another 9 minutes, cooled and 10 mls. of PB - Buffer added. Thus was then connected to the distillation apparatus and steam distillation done as above (3.73, 3.74 and 3.75). Liberated ammonia gas was collected in 1% boric acid and titrated with 0.01 N H_2SO_4. Mixed indicator was used and the end point was a pinkish colour. Calculations as above (section 3.73).

3.8 Bulk Density

Core rings were hammered into the soil and carefully removed so as not to disturb the soil samples. These were then taken to the laboratory for bulk density determination. The soil samples were dried in the oven (105°C) for 24 hours. The soil was then weighed and bulk density calculated using the volume of the core ring.

$$\text{Calculation: Bulk density } (g/cm^3) = \frac{\text{Weight of oven dry soil } (g)}{\text{Volume of core ring } (cm^3)}$$

3.9 Analysis of other Chemical and Physical Properties of the soils

3.9.1 Soil pH determination

Soil pH was determined following the procedure outlined by Black (1965). 10g of air dry soil (passed 0.5 mm sieve) was weighed into a plastic specimen bottle and 25 ml of distilled water was added. The bottle was then tightly stoppered and shaken for 30 minutes in a mechanical reciprocating shaker. The suspension was allowed to stand for 10 minutes and then the soil pH was measured by inserting electrode of a pH meter into the partly settled suspension. The pH was then reported as "soil pH measured in water". Similarly soil pH was measured in N KCl,using the ratios 1: 2.5 soil to KCl ratio.(v/v)

3.9.2 Mechanical analysis

Particle size distribution was determined using the hydrometer method as described by Black (1965). 51 g of air dry soil which had been ground to pass 2 mm sieve was transferred to a milk shake cup. This soil was free of organic matter, having been previously destroyed by adding small portions of 30 % hydrogen peroxide solution. 5 % sodium hexametaphosphate was added to raise the electrokinetic potential of the soil and the soil was then dispersed by mechanical shaking. The suspension was transferred to a 1000 ml glass cylinder and with the

hydrometer in the suspension, water was added to the lower blue line. The hydrometer was then removed and calibrated in distilled water.

After shaking several times the hydrometer was inserted and the first hydrometer reading taken 40 seconds later. The second hydrometer reading was taken 3 hours later. Before each reading, temperature of the soil suspension was recorded.

3.9.3 Organic carbon

Organic carbon in the soils was determined using the Walkley-Black method using the procedure outlined by Black (1965). A known amount of the soil was put in a 250 ml Erlen meyer and 1 N potassium dicromate solution added, swirling the flask gently. Then rapidly 20 ml of concentrated sulphuric acid was added, directing the stream into the suspension and the flask was allowed to stand for 30 minutes. Then 3 to 4 drops of O-phenophthaline complex indicator was added and the solution titrated with 0.5 N ferrous sulphate. A blank titration was done in the same manner but without soil to standardize the dicromate.

3.9.4 Cation exchange capacity and the exchangeable bases

Cation exchange capacity and exchangeable bases were determined using procedures outlined by Black (1965). First the soil was leached with 100 ml N ammonium acetate pH 7.0 to replace all naturally occurring exchangeable cations with ammonium ions. The leachate obtained was used for the analysis of exchangeable bases (Na, Mg, K and Ca). Secondly the soils were washed with alcohol to remove excess ammonium acetate and was then leached with 100 ml N KCl. The leachate obtained was titrated with 0.1 N HCl and this gave the cation exchange capacity of the soils. Ca and Mg were determined by titration with 0.05 M EDTA, while K and Na were determined by flame photometer. Exchangeable aluminium and hydrogen were determined by leaching the soil with 100 ml N KCl. Effective cation exchange capacity was then obtained as the sum of the

exchangeable bases, thus: effective cation exchange capacity = Na + K + Mg + Ca + Al + H.

3.9.5 Available phosphorus in the soil

Available phosphorus in the soil was determined according to the double acid method of Mehlich (1962). This procedure involves extraction of phosphorus by a double acid of 0.5 N HCl in 0.25 N sulphuric acid. A 5 g sample of air - dry soil ground to pass 0.5 mm sieve was extracted with 50 ml of the double acid on a shaking machine for 30 minutes. Colorimetric determination was done by Ascorbic Acid method and the colour measured in a spectrophotometer.

3.1.0 Moisture content

This was done after every soil sampling of the incubated soils. The soils were weighed and then oven dried (at 105^{o}C) for 24 hours. This soil was then weighed. The difference in grams between the moist and dry soils gave the moisture content of the soils. This moisture content was then used to convert the moist soils used to dry soils for the sake of reporting the analysed ammonium N and Nitrate N on dry soil basis.

3.1.1.1 Statistical Analysis

The design of the experiment was Randomised Complete Block Design. Analysis of variance (ANOVA) was done according to Thomas M. Little and Jackson Hills (1975). The means were then separated using least significant difference (LSD) at 95% confidence limit.

CHAPTER FOUR

4.0 RESULTS AND DISCUSSION

4.1 Soil characteristics

The behaviour of nitrogen in soil is controlled by the physical and chemical properties of the soil. This being the case, it was therefore necessary to know some of the soil properties of the three soils which were used.

Tables (1-3) show the physical and chemical characteristics of the three soils, i.e, Gituamba Andosols, Kitale Ferralsols and Katumani Luvisols. The pH varied from one soil to another. It ranged from 4.0 in Gituamba (0-15 cm) to 7.0 in Katumani (15-30 cm) soils. The pH was 4.0 and 4.1 for Gituamba (Table 1), 5.6 and 5.6 for Kitale (Table 2), 6.6 and 7.0 for Katumani (Table 3) for the 0.15 cm and 15-30 cm depths respectively. Low pH has a marked influence on exchangeable aluminium. This is clearly seen in Table 1 where Gituamba soils with lowest pH value tends to have the highest aluminium content of 4.6 me/100g in 0-15 cm and 3.3 me/100 g soil in the 15-30 cm depths. The analysed aluminium was found to be highly negatively correlated to pH ($r = -0.88$, $P \leq 0.05$) (Appendix X) that is, the higher the aluminium content as in the case of Gituamba soil (Table 1), the lower the soil pH especially in the 0-15 cm depth. pH was found to be significant and positively correlated to percent base saturation ($r = 0.86$, $P \leq 0.05$) (Appendix X).

The soils (Tables 1-3) also gave different but expected pattern of organic carbon and total nitrogen distribution in the soil profiles. The organic carbon decreased down the soil profiles. This phenomenon is undoubtedly due to the addition of organic matter mainly at the top. Gituamba soils had the highest of both percent organic carbon (7.9%) and total nitrogen (0.6%). Soil nitrogen percent also decreased with depth. This could be due to the decline in organic matter down the soil profiles. This is because nitrogen is an integral part of organic carbon.

There was a difference in C:N ratios between the two depths in all the three soils. The 15-30 cm depth had a lower C:N ratio than the 0-15 cm depth in all the

three soils. The low C:N ratios observed ranged from 5.3 in Katumani (15-30 cm) to 22.5:1 in Kitale (0-15 cm) soils. All these C:N ratios are within the range which favour net nitrogen mineralization (Janssen, 1929).

Exchangeable potassium was very low in Kitale soils followed by Katumani. In Gituamba soils, there was high exchangeable potassium of more than 3 me/100 g soil. These soils are derived from volcanic ash hence the reason for the high potassium content observed. Keter (1974) suggested that East African rocks are often rich in this element especially when derived from young volcanic rocks.

Generally Ca, Mg, K and Na are higher in top soil than sub soil. The increase down the soil profile in Katumani soils could partly be due to leaching down from above, but may also simply reflect supply of cations from parent rock. CEC is another measure of soil fertility. In all these soils, CEC was higher in the 0-15 cm depth than 15-30 cm with the exception of Katumani soil.

The clay content was found to be highest in Kitale soils (52.9% and 55.0% for 0-15 and 15-30 cm) and lowest in Katumani soils (23.9% and 22.4% for 0-15 cm and 15-30 cm respectively). Katumani soils had the highest percent sand (68.6% and 74.3%) while Gituamba soils had the highest amount of silt (39.8% and 33.9%) for the 0.15 cm and 15.30 cm depths, respectively. The texture of the three soils varied greatly and could have been influenced by such factors as the vegetation of the location as well as the parent material from which these soils were derived. Gituamba soils are loamy, Kitale clayey and Katumani soils are sandy clay loam.

Table 1 Some chemical and physical characteristics of Gituamba Andosols (i.e. Andosols)

Soil properties	Depth (cm)	
	0-15	15-30
pH water(1:2.5)	4.0	4.1
" In Kcl(1:2.5)	3.9	4.0
CEC (me/100 g soil	28.6	26.7
ECEC "	11.6	10.2
Ca "	0.7	0.3
Mg "	0.5	0.1
Na "	0.5	0.4
K "	4.3*	3.3*
% Base saturation	21.0	19.5
Exch. Al^{3+}	4.6	3.3
" $H+$	1.0	0.7
Available P (ppm)	12.5	10.0
% Total N	0.6	0.5
% Organic Carbon	7.9	4.8
C:N Ratio	12.8	9.2
Bulk density g/cm^3	0.6	0.8
% Sand	40.3	38.3
% Clay	19.9	27.9
% Silt	39.8	33.8
Textural class	Loam	Loam

*The figure obtained looks excessive for the soil and most probably potassium fertilizers had been applied. There was no reliable information of the previous history of the site.

Table 2: Some chemical and physical characteristics of Kitale soils (i.e. ferralsols)

Soil properties	0-15 cm	15-30 cm
pH water(1:2.5)	5.6	5.6
" In Kcl(1:2.5)	4.4	4.5
CEC (me/100g soil)	15.3	13.4
ECEC (me/100g soil	11.4	9.1
Exch. Ca	4.7	2.9
" Mg	2.4	2.0
" Na	1.0	0.5
" K	1.5	1.2
% Base saturation	62.7	55.2
Exch.Al(me/100g soil)	1.0	0.8
" H^+	0.9	0.9
Available P (ppm)	2.5	1.5
% Total-N	0.2	0.1
% Organic Carbon	4.5	1.8
C:N Ratio	22.5	13.9
Bulk density (g/cm^3)	1.2	1.1
% Sand	41.9	37.3
% Clay	52.9	55.0
% Silt	5.2	7.7
Textural class	Clay	Clay

Table 3: Some chemical and physical characteristics of Katumani soils (i.e. luvisols)

Soil properties	Depth (cm) 0-15	15-30
pH water (1:2.5)	6.6	7.0
" In Kcl (1:2.5)	4.8	5.6
CEC (me/100g soil)	13.4	12.1
ECEC " "	9.5	10.7
Ca " "	5.7	6.3
Mg " "	1.3	1.9
Na " "	0.6	0.4
K " "	1.5	0.9
% Base saturation	61.4	78.5
Exch. Al^{3+} (me/100g soil)	1.1	0.9
H^+ " "	0.4	0.3
Available P (ppm)	46.0	29.0
% Total-N	0.2	0.1
% Organic Carbon	1.0	0.5
C:N Ratio	5.7	5.3
Bulk density (g/cm^3)	1.4	1.3
% Sand	68.6	74.2
% Clay	23.9	22.4
% Silt	7.5	3.4
Textural class	Sandy clay loam	Sand clay loam

The bulk density was highest in Katumani soils (1.4 and 1.3 g/cm^3) and lowest in Kituamba soils (0.6 and 0.8 g/cm^3) for the 0-15 cm and 15-30 cm depths respectively. This seems to reflect on the soil texture. Soils low in clay content and high in sand content like Katumani have a higher bulk density and vice versa. However the influence of organic matter content on bulk density in these soils cannot be ruled out.

4.2 Nitrogen forms in the soils

Table 4 shows various forms of nitrogen in three soils in kg N/ha. The total nitrogen ranged from 1038.5 kg N/ha in Kitale (15-30 cm) to 6558.6 kg N/ha in

Gituamba (0-15 cm) soils. In all the three soils, the total nitrogen decreased with depth, i.e. from 6,558.6 Kg/ha in the 0-15 cm to 5832.7 Kg/ha in the 15-30 cm depths for Gituamba soils. For Kitale, it was from 1627.0 kg N/ha in 0-15 cm to 1038.5 Kg N/ha in the 15-30 cm depths.The total N for Katumani soils ranged from 3582.2 Kg N/ha in the 0-15 cm to 1663.2 kg in the 15-30 cm depths. Gituamba soils had the highest amount of total nitrogen in the 0-30 cm layer of 12,391.3 Kg while Kitale had the lowest (2665.5 Kg N/ha).

The percentages of total nitrogen in all the three soils appear in the soil property Tables 1-3. This ranged from 0.6% in Gituamba (0-15 cm) to 0.1% in Katumani (15-30 cm) soils. This range has been reported in most cultivated soils (Bremner, 1960). The available nitrogen ($NH_4^+ + NO_3^-$) was also higher in the 0-15 cm than 15-30 cm depth, in all the three soils. This same trend was observed in exchangeable ammonium nitrogen and nitrate nitrogen. Gituamba had the highest amount of available nitrogen (319.6 Kg N/ha) in the 0-30 cm layer followed by Kitale (129.3 Kg N/ha) and then Katumani with 28.6 Kg N/ha.

The exchangeable ammonium nitrogen (Appendix I) was very low and ranged from 0.2% in Katumani (0-15 cm) to 3.2% in Kitale (15-30 cm) of the total nitrogen in each depth. Nitrates ranged from 0.2% in Katumani (0-15 cm) to 1.6% in Kitale (0-15 cm) soils. The nitrates were lower than exchangeable ammonium nitrogen in all the three soils (Appendix I).

Organic nitrogen showed the same trend as total nitrogen (Table 4). Gituamba had the highest amount of 7241.4 Kg N/ha followed by Katumani with 2244.4 Kg N/ha and then Kitale with 1444.5 Kg N/ha in the 0-30 cm depths. Organic nitrogen formed the highest portion of the various forms of nitrogen (Appendix I). This ranged from 75.3% to 95.3% of the total nitrogen. Gituamba had 75.4% and 75.3%, Kitale 92.3% and 91.8% and then Katumani with 95.3% and 91.7% in the 0-15 cm and 15-30 cm depths, respectively.

Table 4: Nitrogen distribution in the three soils (Kg N/ha)

Site	Depth (cm)	Total Nitrogen	organic nitrogen Hydrolyzable	Available nitrogen NH_4^+ + NO_3^-	Fixed nitrogen NH_4^+-f
Gituamba	0-15	6558.6	3742.2	167.1	1305.2
	15-30	5832.7	3499.2	152.5	1403.9
	Total	12391.3	7241.4	319.6	2709.1
	%		58.4	2.6	21.9
Kitale	0-15	1627.0	826.5	72.5	54.0
	15-30	1038.5	618.0	56.8	40.4
	Total	2665.5	1444.5	129.3	94.4
	%		54.2	4.9	3.5
Katumani	0-15	3582.2	1476.3	15.1	152.5
	15-30	1663.2	768.0	13.5	116.9
	Total	5245.4	2244.3	28.6	269.4
	%		42.8	0.5	5.1

Fixed nitrogen ranged from 40.4 kg N/ha in Kitale (15-30 cm) to 1403.9 Kg N/ha in Gituamba (15-30 cm) and was relatively higher in the 0- 15 cm depth compared to 15-30 cm depth. This fixed nitrogen in percent was 4.3% and 7.5% for Katumani, 22.1% and 22.05% for Gituamba and then 3.3% and 3.8% for Kitale soils in the 0 15 cm and 15-30 cm depths respectively (Appendix I). The the reason for low levels of fixed nitrogen in Kitale soils is not very clear but could be due to the inherent clay minerals.

The high amounts of all forms of nitrogen in Gituamba soils seem to reflect on the vegetative cover present in this area. Much of the litter fall is decomposed by microorganisms as it falls giving rise to organic matter. This is clearly shown in Table 1, where Gituamba had the highest per cent organic carbon and in Table 4

where all nitrogen forms are highest in Gituamba soils. The decline in organic matter down the soil profiles had the same effect on all nitrogen forms. As organic matter accumulates on the top, more nitrogen is released but as it declines down the soil profiles, all other forms of nitrogen decrease.

In Katumani soil where vegetation is scarce, there was a marked reduction in organic matter which also gave rise to low nitrogen forms. Kitale soils were sampled from an area under grass and this could have been the reason for the higher accumulation of organic matter on the surface. On the other hand, Katumani soils were sampled from an area which was bare and which had been under cultivation for many years.

It can therefore be concluded that the high accumulation of nitrogen forms in Gituamba was as a result of high vegetation cover and probably due to low pH values which limit the work of microorganisms in decomposing organic matter quickly. Organic nitrogen accumulates in acid soils as a result of slow mineralization but mineralization becomes rapid when the soils are limed (Harmsen and Van Schreven, 1955).

4.3 Organic forms of nitrogen in the soils

Table 5 shows the organic nitrogen fractions of Gituamba, Kitale and Katumani soils. The total hydrolysable nitrogen ranged between 39.0 - 43.9% and 41.7-42.8% for Katumani, 54.6-59.4% and 61.3-63.0% for Kitale and then 55.8-58.4% and 57.4-60.9% for Gituamba in the 0-15cm and 15-30cm depths respectively.Amide N obtained was 16.1-18.7% and 16.7-18.8% for Gituamba; 18.1-20.8% and 20.3-22.5% for Kitale and 10.9-12.6% and 17.0-18.1% for Katumani soils of total nitrogen in the 0-15 cm and 15-30 cm depths respectively.

Hexosamines were 7.5-8.6% and 8.0-8.2% for Gituamba; 5.6-6.5% and 9.9 - 10.6% for Kitale and then 4.9-5.4% and 8.8-10.4% for Katumani soils. Following the same trend, amino acid nitrogen as percent of total nitrogen was 34.8

- 35.4% and 32.7-33.8% for Gituamba, 37.0-37.6% and 35.2-38.9% for Kitale and 23.1-29.2% for Katumani in the 0-15cm and 15-30cm depths respectively.

Also from Table 5 the 0-15 cm depth had a slightly lower total hydrolysable nitrogen than 15-30 cm depth in all the three soils. The same trend was observed for amide nitrogen and Hexosamine nitrogen. Amino acid nitrogen was higher in the upper than in the lower depth.

Table 5: Organic forms of nitrogen as percentage of total nitrogen (i.e. the Range and mean)

SITE	GITUAMBA		KITALE		KATUMANI	
DEPTH	0-15cm	15-30cm	0-15cm	15-30cm	0-15cm	15- 30cm
ORGANIC FORMS OF -N						
Hydrolysable Total N	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	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	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	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

The total hydrolysable nitrogen together with other nitrogen fractions in the soils could have been influenced by the amount and extent of decomposition and humification of organic matter 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 nitrogen fall within the range already obtained by Shigeyoshi and Co-workers (1986). These workers observed a total hydrolysable nitrogen of between 40%and 80%. Amount of amide-nitrogen (Table

5) agrees with those obtained by Bremner (1949a); Keeney and Bremner (1964) who observed values ranging from 15% to 25% of total nitrogen.

Values obtained for hexosamine N were in the 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% to 5%.

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

The types of organic matter added in these soils are mostly of diverse nature. Also the soils have been under different agricultural land use and this tends to influence nitrogen turnover differently. Some forms such as amino acids could be hydrolysed to available forms of nitrogen (NH_4^+ and NO_3^- ions) for plant and microorganism uptake.

4.4 Nitrogen mineralization experiments

4.4.1 Effect of lime ($CaCO_3$) N and P salts on nitrogen mineralization in Gituamba soils

Figure 1 show the effect of various treatments on nitrogen mineralization. Where soils were limed with calcium carbonate, the nitrogen mineralized increased from 75.9 µgN to 169.0 µgN/g soil from 0 day to 60 days reaching the peak of 170.2 µgN/g soil at 120th day of incubation in the 0-15 cm depth. For the 15-30 cm depth, the increase was from 69.3 µgN to 150.2 µgN/g soil from 0 day to 60 days and then declined to 147.7 µN/g soil at 120th day of incubation. The same trend was seen in the control treatment in both depths but with lower amounts than in limed soils. In the control, the increase was from 75.9 µgN to 130.2 µgN/g soil

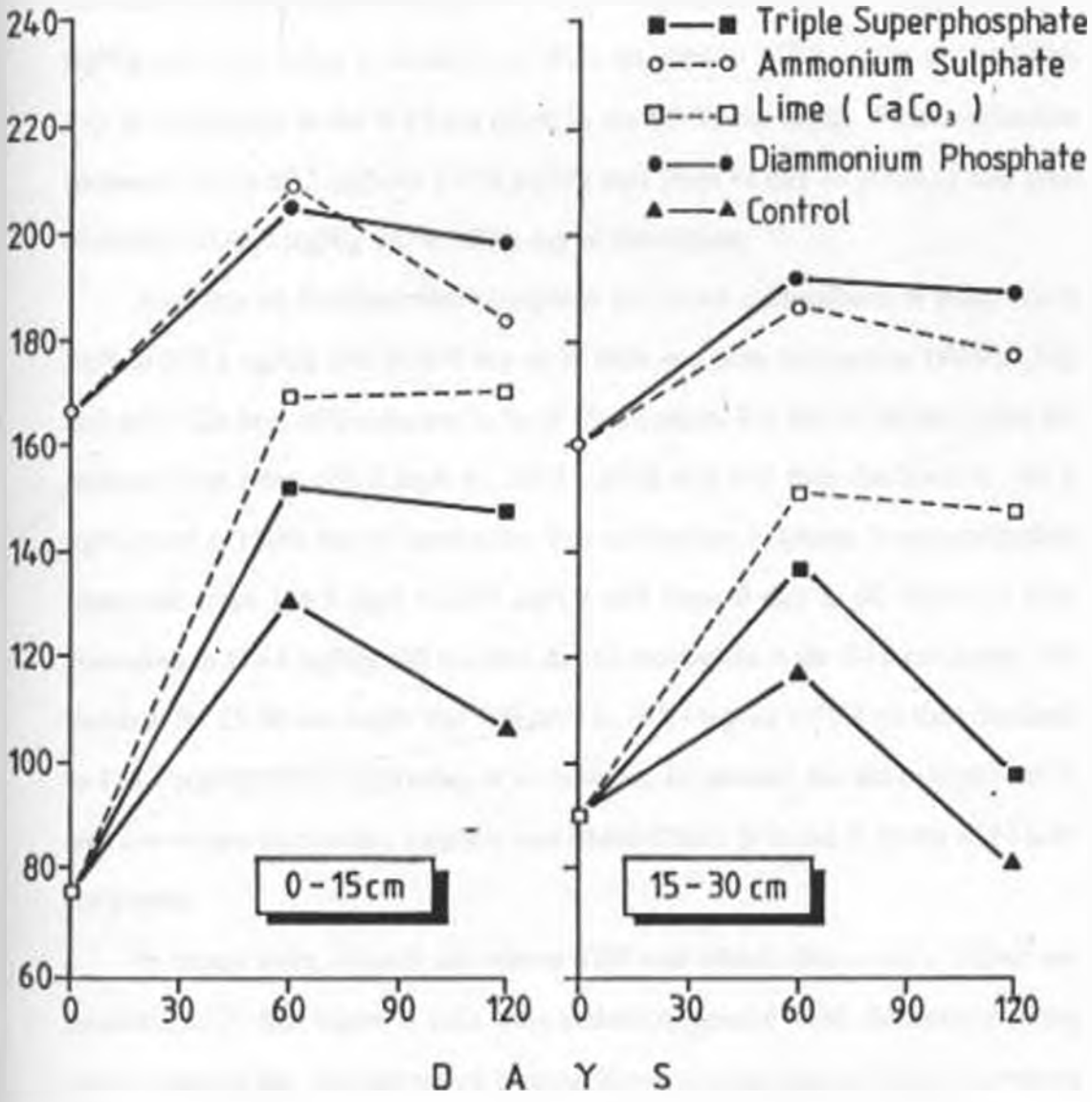

Fig.1. : Effect of Lime, N and P salts on nitrogen mineralization in Gituamba andosols during incubation.

from 0 day to 60 days and then declined to 106.2 µgN/g soil in the 0-15 cm depth. For the 15-30 cm depth, the increase was from 69.3 µgN to 116.7 µgN/g soil from 0 day to 60 days and then declined to 81.7 µgN/g soil at 120th day of incubation.

All other treatments showed the same trend. Addition of phosphorus salt increased N-mineralization (Table 6.0 and Figure 1) from 75.9 µgN to 152.5 µgN/g soil from 0 day to 60 days and then declined to 147.9 µgN/g soil at 120th day of incubation in the 0-15 cm depth.In the 15-30 cm depth, N mineralization increased from 69.3 µgN/to 137.9 µgN/g soil from O day to 60 days and then decreased to 98.2 µgN/g soil at 120th day of incubation.

Addition of diammonium phosphate increased mineralized N from 166.8 µgN to 205.1 µgN/g soil from 0 day to 60 days and then declined to 199.9 µgN/g soil atfer 120 days of incubation in the 0-15 cm depth. For the 15-30 cm depth, the increase was from 160.2 µgN to 193.5 µgN/g soil and then declined to 191.2 µgN/g soil at 120th day of incubation. For ammonium sulphate, N mineralization increased from 166.8 µgN to 209 µgN/g soil from 0 day to 60 days and then decreased to 184.6 µgN/g soil at 120th day of incubation in the 0-15 cm depth. The increase for 15-30 cm depth was 160 µgN to 185.1 µgN/g soil. This then declined to 178.2 µgN/g soil at 120th day of incubation. In general, the net mineralized N was low where ammonium sulphate was added (Table 6) in the 0-15 cm and 15-30 cm depths.

In limed soils, control and where TSP was added, there was a higher net mineralized N than where N salts were added (Appendix Viic). Seperation of the means showed that lime increased N mineralization more than all other treatments and was significant ($P \leq 0.05$) (Appendix Viic). Triple superphosphate increased N mineralization above the control and N salt treatments and was significant ($P \leq 0.05$) after 60 days of incubation. Among the two N salts, DAP increased net N mineralization more than AS, but not significantly ($P \geq 0.05$) (Appendix Viic).

Table 6.0: Nitrogen mineralization under different treatments during incubation for Gituamba Andosols (µgN/g soil)

Treatment	Depth (cm)	Initial (0 days)	After 60 days	After 120 days	µgN/g soil Difference	µgN/g/week 120-0 days
Control	0-15	75.98	130.2	106.2	30.3	1.75
	15-30	69.3	116.7	81.7	12.4	0.70
LIME	0-15	75.9	169.0	170.2	94.3	5.53
	15-30	69.3	150.2	147.7	78.4	4.45
TSP	0-15	75.9	152.5	147.9	72.0	4.20
	15-30	69.3	137.9	98.2	28.9	1.68
DAP	0-15	166.8	205.1	199.9	33.1	1.96
	15-30	160.2	192.5	191.2	31.0	1.82
AS	0-15	166.8	209.4	184.6	17.8	1.05
	15-30	160.2	185.1	178.2	18.0	1.0

In all treatments, production of mineral nitrogen decreased with depth (Table 6.0). The nitrogen mineralization rates also showed the same trend. That is, they were higher in the 0-15 cm than 15-30 cm depth. The highest nitrogen mineralization rates were observed in the limed soils. These were 5.53 and 4.45 µgN/g/week for the 0-15 cm and 15-30 cm depths respectively. The lowest values were observed in soils treated with ammonium sulphate (1.05 µgN/g/ week in both depths). Addition of TSP also produced a high amount of N-mineralization and high mineralization rates of 4.20 µgN/g/week in the 0-15 cm depth. DAP had mineralization rates of 1.96 and 1.82 µgN/g/ week while control had 1.75 and 0.7 µgN/g/week in the 0-15 cm and 15-30 cm depths respectively (Table 6).

Liming of Gituamba soils increased N mineralization significantly ($P \leq 0.05$). Soil pH also increased with the liming of these soils from 4.0 to 5.7 in the 0-15 cm

and from 4.1 to 5.8 in the 15-30 cm depths (Table 11). Where TSP was added and also in the control, there was only a very slight variation in pH from 4.0 to 4.2 in both depths. Where nitrogen salts were added, there was a decline in pH from 4.0 to 3.9 from 0 day to 60 days and then increased to approximately 4.0. The increase in soil pH where lime was added created a favourable environment for microbial activities. The microbes present therefore acted on the accumulated organic nitrogen in this soil hence releasing mineral nitrogen (Sarathchandra and Upsdell, 1981; Smillie and Curtin, 1983). Increase in soil pH could also have increased nutrient availability for the microorganisms hence contributing to the high mineralization rates observed.

Addition of N salts resulted in high production of mineral nitrogen although net mineralized N was very low. The apparent high production of mineral nitrogen was as a result of the added nitrogen fertilizer. This was acted upon by the microorganisms hence releasing mineral nitrogen (NH_4^+ and NO_3^-). Also with addition of fertilizers, there was a drop in soil pH (Table 11.0). However, this drop in soil pH did not hinder the production of mineral N. The low net mineralization of organic N in soils treated with N fertilizers is due to the added N which led to a decrease in soil pH (Table 11). The decline in N mineralization at the end of incubation for virtually all treatments and soil depths could probably be due to reduction of mineralizable N substrates towards the end of the experiment. This could also be due to immobilization of nitrogen by microorganisms whose population could have greatly increased during this long incubation period (Budimir, 1980). The immobilization phenomena might also represent a shift in microbial populations or species that could result from an extended incubation period at constant temperature.

Among the two nitrogen salts, DAP promoted more mineral nitrogen production than ammonium sulphate (Appendix VIIc). This increase in net N mineralization was not significant ($P \geq 0.05$). The presence of phosphorus in the diammonium phosphate could have led to this difference. The presence of P could

have boosted microbial growth (Munevar and Wallum, 1977; Stotzky and Norman, 1961) especially in acid soils where native P is likely to be fixed. This also applies to liming of soils which reduces the chances of P fixation as pH increases. Phosphorus deficiency in some soils limits microbial population which in turn depresses N mineralization.

The decline in mineralized N with depth could be attributed to low organic matter content down the soil profile or a decline in microbial population. It shows therefore that addition of N salts did not have any stimulating effect on organic N mineralization in the soil while addition of P salt had stimulating effect on organic N mineralization. This led to relatively high mineral N production in Gituamba soils (Table 6).

4.4.2 Effect of lime ($CaCO_3$) N and P salts on nitrogen mineralization in Kitale soils

Figure 2 shows the effect of various treatments on N mineralization in Kitale soils. Lime addition showed an increase in nitrogen mineralized above the control. This increase was from 32.9 µgN to 55.4 µgN/g soil from 0 day to 60 days and then decreased to 45.5 µgN/g soil after 120 days of incubation in the 0-15 cm depth. The same trend is seen in the 15-30 cm depth but with lower amounts. In the control, N-mineralization increased from 32.9 µgN to 50.7 µgN/soil from 0 day to 60 days and then decreased to 43.3 µgN/g soil at the end of the experiment in the 0-15 cm depth. The 15-30 cm depth showed the same trend but with lower amounts of mineralized nitrogen.

Triple superphosphate showed the same trend as lime treatment in both depths but the amounts of mineral nitrogen was higher than in limed soils. Here the mineral nitrogen increased from 32.9 µgN/g to 57.1 µgN/g from 0 day to 60 days and then declined to 51.2 µgN/g soil in the 0-15 cm depth at the end of the experiment (120 days). In the 15-30 cm depth, the increase was from 25.8 µgN to 52.6 µgN/g soil and then decreased to 44.6 µgN/g soil at the end of the experiment.

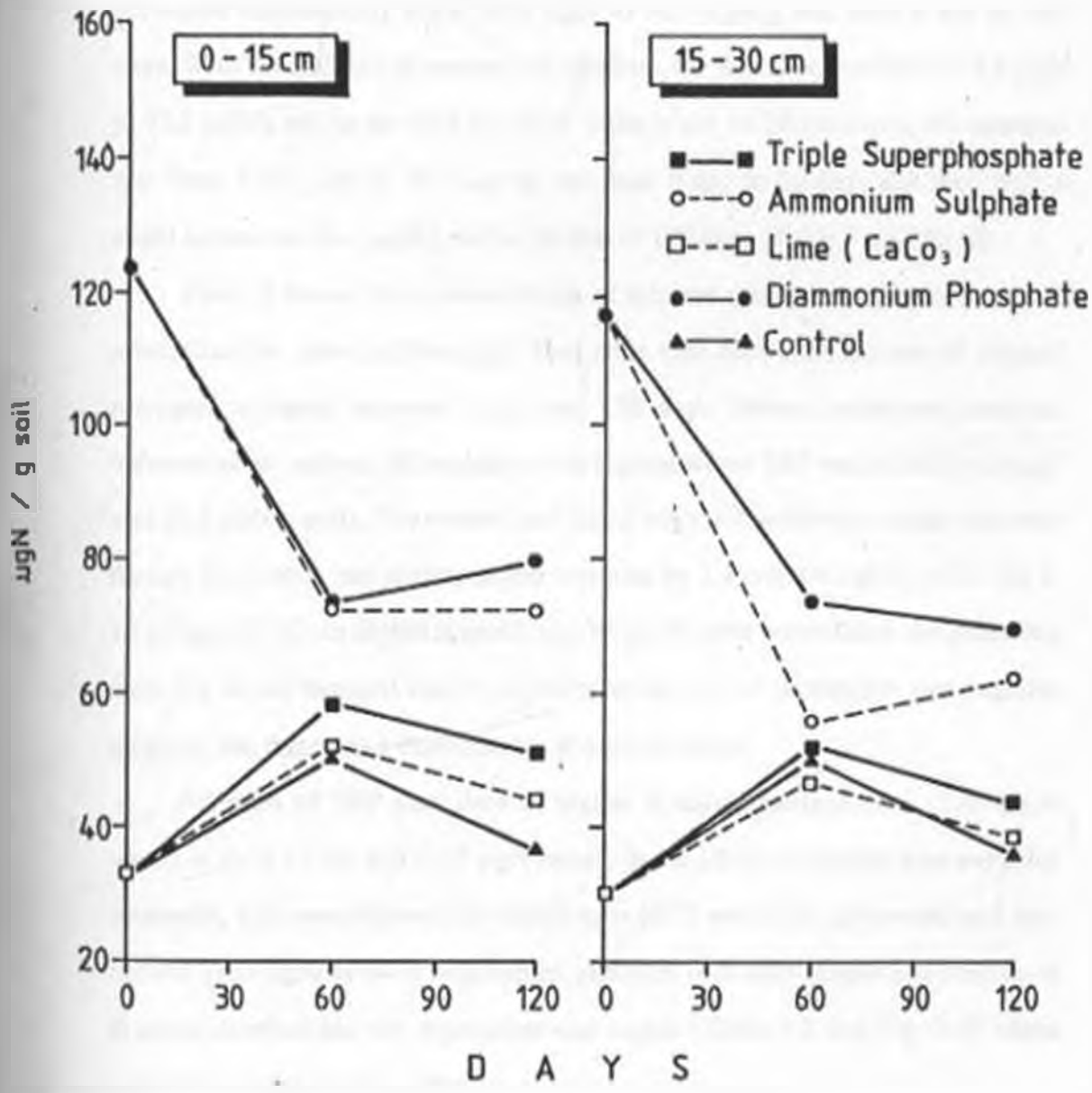

Fig. 2.: Effect of lime, N and P salts on nitrogen mineralization in Kitale ferralsols during incubation.

Addition of nitrogen salts depressed production of inorganic nitrogen. Where Diammonium phosphate was added, the decrease was from 123.9 µgN to 74.3 µgN/g soil from 0 day to 60 days and then increased slightly to 80.1 µgN/g soil after 120 days in the 0-15 cm depth. In the 15-30 cm depth, the N-mineralization decreased continuously from 116.7 µgN to 70.7 µgN/g soil from 0 day to 120 days. With the addition of ammonium sulphate, the decrease was from 123.9 µgN to 73.2 µgN/g soil in the 0-15 cm depth while in the 15-30 cm depth, the decrease was from 116.7 µgN to 56.3 µgN/g soil from 0 day to 60 days and then with a slight increase to 62.1 µgN/g soil at the end of 120 days (Table 7 and Fig. 2).

Table 7 shows the concentrations of mineral nitrogen (µgN/gsoil), and N mineralization rates (µgN/ha/day). This table also shows the amount of mineral nitrogen produced between 0 day and 120 days. Mineral nitrogen produced between initial and end of incubation was highest where TSP was added (18.3 µgN and 18.8 µgN/g soil). The control and limed soils had relatively similar amounts though limed soils had slightly higher amounts by 2.4 and 0.4 µgN/g soil in the 0-15 cm and 15-30 cm depths respectively. Where N-salts were added, the difference from the initial nitrogen and N produced at the end of incubation was negative meaning that there was a depression in N mineralization.

Addition of TSP also showed higher N mineralization rates (1.05 µgN/ week) in the 0-15 cm and (1.12 µgN /week) in the 15-30 cm depths than any other treatment. This was followed by limed soils (0.77 and 0.70 µgN/week) and then control (0.63 µgN/week) in both depths. Addition of N salts caused a depression in N mineralization and the depression was higher (Table 7.0 and Fig. 2.0) where ammonium sulphate was added.

Lime addition showed increase of N mineralization in Kitale soils when compared with the control but not significant. Through this liming, pH in soil was raised from 5.6 to 7.7 and from 5.6 to 7.8 (Table 12) in the 0-15 cm and 15-30 cm depths respectively. Addition of TSP affected soil pH slightly but not significantly (Table 12) while addition of N salts lowered soil pH. With DAP, the pH

decreased from 5.6 to 5.0 and from 5.6 to 4.9 whereas with ammonium sulphate, the pH decreased from 5.6 to 4.8 and from 5.6 to 4.9 in the 0-15 cm and 15-30 cm depths respectively.

Table 7: N-mineralization under different treatments during incubation for Kitale soils (µgN/g soil)

Treatment	Depth (cm)	Initial O days	After 60 days	After 120 days	Difference 120 days	N-min rates µgN/week
Control	0-15	32.9	50.7	43.3	10.4	0.63
	15-30	25.8	50.3	37.1	11.3	0.63
Lime	0-15	32.9	55.5	45.5	12.6	0.77
	15-30	25.8	47.3	37.5	11.7	0.70
TSp	0-15	32.9	57.7	51.2	18.3	1.05
	15-30	25.8	52.6	44.6	18.8	1.12
DAP	0-15	123.9	74.3	80.1	- 43.8	-
	15.30	116.7	72.8	0.7	- 46.0	-
AS	0-15	123.9	73.5	73.2	- 60.3	-
	15.30	116.7	56.3	62.1	- 54.6	-

The non-significant effect on N mineralization after liming of Kitale soils simply qualifies the fact that it is not necessary to lime these soils (Anon, 1976). This is because NH_4^+ ions in this soil are very elusive, hence difficult to trace due to inherent micaceous clay minerals. The mineralizable organic nitrogen (Table 4) was also low. So addition of lime could not make any impression here. The low organic N content could have played a major part in the low mineralization N values obtained. This is so because the substrate comprising the nitrogen to be mineralized normally influences the amount of nitrogen that is to be mineralized (Stanford and Smith, 1972). It could also be due to low microbial activity.

Addition of N salts depressed the amount of mineral N released from the original mineralizable organic nitrogen. The initial depression could have been caused by the acidifying effect (Table 12) of N salts. This could have greatly affected the microorganisms responsible for mineralization process. The increase in soil pH towards the end of incubation could account for the slight increase in nitrogen mineralization for ammonium sulphate (15-30) and Diammonium phosphate (0-15 cm) treatments.

Nitrogen mineralization increased above the control (Tables 12 and Fig. 2). This increase was experienced only after the first 60 days of the experiment and then declined towards the end of incubation. The insignificant increase in production of mineral N in this case could have resulted from the added P which promotes mineralization from soil organic nitrogen. Potassium and phosphorus contents (Table 2) in this soil were low and this could also have affected the mineralization process. These two elements affect microbial biomass and activity (Bartholomew and Clark, 1965).

The higher N-mineralization in the 0-15 cm than 15-30 cm depths could be due to, first; the lower percent organic carbon in the 15-30 cm than 0-15 cm depths. Secondly, the 0-15 cm depth had a higher nitrogen mineralization potential (Table 10) hence, a higher rate and bigger magnitude of mineralizable N than in the 15-30 cm depth.

4.4.3 Effect of N and P salts on nitrogen mineralization in Katumani soils

Addition of N salts in Katumani soils increased N mineralization above the control (Fig. 3). The increase was significant ($P \leq 0.05$) throughout the incubation period. As table 8 shows, N mineralization increased from 97.6 µgN to 200.4 µgN and from 97.0 µgN to 174.6 µgN/g soil from 0 day to 120 days in the 0-15 cm and 15-30 cm respectively.

Where ammonium sulphate was added as a fertilizer, the increase was from 97.6 µgN to 181.7 µgN/g and from 97.0 µgN to 149.9 µgN/g soil in the 0-15 cm

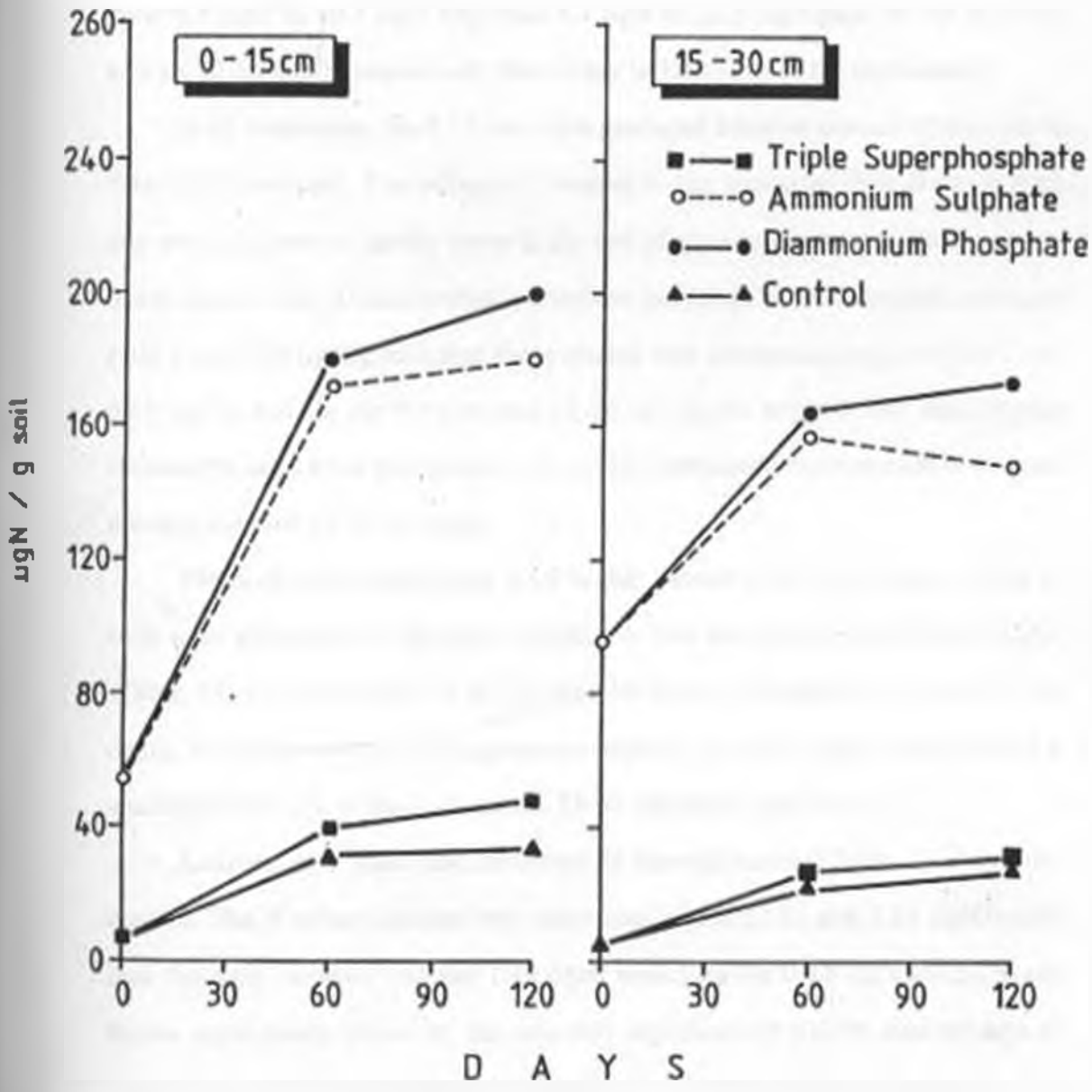

Fig. 3. Effect of N and P salts on nitrogen mineralization in Matomani Luvisols during incubation.

and 15-30 cm respectively. The same trend was seen in the control where mineralized N increased from 6.9 µgN to 35.3 µgN and from 6.1 µgN to 29.9 µgN/g soil in the 0-15 cm and 15-30 cm depth respectively from 0 day to 120 days of incubation. Addition of triple superphosphate increased production of mineral-N from 6.9 µgN to 49.7 µgN and from 6.1 µgN to 32.3 µgN/gsoil in the 0-15 cm and 15-30 cm depths respectively from 0 day to 120th day of the experiments.

In all treatments, the 0-15 cm depth produced a higher amount of mineral N than 15-30 cm depth. The increase in mineral N was very steep from 0 day to 60th day and then became gentle towards the end of the experiment in all treatments. Soils treated with Diammonium phosphate produced more inorganic nitrogen (102.6 and 77.6 µgN/g soil) than those treated with ammonium sulphate (84.1 and 52.9 µgN/g soil) in the 0-15 cm and 15-30 cm depths respectively. Both depths showed the same trend though the 0-15 cm depth produced more amount of mineral nitrogen than the 15-30 cm depth.

The N mineralization rates were higher (above 3.08 µgN/ week) where N salts were added than in all other treatments. The soil pH after addition of DAP (Table 13) dropped from 6.6 to 5.5 after 60 days of incubation in the 0-15 cm depth. With soils treated with ammonium sulphate, the pH dropped from 6.6 to 5.4 and from 7.0 to 5.0 in the 0-15 cm and 15-30 cm depths respectively.

Addition of P salts also increased N mineralization (Table 8) above the control. The N mineralization rates were also higher (2.52 and 1.54 µgN/week) than from the control (1.68 and 1.40 µgN/ week) for the 0-15 cm and 15- 30 cm depths respectively. However, this was only significant ($P \leq 0.05$) after 60 days of incubation. Also the pH dropped with addition of P from 6.6 to 6.4 and from 7.0 to 6.1 in the 0-15 cm and 15-30 cm depths respectively (Table 13). Liming was not applied in this soil because of its high pH.

Table 8: Nitrogen mineralization under different treatments for Katumani luvisols µgN/g soil

Treatments	Depth (cm)	Initial 0 days	After 60 days	After 120 days	Difference 120-0 days	N-min rates µgN/week
Control	0-15	6.9	32.4	35.3	28.4	1.68
	15.30	6.1	22.3	29.9	23.8	1.40
TSP	0-15	6.9	39.8	49.7	42.8	1.52
	15.30	6.1	27.2	32.3	26.2	1.54
DAP	0-15	97.6	180.1	200.4	102.6	6.02
	15.30	97.0	163.2	174.6	77.6	4.55
AS	0-15	97.6	173.9	181.7	84.1	4.90
	15.30	97.0	158.4	149.9	52.9	3.08

The differences obtained of mineralized nitrogen between the 0 day and 120 days (Table 8) after the treatments were all higher than those obtained from the control. DAP had the highest (102.6 µgN) followed by Ammonium sulphate (84.1 µgN/g soil); TSP (42.8 µgN/g soil) and then the control (28.4 µgN/g soil) in the 0-15 cm depth.

Since Katumani soils have very low nitrogen mineralization potential (Table 10), addition of N salts increased the nitrogen content in the substrate to be mineralized (Stanford and Smith, 1972). This in turn led to the high amount of mineralized N (Table 8) when compared to the control and in soils treated with Tripple superphosphate.

Among the two nitrogen sources, DAP increased mineralized N much more than ammonium sulphate. This is attributed to the N and P interaction (Munevar and Wallum, 1977). This increase was significant ($P \leq 0.05$) after 60 days and 120 days of the experiment. The presence of P could have influenced microbial proliferation and activity hence high release of mineral N in soils treated with

diammonium phosphate than those treated with ammonium sulphate. This same trend was seen in the 15-30 cm depth.

4.4.4 Comparison of treatment effects (lime, N and P salts) on N mineralizatiopn in the three soils

Increase in N mineralization is shown in table 9 where a positive sign indicates stimulation of N- mineralization and a negative sign retardation. The highest stimulation was observed in limed Gituamba soils (140.9 and 145.3 kg N/ha) in the 0-15 cm and 15-30 cm depths respectively. Kitale had only 4.8 and 0.9 kg N/ha in the 0-15 cm and 15-30 cm depths respectively.

The increased N mineralization (positive sign) suggests a priming effect. Priming is the stimulation effect of stable humus in soils (Broadbent, 1948; Broadbent and Bartholomew, 1948; Walker et al., 1956). In this case, it was obtained by first subtracting the nitrogen mineralized between 0 day and 120 days in all treatments. The values obtained were then subtracted from the values obtained in the control within this period. This gave either positive or negative values. Positive values indicate that the treatment had a priming effect on N mineralization while negative values indicate a depression.

Addition of triple superphosphate produced stimulation effect in all the three soils. Gituamba soils had the highest stimulation effect of 91.8 kg N and 36.4 kg N/ha in the 0-15 cm and 15-30 cm depths respectively. Kitale had 17.5 kgN and 16.6 kgN/ha while Katumani had 31.7 kgN and 5.4 kgN/ha in the 0-15 cm and 15-30 cm depths respectively.

Where DAP was added stimulation effect was only observed in Gituamba and Katumani soils. This stimulation was 6.2 kgN and 1.7 kgN/ha for Gituamba; 163.3 kgN and 118.4 kgN/ha for Katumani in the 0-15 cm and 15-30 cm depth. In Kitale soils, DAP had a retardation effect. It was -119.1 kgN and -126.0 kgN/ha for the 0-15 cm and 15-30 cm depths respectively.

Table 9: Priming effect on N mineralization by treatments (Lime, N and P-salt) in the three soils (KgN/ha)

SITE	Treatment	Depth (cm)	Initial 0 days	Final 120 days	Difference 120-0 days	Stimulation
GITUAMBA	Control	0-15	167.1	233.6	66.5	-
	"	15-30	152.5	177.7	27.2	-
	Lime	0-15	167.1	374.5	207.4	+140.9
	"	15-30	152.5	325.0	172.5	+145.3
	TSP	0-15	167.1	325.4	158.3	+ 91.8
	"	15-30	152.5	216.1	63.6	+ 36.4
	DAP	0-15	367.1	439.8	62.7	+ 6.2
	"	15-30	352.5	420.7	68.2	+ 1.70
	AS	0-15	367.1	406.1	39.1	- 27.4*
	"	15-30	352.5	392.1	39.6	12.4
KITALE	Control	0-15	72.5	95.2	22.7	-
	"	15-30	56.8	81.6	24.8	-
	Lime	0-15	72.5	100.0	27.5	+ 4.8
	"	15-30	56.8	82.5	25.7	+ 0.9
	TSP	0-15	72.5	112.7	40.2	+ 17.5
	"	15-30	56.8	98.2	41.4	+ 16.6
	DAP	0-15	272.5	176.1	- 96.4	-119.1
	"	15-30	256.8	155.6	- 101.2	-126.0
	AS	0-15	272.5	160.9	- 111.5	-134.2
	"	15-30	256.8	136.6	- 120.2	-145.0
KATUMANI	Control	0-15	15.1	377.7	62.6	-
	"	15-30	13.5	65.7	52.2	-
	TSP	0-15	15.1	109.4	94.3	+31.7
	"	15-30	13.5	71.1	57.6	+ 5.4
	DAP	0-15	215.1	440.9	225.9	+163.3
	"	15-30	213.5	384.1	170.6	+118.4
	AS	0-15	215.1	399.9	184.7	+122.1
	"	15-30	213.5	329.9	116.4	+64.2

Addition of ammonium sulphate had a stimulation effect in Katumani soils (122.1 kgN and 64.2 kgN/ha) in both depths and in Gituamba (12.4 kgN/ha) in the 15-30 cm depth. Kitale soils treated with ammonium sulphate showed a negative effect. This N was probably immobilized or lost through denitrification processes. The priming effect in Gituamba soils could have been due to increased pH (Table 11) after liming. This could have favoured the microbial growth and their activities in the soil. For Kitale soils, the pH increased to above 7.0 (Table 12) after liming. This therefore could have probably led to nitrogen losses through volatilization and denitrification hence the low stimulation effect. Terman (1979) observed losses of ammonia gas as pH increased. Also nitrification following liming favours N_20 losses especially in alkaline pH (Focht and Verstraete, 1977).

In all the three soils, addition of P salts showed an increase in net N mineralization. This is so even in Kitale soils where TSP had the highest amount (17.5 kgN and 16.6 KgN/ha for 0-15 cm and 15-30 cm respectively) of net mineralized N. This was higher than in any other treatment in this soil. Addition of phosphorus provides an essential nutrient to the microorganisms especially in very acid soils thus boosting their population and activity (Stotzky and Norman, 1961).

Generally, DAP had a higher amount of priming effect than ammonium sulphate (except for Gituamba soils, 15-30 cm). This could be attributed to the presence of phosphorus and its interaction with nitrogen which boosts microbial population. (Munevar and Wallum, 1977). The same trend was observed in the other two soils in both depths. For Kitale soils, the depression caused on N mineralization was higher with ammonium sulphate than with DAP. In Katumani soils DAP had a higher stimulating effect than TSP. The reason could probably be due to the presence of N and P. Addition of extra P did not have any effect in this soil probably because of its high inherent P content (Table 3). It can therefore be concluded that addition of P and N salts have a priming effect on N mineralization. This appears to reflect on the physical and chemical properties of the soil.Lime addition to soils also has a positive priming effect more especially with acid soils.

4.5 Nitrogen mineralization potential (N_o) in the three soils

Table 10 gives values of nitrogen mineralization potential (N_o), regression coefficients (r), mineralization rate constants (k) and time taken for 50 percent of potentially mineralizable nitrogen (t1/2) to be mineralized. t1/2 is given by 0.693/k and k is given by 2.303 multiplied by slope of log (N_o - N_t). The calculated mineralizable N (Table 10) was 138.8 µgN and 116.4 µgN/g for Gituamba; 46.0 µgN and 46.4 µgN/g for Kitale and then 260.1 µgN and 197.3 µgN/g soil for Katumani in the 0-15 cm and 15-30 cm depths respectively. The calculated values, compared well with the actual cumulative mineralizable N (Table 10).This was 127.7 µgN and 74.1µgN/g for Gituamba, 48.0 µgN and 54.1 µgN for Kitale and then 80.6 µgN and 47.7 µgN/g soil for Katumani soils in the 0-15cm and 15-30 cm depths, respectively.

Table 10: Nitrogen mineralization potential (N_o), mineralization rate constant (k) and time taken for half of mineralizable N to be mineralized (t 1/2)in the three soils

SITE	Depth (cm)	N_o µgN/soil	% of total Nitrogen	(r) Regression coefficient	k week^{-1}	t1/2 weeks	Calculated N-after 17.1 wks^{-1}	Observed cummulative Nitrogen
GITUAMBA	0-15	392.3	13.2	0.002	0.031	22.3[a]	138.8	124.7
	15-30	162.5	6.1	0.023	0.053	13.1	116.4	74.1
KITALE	0-15	195.6	12.0	0.01	0.023	30.1*	46.0	48.0
	15-30	178.7	17.2	0.01	0.027	25.7	46.4	54.1
KATUMANI	0-15	198.0	12.2	0.022	0.051	13.7	260.1	80.6
	15-30	75.8	10.0	0.003	0.112	6.3	197.3	47.7

The nitrogen mineralization potential (N_o) using Stanford et al., (1974) criteria was 392.3 µgN and 162.5 µgN/g for Gituamba; 195.6 µgN and 178.7

µgN/g for Kitale and then 198.0 µgN and 75.8 µgN/g soil for Katumani soils in the 0-15cm and 15-30cm depths, respectively.

The mineralization rate constant ranged from 0.023 week^{-1} in Kitale (0-15 cm) to 0.112 week^{-1} Katumani (15-30 cm) soils. Kitale soils had the lowest mineralization rate constant (0.023 and 0.027 week^{-1}) in the 0-15 cm and 15-30 cm depths respectively. These mineralization rate constants increased with depth in all the three soils. That is from 0.031 to 0.053 week^{-1} for Gituamba; 0.023 to 0.027 week^{-1} for Kitale and then 0.051 to 0.112 week^{-1} for Katumani in the 0-15 cm and 15-30 cm depths respectively. The time taken for 50 percent of the potentially mineralizable nitrogen (t1/2) to be mineralized ranged from 6.3 weeks for Katumani (15-30 cm) to 30.1 weeks for Kitale (0-15 cm) soils. An interesting observation is that as k increased, t1/2 decreased, that is soils with highest k had the least t1/2 (Table 10.0). For example, Katumani soil with t1/2 of 6.3 weeks (15-30 cm) had the highest k of 0.11 week^{-1} while Kitale with t1/2 of 30.1 weeks had the lowest k (0.023 week^{-1}) in the 0-15 cm depth.

The N_0 as percent of total nitrogen ranged from 6.1% in Gituamba (15-30 cm) to 17.2% in Kitale (15-30 cm) (Table 10) soils. The nitrogen mineralization potential in all the three soils showed the same trend as percent total nitrogen and percent organic carbon (Tables 1, 2 and 3) for both depths. There was a positive significant correlation ($r = 0.82$; $P \leq 0.05$) between organic carbon and nitrogen mineralization potential (N_0). The signficant positive correlation between percent organic carbon and nitrogen mineralization potential (N_0) explains the fact that the 0-15 cm depth with a higher organic matter had a higher N_0 than the 15-30 cm depth. This continues to explain the trend for cumulative mineralizable N.

The decrease of nitrogen mineralization potential with depth reflects on the decline of organic matter down the soil profiles. This is so in all soils even with actual cumulative nitrogen which also decreased with depth except for Kitale soils. For Katumani soil, there was low percent total nitrogen (Table 3), low net

mineralization and low N_0 (Table 10). This reflects the levels of organic matter and organic nitrogen in this soil.

The difference in N_0 between the three soils could be attributed to the differences in the organic matter contents and the cropping systems in the three soils. Katumani soil which has been under cultivation for a long time had the lowest N_0 especially in the 15-30 cm depth. Gituamba with high organic carbon (Table 1) had the highest N_0 in both depths.

Soils having higher N_0 eventually led to higher amounts of N-mineralization rates (Appendix III). For Katumani (both depths) and Gituamba (15-30 cm) soils, half of the nitrogen is susceptible to mineralization and therefore would be mineralized in a growing season of a crop. This is so as their t1/2 was less than 20 weeks which is the normal growing period of annual crops. For Kitale (both depths) and Gituamba (0-15 cm), this will not be the case. However, when the actual mineralized N (48.0 µgN and 54.1 µgN/g soil) is compared with the calculated N (46.0 µgN and 46.4 µgN/g soil) for the 17.1 weeks of incubation, they are more or less the same and therefore verifies the use of the formula in this soil.

For Katumani soils, very little of the actual nitrogen (80.6 µgN and 47.7 µgN/g soil for 0-15 and 15-30 cm depths, respectively) was mineralized when compared with the calculated N (260.1 µgN and 197.3 µgN/g soil for 0-15 cm and 15-30 cm depths, respectively). This could be due to the very low nitrogen content present initially and the low organic matter content in the soil. This however was not detected in the formula. It appears therefore that at low organic content in soils, the formula cannot be used because it exaggerates the mineralizable N.

The variations observed in the three soils (Luvisols, Ferralsols and Andosols) could be attributed to pH of the soil, soil texture (Craswell and Waring, 1972) and organic matter content (Huntjens, 1972) in these soils. The values of N_0 in the three soils ranged from 80 µgN to 392 µgN/g soil while Stanford and Smith (1972) obtained values ranging from 20 µgN to 300 µgN/g soil. Herlihy (1979) observed

variations in nitrogen mineralization potential and mineralization rate constant with seasonal changes.

4.6 Nitrification in the three soils and its effect on soil pH during incubation

Table 11 show nitrate levels and pH changes during incubation of Gituamba Andosols. The soil pH increased as incubation period progressed in lime treatment. In the same manner, nitrate levels showed the same trend. Where N salts were added, nitrification progressed although there was a drop in soil pH, which later increased towards the end of the experiment.

In limed Gituamba soils, the pH and nitrates produced were higher than in any other treatment. This suggests a priming effect through liming of acid soils and also suggests that acidification does depress nitrification but does not stop it completely (Ishaque and Cornfield, 1972).

Table 11: pH changes during incubation in relation to nitrates production (μgN/g soil) in Gituamba soils

Days		0		60		120	
Treatments	depth (cm)	NO_3-N	pH	NO_3-N	pH	NO_3^--N	pH
Control	0-15	37.8	4.0	155.9	4.0	196.7	4.1
	15-30	31.4	4.1	134.8	4.1	137.5	4.2
Lime	0-15	37.8	4.0	232.6	5.5	243.5	5.7
	15-30	31.4	4.1	192.2	5.8	193.1	5.8
TSP	0-15	37.8	4.0	205.6	4.0	222.8	4.2
	15-30	31.4	4.1	196.9	4.2	176.7	4.2
DAP	0-15	37.8	4.0	137.0	3.9	108.4	4.0
	15-30	31.4	4.1	145.6	3.9	117.8	4.1
AS	0-15	37.8	4.0	152.6	3.8	137.6	4.0
	15-30	31.4	4.1	134.3	3.9	139.8	4.0

($r = 0.46$; $P \leq 0.05$)

r = Correlation between pH and nitrates.

Table 12: pH changes during incubation in relation to nitrate produced(μgN/g soil) in Kitale soils

Treatments	Days depth (cm)	0 days NO_3^--N	pH	60 days NO_3^--N	pH	120 days NO_3^--N	pH
Control	0-15	26.4	5.6	24.2	6.3	20.6	6.1
	15-30	17.3	5.6	18.8	6.2	10.0	6.1
Lime	0-15	26.4	5.5	25.2	7.6	49.7	7.7
	15-30	17.3	5.6	19.6	7.6	33.7	7.8
TSP	0-15	26.4	5.6	29.3	5.3	37.8	5.9
	15-30	17.3	5.6	25.1	5.6	24.4	6.0
DAP	0-15	26.4	5.6	54.0	4.8	94.9	5.0
	15-30	17.3	5.6	48.9	4.0	79.6	4.9
AS	0-15	26.4	5.6	70.5	4.0	79.6	4.8
	15-30	17.3	5.6	47.4	4.6	73.9	4.9

$r = -0.46$; $P \geq 0.05$)

r = Correlation between pH and nitrates.

Table 13: pH changes during incubation in relation to nitrates production (μgN/g soil) in Katumani soils

Treatmnts	Days depth (cm)	0 days NO_3-N	pH	60 days NO_3-N	pH	120 days NO_3-N	pH
Control	0-15	7.6	6.6	63.4	6.0	61.9	6.6
	15-30	5.8	7.0	41.4	6.6	48.1	6.9
TSP	0-15	7.6	6.6	71.2	6.4	90.8	6.8
	15-30	5.8	7.0	51.4	6.1	62.3	6.6
DAP	0-15	7.6	6.6	228.9	5.5	233.1	5.7
	15-30	5.8	7.0	193.2	4.3	199.1	5.0
AS	0-15	7.6	6.6	219.6	5.4	222.5	5.9
	15-30	5.8	7.0	173.6	5.0	162.8	5.5

$r = -0.66$; $P \geq 0.05$)

r = Correlation between pH and Nitrates.

Nitrification after addition of TSP in Gituamba soils showed a priming effect. Here the pH (Table 11) fluctuated very slightly. With addition of DAP and AS, pH dropped slightly and then increased toward the end of the experiment.Nitrification took place on the applied ammonium nitrogen and also from the soil N pool. This same observation was seen in Kitale and Katumani soils, where both the ammonium nitrogen applied and that from the soil were nitrified. Nitrate levels correlated positively to pH ($r = 0.46$; $P \geq 0.05$) for Gituamba soils and negatively for Katumani ($r = -0.66$; $P \geq 0.05$) and Kitale ($r = -0.46$; $P \geq 0.05$) soils. This correlation between pH and nitrate production was not significant.

Kitale soils had the least production of nitrates (Table 12) as it tended to retain nitrogen in form of ammonium nitrogen (Appendix V). Though pH decreased after 60 days where soils were treated with DAP, AS and TSP, there was still some nitrification process occuring. Where soils were limed, pH increased to above 7.6 and thus could have led to volatilization of nitrogen in form of NH_3 gas.This could also have contributed to the low nitrate levels observed. Also Kitale soils being under grass cover and the soils contained very many small roots could have had inhibitory effects to the nitrifiers (Ellis, 1954). This soil tends to retain more ammonium nitrogen than nitrates as it is developed under grass cover (Robinson, 1963). Perennial grass secretes small quantities of toxic substances which specifically inhibit activities of nitrifiers (Theron, 1951). Nitrification increased throughout the incubation period (Table 13) in Katumani soils.This is so even where there was a drop in pH after addition of N and P salts. The drop in pH occurred after 60 days of incubation. Nitrification was highest where N salts (especially DAP) were added and lowest in the control.Since Katumani soils are poor in both organic matter and total nitrogen, addition of N salts increased nitrifiable nitrogen in the soil when compared to the control.

There was higher production of nitrates where DAP was applied than with ammonium sulphate application in all the three soils. This difference is attributed to the presence of P in DAP. This P with N interaction boosted microbial proliferation

and activity (Munevar and Wallum, 1977). In all the three soils, nitrification decreased with depth while the pH increased with depth. The variations in pH in the three soils could be due to organic matter present in each soil. Gituamba with higher organic matter content had slight pH changes due to high buffering capacity. Kitale and Katumani soils had drastic pH changes due to low organic matter content which exhibit low buffering capacity.

5.0 CONCLUSIONS

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 soil with the highest amount of organic matter also had the highest quantity of all forms of nitrogen. On the other hand, Katumani soil with the least amount of organic matter had the least quantity of the various forms of nitrogen. Other factors such as soil pH seemed to influence the rate and extent of decomposition of organic matter. This in turn had an influence on the various forms of nitrogen in each soil. Organic form of nitrogen was highest among the various forms of N. Nitrates formed the least portion of nitrogen forms. Non-exchangeable ammonium ion was highest in Gituamba soils. This could be attributed to the high amount of nitrogen present in Gituamba soils and probably the type of clay mineral of this soil. For organic nitrogen fractions, hydrolyzable total N ranged from 40.0% to 61.9%. Amide N 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 nitrogen in the soils. Amino acid N formed the highest portion of the organic N fractions.

Those fractions that are easily hydrolysed, for example Amide N and Amino acid N, were lower in Katumani soil which had been cultivated for a long time than in Kitale soil which was under grass cover.

Liming of soils raised the pH and had a marked effect on N mineralization in Gituamba soils. Since Gituamba soils are acidic, liming created favourable conditions for microbial activity. In Kitale soils, liming did not produce significant N mineralization. This could be due to denitrification and volatilization of ammonia gas as pH increased above 7.0.

Addition of N salts increased production of mineral nitrogen, partly coming from added N and partly from soil nitrogen. Net N mineralization above the control was only observed in Katumani and Gituamba soils. This suggested some priming effect where DAP, AS and TSP were added. But in Kitale soils, N mineralization

was depressed by addition of N salts. However, addition of TSP showed some stimulating effect in this soil.

The nitrogen mineralization potential was highly correlated with percent organic carbon. N_0 showed the same trend as total nitrogen both with depths and soils. Soils with high percent organic carbon and organic nitrogen such as Gituamba had also the highest N_0 and vice versa. The percent organic carbon for Gituamba was significantly correlated ($r = 0.82$, $P \leq 0.05$) to N_0. This means that the higher the organic carbon in the soil, the the higher the nitrogen mineralization potential N_0.

The mineralization rate constant (k) ranged from 0.023 $week^{-1}$ to 0.11 $week^{-1}$. Time taken for half of the nitrogen to be mineralized (t1/2) ranged from 6.3 to 30.1 weeks. An interesting observation was that as the mineralization rate constant increased, t1/2 decreased. Also the formula used in predicting N_0 cannot be used where soils are low in organic matter content as it tends to exaggerate mineralizable N.

Mineralization rates showed the same trend as both total and organic N contents of the soils. High mineralization rates were observed after 60 days of incubation in all depths in the three soils.

Raising soil pH by liming favoured nitrifiers in Gituamba soils. Addition of N salts produced considerable amount of nitrates from the added salts and some from the soil in both Gituamba and Kitale soils. However, in Katumani soils, production of nitrates was very high with the addition of N salts. Nitrate production was positively correlated with soil pH in Gituamba soils only. Nitrification in acid soils (Gituamba and Kitale) could be attributed to presence of acid adapted strains which are active at low pH levels. Kitale soils had the least amount of nitrates produced. This was thought to be due to the nature of this soil being under grass cover and tends to retain more ammonium nitrogen than nitrates.

It can therefore be recommeded to lime Gituamba soils than add N fertilizers. This will help in exploiting the accumulated organic nitrogen which, when released

becomes available for crops. This will reduce the farmers production cost and hence increase his profits. It is not neccessary to lime Kitale soils as liming does not increase N mineralization significantly. Nevertheless future studies should determine threshold levels of liming various acid soils for maximum N mineralization. At the same time liming of such soils should be done at the field and then studies on microbial and N turnover to be carried on.

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APPENDICES

Appendix I: Nitrogen distribution in the three soils as percentage of total nitrogen

SITE	Depth (cm)	NH_4-N_e	NO_3-N	NH_4-Nf	Organic nitrogen
Gituamba	0-15	1.9	0.6	22.1	75.4
	15-30	2.1	0.5	22.1	75.3
Kitale	0-15	2.8	1.6	3.3	92.3
	15-30	3.2	1.2	3.8	91.8
Katumani	0-15	0.2	0.2	4.3	95.3
	15-30	0.5	0.3	7.5	91.7

NH_4-N_e = Exchangeable ammonium nitrogen
NO_3-N = nitrates
NH_4-N_f = fixed nitrogen.

Appendix II: Cummulative net N mineralization (N_t) of the three soils (μgN/g soil)

Site	Depth (cm)	Days 0 Weeks 0	60 8.6	120 17.1
Gituamba	0-15	-	94.5	124.7
	15-30	-	61.8	74.1
Kitale	0-15	-	37.7	48.0
	15-30	-	42.8	54.1
Katumani	0-15	-	45.8	80.6
	15-30	-	24.0	47.7

Appendix III. Actual Net N-mineralization rates of the three soils (µgN/g/week)

Site	Depth (cm)	Days 0 weeks 0	60 8.6	120 17.1
Gituamba	0-15	-	11.0	7.3
	15-30	-	7.4	4.3
Kitale	0-15	-	4.4	2.8
	15-30	-	5.0	3.1
Katumani	0-15	-	5.3	4.7
	15-30	-	2.8	2.8

Appendix IV: Comparison between ammonium-N and nitrates produced (Kg N/ha) during incubation in Gituamba soils

Treatments	Depth	0 days		60 days		1200 days	
	(cm)	NH_4^+-N	NO_3^-N	NH_4^+-N	NO_3^--N	NH_4^+-N	NO_3^--N
Control	0-15	129.2	37.8	130.6	155.9	36.8	196.7
	15-30	121.7	31.4	121.8	134.8	42.3	137.5
Lime	0-15	129.2	37.8	139.3	232.6	131.0	243.5
	15-30	121.1	31.4	137.9	192.2	131.9	193.1
TSP	0-15	129.2	37.8	129.9	205.6	102.6	222.8
	15-30	221.1	31.4	106.5	196.9	39.3	176.7
DAP	0-15	229.2	37.8	314.3	137.0	302.2	137.6
	15-30	221.1	31.4	278.0	145.6	280.9	139.8
AS	0-15	229.2	37.8	308.1	152.6	297.8	108.4
	15-30	221.1	31.4	272.9	134.3	274.4	117.8

Appendix V: Comparison between ammonium-N and nitrates produced (KgN/ha) during incubation in Kitale soils

Treatments	Depth (cm)	0 days		60 days		120 days	
		NH_4^+-N	NO_3^--N	NH_4^+-N	NO_3^--N	NH_4^+-N	NO_3^-N
Control	0-15	46.1	26.4	87.3	24.2	30.6	78.3
	15-30	39.5	17.3	91.9	18.8	10.0	71.6
Lime	0-15	46.1	26.4	96.7	25.2	50.3	49.7
	15-30	39.5	17.3	84.6	19.6	48.7	33.7
TSP	0-15	46.1	26.4	96.4	29.3	79.9	32.8
	15-30	39.5	17.3	90.5	25.1	73.8	24.4
DAP	0-15	246.1	26.4	109.4	54.0	81.2	94.9
	15-30	239.5	17.3	111.2	48.9	76.0	79.6
AS	0-15	246.1	26.4	91.3	70.5	81.3	79.6
	15-30	239.5	17.3	76.4	47.4	62.7	73.9

Appendix VI: Comparison between ammonium-N and nitrates produced (KgN/ha) in Katumani soils during incubation

Treatments	Depth (cm)	0 days		60 days		120 days	
		NH_4^+-N	NO_3^--N	NH_4^+-N	NO_3^--N	NH_4^+-N	NO_3^--N
Control	0-15	7.51	7.6	7.8	63.4	15.8	61.9
	15-30	7.7	5.8	7.7	41.4	17.6	48.1
TSP	0-15	7.5	7.6	8.3	79.2	18.7	90.8
	15-30	7.7	5.8	8.6	51.4	8.73	62.34
DAP	0-15	207.5	7.6	167.6	228.9	207.9	233.1
	15-30	207.7	5.8	165.8	193.2	185.0	199.1
AS	0-15	207.5	7.6	162.9	219.6	177.2	222.5
	15-30	207.7	5.8	175.0	173.6	167.7	162.8

Appendix VIIa ANOVA for net N mineralization for Gituamba soils after 60 days

Source of variation	df	ss	mss	F-calc.
Total	9	4678.04	833.92	
Treatment	4	4373.17	1093.29	94.82**
Depth	1	258.06	258.06	22.38**
Error	4	46.81	11.53	

LSD (95%) = 7.7

S.E =22.8; C.V. = 6.07

Appendix VIIb ANOVA for net N mineralization for Gituamba soils after 120 days of incubation

Source of variation	df	ss	mss	F-calc.
Total	9	7505.32	833.92	
Treatment	4	6287.68	1571.92	10.54**
Depth	1	620.95	620.95	4.16 N.S.
Error	4	596.69	149.17	

LSD(95%)=27.68

S.E.=28.89;C.V.=29.35.

Appendix VIIc Mean seperation using LSD at 95% confidence limit

T1 =Control. T2= Lime. T3= TSP. T4=DAP.T5 =AS. D1=0-15Cm. D2=15-30Cm

60days	120days
T2-T5=53.25*	T2-T5=68.45*
T2-T4=51.70*	T2-T1=65.00*
T2-T1=36.15*	T2-T4=54.30*
T2-T3=14.40*	T2-T3=35.90*
T3-T5=38.85*	T3-T5=32.55*
T3-T4=37.30*	T3-T1=29.10*
T3-T1=21.75*	T3-T4=18.40 N.S.
T1-T5=17.10*	T4-T5=14.15"
T1-T4=15.55*	T4-T1=10.70"
T4-T5=1.55N.S.	T1-T5=3.45N.S.
D1-D2=10.60*	D1-D2=15.76*

Appendix VIIIa ANOVA for net N mineralization for Kitale soils after 120days of incubation

Source of variation	df	ss	mss	F-calc.
Total	9	10392.86	1154.76	
Treatment	4	10373.26	2593.32	576.29*
Depth	1	1.60	1.60	0.36N.S.
Error	4	18.00	4.50	

LSD(95%)=4.81

S.E.=33.98; C.V.=·17.45.

Appendix VIIIb ANOVA for N mineralization for Kitale soils after 60 days of incubation

Source of variation	df	ss	mss	F calc.
Total	9	13359.50	1484.39	
Treatment	4	13268.21	3317.05	147.10**
Depth	1	1.10	1.10	0.05N.S.
Error	4	90.20	22.55	

LSD(95%)=10.76.
S.E.=38.53; C.V.=-71.62.

Appendix VIIIc Mean seperation for Kitale soils (LSD at 95%C.L.)

60 days	120 days
T3-T5=81.2*	T3-T5=76.00*
T3-T4=72.55*	T3-T4=63.45*
T3-T1=4.65 N.S.	T3-T1= 7.7*
T3-T2=3.75 N.S.	T3-T2= 6.4*
T2-T5=77.45*	T2-T5=69.6*
T2-T4=68.80*	T2-T4=57.05*
T2-T1=0.9 N.S.	T2-T1=1.3 N.S.
T1-T5=76.55*	T1-T5=68.30*
T1-T4=67.90*	T1-T4=55.75*
T4-T5=8.65 N.S.	T4-T5=12.55*
D1-D2=8.64 N.S.	D2-D1=0.8 N.S.

Appendix IXa ANOVA for net N mineralization for Katumani soils after 60 days

Source of variation	df	ss	mss	F-calc.
Total	7	4618.09	659.73	
Treatment	3	4299.5	1433.17	16.11*
Depth	1	266.8	266.8	2.99N.S
Error	3	51.79	17.26	

S.E.=25.69

LSD(95%)=24.5,C.V.=19.43

Appendix IXb ANOVA for net N mineralization for Katumani soils after 120 days of incubation

Source of variation	df	ss	mss	F-calc.
Total	7	6286.7	898.1	
Treatment	3	5339.12	1779.71	26.87*
Depth	1	748.85	748.85	11.31*
Error	3	198.73	66.24	

S.E. =29.97

LSD = 21.15; C.V.=14.85.

Appendix IXc Mean seperation for Katumani soils LSD (95%)

60 days	120days
T4-T1=50.45*	T4-T1=64.0*
T4-T3=47.35*	T4-T3=55.6*
T4-T5= 5.5 N.S.	T4-T5=21.6*
T5-T1=44.95*	T5 T1=42.4*
T5-T3=41.85*	T5-T3=34.O*
T3-T1= 3.1 N.S.	T3-T1= 8.4 N.S.
D1-D2=11.55 N.S.	D1-D2= 19.35 N.S.

* Significant at 5%
N.S. Not significant.

Appendix X Correlation between pH and other factors

Base saturation	0.86
Aluminium	-0.88
Nitrates	±0.46, -0.66
N_O	-0.82