ASSESSING THE SOIL CARBON SEQUESTRATION POTENTIAL OF DIFFERENT PLANT RESIDUES

BY

NAA KOTEIKOR AMON

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Department of Soil Science
College of Agriculture and Consumer Sciences
University of Ghana
Legon,Accra,Ghana

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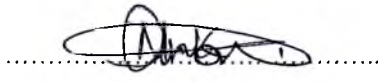
DEDICATION

Dedicated to my parents; Mr. and Mrs. G.K. Hammond and my sisters; Mrs. Naa Adei Essuman and Koteikai Amon

DECLARATION

I hereby declare that, the thesis herein presented for a degree of Master of Philosophy in Soil Science is a result of my investigation. All references to other authors' works as sources of information are duly acknowledged.



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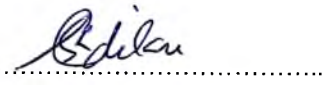
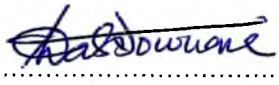


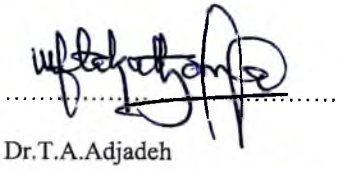
Prof. S.G. K. Adiku

(Supervisor)



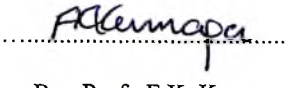
Prof..G.N.N.Dowuona

(Member)



Dr.T.A.Adjadeh

(Member)



Rev.Prof. F.K. Kumaga

(Member)

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ABSTRACT

This study investigated the use of fallow residue management as a means of sequestering soil carbon to mitigate the build up of atmospheric carbon dioxide.

The study involved the analysis of the effect of three soil moisture levels (W1= Field Capacity (FC), W2 = 70% FC and W3 = 40 % FC) on the decomposition rate of five different fallow plant residues.(i) *Pennisetum spp (*elephant grass) from natural bush fallow, RT1, (ii) *Cajanus cajan* (pigeon pea) residue, RT2, (iii) *Vigna unguiculata* (cowpea), RT3, (iv) *Mucuna pruriens* (mucuna) residue, RT4 and (v*) Pennisetum spp* (elephant grass which had benefited from residual fertilizer) under greenhouse conditions. The residues were incubated for 180 days in potted soils (Haplic Lixosols) and monitored over a 6-month period and the amount of organic carbon added to the soil was determined. Limited study of residue decomposition under field conditions was also carried out and compared with the greenhouse conditions.

Results showed that water had a significant effect ($P<0.05$) on the decomposition rates of plant residues. The order of water treatment on the decomposition of plant residues generally was: W1 > W2 > W3.

Decomposition was also different for the residue types in the order: RT1 < RT5 < RT3 < RT4 < RT2. This study showed that the grasses; RT1and RT5 having high C:N ratios were better contributors to soil carbon storage.

It was also observed that at the end of the incubation period all the plant residues added significantly higher carbon to the soil than the control. (RT6). A similar response was also observed in the field even though water and temperature could not be controlled.

A simple equation developed to describe the variation of residue decomposition with soil water gave reasonable predictions of residue weight loss with time. It is concluded that residue type and soil moisture management offer measures for improving soil carbon sequestration in tropical agricultural systems.

TABLE OF CONTENTS

	Page
Title page	i
Dedication	ii
Declaration	iii
Acknowledgements	iv
Abstract	v
Table of content	vii
List of Tables	xi
List of Figures	xiii
CHAPTER 1	1
1.0 INTRODUCTION	1
1.1 Background	1
1.2 Problem specification	2
1.3 Objectives	4
CHAPTER 2	5
2.0 LITERATURE REVIEW	5
2.1 Introduction	5
2.2 The carbon cycle	6
2.3 The role of plants in the carbon cycle	7
2.4 Residue decomposition	8
2.4.1 Factors governing residue decomposition	9

2.4.1.1	Plant (substrate) characteristic	9
2.4.1.2	The nitrogen content of substrate	10
2.4.1.3	Relative size of substrate	11
2.4.1.4	Method and depth of placement of residue	12
2.4.1.5	Environmental factors affecting residue decomposition	12
2.4.1.5.1	Temperature	12
2.4..1.5.2	Moisture content of soil	14
2.4.1.5.3	Soil pH	15
2.4.1.5.4	The carbon: nitrogen ratio of the soil	16
2.4.1.5.5	Cultivation/Tillage	17
2.4.1.5.6	Texture and mineralogy	18
2.5	Processes involved in organic matter decomposition	19
2.6	Contribution of cereal and legumes residues to carbon sequestration	21
2.7	Techniques for estimating carbon sequestration potential	23
2.8	Modeling residue decomposition	25
2.9	Importance of soil organic matter	29
CHAPTER 3		30
3.0	MATERIALS AND METHODS	30
3.1	Experimental site	30
3.1.1	Location and Physiography	30
3..2	Soils and sampling	30
3.3	Determination of physico-chemical properties of the Haplic Lixosol	31

3.3.1	Soil texture	31
3.3.2	Field capacity determination	32
3.3.3	Soil pH	33
3.3.4	Organic Carbon	33
3.3.5	Total Nitrogen	34
3.3.6	Exchangeable Bases	35
3.3.7	Cation exchange capacity	36
3.3.8	Available phosphorus	36
3.4	Greenhouse residue incubation studies	37
3.4.1	Introduction	37
3.4.2	Plant residue	38
3.4.3	Pot experiment	39
3.5	Field studies	40
3.6	Statistical analysis	41
CHAPTER 4 RESULTS AND DISSCUSSION		42
4.0	Greenhouse studies	42
4.1	Soil physical and chemical properties	42
4.2	Some chemical characteristics of the plant residues	44
4.3	Decomposition of residues during greenhouse studies	47
4.3.1	Effect of residue type	47
4.3.2	Effect of soil water on residue decomposition	56
4.4	Modelling the dynamics of residue decomposition	60

4.5 Soil carbon and nitrogen accumulation during incubation studies 71

4.6 Field Studies 77

4.6.1 Some physical and chemical properties of Haplic luvisol 77

4.7 Field residue decomposition study 77

4.7.1Some chemical charctericterics of plant residues obtained from the field 77

4.7.2. Effect of residue type on decomposition 80

4.7.3Comparison between greenhouse pot experiment and field residue studies 82

4.7.4 Soil carbon accumulation during field study 86

CHAPTER 5 88

CONCLUSION 88

Recommendation 89

REFERENCES 90

APPENDIX 112

LIST OF TABLES

3.1	Description of plant residues used in the incubation study	38
4.1	Some physical (a) and chemical (b) properties of Haplic Lixosol.	43
4.2	Some chemical properties of plant residues obtained from Kpev	44
4.3	Half-life of residue dry weight (days) for different water treatments	55
4.4	Half-life of residue dry weight (days) for three water treatments	58
4.5	Effect of soil water on the decay constants (k) g/d for various plant residues obtained from fitted equation	58
4.6	Effect of soil moisture;W1,W2,W3 on decay constants; k1and K2 (g/day) pools for the various residue types obtained from fitted equation	62
4.7	Effect of various residue treatments on soil organic carbon and nitrogen / % accumulation during the incubation period at soil moisture W1	71
4.8	Effect of various residue treatments on soil organic carbon and nitrogen / % accumulation during the incubation period at soil moisture W2	75
4.9	Effect of various residue treatments on soil organic carbon and nitrogen / % accumulation during the incubation period at soil moisture W3	76
4.10	Some physical (a) and chemical (b) properties of the field soil from the University farm	78
4.11	Some chemical properties of the plant residues used for the field study	79

4.12 Comparison between decay rate constant (k; g/day) for the greenhouse pot experiment and that of the field for some of the plant residue treatments 83

4.13 Oxizable carbon % during field studies 86

LIST OF FIGURES

2.1 The Carbon cycle 6

4.1 Variation of residue dry weight with time for water treatment W1 (a) grass residue (b) Legume residue; observed data are symbols and fitted curves are lines. 48

4.2 Variation of residue dry weight with time for water treatment W2 (a) grass residue (b) Legume residue; observed data are symbols and fitted curves are lines. 51

4.3 Variation of residue dry weight with time for water treatment W3 (a) grass residue (b) Legume residue; observed data are symbols and fitted curves are lines. 52

4.4 Variation of observed (symbols) and calculated (lines) plant residue dry weight for RT1,RT5,RT2,RT3 and RT4 at 70 % field capacity, W2 65

4.5 Variation of Observed (symbols) and calculated (lines) plant residue dry weight for RT1,RT5,RT2,RT3 and RT4 at 40 % field capacity, W3 67

4.6 Predicted versus observed plant residue dry weight for RT1,RT5,RT2,RT3 and RT4 at 70% field capacity,W2 69

4.7 Predicted versus Observed plant residue dry weight RT1,RT5,RT2,RT3 and RT4 at 40 % field capacity,W2 70

4.8 Variation of residue dry weight with time for RT2, RT4 and RT5 used for field studies. 81

4.9 Variation of observed (symbols) and calculated (lines) plant residue dry weight for RT2, RT4 and RT5 with time for the field study 85.

CHAPTER 1

INTRODUCTION

1.1 Background

Carbon is a major constituent of all living things and the cycling of carbon between the atmosphere and the biosphere is one of the bases for life on earth. However, as a result of man's exploitative activities such as deforestation, automobile emission and power generation to increase productivity to meet energy demands, the quality of the natural environment has and continues to deteriorate in many parts of the world at a time when the human population is growing at an accelerated rate.

The stable carbon dioxide concentration in the atmosphere has for a long time been 0.034 % (Alexander, 1977). Report by Paustian (1998) indicates that carbon dioxide in the atmosphere has increased by about 30 % since 1800 and continues to increase by about 0.5 % per year. According to a special report on the contribution of carbon pool to the global carbon cycle by the Intergovernmental Panel on Climate Change (IPCC, 2000) over the period 1989-1998, activities in the energy and building sectors of the global economy increased atmospheric carbon levels by 6.3 Gigatonnes of carbon per year (Gt C yr^{-1}). Other reports from the Science Daily Magazine (1995 -2003) also indicate that deforestation and the use of fossil fuels release about 8 billion metric tonnes of carbon dioxide into the atmosphere annually. Although most of the carbon dioxide gas is removed from the atmosphere by plants or by the world's oceans, a significant portion remains air borne. The net result of these fluxes over the last 10-15 years is that atmospheric carbon levels have increased by about 3.3 Gt C yr^{-1}

Carbon dioxide addition to the atmosphere is caused not only by burning of fossil fuel through industrial and agricultural activities, but also by soil organic carbon decomposition and vegetation burning. Methane production, volatilisation and mineralization of soil carbon can also lead to carbon loss from the soil. Consequently, atmospheric carbon dioxide continues to increase at the rate of 3.5 billion tonnes every year (Rice *et al.*, 2004) which presents a problem of global warming.

1.2 Problem specification

Increased atmospheric carbon dioxide in the environment could lead to global warming and changes in climate pattern (Jenkinson *et al.*, 1991; IPCC, 2001). To mitigate these effects various methods have been proposed to control carbon dioxide accumulation in the atmosphere. One of these is to legislate a policy framework, which ensures that the fossil burning industries change their technology to reduce carbon dioxide concentration in the atmosphere (Wise and Cacho, 1999). In other cases, heavy levying of culprit industries to deter their production of carbon dioxide has also not been effective. The Department of Energy in U.S.A used carbon to bind common minerals like serpentine and peridotites and this was believed to result in a mineral carbonate which could be used to prevent carbon dioxide from fossil fuel from reaching the atmosphere (McNelly, 1999). This helps in reducing carbon dioxide concentration in the atmosphere. However, this method is very expensive and could not be used on a wide scale.

Carbon credit trading, incentives for development and application of new technology, education and technical assistance for producers and tax credits for conservation practices are also being used (Bruce *et al.*, 1999) as methods to reduce carbon load of the atmosphere.

Carbon sequestration by soil provides a better means of storage of carbon in a stable solid form and this occurs through direct and indirect fixation of atmospheric carbon dioxide. (Wielopolski *et al.*, 2004). Direct soil carbon sequestration occurs by inorganic chemical reactions that convert carbon dioxide into soil inorganic carbon compounds such as calcium and magnesium compounds. Indirect carbon sequestration by plants through photosynthesis could finally be transformed to the soil when the plant biomass is added to the soil as residue. This process is considered by many as the most cost effective way to reduce the concentration of carbon dioxide in the atmosphere. Thus, increasing soil organic C stocks (i.e. soil carbon sequestration), as a means to mitigate increasing CO_2 concentrations in the atmosphere, requires increasing C inputs and /or decreasing decomposition rates Paustian *et al.*, (2002).

Different plant residues decompose at different rates. Slow decomposing residues will result in more carbon accretion to the soil and being released slowly into the atmosphere (Bouldin, 1988). As noted, carbon additions are mainly through plant residue input and their decomposition rates (Bruce *et al.*, 1999). The quality and quantity of plant residue input will determine the status of the soil as studies show that addition of organic matter to soil improves many of the soil properties that influence yield (Mulongoy and Merckx., 1991). Other factors that affect the residue decomposition rate include the moisture status of the soil (Paustian *et al.*, 2002). The slowly decomposing residues may have an important effect on the organic matter content of the soil but the effect will occur only after a period of several years (Van Faasen and Smilde, 1985).

The storage of carbon in soils if only managed well would therefore offer a more permanent storage of carbon. Whereas most studies on land fallowing in Ghana has focused mainly on soil fertility restoration, there is a general lack of detailed studies on how different

plant types and environmental factors interact to determine soil carbon storage. In deciding on the ultimate goal to get more carbon stored in the soil through plants which are commonly left on the soil in various fallow systems, as well as improve the productivity of soil within cropping systems, it is necessary to evaluate the carbon sequestration potential of various fallow plant residues. The amount of carbon sequestered at a site reflects the long-term balance between carbon addition and release mechanism (Christensen, 1996; Turner and Lambert, 2000; Paustian *et al.*, 2002).

1.3 Objectives

The objectives of this research are to:

i) determine the effect of five fallow residue types obtained from a maize-cropping system at Kpeve in the Volta Region of Ghana on the decomposition rates under greenhouse conditions, and compare with limited field studies at the University Farm, Legon,

ii) determine the effect of soil moisture levels on the decomposition rate of these plant residues and their contribution to soil carbon storage,

(iii) determine the additions of organic carbon to the soil by the various residue types.

CHAPTER 2

LITERATURE REVIEW

2.1 Introduction

The increasing level of carbon dioxide in our atmosphere has caused scientists to investigate ways of limiting carbon emissions from agriculture. Agriculture has a tremendous potential to reduce carbon present in the atmosphere due to assimilation of atmospheric carbon by growing plants (Cole *et al.*, 1997). Plant residues returned to the soil would add carbon to the soil and part of this will be converted by microbial decomposition processes to organic matter.

The amount of carbon (C) stored in soils depends primarily on the balance between C inputs from the plant residues and C emissions from decomposition. Thus, increasing soil organic C stocks (i.e. soil carbon sequestration), as a means to mitigate increasing CO_2 concentrations in the atmosphere, requires increasing C inputs and/or decreasing decomposition rates. Both inputs and decomposition rates are affected by environmental factors such as climate (temperature and rainfall) and soil physical factors (soil texture, clay mineralogy, profile development), as well as agricultural management practices (Paustian *et al.*, 2002)

Different plant residues decompose at different rates. Slow decomposing residues will result in more carbon accretion to the soil and being released slowly into the atmosphere. Biodegradation of plant residues have considerable contribution in carbon evolution in natural and agro ecosystems. The chemical composition of plant residues determines their contribution to carbon sequestration and the fertility status of the soil. Thus, increased understanding of residue decomposition and its associated N mineralization may improve the management of cover crops in these systems (Quemada and Cabrera, 1995).

2.2 The carbon cycle

The carbon cycle is the various interlocking processes of synthesis and decomposition by which carbon is circulated through the biosphere, geosphere, hydrosphere and atmosphere of the earth (Merritts *et al.*, 1998; Purves *et al.*, 1995; White, 1979). All these components are reservoirs of carbon. Figure 2.1 below describes the process of the carbon cycle.

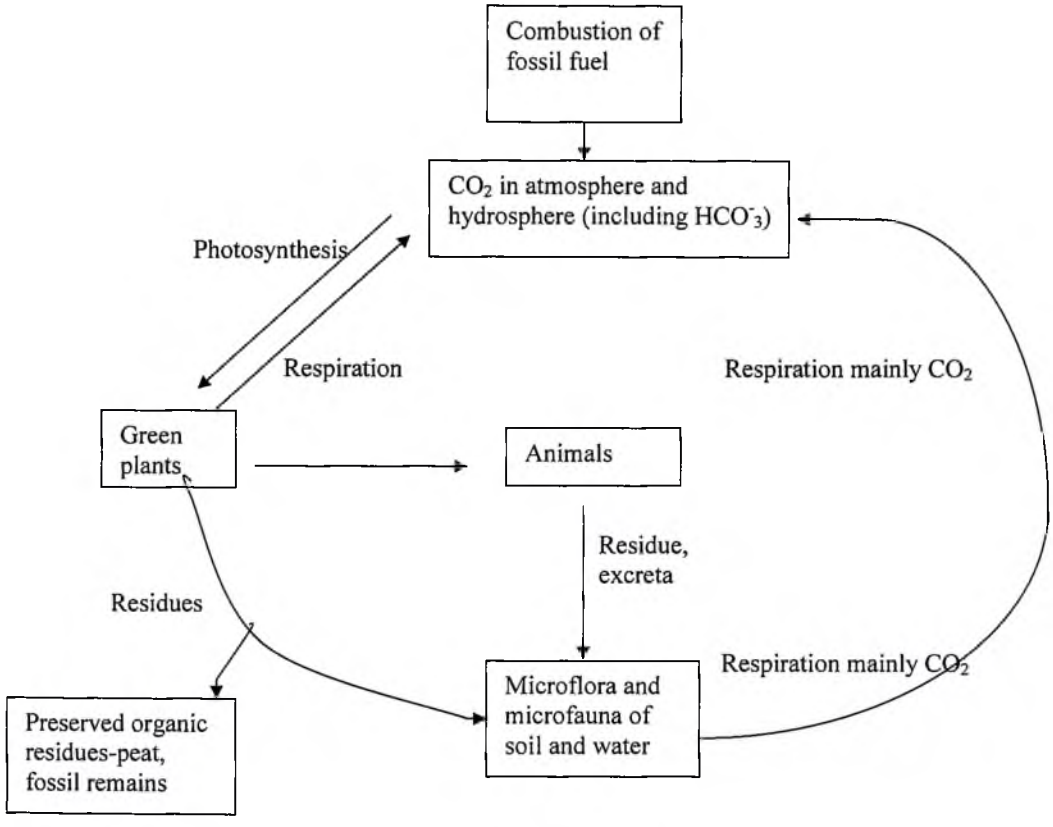


Fig. 2.1. The Carbon cycle (White, 1979)

Carbon exists in the earth's atmosphere primarily as the gas carbon dioxide and plays an important role in supporting life (Alexander, 1977). Through photosynthesis carbon dioxide is

converted to plant material and this carbon is lost when the crop is harvested. Animals consume the plants and carbon is returned to the soil in the form of manure or excreta which are admixed to a variable extent with the mineral component. The dead organic matter is colonized by a variety of soil organisms which derive energy for growth from the oxidative decomposition of complex organic molecules. During decomposition, essential elements are converted from organic combination to simple inorganic forms through the process of mineralization (White, 1979). Mineralization, especially the release of carbon dioxide is vital for the growth of succeeding generations of green plants which convert organic carbon to carbon dioxide. When the micro organisms respire, carbon dioxide is released into the atmosphere.

The remainder of the substrate carbon used by the micro organisms is incorporated into their cell substance or biomass, as a variable proportion of the other essential elements N, P and S. This incorporation renders these elements unavailable (immobilized) for plant growth until the organisms die and decay. The residues of the organisms, together with the more recalcitrant parts of the original substrate, accumulate in the soil (White, 1979). Soil carbon losses are exacerbated through erosion and to a lesser extent through leaching of dissolved organic carbon. Hence the basic processes of the carbon cycle are carbon dioxide in through photosynthesis and carbon dioxide out through decomposition (Goings, 2001).

2.3 The role of plants in the carbon cycle

Green plants play a very important role in the carbon cycle. Carbon dioxide is used by plants for photosynthesis to build up carbon compounds. The amount of carbon taken up by photosynthesis and released back to the atmosphere by respiration each year is 1000 times greater than the amount of carbon that moves through the geological cycle on annual basis

(Merritts *et al.*, 1998; Purves *et al.*, 1995). During the process of photosynthesis, sunlight energy is first trapped by the chlorophyll of the plant. The energy is converted to a chemical form called Adensosine triphosphate (ATP). The plants then use the energy from ATP to produce sugar ($C_6H_{12}O_6$). This process of photosynthesis requires water and produces oxygen as shown below

$$6\,CO_2 + 6\,H_2O + \text{Sun light} \longrightarrow C_6H_{12}O_6 + 6\,O_2 \qquad (2.1)$$

Animals eat plants to obtain the energy trapped during photosynthesis. As the animals bodies break down the carbohydrates in the plant tissue, carbon dioxide is released to the atmosphere through the process of respiration.

$$C_6H_{12}O_6 + 6O_2 \longrightarrow 6CO_2 + 6\,H_2O \qquad (2.2)$$

Plants too respire as they breakdown the organic molecules in themselves to release the stored energy. Plants also release CO_2 to the atmosphere when they decompose. When dead plants slowly decay under high pressure and high temperature, they may form pools of energy known as fossil fuels (White, 1979). Fossil fuels, as well as fresh vegetation through the process of combustion release the energy stored in them in the form of CO_2 into the atmosphere.

2.4 Residue decomposition

According to Thien and Graveel (1997), decomposition describes a series of processes that ultimately reduce the complexity of a material. Residue decomposition will therefore involve the constituent parts of the material being released or synthesized into new compounds. This act of releasing and synthesizing of materials is the main process that recycles nutrients back into the soil.

2.4.1 Factors governing residue decomposition

Decomposition of organic materials and the release of carbon dioxide from native soil organic matter or decaying litter is the result of complex interactions between the microbial population and various factors. The major factors include the quantity and quality of substrate, moisture, temperature, soil pH, soil texture and management practices (Alexander, 1977).

2.4.1.1 Plant (substrate) characteristic

The rate of decay of added organic materials is affected by their major constituents (Rice *et al.*, 2004). It is known that materials with high contents of lignin degrade very slowly due to the paucity of organisms that degrade it and the more resistant nature of lignin (Duncan, 1996). Similarly, plant materials high in polyphenols and tannins degrade more slowly than those with high contents of water soluble materials and cellulose (Taylor *et al.*, 1989). Plant residue is a complex mixture consisting of soluble sugars, free amino acids, proteins, cellulose, hemicellulose and lignin (White, 1979; Rice *et al.*, 2004). Typical components and their proportion within the whole tissue vary among plant species (Broder and Wagner, 1988) and within plant species (White *et al.*, 2003). The residue of different plant species can decompose at different rates. According to reports by Kaboneka *et al.* (1997), 48%, 56 % and 60 % of wheat straw, corn stover and soybean stubble were mineralized during a 30 day incubation period, respectively. Thus, the chemical and physical characteristics of crop residues affect their decomposition rate.

The age of a plant affects its composition, and therefore its rate of decomposition. As the plant ages, its chemical composition changes; the amounts of N, proteins and water soluble

substances decrease while the proportion of cellulose, lignin and hemi cellulose increase (Brady, 1990).

2.4.1.2 The nitrogen content of substrate

The nitrogen content of plant material has been shown to be an important factor controlling the rate of decomposition in many studies (Cowling and Merrill, 1966; Aber and Mellilo, 1980; Campbell, 1978). Residues that are low in nitrogen (N) but high in fibre are resistant to decay and therefore decompose more slowly than crop residues that are high in N and low in fibre. For example, corn cobs, corn stalks, sorghum stalks, soybean stems and sunflower stems are crop residues that are low in N, high in fibre and are slow to decompose even in warm moist soils (Vigil *and* Sparks, 2002). On the other hand, the leaves of corn, sorghum, soybean and wheat are all relatively high in N and low in fibre and so decompose rapidly.

There are also differences among species of grasses regarding the amount of nitrogen that is available for decomposition. Vallis and Jones (1973) indicated that leaves and litter of legumes *Desmodium intorturm* cv Greenleaf and *Phaseolus atropurpureus* cv Siratro had similar N and lignin content. The N mineralized from the former was less than that from *P. atropurpureus* and therefore attributed it to a much higher polyphenol content of the *D. intorturm.* Bartholomew (1965) and Mahendrappa (1978) also demonstrated that the addition of elemental nitrogen to natural litter materials and incorporated crop residues respectively enhance their rate of decomposition. Thus, a high N content in litter facilitates N mineralization by encouraging a high rate of decomposition and ensuring that N mineralization exceeds immobilization by considerable extent (Haynes *et al.*, 1986). Increased nitrogen deposit may

temporarily enhance forest carbon sequestration in nitrogen-limited ecosystems, leading to a short-term carbon gain in net primary productivity.

Fertilization with nutrients sometimes increases rates of decomposition (Gill and Lavender 1983; Hunt *et al.*, 1988; Prescott *et al.*, 1992; O'Connell, 1994; Downs *et al.*, 1996; Hobbie and Vitousek, 2000). However, fertilization often has little or no effect on decomposition (Staaf, 1980; Pastor *et al.*, 1987; Hunt *et al.*, 1988; Theodorou and Bowen, 1990; Van Vuuren and Vander Eerden, 1992; Andren *et al.*, 1993; Prescott, 1995; Downs *et al.*, 1996; King *et al.*, 1997; Hobbie and Vitousek, 2000). Indeed it may even decrease the rates of decomposition (Gill and Lavender, 1983; Titus and Malcolm, 1987; Fog, 1988; O'Connell, 1994; Prescott, 1995; Berg and Matzner, 1997; Magill and Aber, 1998). Studies have shown that N addition speeded decomposition of low-lignin but not high lignin leaves (Hobbie, 2000).

2.4.1.3. Relative size of substrate

The relative size of a substrate is the ratio of the surface area to the weight. The relative size of plant residues affects their rate of decomposition and build-up of organic matter. Crop residues that are chopped and broken up by stalk choppers or tillage equipment have smaller particle sizes than residues that are left undisturbed. A study by Vigil and Sparks (2002) has shown that the breaking up of a crop residue into smaller particles sizes exposes more residue surface area to microbial attack. Thus, broken crop residues decompose faster than residues that are left intact. For example, wheat or millet stems are inherently smaller and have greater specific surface area of residue material than corn, sorghum or sunflower stalks. A given weight of the smaller wheat and millet stem residues decomposes faster than the same weight of corn, sorghum or sunflowers under the same conditions of soil temperature, water and tillage.

Crop residues that have also been chopped and tilled are more intimately mixed with the soil microbial population. These residues are physically in a more favorable position for greater microbial attack and should decompose faster than residues that are left standing and undisturbed.

2.4.1.4. Method and depth of placement of residue

The method of addition of plant residues to soil affects the rate of their decomposition and build-up of organic matter reserves. When residues are on the surface as a mulch, they often become dessicated and decompose more slowly than if they are incorporated (Parker, 1962; Brown and Dickey, 1970; Shields and Paul, 1973).

The rate of decomposition also varies with depth of placement. This is particularly true since depth affects temperature, aeration, and moisture conditions of decomposition. At greater depths, the rate of carbon dioxide production diminishes and little is volatilised at lower depths. This decrease in activity with depth corresponds to the drop in organic carbon level such that the proportion of the total carbon oxidised in a given time interval remains relatively constant (Campbell, 1978). Generally, less humus are left at shallow depths than at much lower depths as organic residues decomposed more rapidly. Burial of residues under wet, cold conditions (Kononova, 1966) or very dry conditions tends to preserve organic residues (Shields and Paul, 1973).

2.4.1.5 Environmental factors affecting residue decomposition

2.4.1.5.1 Temperature

Temperature is one of the most important environmental conditions determining how rapidly natural materials are metabolised. Individual microbial species and the biochemical

capacities of the population as a whole have temperature optima. The mesophillic bacteria actinomycetes and fungi have a temperature optima range of 0-45 °C while the thermophillic types have a range of 45-60 °C, (Alexander, 1977). Thus, a change in temperature will alter the composition of the species of the active flora, the total number of species and also the total number of microbial cells.

According to White (1979) decomposition rates increase in an exponential fashion, as described by an Arrhenius or Q10 type relationship, with increasing temperature across the range of temperatures occurring in most soil environments (e.g. $\leq$ 0-35 °C). Short-term laboratory incubations from numerous studies support this basic control on the metabolism of soil biota, whereby respiration typically increases by a factor of 2 or more for every 10° C increase in temperature (Paul and Clark, 1989). This response to temperature is the basis for many predictions that global warming will greatly increase C loss from many terrestrial ecosystems (Jenkinson *et al.*, 1991)

Brady (1990) also indicated that the decomposition of organic matter in general is accelerated in warm climates; a lower rate of decay is the rule in cool regions while within belts of uniform moisture condition and comparable vegetation, the average total organic matter and nitrogen increases from two to three times for each 10 °C decline in mean annual temperature. Hence decomposition is slower at lower than at higher temperatures. Increased temperature is associated with greater carbon dioxide release.

Appreciable organic matter decomposition can occur at 5 °C and probably at cooler values, but decomposition of plant residue is increased with progressively warmer conditions (Campbell, 1978). Moreover, increasing the temperature shortens the time required before maximum rate of carbon dioxide evolution is attained. Hence, since the composition of the

microbial flora varies from one locality to the other and is also altered even in single site treated with different plant residues, a single optimum for organic matter decomposition cannot be found. Rather, a wide range, 28 to 40 °C, has been postulated, (Alexander, 1977). Below 25 °C, the rate of decomposition will accelerate with increased temperature. The high rate of organic matter decline in tropical soils has been attributed to the higher temperatures than in the temperate regions. Above 40° C, organic matter decomposition again slows down, except where thermophillic organisms abound (Alexander, 1977).

Liski *et al.* (1999) also showed that carbon storage in soils of both high- and low-productivity boreal forests in Finland actually increased with increasing temperature, thereby putting to rest the idea that rising temperatures will enhance carbon losses from soils and trees and exacerbate global warming.

The effect of temperature on microbial activity is that activity increases with rising temperature until there is some interference with life processes of the microbes or unless the soil first becomes relatively dry. Microbial activity normally halts in dry soil (Bowman *et al.* 2002). The higher rates of decomposition above 35 °C are associated with higher rates of respiration. Carbon dioxide evolution continues to increase and remains higher at temperatures up to about 70 °C but the decomposition is probably more chemical than biological at temperature above 50 °C.

2.4.1.5.2 Moisture content of soil

Organisms differ in their response to the moisture content of their environment. In general, fungi and actinomycetes are relatively tolerant to low moisture potentials (White, 1979). According to Wilson and Griffin (1975), an active micro-flora is maintained down to a soil

moisture potential of approximately -1500 kPa while bacteria become inactive below -800 to 1500 kPa. Maximum microbial growth and activity require the presence of sufficient water and therefore decomposition of organic matter is very slow in dry soils. On the other hand, because oxygen is required in microbial metabolism, decomposition is faster under aerobic conditions. At very high soil moisture contents the rates of microbial activity and decomposition are decreased due to lack of oxygen (Yoshida, 1975). Thus, saturation of the soil with water impedes the diffusion of oxygen into the soil hence maximum decomposition occurs in soils that are near field capacity.

Vigil and Sparks (2002) noted that decomposition is slow at soil water contents with less than 40 % water- filled pore space (barely moist to the touch but not dusty dry) and stops in soils that are air dry. Glenn *et al.* (1993) also indicated that soil carbon decomposition is dependent on soil moisture and so dry soils are less likely to lose carbon. In swampy areas, the slow rate of decomposition results in the formation of peaty soils, containing very high amount of organic matter (White, 1979). Water–logging impede loss in organic matter with the accumulation of large amounts of organic acids as intermediates of the decomposition process.

2.4.1.5.3. Soil pH

Many changes in soil microbial populations and activities occur as soil pH changes. Characteristically, the population shifts from bacteria to actinomycetes and then to fungi as soil pH declines, although acid tolerances of individual species vary widely, (Alexander, 1980). Carbon mineralization is most rapid in neutral to slightly alkaline soils, (Alexander, 1977). Soil pH has little effect, except below 4 when the decomposition rate slows as in the case of mor humus and many upland peats, (White, 1979). The treatment of acid soils with lime accelerates

the decay of soil organic matter, (Edmeades *et al.*, 1981). Thus liming of acid soils enhances carbon dioxide volatilisation.

2.4.1.5.4 The carbon: nitrogen ratio of the soil

The ratio of the percentage of carbon to that of nitrogen is termed the carbon: nitrogen ratio, or the C:N ratio, which is the relative quantities of these two elements in fresh organic materials, humus, or in the whole soil body (Thompson, 1957). Considering the diversity of soil, climate conditions and the plant materials entering soils, it is truly remarkable that the C:N ratio of soil organic matter as a whole is quite stable and differs little from that of humus which ranges from about 8 10:1 (Miller *et al.*, 2004) to 10-12:1 (McGill *et al.*,1981; Juma and McGill,1986).

When organic materials with a C:N ratio of greater than 30 are added to soils, there is immobilization of soil nitrogen during the initial decomposition process. For ratios between 20 and 30, there may be neither immobilization nor release of mineral nitrogen. If the organic materials have a C:N ratio of less than 20 there is usually a release of mineral nitrogen early in the decomposition process (Thompson and Troeh,1978). During the initial stages of the decomposition of fresh organic material, there is a rapid increase in the number of heterotrophic organisms accompanied by a large evolution of carbon dioxide. If the C: N ratio of the fresh material is wide there will be net immobilization. As decay proceeds, the C: N ratio narrows and the energy supply (carbon) diminishes. Some of the microbial populations die because of the decreased food supply, and ultimately, a new equilibrium is reached (Tisdale and Nelson, 1966). The attainment of this new equilibrium is accompanied by the release of mineral nitrogen resulting in the final soil level having nitrogen higher than the original level. There

may also be an increase in the level of stable organic matter or humus, depending on the quantity and type of fresh organic material originally added. The time required for the decomposition cycle to run depends on the quantity of organic matter added, the supply of utilizable nitrogen, the resistance of the material to microbial attack (the amount of lignin, waxes and fats present) and temperature and moisture level in the soil, (Tisdale and Nelson, 1966). Alexander (1977) stated the critical C:N ratio for net N mineralization to occur to be less than 20 -30, whereas a C:N ratios greater than 30 would favour net N immobilization.

2.4.1.5.5. Cultivation /Tillage

The physical condition of a soil affects crop residue decay. Severe soil compaction caused by improper tillage or intense traction impedes both water and air movement into a soil. Consequently, if the soil is left in that condition for an extended period, decomposition will be less (Bowman *et al.*, 2002). Cultivation enhances organic matter destruction. This is because apart from chopping plants parts into smaller fractions, aeration is enhanced for fast microbial activity. Cultivation and tillage of soil tends to break down the structure so that organic matter in sterile pore is exposed to micro organisms and its decomposition rate is accelerated, (White, 1979). Frequent and intensive tillage often accompanied by increased soil erosion tends to accelerate decomposition and thereby reduces the amount of carbon dioxide absorbed by vegetation such that much of the carbon dioxide remains in the atmosphere.

Disturbance of soil aggregates by natural or anthropogenic (tillage) forces increases the decomposition of physically protected organic materials (Rice and Angle, 2004). Tillage practices that conserve soil structure are important for increasing soil carbon storage and retaining sequestered carbon. Long-term studies elsewhere showed that a no-tillage practice

accumulated more soil carbon compared with tilled soils and the amount of carbon stored was also related to the amount of precipitation on the site and crop rotation (Fabrizzi *et al.*, 2004).

2.4.1.5.6. Texture and mineralogy

Texture affects drainage and aeration. Thus, because of the high moisture content and relatively poor aeration of poorly- drained soils, organic matter and nitrogen content are generally much higher in them than their better drained equivalent. Generally, soils high in clay and silt are able to protect the protein nitrogen from degradation through organomineral complexes that are formed, which then result in a high organic matter content of the soil (Nichols, 1984). Decomposition and humus formation is therefore low in coarse- textured soils than in fine- textured soil (Brady, 1990). Thus, fine textured soils contain about twice as much total organic matter as do sandy soils (Brady, 1990).

According to White (1979), the adsorption of various compounds by clays and sesquioxides generally serves to slow down their rate of decomposition. The organic matter held in the relatively stable pores in clay soils of diameter $< 1\mu m$ is less accessible to microbial attack. Positive correlations between soil organic carbon and clay contents have also been observed (Schimel *et al.*, 1985; Spain, 1990; Feller *et al.*, 1991).

Soil texture and mineralogy affect the micro and macro structure of soils and the binding forces in soils that enable the formation of mineral-organic matter complexes in soil (Hassink, 1996). Since all primary plant compounds are susceptible (to varying degrees) to breakdown by enzymes produced by soil micro organisms, the role of soil minerals in 'protecting' organic matter (by restricting its accessibility to enzymatic action) is crucial in stabilizing organic matter in soil over longer periods of time.

It's been well accepted that soil texture is a key variable affecting soil organic carbon (SOC) stocks– sandy soils have low organic matter contents and SOC stocks tend to increase with increasing proportions of silt and clay (Burke *et al.*, 1989). Greater attention in the past has been placed on organo-mineral complexation due to negative charges on clay surfaces that enable an electrostatic binding to negatively charged organic colloids via positively charged metal cations (e.g. Ca^{2+}, Mg^{2+}). Thus, 2:1 clays (e.g. smectites), have more negative surface charge,and more effectively stabilized organic matter compared to more low charged clay minerals such as 1:1 clays (e.g. kaolinite) (Hassink *et al.*, 1997).

2.5 Processes involved in organic matter decomposition

The dynamic nature of soil organic matter cannot be over emphasized. Plant residues undergo extensive alteration in the soil before they become humus. Various types of micro organisms attack the residues and decompose their constituents. The residues serve as a source of nutrients and energy for the life processes of the micro organism. As plant materials are added to the soil, as much as two-thirds of the carbon may be lost to the atmosphere as CO_2 in a single season as a result of decomposition (Feng and Li, 2001a). Subsequent decomposition, however, slows resulting in accumulation of stable organic carbon in soils. The gross turnover time of soil organic carbon, expressed as the ratio of total amount of organic carbon in the soil at steady state to the annual rate of addition, can be more than 20 years in temperate regions (Stout *et al.*, 1981).

Residues added to the soil are first broken down to their basic organic components by the extracellular enzymes produced by heterotrophs (White, 1979). The number and type of flora

involved in the decomposition process depends on the type, quantity and availability of the organic matter (Campbell, 1978).

Each individual organism has its own complex enzymes which allows it to decompose certain chemical compounds. The first group of heterotrophs (the primary flora) attack the basic components of the added carbonaceous substances: these are succeeded by the secondary flora which thrives on the cells and by-products of the primary flora. Addition of simple sugars causes proliferation of bacteria, starch stimulates actinomycetes, cellulose benefits fungi while the proteins and amino acids influence spore-forming bacilli (Alexander, 1961). The water-soluble material decomposes first followed by cellulose and hemicellose at equal rates while the lignin fractions become the most resistant and tend to accumulate in the soil (Alexander, 1961). Thus, the percentage of available carbon in a substrate decreases with time while the percentage of resistant materials increases (White, 1979). There is an increase in lignin percentage in the substrate with time regardless of the environment. An increase in microbial cell constituents also occurs. There is an apparent decrease in OH groups and OCH_3 (in lignin) and an increase in COOH groups which are reactive and responsible for cation exchange (McBridge, 1994). Cation exchange capacity of decomposing material increases soil nutrient content.

There is also a change in the elemental composition of the decomposing material during decomposition. The mineralization of carbon is unique because its end-products (carbon dioxide and methane) are volatile and so can be lost to the atmosphere (Thien and Graveel, 1997). The N:P ratios tend to increase as decomposition proceeds, except in cases where nitrogen products are volatile so that only a small percentage remains. On the other hand, higher litter nitrogen concentrations generally lead to higher initial rates of CO_2 loss from decomposing plant litter. This might be viewed as a negative outcome, but over the longer term it appears that initially

enhanced loss of CO_2 from the soil-litter ecosystem is more than compensated for by enhanced carbon savings stimulated by the addition of nitrogen (Hu *et al.*, 2001).

After a quarter to a third of the original plant litter has decomposed and has disappeared, decomposition tends to become slower where there is a higher N concentration in the litter. A reason for the deceleration has been attributed to an increase in concentration of the stable lignin. Also, several species of fungi with the ability to decompose lignin via lignin-degrading enzymes do not seem to be able to produce the necessary enzymes in the presence of plentiful N-rich compounds. This failure to synthesize the enzymes may be related to a scarcity of manganese, the concentration of which often has been observed to decline as soil nitrogen availability rises (Berg and Matzner, 1997).

The C:N ratio of plant material entering the soil may vary from 10 for green legumes to more than 50 for straws. (Thompson and Troeh, 1978). The C: N, C: P, C: S ratios therefore decrease with time until equilibrium is reached. According to Larson *et al.* (1972) the C: N, C: P, N: P and N: S ratios increased with increasing rate of residue addition while the S: P ratio remained relatively constant. The equilibrium level is therefore determined by the composition and demands of the microbial cells. While a high rate of organic matter decomposition is a good index of microbial activity, it may not correspond to maximum number of micro organisms. Studies indicate that the maximum number of microbes is between 300 and 350 (Thompson, 1957).

2.6. Contribution of cereal and legume residues to carbon sequestration

The chemical compositions of various crop residues differ greatly in the amount of carbon dioxide produced when added to the soil. Studies by Abdurahman *et al.* (1998) illustrate that even within one crop group, large differences in organic matter production occur. The

studies showed that dry leaf production from pigeon pea yielded 3 t ha^{-1} whilst that of cowpea produced only 0.14 t ha^{-1}. This shows that the choice of crop has a major influence on how much carbon can be sequestered by an agricultural system. Curtin *et al.* (2000) demonstrated the advantage of cereals over legumes for achieving maximum carbon sequestration. According to them whilst black lentil fallow in semi-arid regions of Canada added between 1.4 and 1.8 t C ha^{-1}, a wheat crop would add 2-3 times this amount of carbon annually.

Works done by Grant and Bailey (1994) also indicate that residues from cereal crops contain much more carbon than nitrogen and may stimulate micro organism to tie up nutrients. In contrast, legume residue has more nitrogen, which favours nutrient release by micro organisms. Of the plant residue returned to the soil, about 15 % can be expected to be converted to passive soil organic carbon (Lal, 1997). Gregorich *et al.* (1998) reported that although the chemical composition of the plant residue affects its rate of decomposition there is little effect on the organic matter formed.

According to Hu *et al.* (2001), the key factor responsible for enhanced carbon storage in grassland sites is the high carbon input derived from plant roots. It is this high root production that provides the potential to increase soil organic matter in pastures and vegetated fallows compared to cropped systems. Root debris tends to be less decomposable than shoot material because of their higher lignin content (Woomer *et al.*, 1994). Consequently, the key to maintaining and increasing carbon sequestration in grassland systems is to maximise grass productivity and root inputs (Trumbore *et al.*, 1995).

Grasses have also been shown to sequester more carbon than leguminous cover crops (Lal *et al.*, 1999). Hence, because of the high C: N ratio of grasses, their residues decompose slowly and soil nitrogen availability may be substantially decreased following their

incorporation into the soil. Grasses also have the potential to sequester carbon on previously degraded land. Garten and Wullschleger (2000) using a modelling approach estimated that a 12 % increase in soil carbon could be obtained under switchgrass (*Panicum virgatum L.*) on a degraded land for ten years.

2.7 Techniques for estimating carbon sequestration potential

Rapid and accurate measurement of carbon in soil samples is essential to evaluate the amount of carbon that can be sequestered in soils and for modelling global change. Two options are available to this purpose, direct experimental measurement and monitoring and predictions with soil organic carbon models (Bruce *et al*,. 1999). To be of practical value to the producers and farmers, these changes need to be evaluated over relatively short periods, from a few years to a decade. Changes of soil organic carbon, however, is slow and occurs over much longer time periods (Bruce *et al*., 1999).

Direct measurement and monitoring of soil organic carbon changes over short periods must deal with uncertainties of sampling and measurement errors, and more importantly, uncertainties resulting from non-uniformity of field soils. The most direct means of determining soil carbon sequestration is to measure over time sequential changes in soil carbon. Such measurements are complicated by the spatial and temporal heterogeneity of soil carbon contents and its slow relative rate of change.

Soil carbon can exhibit significant field-scale variability due to spatial varying topography and parent material or past differences in vegetation or management history. Even in seemingly 'uniform' fields, soil carbon contents can vary by as much as 100 % (Elliott, 1994). In addition, the seasonality of plant growth and decomposition processes can cause temporal variability in carbon fractions, in roots, plant litter and microbial biomass. Finally, short-term

changes in total carbon can be difficult or impossible to detect against the large 'background' of carbon already present in the soil.

Many of these problems can be overcome through the use of well-designed sampling and analysis procedures that minimize effects of spatial and temporal variability and standardize sample preparation (Bruce *et al.*, 1999). Measurement of specific soil carbon fractions may be useful as early indicators of change, although it may still be necessary for monitoring periods of several years to verify changes in total soil carbon. Most data on soil carbon changes in agricultural soils are derived from long-term field experiment (Paul *et al.*, 1997). Where treatments have been properly randomized, significant differences in soil carbon as a function of different agricultural management practices can be statistically inferred.

Prediction based on models validated against available experimental evidence is another option. Models are used to investigate how soil organic matter varies across regions and landscapes as a function of climate, vegetation, topography, soils and other environmental factors. The most widely used simulation models conceptualize soil organic matter as being composed of 3 to 4 fractions, which vary in physical and chemical properties affecting turnover rates (McGill, 1996). Crop residues are similarly subdivided into 2 to 3 fractions varying in decomposability.

The environmental factors controlling soil organic matter in most models include soil temperature and moisture, soil aeration or drainage class, soil texture and mineralogy. The rate of organic matter input as crop residues, manure, sludge, or other amendments is a major determinant of soil carbon levels. Some models simulate crop growth and residue inputs directly, while others require that organic matter addition rates be specified as inputs to the model. Intergovernmental Panel on Climate Change guidelines addresses empirical modeling.

The use of soil organic matter models range from the field level (Jenkinson *et al.*, 1987) to regional (Parton *et al.*, 1987) and global applications (King *et al.*, 1996).

Thus, in formulating models, the following must be noted: The model must be easy to use, based on sound theoretical description of soil organic carbon processes and contains only parameters that are physically meaningful and experimentally measurable. The models must also be able to reproduce both the quick decomposition of plant residues within a single season and the very old organic carbon deposits in soils with which it could be considered valid for predictions in both the short term changes in soil organic carbon, i.e., a few years to decades, and for predictions in much longer term stabilization of soil organic carbon (Bruce *et al.*, 1999).

2.8 Modeling residue decomposition

Residue decomposition depends on the biochemical fractions of the material (Heal *et al.*, 1997). The concentrations of nutrients, structural carbohydrates, and other compounds (i.e., lignin and other polyphenols) ratios have been used as indices of biochemical quality. More specifically, great efforts have been devoted to develop a residue quality index that best describes C and N residue release rates. For example, in incubation studies, total N concentration (Frankenberger and Abdelmagid, 1985) or its inverse (Quemada and Cabrera, 1995) were reported to be the best indices for C and N residue release rates of legume and grass residues. Others identified soluble C (Oglesby and Fownes, 1992; Kuo and Sainju, 1998), cellulose (Bending *et al.*, 1998), or lignin (Müller *et al.*, 1988; Giller and Cadisch, 1997) to be most closely related to residue decomposition or C and N mineralization rates. Furthermore, some ratios, such as lignin to N (Vigil and Kissel, 1991) or polyphenol plus lignin to N (Constantinides and Fownes, 1994), have also been used as indice of residue nutrient release. Mechanistic models such as Century (Parton *et al.*, 1994) use the lignin/N ratio to partition

residue biomass into easily decomposable (soluble carbohydrates and proteins) and recalcitrant (fibres and lignin) pools.

The biochemical components controlling residue decomposition change with time. Soluble nutrients are more relevant at earlier decomposition stages and structural carbohydrates or lignin at later stages (Heal *et al.*, 1997). Consequently, the length of the decomposition period being analyzed will determine which fractions have more control or are more relevant in residue decomposition. The C: N ratio (C: N) is the most widely used index of residue quality and predictor of decomposition rate (Heal *et al.*, 1997). However, the use of the initial C:N of the residues does not consider the availability of these nutrients for microbial growth. Consequently, it has failed to be a reliable predictor of decomposition or, mineralization (Smith *et al.*, 1992; Honeycutt *et al.*, 1993; McKenney *et al.*, 1995). Vigil and Kissel (1995) concluded that N mineralization parameters were estimated poorly by C: N, especially when C: N ranged from 10 to 28.

Gilmour *et al.* (1998) also concluded that decomposition rate variations among years and type of residues were not related to crop species, year, N content, and /or C: N. These authors intimated that the variability in the kinetic parameters needs to be explained. It is therefore accepted that dynamic models that include a more detailed description of decomposition of the various chemical compounds are needed to improve prediction of C and N turnover (Dendooven *et al.*, 1997; Heal *et al.*, 1997). Earlier report suggests the use of C and N concentration in the residue soluble fractions as a better indicator of residue C and N release processes (Cochran *et al.*, 1980; Reinertsen *et al.*, 1984; Henriksen and Breland, 1999).

Bowman (1990) described a progression in the efforts to model plant residue decomposition and soil organic matter dynamics. The earliest models of litter decomposition and

soil organic matter dynamics were first-order decline functions. According to Swift *et al.* (1979) this type of model is useful in the description of rapidly decomposing leaf litters and for readily metabolisable substrate additions such as carbohydrate. An advantage of this mathematical approach is that the exponential decline coefficient (k) can be compared between substrates and sites when this coefficient is calculated on a relative basis (litter remaining/ initial litter) and the units of time are the same.

Work done by Jenny *et al.* (1949) and Greenland and Nye (1959) indicate that the first-order exponential decline functions assume a constant decline in absolute terms. Swift *et al.* (1979) also showed that many coniferous and broad leafed litter materials are well described by this decline function, as are the individual components of the litter material. A difficulty with this approach is that while the function is asymptotic, the remaining material approaches zero at the same proportional rate as does the increment of decomposed material, not allowing for the effect of a more recalcitrant fraction as decomposition progresses. The assumption is that all added organic materials become entirely mineralized at the same rate throughout the decomposition process.

The K-Model developed by Feng and Li (2002) takes a different approach. The K model recognised that carbon dynamics in soils can be represented by three basic processes. Thus, plant material entering the soil is divided into compartments with different rate constants to account for both the initial, fast decomposition and subsequent slow decomposition (Jenkinson and Rayner, 1977; McGill *et al.*, 1981; Van Veen and Paul, 1981; Parton *et al.*1983, Parton *et al.*, 1987; Jenkinson, 1990; Smith *et al.*, 1997).

Plant residue added to the soil, including manure and other organic materials, is characterized by a metabolic fraction of relatively fast decomposition and a more resistant

structural fraction. The relative quantities of the two fractions are determined by the C: N ratio of the plant residue. For example in Roth Carbon model (Jenkinson, 1990), soil organic carbon is divided into decomposable and resistant plant materials (representing the annual input into the soil) and biomass and physically and chemically protected soil organic matter. The rate constants used to describe these compartments range from 4.5 /year for decomposable plant material to 3.5-10 /year for resistant soil organic matter.

Particularly the Century model has been used widely in climate change compartments. Frissel and Van Veen (1981) developed an N transformation model based on four stages of organic matter transformation. The stages used in the model are: (1) C: N ratio controls mineralization and immobilization. (2) Consideration was made for differences in decomposition rates of organic compounds in plant residues for amino acids, cellulose, lignin fractions and microbial mass (in this stage the authors assumed that the organic matter decomposition is controlled by C uptake by the biomass rather than the C/N ratio). (3) N transformations were incorporated into a multi-layer soil model. (4) Soil organic matter (SOM) fractions were incorporated into the C and N pool.

The model was based on the assumption that soil organic matter can be represented by several carbon and nitrogen pools. The biomass growth rate was controlled by the carbon availability from the added soil pool, and it was assumed that there was no change in the microbial population if no carbon was added to the soil. Nitrogen immobilization was proportional to biomass growth and it was assumed that mineralization occurred simultaneously and independently of immobilization. The authors used the C:N ratio to characterize mineralization and immobilization. They assumed that if the C:N ratio was less than 20 or 30, then net mineralization occurs; otherwise, net immobilization occurs (Donigan, 1994).

2.9. Importance of organic matter

The significance of soil organic matter to soil fertility is that it influences so many different soil properties. It is a dual source and a sink for nutrient elements which can form organic moieties (Mulongoy and Merckx, 1991). It has physical and chemical properties which facilitate aggregation with mineral particles, particularly clays, and in turn modify soil physical structure and influence soil water regimes. It is a source of energy for the soil biota and thus influences many of the biologically-mediated processes of soil. Thus, soil organic matter itself represents a set of attributes rather than an entity.

Organic matter content is higher under grassland vegetation than under forest cover (Hu *et al.*, 2001). Some of the functions of organic matter are that in fine textured soil it helps to maintain good soil structure (Allison, 1973). It also increases the cation exchange capacity, thereby reducing leaching losses of elements such as potassium, calcium and magnesium. Report by Allison (1973) indicate that the organic matter of most mineral soils accounts for about 30-65 % of the total cation exchange capacity (CEC). For instance, in sandy and organic soils, more than 50 % of the CEC is likely to be due to the organic component of the soil. Allison (1973) stated that the organic matter of different soils vary greatly in their CEC. The more humified the organic matter the higher its CEC. Grim (1953) also gave values for kaolinitic, illitic, vermiculitic and montmorillonitic clays as 3-15, 10-40, 100-150 and 80-150 c mol/kg, respectively. Organic matter serves as a reservoir for soil nitrogen and improves soil-water relation; its mineralization provides a continuous though limited supply of nitrogen, phosphorus and sulphur to the crop.

CHAPTER 3

MATERIALS AND METHODS

3.1. Experimental site

3.1.1 Location and physiography

This study was conducted within the framework of an on-going experiment to assess the contribution of 7 fallow management systems involving different plant species to soil carbon storage within a maize-cropping system at Kpeve (Lat, 6 ° 43.45'N, Long 000 ° 20.45'E) in the Volta Region of Ghana, and also within a similar cropping system at the University of Ghana Farm, Legon (Lat 5° 39'N and Long 0° 11'W). The location of the project at Kpeve falls within the Forest-Savanna transition zone which receives about 1400 mm rainfall annually. The vegetation consists of scattered trees and grasses such as elephant grass. At the University Farm, Accra, annual rainfall received is between 635-1143 mm and the ecological zone is the Coastal Savanna.

3.2 Soils and sampling

The soil from Kpeve is classified as Haplic Lixisol (WRB, 1998) which has a dark greyish brown top soil and greyish brown to brown sub soil. The soil contains abundant small to large sized quartz stones and moderate amounts of low activity clay, (Adiku *et al.*, 2003 unpublished). The soil was sampled from a 0-20 cm depth at the Experimental Site of the Ministry of Food and Agriculture Station at Kpeve in the Volta Region.

The soil from the University of Ghana Farm, Legon is classified as Haplic Luvisol (WRB, 1998). Morphologically, the soil is deep to very deep, varies from red to brown, moderately heavy to medium-textured soil and devoid of concretions and gravel at least to 0.6 m

from the surface. The soil has developed from weathered products of tertiary sands (Brammer, 1962). The soil, among others, is good for both mechanized and hand cultivation in the coastal savanna areas of Ghana (Acquaye and Laryea, 1982; Ahenkorah *et al.*, 1994). Both the soil at Kpeve and the University Farm are well drained and contain large amounts of low activity clays.

The soils sampled from Kpeve were air-dried, roots and other debris removed and then lightly ground and passed through a 2mm sieve. The sieved soil samples were stored in sacks and kept in cupboard for further use. The physico-chemical properties of the soil are determined as described below.

3.3 Determination of physico-chemical properties of the Haplic Lixosol

3.3.1 Soil texture

The various particle fractions were determined using the hydrometer method (Bouyoucos, 1962). Forty (40) grammes of the fine earth fraction of the (< 2mm) soil was dispersed in a 5 % 100-ml sodium hexametaphosphate solution prepared by dissolving 50g of calgon in a litre of water.

The suspension was allowed to stand for about 10 min followed by mixing for 5 min with a Vortex mixer after which the suspension was transferred into a sedimentation cylinder and brought to 1 Litre mark wht the addition of distilled water. The suspension was allowed to equilibrate and the initial temperature taken. A plunger was inserted close to the bottom of the cylinder and the suspension stirred vigorously by moving it up and down several times (about 10 times). Timing was started immediately with a stopwatch and the hydrometer reading was taken at 5 mins and at 5h from the time of mixing the suspension. The sand fraction was recovered by decantation and the dry weight recorded after it had been oven-dried for 2 days and cooled in a

dessicator. The clay and silt fractions were determined by the difference in the 5 mins and 5hr readings. The percentage clay and silt were estimated by the fraction contained in the amount of soil taken. The textural class of the sand, silt and clay were determined using the USDA textural triangle.

3.3.2 Field capacity determination

A 1300 g soil sample was weighed into a 1.2 L plastic pot with drainage holes underneath. The soil sample was packed in the pot to an average bulk density (D_b) of about 1.3 kg/m^3 which approximated that of the field soil and about 1000 ml of water was added until the soil became saturated and began to drain from the bottom. The wet soil was allowed to drain for 48 hours, while the surface was covered with plastic sheets to prevent evaporation. Thereafter, the gravimetric water content (θ_g) of the soil on was determined after oven drying at 105° C for 24 hours according to the formula

$$\theta_g = \frac{Mw}{Ms} \quad g/g \qquad (3.1)$$

where: Mw = Water in soil (Initial weight of soil when moist - Oven dry weight) / g

Ms = Weight of oven dry soil/ g

The results obtained multiplied by the bulk density (D_b) gives the volumetric moisture content (θ_v), D_w = Density of water. (Phogat *et al.*, 1999).

$$\theta_v = \frac{\theta_g * D_b}{D_w} \qquad (3.2)$$

3.3.3 Soil pH

Soil pH was measured in 1:1 (soil: water) suspension using the electrode MV 88 Praitronic pH meter. A 20 g soil sample was weighed in a 50-ml beaker and 20 ml of distilled water added. The mixture was stirred with a glass rod for 30 minutes and allowed to stand for 1 hour. The pH of the suspension was read on the electronic pH meter and recorded as pH in 1:1 soil: water ratio.

3.3.4 Organic carbon

The Walkey - Black method as modified by Allison (1965) was used to determine the organic carbon content of the soil. Potassium dichromate (1N, 10 ml) solution and 20 ml of concentrated (98 %) sulphuric acid (H_2SO_4) were added to a 0.5 g soil (which had been passed through a 0.5 mm sieve) in an Erlenmeyer flask. The flask was swirled round and allowed to stand for 30 minutes and 200 ml of distilled water was added, mixed and allowed to cool. The residual dichromate remaining in solution after the oxidation of the oxidizable organic material in the soil sample was titrated against 0.2 N ammonium ferrous sulphate solution after 10 ml of orthophosphoric acid (85 %) and 2 ml indicator solution (barium diphenylamine sulphate) have been added and titrated to a green end-point. A blank in which the same procedure was followed but without any soil sample preceded the soil as a check. The carbon content (OC) was calculated as follows.

$$\% \ OC = \frac{(10 - X * N)}{W} 0.3 \ * \ 1.33 \qquad (3.3)$$

Where X =Volume of ferrous ammonium sulphate solution titrated.

N = Normality of ferrous ammonium sulphate solution (10/ Volume of ammoniun sulphate titrated with the blank.

W = Weight of soil sample taken.

0.3 = milliequivalent weight of Carbon

1.33 = correction factor. Walkey and Black averaged 77 % recovery of organic carbon (OC) by this method and introduced this correction factor.

The percentage organic matter (OM) was estimated by multiplying percent organic carbon by 1.724, the Van Bemmelen factor.

3.3.5 Total nitrogen

The Kjeldahl method (Hesse, 1971) was used to determine total nitrogen. A 2 g soil sample was put into a micro Kjeldahal flask and 1g of digester accelerator (10g K_2SO_4 + 1g $CuSO_4.5H_20$ and 0.1 g selenium) added. About 1 ml distilled water was added to moisten the soil and 5 ml concentrated Sulphuric acid also added.The flask was put on a digestor and the mixture was allowed to digest for at least two hours until the digest became clear. It was then allowed to cool and then transferred with distilled water into a 50 ml volumetric flask and made up to the volume. A 5 ml aliquot was put into a Markham distillation apparatus and 5 ml of 40 % NaOH was added and distilled. The distillate was collected into a 5 ml (2 % boric acid) to which about three drops of a mixture of methl red and methelene blue indicator solution had been added. The distillate was titrated with 0.01 N HCl from a green to an indicator reddish end point. Total Nitrogen was calculated using the formula.

$$\% \text{ Nitrogen} = \frac{N * X * 50 * 0.014 * 100}{W * V} \qquad (3.4)$$

N= Normality of HCl used. (N)

X= Volume of HCl used for the titration (ml)

V= Volume of filtrate (aliquot used for the distillation (ml)

W = Weight of soil used for the digestion (g)

3.3.6 Exchangeable Bases

10 g soil was weighed into an extraction bottle and 100 ml of 1N ammonium acetate solution was added. The mixture was shaken for one hour after which the content was filtered through a No. 42 Whatman filter paper. Aliquots of the extract were used for the determination of Ca^{2+}, Mg^{2+}, K^{+} and Na^{+}.

Calcium was determined by taking 10 ml aliquot of the sample solution and adding 10 ml of potassium hydroxide and 1ml of 1N triethoanolamine. About three drops of potassium cyanide (10 % w/v) solution and a few crystals of cal-red indicator were added. The mixture was then titrated with 0.02N EDTA solution from red to a blue end point.

Exchangeable Ca and Mg were determined by pipetting 10 ml aliquot of the sample solution and adding 5ml of ammonium chloride -ammonium hydroxide buffer solution and 1ml triethanolamine. Potassium cyanide solution and Erichrome Black T solution (0.2 g Erichrome Black T indicator dissolved in a mixture of 50 ml methanol and 2g hydroxylamine hydrochloride) of about three drops each were also added. The mixture was then titrated with 0.02 N EDTA solute ion from red to a blue end point. The amount of exchangeable Mg was estimated as the difference between this titration and that of Calcium.

Exchangeable Na and K were determined using the flame photometer by calibrating the photometer with standard 10 ppm Na^{+} and K^{+} solutions and reading the Na^{+} and K^{+} concentrations of the extractant.

3.3.7 Cation exchange capacity

A 10 g soil sample was put into an extraction bottle and 100 ml of 1N ammonium acetate solution was added. The bottle with its content was shaken for 30 mins on a mechanical shaker. The content was filtered through a No.42 Whatman filtered paper. The sample was then leached four times with 25 ml of 0.01N methonol to wash off excess ammonium. Another 25ml portion of acidified potassium chloride was used to leach the soil four times. An amount of 5 ml of the leachate was transferred into a Kjeldahl flask, and 5ml of 1N sodium hydroxide and some distilled water were added. The distillate was collected into 5 ml boric acid and then titrated with 0.01N hydrochloric acid from greenish to a violet end-point. The cation exchange capacity (CEC) was calculated using the principle of normality and the value expressed in c mol $^{(+)}$/kg soil.

3.3.8. Available phosphorus

The available phosphorus was determined according to the method by Bray and Kurtz (1945). A 10 g soil sample was put into an extraction bottle and 50 ml of extractant (0.03N NH_4F in 0.025N HCl) was added and shaken for about two minutes on a mechanical shaker. It was filtered and an aliquot of 5 ml was used to develop the colour using the Murphy Riley (1962).method

A 20 ml aliquot of the sample was put into a 50 ml volumetric flask. The pH was adjusted by adding drops of *p-nitro phenol* indicator and few drops of 4 N NH_4OH until the solution turned yellow. Then 2 ml of reagent B (1.056g of ascorbic acid in 200 ml of reagent A) was added. Reagent A was made by dissolving 12 g of ammonium molybdate in 250 ml of distilled water and adding 0.2998 g of antimony potassium tartarate. The dissolved reagents

were added to 1000 ml of 5N H_2SO_4 (148 ml conc.H_2SO_4 per litre) and mixed thoroughly and made up to 2 L. The solution was made up to volume with distilled water, and a blank was also prepared using the same procedure but without the soil. The spectrophometer was calibrated using standard phosphorus solution by pipetting 5 ml of the standard phosphorus solution (5 ppm) into a 50 ml volumetric flask and adding 2 drops of ammonium solution and *p-nitro phenol* solution and 8 ml of Reagent B and distilled water to develop the colour. The intensity of the colour at a wavelength of 712 nm was measured with the spectrophotometer and recorded.

$$\text{Available phosphorus/mg/kg} = \frac{V * (R1-R2)}{W1 * V1} \qquad (3.5)$$

V = Volume of extractant used

$R1$ = Sphectrophotometer reading for the aliquout used

$R2$ = Sphectrophotometer reading for the blank.

W = Weight of soil used.

$V1$ = Volume of aliquot taken for the reading.

3.4 Green house residue incubation studies

3.4.1 Introduction

Residues of different fallow plants from the fallow management experiment at Kpeve briefly described in Table 3.1 were used for the incubation studies. The fallow plants described in Table.3.1 were planted at the onset of the minor season of September 2003 after the major season (May to June) maize. Only treatment RT5 benefited from the residual fertilizer of the previous maize crop. In all other cases, the previous maize was unfertilized. The fallow plants

were harvested in March 2004 and kept in a cold room until their use in this study in June 2004. Treatment RT6 is included as the control.

Table 3.1. Description of plant residues used in the incubation study

Designation of Residue	Description
RT1	*Pennisetum spp* (elephant grass) obtained from natural- fallow following unfertilized maize.
RT2	*Cajanus cajan* (Pigeon pea) residue following unfertilized maize
RT3	*Vigna unguiculata* (Cowpea) residue following unfertilized maize
RT4	*Mucuna pruriens* (mucuna) residue following unfertilized maize
RT5	*Pennisetum spp* (elephant grass) following fertilized maize.
RT6	Residue free (control)

3.4.2 Plant residue

The fallow residues (essentially made up of leaves and stems) were chopped into about 2-3 cm lengths and thoroughly homogenized. The materials were oven dried in brown paper bags at a temperature of 60 °C for 72 hours until samples attained constant weight. Thereafter, a portion of each residue type was ground for the determination of oxidizable carbon, nitrogen and phosphorus. The carbon content of the plants was determined following the wet oxidation method of Walkey –Black as modified by Allison (1965) but in this case, 0.2 g of ground plant residues was used. The nitrogen content (0.2 g sample) was determined using the Kjeldahl

method. Total phosphorus content was by digesting 0.2 g of plant sample with 5ml concentrated H_2SO_4 and further oxidized with H_2O_2 (35%) reagent on a sand bath. The digest was thoroughly mixed with distilled water and made up to 100 ml. 5 ml of the solution was taken for colour development using the Murphy and Riley (1962) method.

3.4.3 Pot experiment

Eight hundred gram sieved (2 mm) soil samples were packed into 1.2 L pots to the same bulk density as stated in section 3.3.2. The pots had holes at the bottom to allow free drainage and uptake of water from below. Litter bags of 10 cm x 6 cm size were prepared from mosquito netting and 8 g of dried residue samples were placed in them. The plant residues in their litter bags were then placed horizontally at a depth of 1 cm below the soil surface in the pot. The soils in the pots were brought to three moisture levels; field capacity (FC) = W1, 70% FC = W2 and 40% FC = W3 and thereafter maintained by periodically weighing the pots and re-wetting. The treatments were placed in the greenhouse at an average temperature of 30 ^{0}C with a maximum of 32 ^{0}C and minimum of 28 ^{0}C.

The experimental design was completely randomised design with 6 residue types and 3 water treatments. Residues from the pots were retrieved in triplicates 10, 20, 30, 50, 80, 120, 150 and 180 days after the onset of the incubation studies. Thus, in total there were 432 pots in all. At each retrieval, the litter bags were emptied and the dry weight of the material remaining determined after oven drying at 60 °C for 72 hours. Further, the soil in the pots after each litter bag retrieval was homogenized and analysed for carbon and total nitrogen content.

3.5 Field studies

A field study was carried out at the University of Ghana farm from July-September 2005 to compare the results in the green house with that in the field. The fallow management at the University farm was similar to that at Kpeve. However, the soil at the University Farm was different (Haplic Luvisol) from that of Kpeve. Further, soil moisture content and temperature could not be controlled on the field. The pH, CEC, Available P, Organic carbon and nitrogen properties of the soil were also determined.

Plant residues consisting of *Pennisetum spp,* RT5 (Elephant grass following previously fertilized maize), *Cajanus cajan,* RT2 (Pigeon Pea following unfertilized maize), *Mucuna pruriens,* RT4 (*Mucuna spp.* following unfertilized maize) and a control RT6 which was a bare plot kept weed-free were the treatments used. Plant residue treatment for *Vigna unguiculata* (Cowpea, RT3) was not available at the on-set of the experiment and so could not be considered. The plant residues were from the previous minor season (October 2004 planting) harvest in June. Plant residues grown in situ from an estimated plot area of 1 m^2 were used for the study.

Unlike the greenhouse, fallow vegetation was planted in situ in the field. Plants growing in-situ were harvested and their biomass determined. The biomass were equivalent to 2000 kg/ha, 2200 kg/ha and 3500 kg/ha for RT2, RT4 and RT5 respectively. The field studies involved the incorporation of 350 g plant residues into litter bags made of 1 mm nylon netting which were 72 cm in length and 42 cm in width. Plant residues were chopped to about 30 cm length and buried 1 cm below the surface of the soil. The experimental lay out was Randomised Complete Block Design with 4 replicates.

The litter bags were retrieved 10, 20, 30, 50 and 80 days after the onset of the field study. On retrieval, the litter bags containing the plant residues were thoroughly shaken to remove all

soil particles and the contents emptied into brown paper bags. The residues were oven dried at 60 °C for 72 hours and weighed. Organic carbon content of the soil taken at a depth of 0-15 cm on the day of retrieval of the residue was also determined. The soil of the control (bare plot), was sampled along side the other treatments on residue retrieval dates.

3.6 Statistical analysis

The data were subjected to analysis of variance using the Genstat software. The significance of the treatments effects were tested at 5 % level of probability. The Least Significant Difference (LSD) was used to separate the means of the treatments.

CHAPTER 4

RESULTS AND DISCUSSION

4.0 Greenhouse studies

4.1 Soil physical and chemical properties

Table 4.1 shows some physical and chemical properties of the Haplic Lixisol from the Kpeve site. Soil texture is sandy loam as is typical of soils of the Togo series (Brammer, 1962) and with a fairly high water holding capacity of 0.23 g/g. Drainage and aeration would be expected to be high in such a soil. Particle size distribution shows a 15 % silt and 20 % clay content which are quite low. The texture is more of the coarse type (Brady, 1990), hence organic carbon content would also be low as the protein nitrogen are not well protected from degradation and drainage.

Soil pH of 7.5 is between neutral and slightly alkaline and therefore expected to support nutrient availability by increasing rate of decomposition since low pH retard decomposition, (White, 1979). The dominance of low activity clays (Adiku *et al.*, 2003 unpublished) may explain the low CEC value of 26.72 cmol $^{(+)}$ /kg and the inherent fertility, even though the available P of 13.48 mg/kg is fairly high for tropical soils. According to Manu *et al.* (1991), CEC for West African soils depends more directly on soil organic carbon ($r = 0.86$) than on soil clay content ($r = 0.46$). Organic carbon content of (1.62 %) coupled with the low exchangeable bases particularly calcium also reflect the generally low fertility of the soil. The low organic carbon content of (1.62 %) is also typical of most Tropical soils (Ankomah *et al.*, 1995). The total nitrogen content of (0.08 %) is also quite low and together with a C:N ratio of $\approx$ 20.3, decomposition rates of added materials could be fairly high.

Table 4.1 Some physical (a) and chemical (b) properties of Haplic Lixisol *

(a) physical

Particle size distribution			Moisture content at field capacity g/g	Textural class
Sand %	Silt %	Clay %		
65	15	20	0.23	Sandy loam

(b) Chemical

pH	OC	TN	Avail.P	Exchangeable bases (c mol$^{(+)}$/kg)				
(1:1)	(%)	(%)	mg/kg	Ca^{2+}	Mg^{2+}	Na^{+}	K^{+}	CEC
7.5	1.62	0.08	13.48	13.9	2.7	0.89	1.18	26.72

pH 1:1 Soil pH measured in 1:1 soil: water ratio CEC: Cation Exchange Capacity

OC % : Organic Carbon % TN %: Total Nitrogen %

Avail. P: Available Phosphorus mg/kg

* : All values are means of 3 observations

Microbial growth in the soil requires the C: N: P ratios to be within the range of 120:10:1. for biodegradation of organic substances (Alexander, 1977; Kowalenko, 1978). For the Haplic Lixisol, the C: N: P ratio of 16.2: 0.8: 0.0135 is quite good. Hence, microbial growth in such a soil is expected to be quite favourable for decomposition processes.

4.2 Some chemical characteristics of the plant residues

Table 4.2 shows some chemical properties of the plant residues from the Kpeve site. The results indicate that the oxidizable carbon content (C) of the residue is between 334.9 g/kg and 470.3 g/kg with the least value obtained for RT4 (M: *Mucuna spp* following unfertilized maize) and the highest, RT1 (EG: Elephant grass following unfertilized maize) as shown below

Table 4.2:

Some chemical properties of plant residues obtained from kpeve.

Residues type Description	Carbon g/kg	Total Nitrogen g/kg	Total Phosphorus g/kg	C: N	C: P
RT1 (EG)	470.3	15.8	43.0	29.77	109.37
RT2 (PP)	404.6	20.3	53.0	19.93	76.34
RT3 (CP)	420.7	17.9	47.0	23.5	89.51
RT4 (M)	334.9	17.9	41.0	18.71	81.68
RT5 (EFF)	401.6	16.1	40.0	24.94	100.4

(EG = Elephant grass; PP = Pigeon Pea; CP = Cowpea; M = *Mucuna spp.*, following unfertilized maize) and EFF = Elephant grass following fertilized maize.

It has been widely reported that the total carbon concentration in plant tissue is very stable and close to 400 g kg^{-1} (Honeycutt *et al.*, 1993; Kuo *et al.*, 1997). The values for carbon in this study are close to this figure. Woomer *et al.* (1994) also indicated that on the average, crop residues contain between 40-50 % carbon which is close to the range of 33.5- 47 % carbon content present in these plant residues. Although RT1and RT5 are grasses, the high carbon

content of RT1 more than RT5 may be due to more structural polysaccharides nature of the cell wall.

The total nitrogen (N) content of the plant residues also ranges from 15.8 g/kg to 20.3 g/kg with RT1 (EG) having the least value and RT2 (PP) the highest value respectively. Generally, the legume residues unlike the grasses tend to have more N content due to the nitrogen fixing ability of the plant. This is through a symbiotic association with the nitrogen fixing bacteria, Bradyr*hizobium*, in their root nodules (Brady, 1990). The N content of the residues particularly the legumes are however low as compared to the 2 % stated by Fox *et. al.* (1990) for decomposition.

The C:N ratio of the plant residues also ranges from 18.71- 29.77 with the lowest value obtained for RT4 (M) and the highest for RT1 (EG). The C: N is dependent on the proportion of carbon and nitrogen present in the substrate as stated by Cowling and Merrill (1966). The higher the proportions of N present, the lower the ratio, while the greater the carbon content, the higher the C: N ratio. The legume materials RT2 (PP), RT3 (CP) and RT4 (M) have lower C: N than the grasses; RT1 (EG) and RT5 (EFF), because of the high N content of the plant obtained from the nitrogen fixing bacteria in the root nodules (Brady, 1990).

It could also be noted that there is variation within the legumes. RT3 (CP) and RT4 (M) have the same N content of 17.9 g/kg (or 1.79 % N) yet their ratios are different. RT2 (PP) has a high N content of 2.03 %, yet it's C: N ratio is more than RT4 (M). The difference in the C: N of the RT3 (CP) and RT4 (M) could be attributed to their carbon content. In addition, considering the morphological nature of the plant materials, the constituents of cowpea cell wall are of more structural polysaccharides which have a relatively stable carbon concentration (Chesson, 1997) unlike the mucuna which is herbaceous and therefore had less structural

polysaccharides (Brady,1990). Thus, the proportion of carbon present in the former will certainly be higher than the latter. The slightly lower C: N ratio of the grasses; RT5 (EFF) than RT1 (EG) may be attributed to the high N %, probably due to the residual fertilizer obtained from the previously fertilized maize plot.

Generally, Buckman and Brady (1969) noted that the C:N ratio of plant material is variable ranging from 20:1 or 30:1 for legumes and farm manure to as high as 90:1 or even more, in certain straw residues and to as high as 400:1 in sawdust. According to Thompson and Troeh (1978) the break-even point for decomposition of organic material is 32:1 while White (1979) stated that C: N ratio > 25 results in net immobilization and < 25 net mineralization occurs. Hence, materials with narrow C: N ratios tend to decompose about 50 % faster than those that are deficient in nitrogen. Ease of decomposition would therefore be RT2 >RT4. According to Fox *et al.* (1990), nitrogen content of plant material should be greater than 2 % for mineralization to proceed while when less than 2 % immobilization generally occurs. This tends to have an important implication for the decomposition processes and nutrient release by different kinds and age of plants. From Table 4.2, it will be only RT2 (PP) which would satisfy this requirement.

Thompson (1957) indicated that P content of organic material is approximately 0.5 % and therefore by this the values obtained (0.4-0.53 % P) for the various residues are reasonable. Generally, the P content of legumes tends to be more than the grasses in agreement with the report of Maikslenien (2000) that cereals remove relatively little phosphorus from the soil. This high P content of legumes may be linked to the ability of its roots to probably have more phosphorus solubilizing bacteria around the root zone and therefore making it more accessible

for the plant to use and conserve and also due to the fact that P is used for nodulation. (Brady, 1990)

It could also be observed that the C: P ratio of the plant residue was high for the grasses with values of 100.4 and 109.37 for RT5 (EFF) and RT1 (EG) respectively whereas the legume residues had lower values of between 76.34 and 89.51. The wide C: P ratio of the grasses is expected to cause immobilization of phosphorus and therefore limit nutrient availiablity. The C: P ratios of the plant residues also show a similar trend like that of the C: N. Lower values of 76.34 to 89.51 were obtained for the legumes and higher values of 100.4 and 109.37 for the grasses. No clear explanation could be given but it could be attributed to the low P content of the grasses and the high carbon content with the exception of the *Mucuna spp* (M) in this case. A study by Nziguheba (2001) reports C: P ratio for dry matter of crop residue to be in the range of 140 -250. This shows that the values obtained were much lower as compared to the values of Nziguheba (2001). It is expected that the C: P ratio like the C: N may also have some important effect on the decomposition of the plant materials although more emphasis is laid on the C: N ratio in this study.

4.3 Decomposition of residues during greenhouse studies

Residue decomposition is affected by many factors such as temperature, moisture, soil pH, substrate quality and quantity. At the greenhouse where temperature of the environment was monitored and substrate quantities were also the same, the effect of the other variables such as residue (type) and moisture could be assessed.

4.3.1 Effect of residue type

Figure 4.1 shows the variation of residue dry weight with time for water treatment W1 (field capacity) for (a) grass residue and (b) legume residue; observed data are symbols and fitted

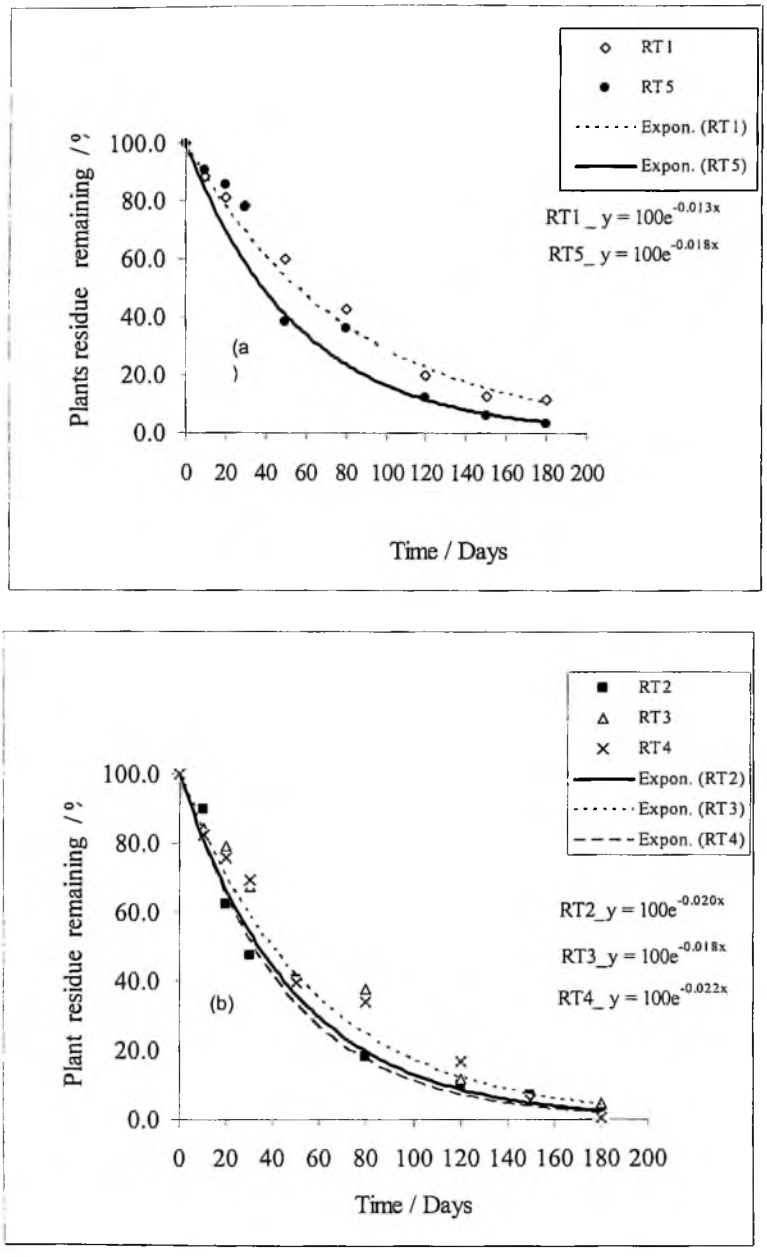


Fig.4.1: Variation of residue dry weight with time for water treatment W1 (a) Grass residue (b) Legume residues; observed data are symbols and fitted curves are lines.

curves are lines. Generally, both graphs are curvilinear and decomposition pattern for the grass (a) seems to be quite slower than for the legumes.

In order to assess the effect of residue type, three dates were selected namely, the 20^{th}, 80^{th} and 150^{th} day to conform to two commonly observed phases of decomposition namely, the initial fast decomposition stage and the subsequent slow decomposition stage (McGill *et. al.*, 1981; Parton *et. al.*, 1987; Jenkinson, 1990; and Smith *et al.*,1997). The results from this study show that for the first 20 days of incubation, decomposition patterns of the grasses; RT1 (EG) and RT5 (EFF) were somewhat similar with Fig.4.(1a), having each lost 15 % of their intial dry weight, with soil water treatment W1. The legume residues generally showed a faster decomposition pattern than the grasses as in Fig 4. (1b). Dry weight lost for RT3 (CP) and RT4 (M) were 21 % and 24 % respectively while RT2 (PP) showed a much faster loss of about 38 % of the original weight by the 20th day of incubation. This trend is in agreement with the general view that lower C: N ratios lead to faster residue decomposition. These results also suggest that residue N availability is more critical in controlling biomass decomposition. (Ruffo and Bollero, 2003)

Significant differences in decomposition patterns became evident with time. It may be deduced from Fig.4.1 (a) and (b) that the days to 50 % loss of weight (half- life) was 58 days for RT1 but only 38 for RT2 as shown in Table 4.3.

The rest of treatments RT3, RT4 and RT5 had half lifes values between 34 to 40 days. By 80 days of incubation, RT1 (EG) lost a little over 50% of its residue while RT2 (PP) lost almost 75 %. The other plant residues lost about 60 % of their initial weight by 80 days of incubation. By 150 days, a great proportion of all the residues had decomposed so that there was no significant difference between residues remaining, irrespective of type. This indicate that

as decomposition proceeds to a certain level, the plant materials irrespective of type tend to decompose at similiar rate thereby approaching that of humus.

Figures 4. 2 and 4.3 also show the variation of residue dry weight with time for water treatment W2 (70 % field capacity) and W3 (40 %field capacity) for (a) grass residue and (b) legume residues, respectively. Residue decomposition patterns for the W2 and W3 water treatments followed the same pattern described for W1 in section 4.3.1, but with this, decomposition was much slower.

The grass residues RT1 (EG) and RT5 (EFF) in Fig.4.2 (a) show quite a similar trend of decomposition. By 20 days, 11.3 % and 10.9 % of RT1 (EG) and RT5 (EFF) have decomposed respectively. By 80 days, 45.5 % and 41.5 % had decomposed and similarly, by 150 days, 84.3% and 81.3 % had decomposed. This probably indicates that, there is no significant difference between RT5 and RT1 when the moisture content is slightly low as at W2. Thus explaining the one line curve for the two plant residue even though RT5 may have benefited from the residual fertilizer.

Fig.4.2 (b) did not also show much variation among the treatments. By 20 days, 25.9 %, 21.3 % and 30.3% of RT2 (PP), RT3 (CP) and RT4 (M) had decomposed respectively showing a faster decomposition than the grasses. By 80 days, 60.9% of RT2, 75.6 % of RT3 and 61% of RT4 had decomposed. These losses in weight of residue seem to be slightly higher than the W1 legumes although values did not differ much except for RT3 (CP) which showed a greater loss in weight. By 150 days, 89.5% of RT2 (PP), 85.6% of RT3 (CP) and 90.9 % of RT4 (M) had decomposed.

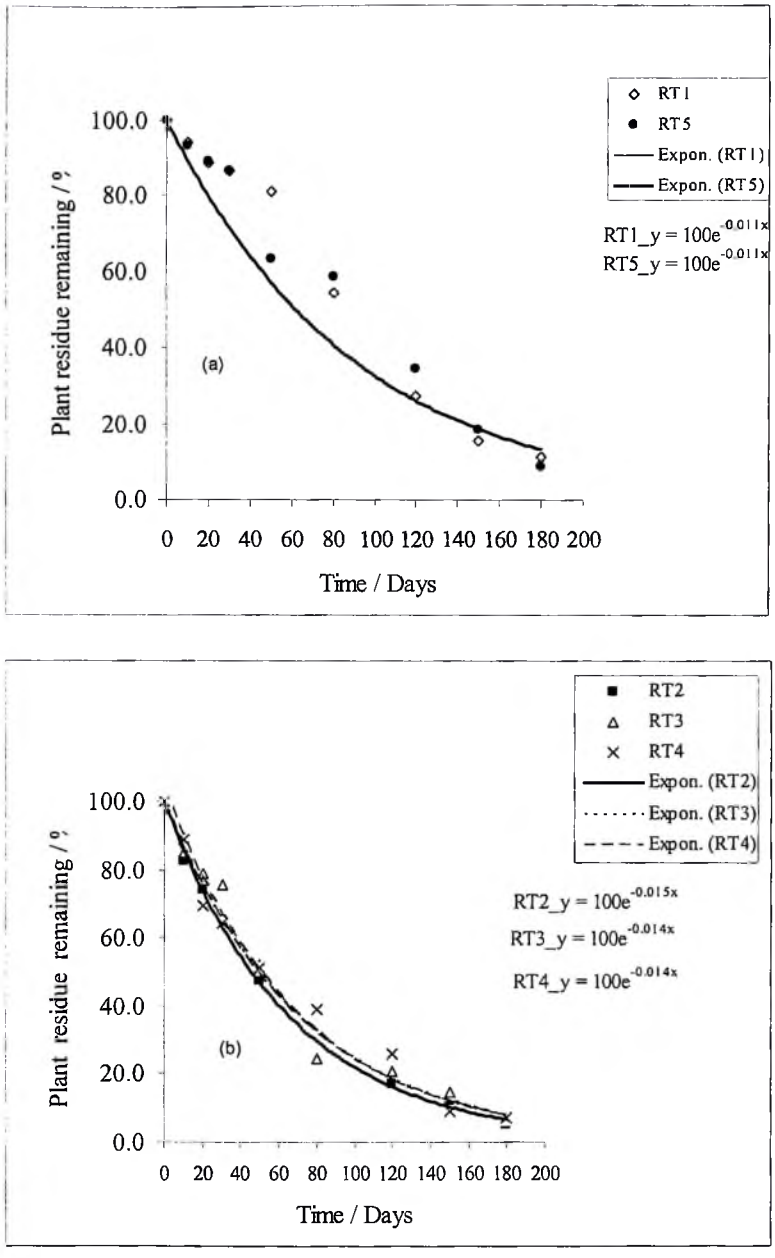
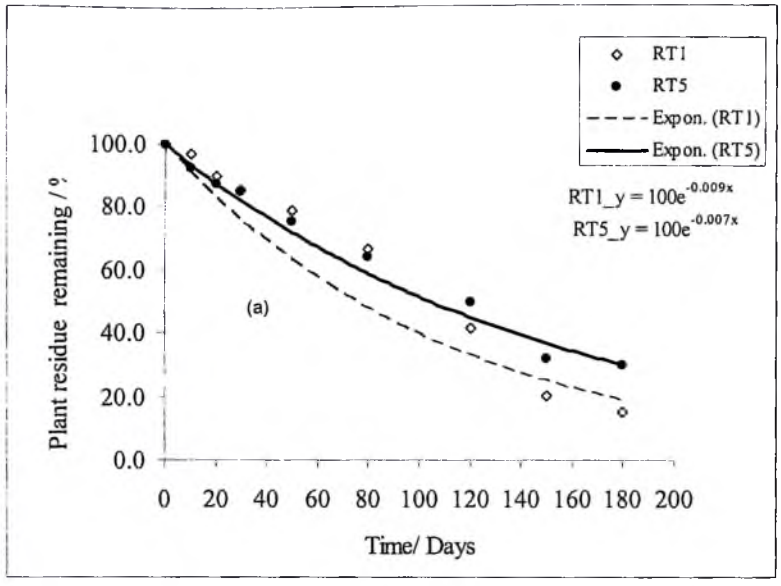


Fig.4.2: Variation of residue dry weight with time for water treatment W2. (a) Grass residues (b) Legume residues; observed data are symbols and fitted curves are lines.



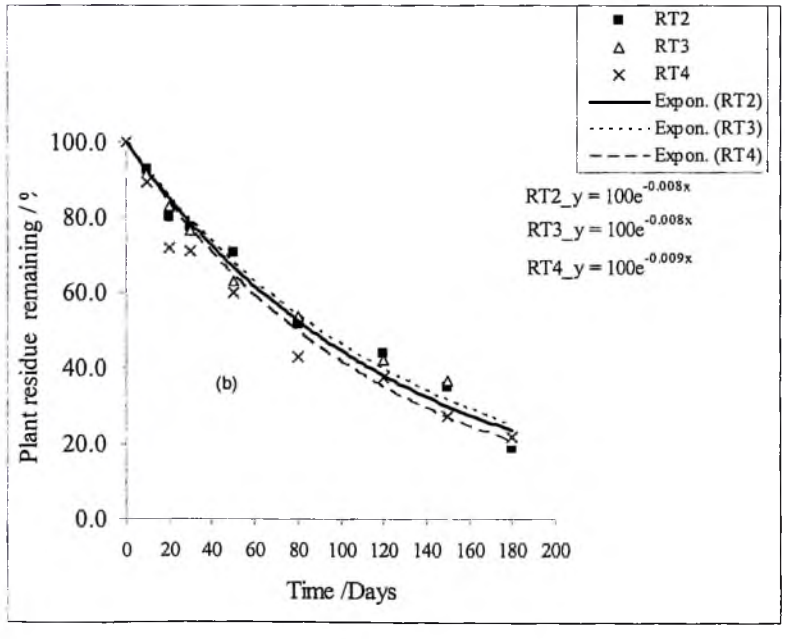


Fig.4.3: Variation of residue dry weight with time for water treatment W3 (a) Grass residues (b) Legume residues; observed data are symbols and fitted curves are lines.

In Fig 4.3 of W3, RT2 (PP) was the least decomposed while RT1 (EG) was the fastest to decay. The slow decomposition of RT2 (PP) could be attributed to a greater portion of the insoluble part of the plant material present in the residue despite its high N content of 2.03 %. It may also be due to the low moisture content of the soil inhibiting decomposition of the woody parts. The fast decomposition of RT1 (EG) was unexpected as its nitrogen content was low and so decomposition was expected to be slow. Hence, the fast decomposition could be attributed to an unknown factor other than nitrogen or phosphorus contents.

The observed differences in residue decomposition patterns may be attributed to the differences in C:N ratios. The C:N ratio of organic matter in cultivated surface soil range from 8:1 to 15:1 and this is relatively uniform among different soils within a climatic region (Brady,1990). According to Thompson and Nelson (1966), heavily leached soils are likely to have higher C:N ratios of 30:1 while forest soils have C:N ratio of 20:1. A C:N ratio > 25 will result in immobilization and mineralization will occur when C:N ratio drops below 20:1. Considering the fairly high C:N ratio of 20:1 of this soil, decomposition was expected to be slow as the low soil nitrogen content of 0.08 % would limit microbial growth leading to an initial immobilization of nutrients (Fox *et al.*, 1990). Thus, the addition of a plant material high in nitrogen or with low C:N ratio will imply the microbes will resort to use the nitrogen in the residue to build up their population, produce more enzymes to speed up decomposition and subsequently, decomposing the material at a faster rate (Aber and Melillo, 1991).

In contrast, grass residues like RT1 (EG) with a high C:N ratio of 29.77:1 are expected to tie up their limited nutrients in the soil as nitrates leading to the priming effect of nitrate depression in the soil. The microbes would therefore not have access to it and therefore may not be stimulated to grow to increase activity and cause decomposition (Grant and Bailey, 1994).

Decomposition was therefore expected to be slow. It is in this regard, that decomposition of plant residues follow the order; RT2 > RT4 > RT3> RT5 > RT1 with time, with the legumes residue obviously decomposing faster than the grasses. The order of decrease is related to the lower C: N ratios of the plants as stated earlier.

At 80 days of incubation, there was no significant difference between the legume decomposition, nor was there any difference between the legumes and RT5 (EFF). Thus, even though RT5 was an elephant grass, the apparent benefit it derived from the residual fertilizer after maize resulted in a lower C: N ratio than RT1 (EG) and hence a faster decomposition rate. By 150 days, a great proportion of all the residues had decomposed so that there were no significant difference among residues remaining, irrespective of type.

It is of interest to note that the rate of decrease of plant residues in soils is influenced not only by the C: N ratio but other residue components such as lignin and polyphenol content (Melillo *et al.*, 1982; Taylor *et al.*, 1989). But since these were not determined, the discussion is limited to C: N ratios alone as it plays a major role in the decomposition process.

Table 4.3 below shows the half-life for the various residue types at the different water treatments derived from Figures 4.1, 4.2 and 4.3. Half life in this case was regarded as the number of days for half of the plants residue to have been decomposed.

Table 4.3 Half –life of residue dry weight (Days) for the different water treatments

	Half –life for water treatments (Days)		
Residue treatments	W1	W2	W3
RT1 (EG)	58_a	62_a	80_a
RT2 (PP)	38_b	48_a	85_b
RT3 (CP)	40_b	50_a	84_b
RT4 (M)	34_b	50_a	76_c
RT5 (EFF)	40_b	62_a	104_d

* Figures bearing same subscripts in each column are not significantly different

Results from the table indicate that at W1, half-life for RT1 (EG) was 58 days, 38 days for RT2 (PP) and only 34 days for RT (M). The rest of the treatments RT3 (CP), and RT5 (EFF) had half-lifes of about 40 days. RT4 (M), RT3 (CP) and RT5 (EFF) also appear to be similar while RT1 (EG) seems to be significantly different from the rest. As moisture content decreases to W2, differences in the half-lifes were not significant for both the grasses and the legumes. The residues may be said to be decomposing at similar rate regardless of the type. At W3, differences between the residues were quite evident with RT1 (EG) decomposing even faster

than the legumes RT2 (PP) and RT3 (CP). It implies that despite the high C:N and C:P ratios of the EG, decomposition was still high, hence the results. RT2 (PP) and RT3 (CP) had almost the same number of days while RT5 (EFF) took 104 days for half of its residue to decompose with RT4 (M) having only 76 days. The lower number of days for RT4 (M) agrees with the results of Brady (1990) and Larson *et al.* (1972) on fast decomposition for lower C: N ratio residues.

4.3.2 Effect of soil water on residue decomposition

It is known that apart from plant factors, residue decomposition also depends on environmental factors such as temperature and soil water, among others (Olson, 1963). With temperature being generally uniform in the greenhouse, soil water was the dominant factor for consideration. Figures 4.1, 4.2 and 4.3 show the decline of residue dry weight with time not only for the various plant residues treatments (RT) but also at soil water treatments W1, W2 and W3. Comparisons between the figures show very important features.

First, decomposition patterns show high probabilities for the higher soil water treatments (W1: Fig. 4. 1) than for the drier soil (W3: Fig.4.3). Secondly, the total dry weight loss at 180 days for each residue type was higher in W1 than W2 and W3. At the end of an incubation period of 120 days, all residue types lost more than 80 % of their initial dry weights in W1 while in W3 water treatments, some residues like, RT2, RT3 and RT5 still had more than 40% of their initial dry weight still remaining. These observations agree with the findings of Glenn *et al.* (1993) that decomposition rate decreases as moisture content of soil reduces. Vigil and Sparks (2002) also observed that, soil moisture must be near field capacity for decomposition to occur, and that at soil moisture less than 40 % of field capacity, decomposition rates become very slow.

Thirdly, as shown in Table 4.4, the half –life (days to 50 % weight loss) of the residue decomposition vary considerably with water treatments; W1, W2 and W3. Results indicate that for W1, half life of RT1 (EG) was 58 days but this increased to 62 days for W2 and 80 days for W3. In RT2 (PP), half life increased from 38 days in W1 to 48 days in W2 and to 85 days in W3. It is worth noting that although half-life for RT3 (CP) at water W1 and W2 were different, the difference was not significant ($P \leq 0.05$). Statistical analysis of the data indicates that the effect of water treatments on half -life was highly significant ($P<0.05$). As can be observed decomposition takes longer time as moisture contents of soil reduces for each residue type. Significant differences in decomposition patterns became evident with time.

Fourthly, the order of decomposition in W3 was also altered with RT1 showing not only relatively faster rate of decomposition but also higher loss of plant material by day 120 than all the other treatments. The order of decomposition at W3 was RT1> RT4> RT2 > RT3 > RT5. No clear explanation for this observation could be given, but it can be suggested that as the soil was slightly dry, only low microbial population could be supported and so, the composition could be altered to favour those that may survive on low N but require high C and P. In that case, RT1 residue may be a more suitable substrate.

However, according to Berg and Matzner, (1997), several species of fungi with the ability to decompose lignin via lignin-degrading enzymes do not seem able to produce the necessary enzymes in the presence of abundant N-rich compounds. This failure to synthesize the enzymes was attributed to a scarcity of manganese, the concentration of which often has been observed to decline as soil nitrogen increases but, considering the nitrogen content of the soil (0.14%) being low, the fungi-concept would fail in this regard.

Table 4.4 Half –life of residue dry weight (Days) for three water treatments

	Half –life (Days)				
	Residue treatments				
Water treatments	RT1	RT2	RT3	RT4	RT5
W1	58_a	38_a	40_a	34_a	40_a
W2	62_b	48_b	50_a	50_b	62_b
W3	80_c	85_c	84_b	76_c	104_c

* Figures bearing same subscripts in each column are not significantly different

Table 4.5. Effect of soil water on the decay constants (*k*) g/d for various plant residues obtained from fitted equation.

	k-values				
	Plant residue type				
Water treatments	RT1	RT2	RT3	RT4	RT5
W1	0.013_a	0.020_a	0.018_a	0.022_a	0.018_a
W2	0.011_a	0.015_a	0.014_a	0.014_b	0.011_a
W3	0.009_a	0.008_b	0.008_b	0.009_c	0.007_a

Figures bearing same subscripts in each column are not significantly different.

Table 4.5 also shows the effect of soil water on the decay constants (k) g/d for the various residue types. As was observed for the half-life, decay constants decline with soil dryness especially for RT2, RT3, RT4 and RT5.

A general trend of decline in the decay constants can however be observed as seen in the Table. 4.5. From W1 to W3, RT2 (PP), RT3 (CP), RT4 (M) and RT5 (EFF) decay constants declined by 0.6%, 0.56%, 0.59 % and 0.61 % respectively. The table also indicates that for W1, decay constant for RT1 (EG) was 0.013 g/d but this decreased to 0.011 g/d for W2 and to 0.009 g/d for W3. This shows a 0.31 % decline from W1 to W3 and therefore a considerably slowing down of the decomposition process. This implies less material decomposing and therefore less carbon dioxide will be released as compared to the rest of the treatments.

Values for the legumes residues seem to be quite higher than the grasses for W1 and W2. Thus, rate of residue decomposition is generally faster for the legumes than for the grasses. Statistical analysis indicates that the effect of water on the treatments for the decay constants was significant ($P< 0.05$) for some of the residue treatments. Generally, the difference between water treatments W1 and W2, W2 and W3 was very small as compared to that between W1 and W3.

It is important to note that as observed from the rate constants (k), decreasing order of decomposition at W1 is RT4 > RT2 >RT3, RT5 >RT1. At W2 order of decomposition was RT2> RT3> RT4>RT5>RT1. At W3, the RT1 had a high value of 0.009 g/d like that of RT4 compared to the rest of the treatments. Rate of decomposition was in the order RT1, RT4 >RT2, RT3, >RT5. Thus if desired to store carbon in agricultural plant residues, then they should be applied in very dry soils at moisture content of 40 % field capacity or less.

4.4 Modelling the dynamics of residue decomposition

The dynamics of carbon in soils, residue decomposition, C and N release rates are often complex and can be improved with models that include biochemical fractions and their interactions (Henriksen and Breland, 1999). Feng and Li (2002) advanced the K-model, recognizing that carbon dynamics in soils can be represented by three basic processes: the initial attack of the plant residue by soil microbial population, the growth and death of the soil microbial biomass and the decomposition of the dead soil microbial biomass residue. The model focuses on the fraction of added residue which remains un-decomposed at any time and observes that this fraction decreases continuously with time but at a diminishing rate, as shown in Figs. 4.1 to 4.3. Gordillo and Cabrera (1997) in an incubation study proposed a two-pool first-order kinetic model to describe N mineralization in broiler litter.

The most common approach to develop a predictive model of decomposition or mineralization rates based on residue quality has been to relate decomposition parameters to the different residue biochemical fractions either by multiple regression (Müller *et al.*, 1988; Trinsoutrot *et al.*, 2000) or correlation (Thomas and Asakawa, 1993; Bending *et al.*, 1998). Since the different biochemical fractions are highly correlated (Müller *et al.*, 1988; Kuo and Sainju, 1998), multiple regression by ordinary least squares is not appropriate for the estimation of parameter coefficients due to the presence of multicollinearity among the predictor variables. When multicollinearity exists among the predictor variables, the variances of the parameter estimates are inflated and statistically unstable (Dillon and Goldstein, 1984; Johnson and Wichern, 1998). In addition, the results are difficult to interpret and very sensitive to the inclusion or lack of inclusion of specific variables or to small changes in data points (Dillon and Goldstein, 1984).

Accordingly, most residue decomposition models employ double exponential decay equation, with two decay constants, describing the fast and slow rates respectively. Using the curve-fit software and following SAS (1991), an equation of the form

$$Y = A^{*}\exp^{(-k1t)} + B^{*}\exp^{(-k2t)} \quad (4.1)$$

where Y = weight of plant residue (g) remaining at t (days). *A, B*, k1 and k2 are constants fitted to data on residue and water treatments.

The results in Table 4.6 indicate the dependence of the decay constants k1 and k2 on water for a given residue type. The values for the various residue types and soil water treatments were obtained from the fitted equation above. Results from the data indicate that k1 which is the initial fast decomposition does not differ much from the k2 which is the slow decomposition stage. There is also a general decline in the k1 and k2 values as water content of soil decreases except for RT2, RT3 and RT4 treatments which have an unusual high k1 values at W3 and particularly RT2 at W3 for the k2 value. The exceptionally high value obtained for the k1 at W3 seem to imply that, rate of decay was obviously faster for the legumes than for the grasses. These observed values at W3 seem to contradict the earlier conception that decomposition tends to decrease as moisture content reduces.

Decay constants (k1 and k2) for the grasses; RT1 and RT5 seem to maintain some form of consistence at their respective water contents. There is however no significant difference between the two pools for all the water treatments. This lack of signifiacant difference may be attributed to an increase in surface area of the plant residues resulting from the cuttings. Studies by Vigil and Sparks (2002) indicate that chopping plant residue to smaller sizes hasten decomposition process and therefore this may have resulted in the lack of difference between the two pools k1 and k2. Another probable factor could also be the high temperture of 30 ^{o}C

which might have hastened the decomposition process. The small amount of residues imputed may also be a contributing factor to the pools.

Table 4.6 Effect of soil moisture; W1, W2, W3 on the decay constants; k1and k2 (g/day) pools for the various residue types obtained from fitted equation.

Water treatments	W1		W2		W3	
k values	k1	k2	k1	k2	k1	k2
Residue type						
RT1	0.012	0.012	0.010	0.010	0.008	0.008
RT2	0.032	0.015	0.014	0.014	0.076	0.076
RT3	0.015	0.016	0.014	0.014	0.056	0.006
RT4	0.015	0.016	0.013	0.013	0.059	0.007
RT5	0.012	0.012	0.012	0.012	0.010	0.010

It is evident from the foregoing discussions that the decay rate constant depend on both the type of residue and the soil water, hence any dynamic description must at least consider these effects. In a study by Ruffo and Bollero (2003) first-order exponential single-pool decay model was used to analyze biomass decomposition, carbon and nitrogen residue released with time either expressed as degree- day with base temperature 0°C (Honeycutt and Potaro, 1990) or decomposition –day (Stroo *et al.*, 1989; Steiner *et al.*, 1999). Thus, it would be appropriate to use a single pool to describe this model. Even though it is known that temperature exerts

tremendous effects on decomposition rates through Arrhenius relations, it is not a major variant in the tropics in comparison to soil water. For simplicity, simple decay functions are used to describe the decomposition in this study. A simple exponential equation of the form below is proposed.

$$Y = Ae^{-kt} \qquad (4.2)$$

Y = dry weight of plant residue remaining un-decomposed in grams, A = initial dry weight of applied residue, k = decay constant (g/day), e is the base of the natural logarithm and t is time/day. Soil water effect on the k was highly significant with $P < 0.05$ (appendix 5 to 7). Using the above equation 4.2, another equation can be formulated using a k_{eff} which depends on soil water.

The exponential equation derived to predict residue decomposition under variable soil water is:

$$Y_r = Ae^{keff(\theta)t} \qquad (4.3)$$

Where Y = dry weight of plant residue remaining un-decomposed, A = initial dry weight of applied residue in grams, k_{eff} = effective decay constant, r is residue type, θ is soil water g/g, e is the base of the natural logarithm and t is time/day.

Assuming that the optimum soil water for decomposition is θ_{FC} (corresponding to field capacity) and that at air dry soil water content, θ_d, decomposition ceases, then at any given soil water content θ, k_{eff} may be expressed as:

$$k_{eff} = k_{FC}\left[\frac{\theta - \theta_d}{\theta_{FC} - \theta_d}\right] \qquad (4.4)$$

with k_{FC} being the decomposition rate constant for field capacity obtained from W1 for each residue type as shown in Table 4.5. Soil water beyond field capacity may incapacitate the

growth of microbes that require good aeration for decomposition. This aspect has not been considered in equation (4.4) but can be easily incorporated. The effect of residue type *r* on decomposition rate may not lend itself to direct formulation but *r* classes can be derived based the on C: N ratios.

Figures 4.4 and 4.5 show predicted patterns for the residue treatments (RT) using the k_{eff} value calculated at soil water content (θ) of 0.16 g/g (70 % FC) and 0.09 g/g (40 % FC). Air-dried soil (θ_d) of 0.019g/g was put into equation 4.4. The result obtained for the k_{eff} was then put into equation 4.3 to give the calculated plant residue dry weight.

Thus, figure 4.4 presents the variation of observed (symbols) and calculated (lines) plant residue dry weight for (a) Natural elephant grass; RT1 (b) Elephant grass from fertilized maize plot; RT5, (c) Pigeon pea; RT2, (d) Cowpea; RT3, (e) *Mucuna spp*; RT5 at 70 % field capacity. The calculated plant residue remaining in (a) and (b) suggest that grass had a higher proportion of the fast decomposing fraction which was reflected in a higher loss of the original dry weight during the 30^{th} and 50^{th} day for (a) and almost through out the period of incubation for (b), respectively. The observed values for (a) were slightly above the estimated for the first 30 days and slightly below the estimated values from the 120 day onwards. This indicate that possible modelling for decomposition of grass may be best fitted at the early stages (0-30 days) of incubation and towards the latter end (120-180 day) as it shows a similar trend to the line.

Figs.4. (c), (d) and (e) of W2 give quite a remarkable trend. The observed did not depart markedly from the calculated values but fell within or close to the lines. This suggests that the decomposition of legumes probably linked to the higher nitrogen content, may have resulted in better prediction of residue decomposition rate at the moisture content of 70 % field capacity. The agreement between the observed and the calculated was quite perfect.

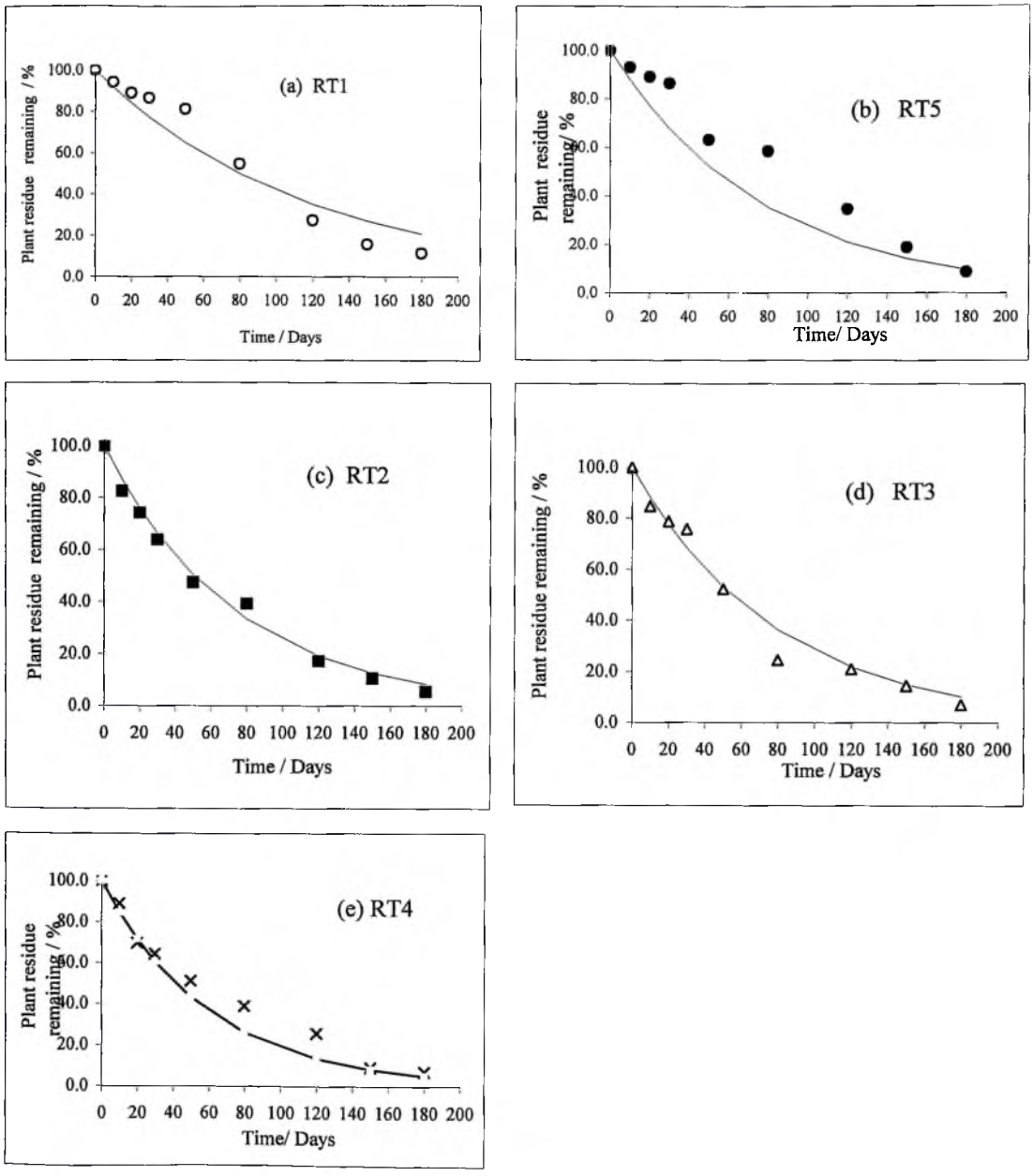


Fig 4.4 Variation of observed (symbols) and calculated (lines) plant residue dry weight for residue treatments ;(a) RT1, (b) RT5; (c) RT2 (d) RT3 (e) RT4 .at 70 % field capacity (W2

Figure 4.5 shows the variation of observed (symbols) and calculated (lines) plant residue dry weight for (a) Natural elephant grass; RT1 (b) Elephant grass from fertilized maize plot; RT5, (c) Pigeon pea; RT2, (d) Cowpea; RT3, (e) *Mucuna sp*; RT4 at 40 % field capacity. The graphs are curvilinear but steep especially for (a). The observed fell within or very close to the calculated (lines) in the entire figure. This implies that the performance of the model in predicting the decompostion of the residue was fairly good.

However, from the 120 day onward, Fig.4.5 (a) showed a deviation from the calculated and therefore much faster rate of decomposition is expected. It is of interest to note that although in Fig. 4.5 (b), RT5 which is elephant grass showed quite a similar trend of decomposition like that of the legumes, (c), (d) and (e) and this could be attributed to the beneficial nitrogen it obtained. The model for determining rate of decomposition is much better when moisture content is much lower as at 40 %. This implies a prediction of decomposition is dependent on soil water content for a residue type.

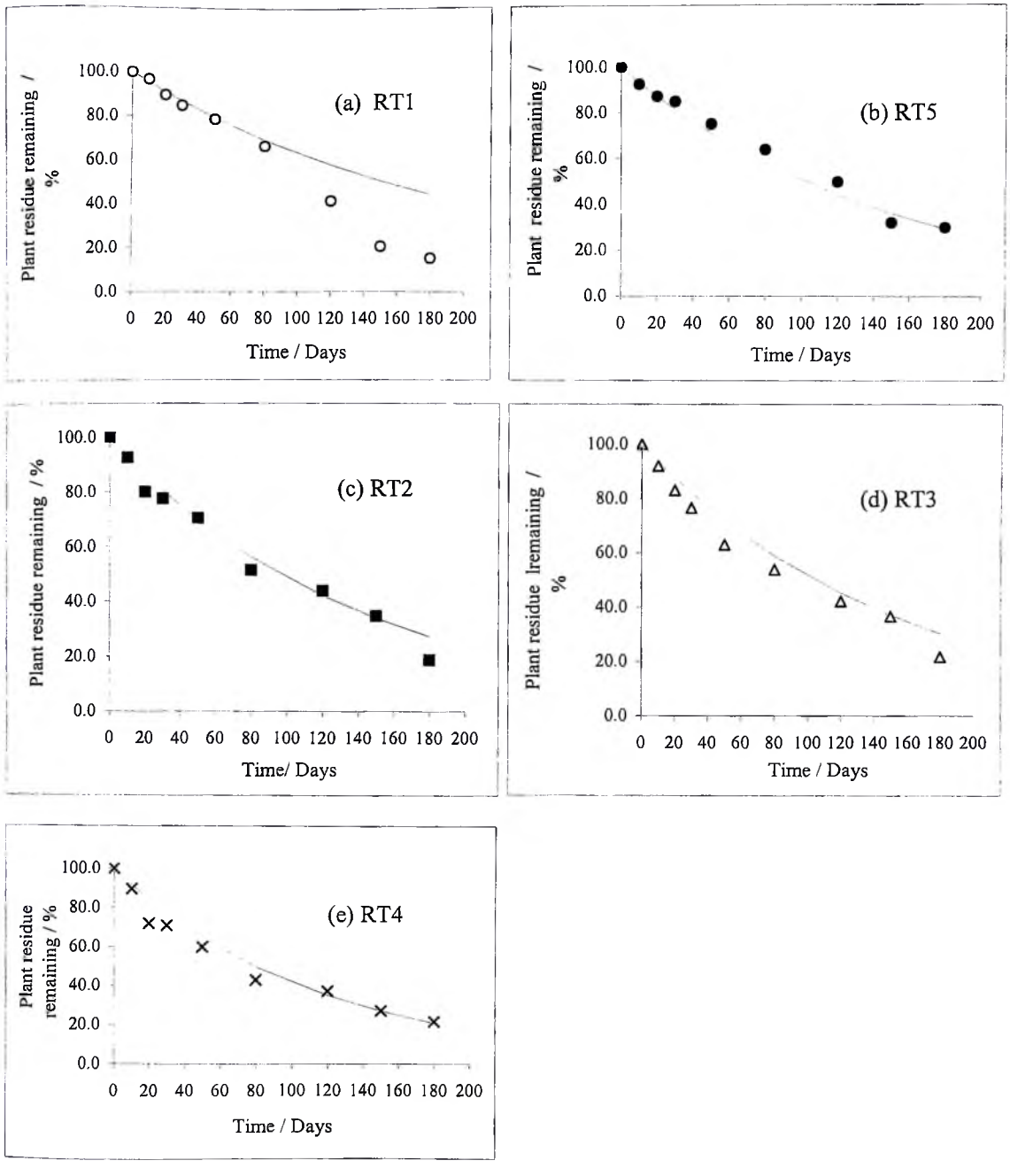
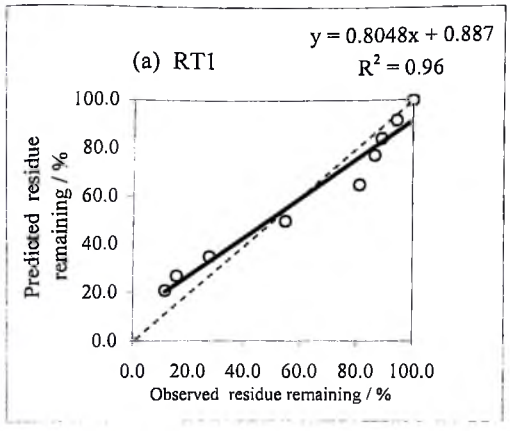
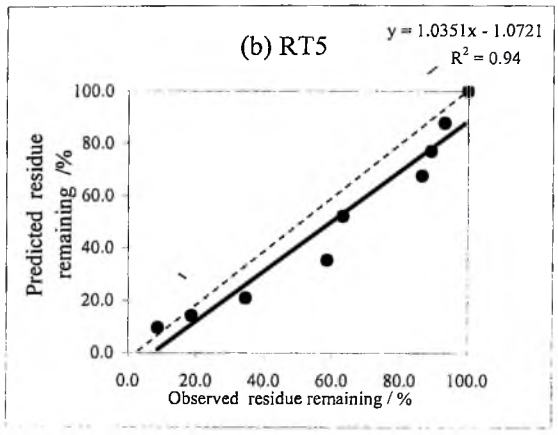


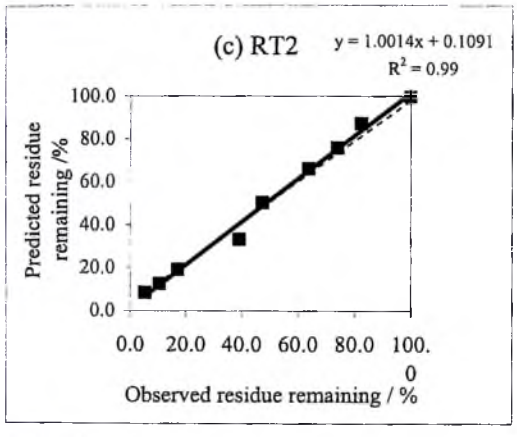
Fig 4.5 .Variation of observed (symbols) and calculated (lines) plant residue dry weight for residue treatments ; (a) RT1, RT5 (b), (c) RT2, (d) RT3, e) RT4 at 40 % field capacity

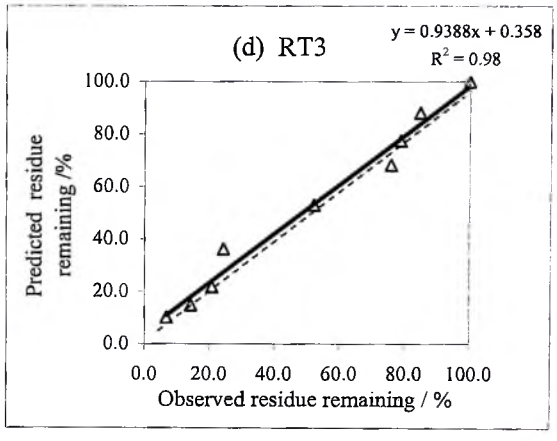
Figure 4.6 shows the predicted versus observed plant residue dry weight of (a) Natural elephant grass; RT1 (b) Elephant grass from fertilized maize plot; RT5, (c) Pigeon pea; RT2, (d) Cowpea; RT3, (e) *Mucuna sp*; RT5 at 70 % field capacity. The dotted line is 1:1 showing the theoretical relationship between the predicted and observed values while the continuous line is a linear regression. The mean coefficient of determination (R^2) using linear regression analysis was high ($R^2 \geq 0.94$) and the slope between 0.80 and 1.05. This shows a positive correlation between the predicted values and the observed. The 1:1 line also shows a similar trend of good relation for all the figures except for Fig.4.6 (b) which had the 1:1 line slightly above the predicted versus observed values. This suggests that Fig. 4.6 (b) was under estimated.

Figure 4.7 shows the predicted versus observed plant residue dry weight of (a) Natural elephant grass; RT1 (b) Elephant grass from fertilized maize plot; RT5, (c) Pigeon pea ; RT2, (d) Cowpea; RT3, (e) Mucuna sp; RT5 at 40 % field capacity. The dotted line is 1:1 and the continuous line is a linear regression. In all the treatments there is perfect agreement between observed and predicted residue dry weight except for RT1. The correlation between the predicted and the observed is high as $R^2 \geq 0.98$ and the 1:1 line was also perfect except for Fig.4.7(a) which deviated much from it. The high values of RT1 show a slight over estimation of the model. The model therefore indicates that decomposition rate of plant residue will be better predicted at lower moisture content than that at higher moisture level. It could also be observed that figures having the dotted lines above them have negative intercepts except for Fig. 4.7(b) while those with a fair distribution of points around the dotted lines gave better predictions.









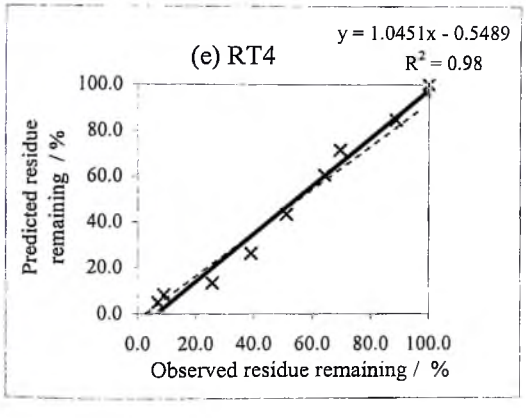


Fig 4.6 .Predicted versus observed plant residue dry weight of (a) RT1; (b) RT5;

(c) RT2; (d) RT3; (e) RT4; at 70 % field capacity (W2), dotted lines; 1:1

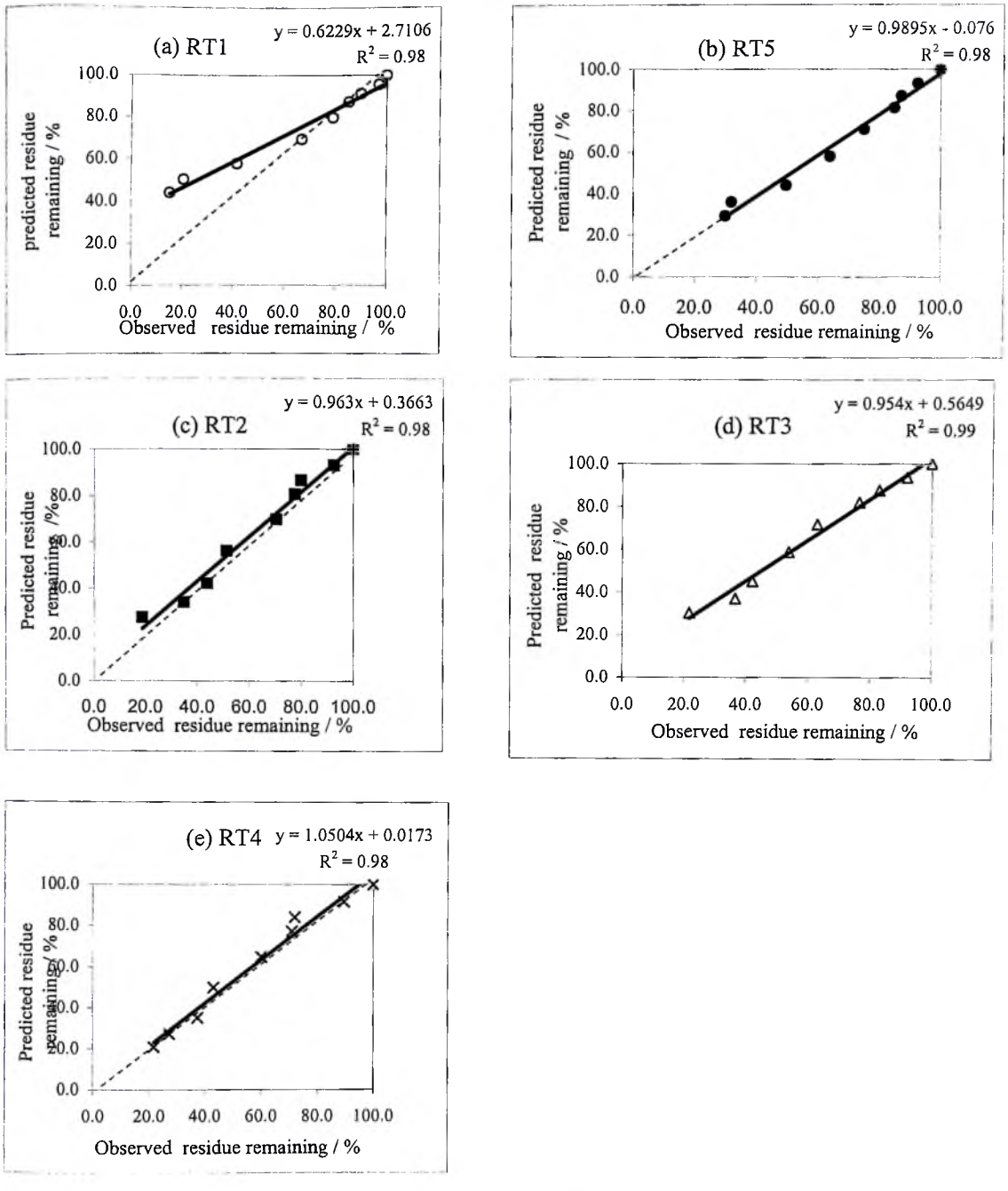


Fig 4.7 . Predicted versus Observed plant residue dry weight of (a) RT1; (b) RT5

(c) RT2; (d) RT3; (e) RT4;. at 40 % field capacity (W3), dotted lines; 1:1

4.5 Soil carbon and nitrogen accumulation during incubation studies

The amount of carbon stored in the soil is a balance between the quality and quantity of material input, amount decomposed and the amount released into the atmosphere as carbon dioxide (Christensen, 1996; Turner and Lambert, 2000; Paustian *et al.*, 2002). Table 4.7, shows the effect of the various residue treatments on the organic carbon and nitrogen accumulated in the soil at field capacity (FC) W1 at different times (A= 20, B= 80, C= 180 days from the commencent of the incubation study). It must be noted that treatment RT6 is the control which received no residue.

Table 4.7. Effect of various residue treatments on soil organic carbon and nitrogen / % accumulation during the incubation period at soil water treatment W1.

	Organic Carbon %			Total Nitrogen %		
	A	B	C	A	B	C
Treatments	%	%	%	%	%	%
RT1	1.74	2.14	1.61	0.13	0.16	0.13
RT2	1.75	1.78	1.65	0.15	0.15	0.18
RT3	1.78	2.08	1.74	0.16	0.16	0.16
RT4	1.83	2.19	1.68	0.14	0.20	0.21
RT5	1.81	2.06	1.71	0.14	0.17	0.14
RT6	1.64	1.64	1.44	0.08	0.10	0.12
Lsd	0.23	0.37	0.22			
P<0.05	0.54	0.04	0.11			

A:20 days after incubation B: 80 days after incubation C: 180 days after incubation .

Initial organic carbon and nitrogen contents of the soil were 1.62 % and 0.08 % respectively at the start of the experiment. There was a general increase in % carbon at the initial phase of decomposition with RT4 (M) having the highest followed by RT5 (EFF), RT3 (CP), RT2 (PP), RT6 (C) and RT1 (EG) in that order. The % carbon in each soil changes with time. A low carbon content was observed at the end of the experiment in all the cases although each was higher than the control. Results from the table indicate that at 20 and 180 days, the organic carbon content did not differ significantly among the treatments. This implies the carbon stored at the early stage of decomposition are the same and this could be attributed to the fast decomposition rate of the residues.

However, soil samples taken 80 days after showed significantly ($P < 0.05$) higher values for the grass RT1 (EG), RT3 (CP), RT4 (M) and RT5 (EFF). The high organic carbon of RT4 (M) and RT3 (CP) were unexpected because their decomposition was faster and so loss of carbon was expected to be high. Comparison among the treatments at 80 days of incubation also indicates that apart from pigeon pea (RT2) which was not significantly different from the control, the legumes and non legumes treatments were similar.

It could also be noted that soil carbon content for the various treatments were all significantly above the control (RT6). This indicates the differences in the contribution of plant residues to carbon storage based on their composition. As stated earlier carbon accumulated at the end of the 180 days of incubation for the various treatments were not significantly different. Thus, it takes quite a long time for carbon storage to show any significant difference with time irrespective of residue treatment which suggests that net carbon accumulation takes quite a long time as noted by Van Faaseen and Smilde, (1985).

Nitrogen content of the soil on the respective days was not significantly different from the control. However it can be observed that nitrogen % of the soil increased with increasing decomposition especially for the legumes and less so for the grasses, with RT5 (EFF) behaving similarly like the legumes although fluctuation could be observed in the trend. In RT1 (EG) N % was stablized. The high amount of N % in the legumes agrees with the report of Vine (1953) that legumes increase the nitrogen content of soil. RT3 (C) however maintained some consistency in % nitrogen at the different times.

Some of the residues typically represent the main situations in the N dynamics of the soil during decomposition : the highest total N (0.21 % N) was obtained for mucuna spp (RT4) and the lowest (0.13% N) for elephant grass from unfertilized maize plot (RT1) at the end of the incubation period.

An N mineralization was observed throughout the entire incubation period in only mucuna spp (RT4) and pigeon pea (RT 2). The incorporation into the soil of the other residues for example RT1 and RT5 appears to cause some N immobilization although the amounts of N immobilized may vary with the residue type. Thus, N mineralization was followed by an N immobilization phase, the amount of which varied between residues types. Generally, 180 days after incorporating the residues into the soil, the concentrations of mineral N present in the treated soils were more than those in the corresponding control.

The C: N ratio for the various treatments shows a general decrease in the carbon to nitrogen content of the soil as decomposition progressed. The rate of mineralization was expected to be high at all the days since C: N ratios were within 8 - 13.4 with the exception of the control which had a higher value of 20. The control however appeared to show some amount of mineralization as the C: N ratio decreased with time. The initial C: N of the residue

was between 24 and 30. Addition of this material will after 180 days result in lower C: N ratio of the soil than the corresponding control.

As decomposition proceeded, the proportion of water soluble compounds like sugar and free amino acid had less influence on the rate of C mineralization because these fractions had been largely degraded. It has been shown that the decomposition of crop residues can be affected by the availability of N since the C: N ratio of the decomposers is far lower than the C: N ratio of many crop residues. Thus, very often, the availability of soil inorganic N will, at least in the short term, control the kinetics of C decomposition, as has been shown with cereal residues (Recous *et al.*, 1995; Henriksen and Breland, 1999; Corbeels *et al.*, 2000). Consequently the relative weight of the carbon coming from the residue by primary decomposition falls during the course of decomposition. Thus only the initial rates of decomposition can be explained by the biochemical characteristics of the residues.

Table 4.8 presents the effect of the various treatments on the organic carbon and nitrogen accumulated in the soil during the incubation period at soil water treatment, W2. A similar trend like that described above could be observed. However, there was a significant difference between the treatments on the 20th and 80th days. By the 80th day, all the legumes and RT5 were significantly different from the control except RT1. By 180 days, all treatments had similar organic carbon content that was significantly different from the control.

It could be observed that soil carbon accrued from the legumes was higher than that for the grasses. In addition, although treatments were not significantly different, RT1 (EG) at the 80 day had a carbon content even much lower than the residue free treatment, (RT6) and this may be attributed to some form of immobilization of the carbon in the soil by the microbes.

Nitrogen content of the soil at the selected days was not significantly different. However, it can be observed that high nitrogen content was obtained for the legumes than the grass (RT1).

Table 4.9 also shows a similar trend for carbon accrued for water treatment W3. Results indicated the amount of carbon stored at 20 and 180 days were significantly different. It could be observed that carbon content for the control (RT6) was lower than the other treatments. The N % of the soil for the respective treatments was highr for the legumes than the grasses at the end of the period. A general C: N ratio of all the treatments at the end of the incubation showed a value of below 20 and hence quite feasible for mineralization process.

Table 4.8. Effect of the various residue treatments on soil organic carbon and nitrogen (%) accumulation during the incubation period at soil water treatment W2

	Organic Carbon %			Total Nitrogen %		
	A	B	C	A	B	C
Treatments	%	%	%	%	%	%
RT1	1.73	1.70	1.68	0.12	0.14	0.16
RT2	2.03	2.08	1.84	0.17	0.27	0.17
RT3	2.12	2.09	1.72	0.18	0.21	0.17
RT4	2.10	2.16	1.71	0.19	0.25	0.18
RT5	1.92	2.15	1.75	0.14	0.16	0.15
RT6	1.68	1.76	1.52	0.09	0.12	0.11
Lsd	0.27	0.16	0.19			
P<0.05	0.02	<0.001	0.06			

A: 20 days after incubation B: 80 days after incubation C: 180 days after incubation period

Table 4.9. Effect of the various residue treatments on soil organic carbon and nitrogen / % accumulation during the incubation period at soil moisture W3

Treatments	Organic Carbon (%)			Total Nitrogen (%)		
	A	B	C	A	B	C
RT1	1.92	1.89	1.75	0.12	0.15	0.15
RT2	2.13	2.14	1.72	0.15	0.16	0.19
RT3	2.00	1.88	1.69	0.13	0.16	0.17
RT4	1.88	2.15	1.82	0.16	0.13	0.18
RT5	1.98	1.80	1.73	0.13	0.16	0.15
RT6	1.71	1.79	1.55	0.11	0.12	0.11
Lsd	0.22	0.36	0.014			
P>0.05	0.03	0.14	< 0.001			

A:20 days after incubation B: 80 days after incubation C: 180 days after incubation

4.6 Field Studies

4.6.1 Some physical and chemical properities of Haplic luvisol

Table 4.10 shows some physical and chemical properities of the field soil from the University of Ghana Farm. Soil texture is sandy loam and like that of the Haplic lixisol from Kpeve, drainage and aeration are expected to be high. The pH of the soil ranges from 6.4 to 6.7 which are near the neutral zone and so availablilty of nutrients are also expected to be very high. Although, organic carbon content of the soil ranged from 0.33 % to 0.41 % and total nitrogen content from 0.052 to 0.059 % which were very low, the C:N ratio of the soil which is between 7.59 and 5.59 was quite low and so mineralization of nutrients would also be expected to be high. The available phosphorus content between 14.9 mg/kg and 15.38 mg/kg are also quite high confirming the high nutrient status of the soil. The Cation exchange capacity (CEC) values between 10-13 $cmol^{(+)}/kg$ may be quite low but considering the geneally low values obtained for it's organic carbon content, the value obtained was quite reasonable. Field capacity of the soil was 0.19 g/g with a wilting point of 0.115g/g.

4.7 Field residue decomposition study

4.7.1 Some chemical characteritics of plant residues obtained from the field

Table 4.11 shows some chemical properties of the plant residue used for the field study. RT2 and RT4 were used to represent a typical legume fallow residue and RT5, a typical grass fallow. Results from the Table indicate that the carbon content of 493.8 g/kg and 459.1 g/kg for the RT2 (PP) and RT4 (M) residues respectively were quite high while that for RT5 (EFF) was low.

Table 4.10 Some physical (a) and chemical (b) properities* of the field soil from the University Farm.

(a) physical

Treatments	Sand %	Silt %	Clay %	Textural class	Field capacity g/g
RT2 (PP)	64.0	25	5	Sandy loam	0.19
RT4 (M)	66.8	22.5	7.5	Sandy loam	0.19
RT5 (EFF)	69.6	25	5	Sandy loam	0.19
RT6 (C)	66.8	25	5	Sandy loam	0.19

(b) chemical

Treatment	pH 1:1	OC %	TN %	Avail.P mg/kg	CEC(cmol $^{(+)}$/kg
RT2 (PP)	6.7	0.37	0.058	15.5	13
RT4 (M)	6.6	0.33	0.059	15.2	12
RT5 (EFF)	6.4	0.38	0.052	15.9	12
RT6 (C)	6.5	0.41	0.054	14.9	10

1:1 Soil/ water suspension
OC: Organic carbon
TN : Total Nitrogen
Avail. P: Available Phosphorus
CEC: Cation exchange capacity
* All values are means of four observations

Table 4.11 Some chemical properties of the plant residues used for the field studies

	C g/kg	N g/kg	P g/kg	C: N	C:P
Residue treatments					
RT2 (PP)	493.8	1.89	2.81	26.11	175.88
RT4 (M)	459.1	1.68	3.19	27.33	143.92
RT5 (EFF)	334.7	0.88	3.41	38.25	98.15

RT2 (PP: Pigeon pea following unfertilized maize), RT4 (.M: *Mucuna spp.* following unfertilized maize) RT5 (EFF: Elephant grass following previously fertilized maize).

Carbon ranged from 33.47 to 49.38 % which conforms to the results of Woomer *et al.* (1994), hence the values obtained are quite reasonable. RT2 (Pigeon pea) had the highest nitrogen content of 1.89 g/kg although according to Fox *et al.* (1990), all the nitrogen content in the treatment are below the 2 % stated for decomposition. The N content of RT5 (EFF) was quite very low 0.88 % as it was expected to have benefited from the residual fertilizer in the soil.

The C: N ratio was also below 30 for the legumes while that of the grass was quite high (38:3). This indicates that decomposition would be faster for the legume as compared to the elephant grass from the fertilized maize plot. Usually phosphorus content of plant residue vary and is much higher in legume than in grasses Maikslenien, (2000). Thus, the high value obtained for the grass was unexpected although the high P content of the soil (15.9 mg/kg) may have contributed to this phenomenon. The C:P ratio for the legumes (RT2 and RT4) was higher

than that for the grass (RT5) and this may be attributed to the high amount of P present in the residue.

4.7.2 Effect of residue type on decomposition

Figure 4.8 shows the variation of residue dry weight with time for the RT2; Pigeon pea, RT4 *Mucuna spp.* and RT5; Elephant grass from fertilized maize plot under field conditions. Decomposition, as observed was faster for RT4 than RT2 and then RT5. Thus decomposition was generally faster for the legume residues than the grass as expected.

Considering the intial fast phase of decomposition to be 20 days, 71.43 % and 66.67 % of RT2 and RT4 were respectively remaining while RT5 had 78.3 % residue left. The time taken for half of the plant residues (half-life) to decompose was 42, 28 and 52 days for RT2, RT4 and RT5 respectively. This shows that decomposition of the grass (RT5) was slightly slower than the RT2 although under green house condition, they seem to be the same. RT4 (M) however decomposed very fast due to the herbaceous nature of the residue. At the end of the 80 day study, only 19.84 % of RT2 and 12.22 % of RT4 residue were left while RT5 had 34.09 % residue remaining. This implies, that grass (EFF) residue would be a better contributor to carbon storage as a result of its slow rate of decomposition (Bruce *et al.*, 1999).

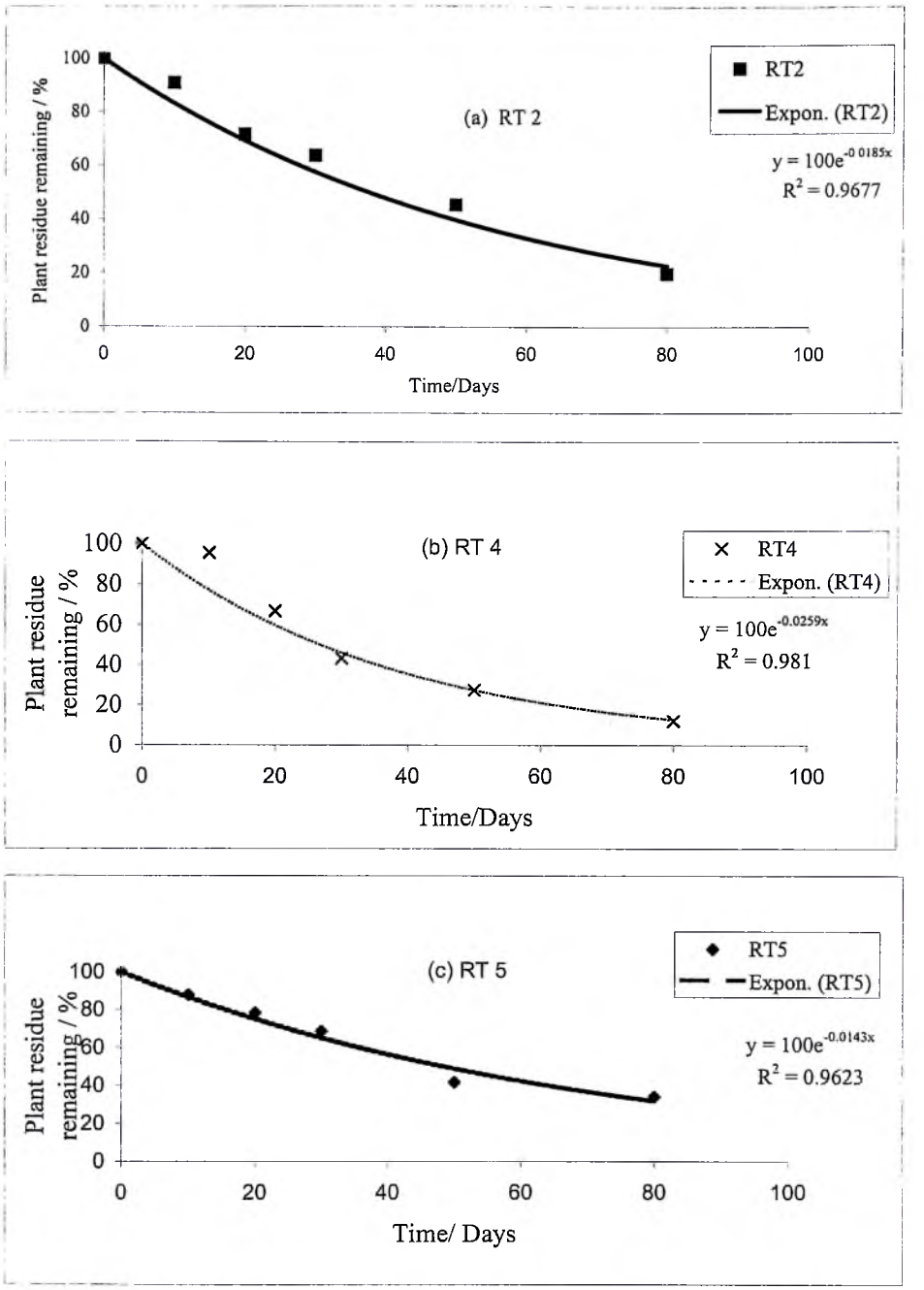


Fig.4.8. Variation of residue dry weight with time for (a) RT2; (b) RT4;

(c) RT5; used for the field studies; observed data are symbols and fitted curves are lines

The rate of decomposition (*k*) from Table 4.12 for RT2, RT4 and RT5 were 0.020g/day, 0.026 g/day and 0.014 g/ day respectively. The effect of residue on decomposition is more related to the C: N of the plant residues. Most authors, Paul and Clark (1989) and White (1979) have suggested that net N mineralization occurs when C/N ratios of residues are < 25. Nevertheless, Thompson and Troeh (1978) stated a C:N ratio of 32 as the threshold. It has been shown that residues with low C/N ratio can cause net immobilization of the soil mineral N Jensen, (1994). In fact, most studies referring to a C/N ratio threshold value carried out in the field did not monitor precisely the changes occurring in soil mineral N with time.

In this study, C:N ratio of all the residues was between 26.11 and 38.25 and therefore was expected to cause net N immobilization in the early stages of decomposition in agreement with the literature. Considering the net effect of the residues after 80 days of incorporation, the legume residues possibly had a C/N ratio below 25 and may have induced net N mineralization, whereas the grass with a C/N ratio slightly above 32 might have caused net immobilization of soil mineral N.

Finally, it seems much more important to consider the kinetics of decomposition rather than just the amounts mineralized by the end of decomposition, as the relationships between C mineralization and N and the intrinsic characteristics of the residues are temporally dynamic (Vanlauwe *et al.*, 1996; Quemada and Cabrera, 1995).

4.7.3 Comparison between greenhouse pot experiment and the field residue studies.

Table 4.12 shows the comparison between the decay rate constant k (g/day) for field and under greenhouse experiments at W1, W2 and W3 for RT2, RT4 and RT5 residues. The results

indicate that values obtained at W1 were slightly quite close to that of the field although decomposition rates of the plant residues was quite faster on the field than with the pot studies.

Table 4.12 Comparison between decay rate constant k (g/day) for the greenhouse pot experiment and that of the field for some of the plant residue treatments

Residue type	W1	W2	W3	On the field
RT2	0.020	0.015	0.008	0.019 ± 0.011
RT4	0.022	0.014	0.009	0.026 ± 0.007
RT5	0.018	0.011	0.007	0.014 ± 0.005

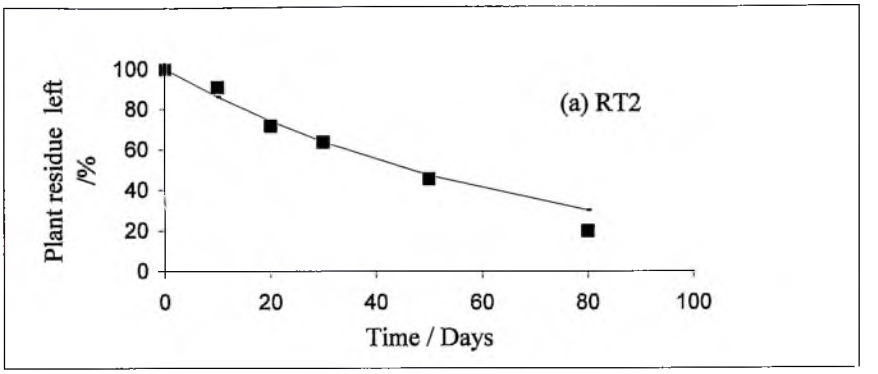
Water content of the soil at field capacity (W1) for the pot experiment in the greenhouse was (0.23g/g) while that for the field was 0.185 g/g with a wilting point of 0.115 g/g. Thus, the soil in the field had a lower water holding capacity compared to the pot although the flutuations in temperature and termites may have hastened its residue decomposition on the field. RT5 (EFF) was however slightly faster at W1 than on the field. Thus, in order to make a reasonable assement between the pot experiment and that of the field, soil water (which was not controlled) was assumed to be at 70 % field capacity (W2) throughout because the study was conducted during the rainy season and this W2 (0.16 g/g) is also much closer to the field capacity of 0.185 g/g . Hence, the decay rate constants could be compared at 70 % FC for both greenhouse and field studies (Table 4.12). The result obtained indicated that decomposition was slightly faster in the field than in the greenhouse pot experiment especially for RT4.

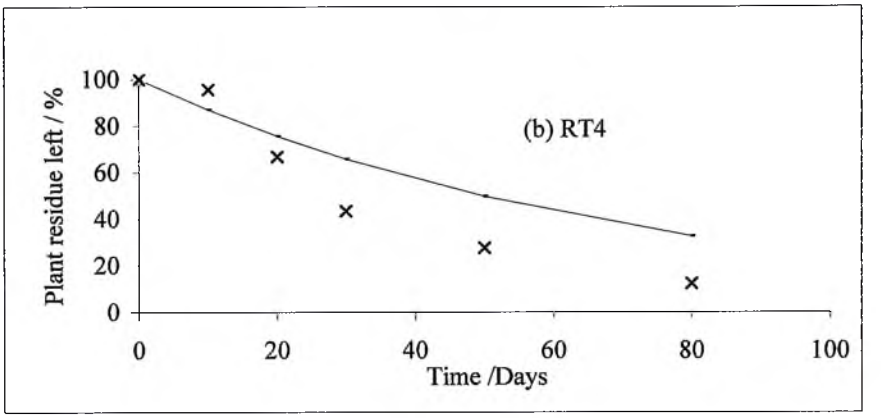
In the field, residue decomposition rates are controlled by many environmental factors (Martin and Haider, 1986). Especially important are water content and temperature (Parr and Papendick, 1978). Temperature of the field fluctuated with time and was between 26 °C and 32 °C. The changes in the temperature may probably have resulted in the fast decomposition of the residue since temperature in the greenhouse hardly exceeded 30 °C and was less variable. Comparison between W3 (40 %FC) and that for the field also showed that under drier conditions, decomposition of plant residue will be even faster in the field than in the greenhouse.

From the greenhouse studies, equation 4.3 on page 63 was applied to calculate residue decomposition for field studies using the rate constant (k)/g/day obtained for W2 observed in the greenhouse (Table 4.12). Figure 4.9 show variation of observed (symbols) and calculated (lines) plant residue dry weight for (a) Pigeon pea, RT2 (b) *Mucuna spp.*, RT4 and (c) Elephant grass from fertilized maize fallow plot, RT5.

The figures illustrate that the calculated decomposition did not depart markedly from the observed as in RT2 and RT5. Thus the values observed in this study fell within or close to the predicted. It therefore gave an indication that comparison between the field and that in the greenhouse is feasible and so modelling of pot experiments could be quite useful for assessing field scale decomposition provided the necessary factors like moisture content are known.

It can also be observed that it was only RT4 which had its observed values generally lower than the calculated for the field. The half life for the predicted RT4 (M) was high about 42 days compared to the observed of about 28 days.





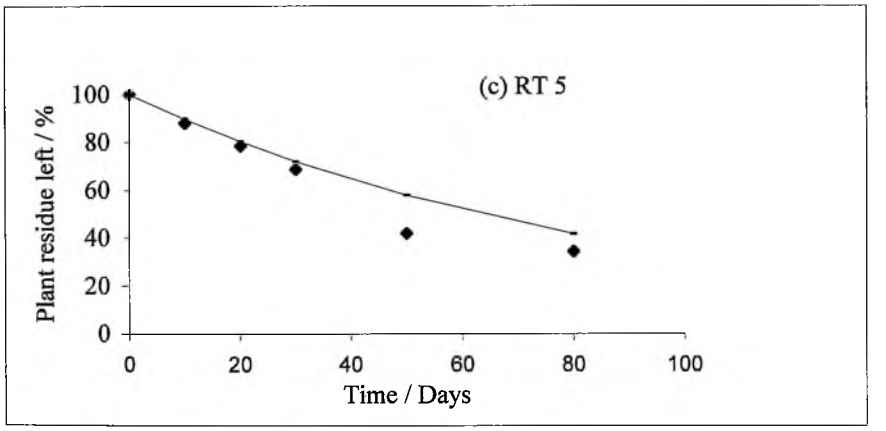


Fig 4.9 .Variation of observed (symbols) and calculated (lines) plant residue dry weight for

(a) RT2; (b) RT4 ; (c) RT5; with time for the predicted field study

4.7.4 Soil carbon accumulation during field study

Table 4.13 shows the amount of carbon stored in the soil up to 80 days under field conditions. The soil was initial sampled just before residue incorporation. Results from the data indicate that organic carbon content for the various treatments at the start of the experiment were not significantly different from each other except for the control. The high organic carbon at the beginning of the study for the control (RT6) may be attributed to a previous cropping of the land with a high biomass plant like *Mucuna pruriens*.

Table 4.13 Oxidizable carbon (%) to the soil during field studies.

	Days after commencement of studies		
Days	0	20	80
Plant Residues treatment			
RT2	0.37_a	0.49_a	0.67_a
RT4	0.33_a	0.46_a	0.60_b
RT5	0.38_{ab}	0.41_b	0.61_b
RT6	0.41_{ab}	0.21_c	0.26_c

Figures bearing same subscript in each column are not significantly different.

$P < 0.001$

Lsd 0.05

It could also be observed that there is general build up of carbon from the 20-80 days as decomposition proceeded except for RT6. The control however seemed to show a state of

decline which may be due to immobilization and perhaps leaching of the carbon in the soil. The RT5 which is from the fertilized maize plot also seemed to accumulate much carbon than the legumes at the begining of the experiment.

From the start of the study to the 20th day, RT2, RT4 and RT5 tended to have a 0.12 %, 0.13 %, 0.02 % increase in carbon storage respectively while the control (RT6) to declined by 0.20 %. All the residue treatments were also significantly higher than the control. The soil under treatment RT5 at the 20th day had the lowest organic carbon content compared to the other residue treatment which was not expected because gramineae (grasses) contain relatively high amount of lignin which tends to decompose slowly hence preserving the carbon. The soil in treatment RT2 had the highest levels of organic carbon stored on the 80th day. This was also not expected as much of the residues were observed to have decomposed. However, this observation could be attributed to some of the resistant plant residue still present.

Results also indicate that RT6 had the highest carbon content at 80 days. At the end of the 80 days period, there was no significant difference between the residue treatments but, each treatment was significantly higher than the control indicating that additions of a plant residue certainly increased soil carbon to the soil although minimal. RT2, RT4 and RT5 had 1.81 %, 0.27, 0.21% carbon respectively above the initial carbon content of their soil. The control plot had a much low carbon content at the end of the period.

CHAPTER 5

CONCLUSION

This study investigated the decomposition of plant residues as affected by substrate quality and other environmental factors such as soil moisture. The C: N ratio of the plant residues was found to greatly influenced decomposition rate such that plant residues with high nitrogen content showed faster decomposition than those with lower nitrogen content. Legume residues with low Carbon: Nitrogen ratios decomposed faster than grass residues. Thus, the order of decomposition decreased with RT2 (Pigeon pea) > RT4; (*Mucuna spp.*) >RT3 (Cowpea) > RT5 (elephant grass from fertilized maize plot) > RT1 (elephant grass from unfertilized maize plot). Although, legumes residues which showed rapid decomposition would contribute less to soil carbon storage in a long term, they would in short term increased soil productivity greatly in agricultural field as compared with the grasses.

. The moisture content of the soil was found to have a remarkable influence on the rate of decay. High soil moisture content resulted in faster decomposition rates of plant residues although some exceptions may occur. Thus, decomposition rate of plant residue generally increased in the order 40 % moisture content < 70 % moisture content < 100 % moisture content. There is therefore increased storage of carbon in soils under conditions of low soil moisture content below the field capacity content.

There is a good relation between rate of decomposition for a plant residue type in the a greenhouse and that in field such that knowing the environmental conditions pertaining on the field, it may be possible to estimate field scale decomposition using parameter derived from greenhouse studies. The amount of carbon stored in the field has a bearing on the type of plant residue incorporated into the soil. Each residue type however added some carbon which is

certainly better the bare control soil. The study has also shown that simple equations could be used to describe the effect of soil moisture on plant residue decomposition. A good agreement between the predicted versus observed residue decomposition under varying soil water was found using a simple model developed in this study.

Recommendation

Even though this study sought to assess the effect of residue type and soil water on residue decomposition, the type effect was only limited to carbon and nitrogen due to lack of facilities. Thus, detailed studies on residue factors such as lignin content, cellulose and polyphenol content would be recommended in future studies since these factors significantly affect decomposition rates.

Also, since grasses have been found to be better contributors of carbon storage in soil under low moisture content below the field capacity, it would be imperative to assess the different types of the grass family which might contribute better to soil carbon. Furthermore farmers are entreated to leave the grasses on the soil when weeding the land to conserve carbon storage instead of burning them.

Furthermore as the microbes are the major agents of decomposition process, the microbial dynamics and diversity with time need to be determined in order to assess which microbes influence decomposition at a particular stage.

Finally, the information gained from such detailed studies could be used to improve the calibration and validation of some carbon turn-over models.

REFERENCES

Abduranhman, M.D., Seeling, B, Rego, T.J. and Reddy B.B. 1998. Organic matter imputs by selected cropping systems on a vertisoil in the semi-arid Tropics of India. Annuals of Arid Zones.37, 363-371.

Aber, J.D. and Melillo, J. M. 1980. Litter decomposition: Measuring relative contributions of organic matter and nitrogen in forest soils.Can.J.Bot. ppl. .416- 421.

Aber, J. D. and Melillo, J. M. 1991. *Terrestrial ecosystems* Philadelphia. Soil Survey of Kalamazoo County, Saunders College Publishing, Austin, FR 1979. Pennsylvania, USA. p 436.

Acquaye, D.K. and Laryea. K.B. 1982. Unpublished soil report of the University of Ghana Farm. Legon. Dept. of Soil Science, University of Ghana.

Adiku, S.G.K., G .N.N. Dowuona and Kumaga, F. K. 2003. *Measuring and Assessing soil carbon sequestration by Agricultural Systems in Developing Countries*. Unpublished Annual report No. 2003/002. Dept. of Soil Science, University of Ghana. 50 pp.

Ahenkorah, Y., Amatekpor, J.K., Dowuona, G.N.N. and Duah-Yentumi, S. 1994. A field survey report on soil and water resources of Ghana. Their conservation, management and constraints to their utilization for sustainable development, United Nations University Institute for Natural Resources in Africa Report. November, 1994. pp. 131.

Alexander, M.1961. Introduction to Soil Microbiology.Wiley, New York, N,Y.,472pp

Alexander, M.1977. Introduction to Soil Microbiology; 2nd ed.John Wiley and Sons, New York, NY. 133-146.

Alexander, M. 1980. Effect of acidity on micro organisms and microbial processes in soil. In effects of acid precipitation on Terrestrial Ecosystems (Hutchinson, T.C and.Havas, M. eds).pp 363-380. Plenum, New York.

Allison, L.E.1965. Organic Carbon, Walkey-Black method. In: C.A. Black, D.D.Evans, J.L.White, L.E. Ensminger and F.E.Clark (eds). Methods of soil analysis.Part 2, Chemical and Microbiological properties. American Society of Agronmy Inc.Madison, Wisconsin. pp.1372-1376.

Allison, F.E.1973. Soil Organic Matter and its Role in Crop Production. Developments in Soil Science, 3. Elsevier, Amsterdam. 637-670 pp.

Andren O., Kalman, R. and Katterer, T. 1993. Water and temperature dynamics in a clay soil under winter wheat: influence on straw decomposition and N immobilization. Biol. Fertil Soils 15:1–8.

Ankomah, A.B., Zapata, F.Z., Danso, S.K.A. and Axmann, H. 1995. Cowpea Varietal Differences in uptake of Phosphorus from Gafsa Phosphate rock-P. Ultilization. Fert.Res 41: 219-22

Bartholomew, W.V.1965. Mineralization and immobilization of nitrogen in the decomposition of plant and animal residues. In: Soil Nitrogen (W.V. Bartholomew and F.E.Clark,eds), pp. 285-306.Am.Soc.Agron., Madison,Wisconsin.

Bending, G.D., Turner, M.K. and Burns, I.G. 1998. Fate of nitrogen from crop residues as affected by biochemical quality and the microbial biomass. Soil Biol. Biochem. 30: 2055–2065.

Berg, B. and Matzner, E. 1997. Effect of N deposition on decomposition of plant litter and soil organic matter in forest ecosystems. Environmental Reviews 5: 1-25.Green Alert April 12, 2002. Vol. 1, No. 25

Bouldin, D.R. 1988. Effect of Green manure on soil organic matter content and nitrogen availability .In International Rice Research Institute,eed, Green manure in Rice farming Proceedings of a Symposium on sustainable agriculture.IRRI,Los Banos, Philippine, pp 151-164, Los Bonos, Philippines.

Bouyoucos, G.J. 1962. Hydrometer method improved for making particle size analysis of soils. Agron. J.54:464-465.

Bowman, A.F. 1990. Exchange of greenhouse gases between terrestrial ecosystems and the atmosphere. In: Bowman, A.F. (ed) Soils and the Greenhouse Effect. Chechester, UK; John Wiley, New York. Pp 66-127

Bowman, R.., Sucik, M., Rosales, M. and Saunders, J. 2002. Conservation Tillage Fact Sheet.No.2-98 .USDA-ARS, USDA-NRCS and Colorado, Central Great Plains Research Station, 2002 Annual Report. pp 1-5

Brady, N.C.1990. The Nature and Properties of Soils. 10th ed. MacMilian publ., New York, NY. pp.96-97,279-300, 328.

Brammer, H. 1962. Soils of Ghana. In: J.B.Wills (ed) Agriculture and Land use in Ghana ,p 88-126,151-154 .Oxford University Press, New York.

Bray, R. H. and Kurtz. L.T. 1945. Determination of total organic and available forms of phosphorus in soils. Soil Sci.59: 39-45.

Broder, M.W and Wagner,G.H. 1988. Microbial colonization and decomposition of corn, wheat, and soybean residue. Soil Sci. Soc. Am. *J.* 52:112-117.

Brown, P.L. and Dickey, D.D., 1970. Soil Sci.Soc.Amer.Proc., 34:118-121

Bruce, J. P., Frome, M., Haites, E., Janzen, H., Lal, R. and K. Paustian. 1999. Carbon sequestration in soil. J. Soil Water Cons. 54:382-389.

Buckman H.O. and Brady, N. C. 1969. The Nature and Properties of Soils. 7th edition, The Macmillian Company/ Collier-Macmillan Limited, London.pp147-148

Burke, I.C., Yonker, C.M., Parton, W.J., Cole, C.V., Flach, K. and Schimel, D.S. 1989. Texture, climate, and cultivation effects on soil organic matter content in U.S. grassland soils. Soil Sci Soc Am J 53:800–5.

Campbell, C.A. 1978. Soil organic carbon, nitrogen and fertility. In Soil organic matter (M.Shnitzer and S.U.Khan eds) pp 173-263. Am. Elsevier, New York

Chesson, A. 1997. Plant degradation by ruminants: Parallels with litter decomposition in soils. p. 47–66. *In* G. Cadisch and K.E. Giller (ed.) Driven by nature: Plant litter quality and decomposition. CAB Int., Cambridge, UK

Christensen B.T.1996. Matching measurable soil organic matter fractions with conceptual pools in simulation models of carbon turnover: Revision of model structure. In: Evaluation of Soil Organic Matter Models (eds Powlson D.S., Smith P, Smith J.U.), NATO ASI Series 1,Vol.38. Springer-Verlag, Berlin.

Cochran, V.L., Elliott, L.F. and. Papendick, R.E. 1980. Carbon and nitrogen movement from surface-applied wheat (*Triticum aestivum* L.) straw. Soil Sci. Soc. Am. J. 44:978–982.

Cole, C.V., Duxbury, J., Freney, J., Heinemeyer, O., Minami, K., Mosier, A.., Paustian, K.,Rosenberg, N., Sampson, N., Saverbeck,D.and Zhao, Q. 1997. Global estimates of potential mitigation of greenhouse gas emissions by agriculture. Nutrient Cycling Agroecosyst. 49: 221-228

Constantinides, M. and Fownes, J.H. 1994. Nitrogen mineralization from leaves and litter of tropical plants: Relationship to nitrogen, lignin, and soluble polyphenol concentrations. Soil Biol. Biochem. 26:49–55.

Corbeels ,M.., Hofman, G. and Van Cleemput, O. 2000. Nitrogen cycling associated with the decomposition of sunflower stalks and wheat straw in a vertisol. Plant Soil; 218:71-82.

Cowling, E.B.and Merrill, W.1966. Nitrogen in wood and its role in wood deterioration. Can. J. Bot. 44, 539-554.

Curtin, D., Wang, H., Selles, F., Zentner, R.P., Biederbeck, V.O. and Campbell, C.A. 2000. Legume green manure as partial fallow replacement in semiarid Saskatchewan: Effect on carbon fluxes. Canadian Journal of Soil Science 80, 499 –505

Dendooven, L., Merckx, R., Verstraetem, L.M.J. and. Vlassak, K .1997. Failure of literative curve-fitting procedure to successfully estimate two organic N pools. Plant Soil 195:121–128

Dillon, W.R. and Goldstein, M. 1984. Multivariate analysis: Methods and applications. John Wiley & Sons, New York. 587 pp

Donigan, A.S. 1994: Assessment of alternative management practices and policies affecting soil carbon in agroecosystems of the Central United States pp 32

Downs, M.R., Nadelhoffer, K.J., Melillo, J.M., Aber, J.D., 1996. Immobilization of a N--labeled nitrate addition by decomposing forest litter. Oecologia 105:141–50.

Duncan, R.R.1996. Breeding and improvement of forage sorghums for the tropics. p. 161-185. In D.L. Sparks (ed) Advances in agronomy. Vol.57. Academic Press, Inc.

Edmeades, D.C., Judd, M. and Sarathchandra, S.U.1981.The effect of lime on nitrogen mineralization as measured by grass growth. Plant soil 60,177-186.

Elliott, E.T. 1994. "Embodying process information in models evaluated with site network information: Nnairobi workshop." 10th World Congress of Soil Science 9 (Supplement): 163–177.

Fabrizzi ,K.P., Rice, C.W, Staggenborg ,S. 2004. Managing the microbial community for soil management. In 2004 Agronomy Abstracts [in press]. ASA, Madison, WI. p 5

Feller, C., Frisch, E., Poss, R. and Valentin, C. 1991. Effects of the texture on the storage and dynamics of organic matter in some low activity clay soils (West Africa particularly) Cah. ORSTOM, Ser.Pedol. XXVI: 25-36.

Feng, Y. and X. Li. 2001a. An analytical model of soil organic carbon dynamics based on a simple hockey stick" function. Soil Science, vol 166: 431-440

Feng, Y. and Li, X. 2001b.Carbon sequestration potential in Agricultural soils. AIDIS-CANADA Environmental Project. Canadian International Development Agency (CIDA), Alberta Research Council Inc. (ARC) and AIDIS. 1- 5

Feng, Y. and Li, X. 2002. A tool to determine Long-term Sustainable Manure Application Rate for Alberta Soils. Report to Canadian-Alberta Beef Industry Development Fund. p120.

Fog, K .1988. The effect of added nitrogen on the rate of decomposition of organic matter. Biol Rev 63:433–62p

Fox, R. H., Myers, R.J.K. and Vallis, I.. 1990. The Nitrogen mineralization rate of legume residues in soil as influenced by their polyphenol, lignin and Nitrogen contents, Plant Soil 29:251-254

Frankenberger, W.T. Jr. and Abdelmagid, H.M. 1985. Kinetic parameters of nitrogen mineralization rates of leguminous crops incorporated into soil. Plant Soil 87:257–271

Frissel, M.J.and Van Veen, J.A. 1981. Simulation model for nitrogen immobilization and mineralization. In: Modelling Wastewater Renovation by Land Disposal. I.K. Iskandar(ed.) John Wiley et al., New York, P. 359-381.

Garten, C.T. and Wullschleger, S.D. 2000. Soil carbon dynamics beneath switchgrass as indicated by stable isotope analysis. Journal of Environmental Quality 29, 645-653

Gill, R.S. and Lavender D.P. 1983. Litter decomposition in coastal hemlock (Tsuga heterophylla) stands: impact of nitrogen fertilizers on decay rates. Can J For Res 13:116–21.

Giller, K.E. and Cadisch, G .1997. Driven by nature: A sense of arrival or departure? p. 393–499. *In* G. Cadisch and K.E. Giller (ed.) Driven by nature: Plant litter quality and decomposition. CAB Int., Cambridge, UK.

Gilmour, J.T., Mauromoustakos, A., Gale, P.M. and Norman, R.J. 1998. Kinetics of crop residue decomposition: Variability among crops and years. Soil Sci. Soc. Am. J. 62:750–755

Glenn, E., Squires, V., Olsen, M. and Frye. 1993. .Potential for Carbon sequestration in Drylands, Water, Air and Soil pollution 70, 341-355

Goings, K.A. 2001. Basic Biological factors of soil carbon and nitrogen. National Soil Survey Center NRCS,USDA, Lincoln, Nebraska.

Gordillo, R.M. and Cabrera, M.L. 1997. Mineralizable nitrogen in broiler litter: I. Effect of selected litter chemical characteristics. J. Environ. Qual. 26:1672–1679

Grant C.A. and Bailey, L.D. 1994. The effect of tillage and KCL addition on pH, Conductance, P,K,CL. Can. J. Soil Sci: 74:307

Greenland, D.J. and Nye, P.H. 1959. .Increase in the carbon and nitrogen contents of tropical soils under natural fallows. J. Soil Science 10:284-99.

Gregorich, E.G., Rochette, P., McGuire, S., Liang, B.C. and Lessard, R. 1998. Soluble organic carbon and carbon dioxide fluxes in maize fields receiving spring-applied manure. Journal of Environmental Quality 27, 209-214.

Grim,R .E.,1953.Clay Mineralogy.McGraw Hill, NewYork.N.Y.,384pp

Hassink, J. 1996. Preservation of plant residues in soil differing in unsaturated protective capacity. Soil Science America Journal 60, 487-491

Hassink, J., Whitmore, A.P. and Kubát, J., 1997. Size and density fractionation of soil organic matter and the physical capacity of soils to protect organic matter. European Journal of Agronomy 7, 189-199.

Haynes, R.J., Cameron, K.C., Goh, K.M. and Sherlock, R.R. 1986. Mineral Nitrogen in the Plant-Soil Systems pp 82-106.Academic Press.Inc.

Heal, O.W., Anderson, J.M. and Swift, M.J. 1997. Plant litter quality and decomposition: An historical overview. p. 3–30. *In* G. Cadisch and K.E. Giller (ed.) Driven by nature: Plant litter quality and decomposition. CAB Int., Cambridge, UK.

Henriksen, T.M. and T.A. Breland. 1999. Evaluation of criteria for describing crop residue degradability in a model of carbon and nitrogen turnover in soil. Soil Biol. Biochem. 31:1135–1149.

Hesse, P.R. 1971. A textbook of soil chemical analysis. New York, John Murray Publishers Ltd.

Hobbie, S.E. 2000. Interactions between litter lignin and soil nitrogen availability during leaf litter decomposition in a Ha-waiian montane forest. Ecosystems 3:484–94.

Hobbie S.E. and Vitousek, P.M. 2000. Nutrient regulation of decomposition in Hawaiian montane forests: do the same nutrients limit production and decomposition? Ecology. 81:1867–1877.

Honeycutt, C.W. and Potaro, L.J.1990. Field evaluation of heat units for predicting crop residue carbon and nitrogen mineralization. Plant Soil 125:213–220

Honeycutt, C.W., Potaro, L. J., Avila, K.L. and. Halterman, W.A .1993. Residue quality, loading rate, and soil temperature relations with hairy vetch (*Vicia villosa* Roth) residue carbon, nitrogen, and phosphorus mineralization. Biol. Agric. Hortic. 9:181–199.

Hu, S., Berkeley, F.S., Chapin, II., Firestone, M.K., Field, C.B and Chiariello, N.R. 2001. Nitrogen limitation of microbial decomposition in a grassland under elevated CO_2. Carnegie Institution of Washington. *Published:* Nature .Jan. 11, 2001.

Hunt, H.W., Ingham, E.R, Coleman, D.C, Elliott, E.T. and Reid, C.P.P. 1988. Nitrogen limitation of production and decomposition in prairies, mountain meadow, and pine forest. Ecology 69:1009–1016.

IPCC .2000. Land-use, Land use Change, and Forestry .Eds: Watson, R.T., Noble, I.R., Bolin,B., Ravindranath, N.H.., Verado, D.J. and Dokken, D.J. *A Special Report of the Intergovernmental Panel on Climate Change. pp.30.* Cambridge University Press, NY.

IPCC. 2001. Climate Change 2001: Impacts, adaptation and vulnerability . Eds: McCarthy, J.J., Canziani, O.F., Leary, N.,A., Dokken D.J and White,K.S. *A Special Report of the Intergovernmental Panel on Climate Change. pp. 30.* Cambridge University Press, NY.

Jenkinson,D.S. and Rayner, J.H. 1977. The turnover of soil organic matter in some of the Rothamsted classical experiments. Soil Sci., 123:298-305

Jenkinson, D.S., Hart, P.B.S., Rayner, J.H. and. Parry, L.C. 1987. Modeling the turnover of organic matter in long-term experiments at Rothamsted. INTECOL Bulletin 15, 1–8.

Jenkinson D.S.1990.The turnover of organic carbon and nitrogen in soil. Philosophical Transactions of the Royal Society .London series.B.329, 361-368.

Jenkinson, D.S., Adams, D.E. and Wild, A. 1991. Model estimates of CO_2 missions from soil in response to global warming. Nature 351:304-306.

Jenny, H., Gessel, S.P. and Bingham, F.T.1949. Comparative study of decomposition rates of organic matter in tropical and temperate regions.Soil Science 68:419-32.

Jensen, E.S. 1994. Mineralization–immobilization of nitrogen in soil amended with low C/N ratio plant residues with different particle size. Soil Biol. Biochem. 26:519-521 b.

Johnson, R.A. and Wichern, D.W. 1998. Applied multivariate statistical analysis. 4th ed. Prentice Hall, Upper Saddle River, NJ. 816p

Juma, N. G. and McGill, W. B. 1986. Decomposition and nutrient cycling in agro-ecosystems. pp 74-136 In M. J. Mitchell and J. P. Nakas (ed) Microfloral and faunal interactions in natural and agro-ecosystems. Martinus Nijhoff/Dr. W. Junk Publishers

Kaboneka, S., Sabbe, W.E. and Mauromoustakos, A. 1997. Carbon decomposition kinetics and nitrogen mineralization from corn, soybean, and wheat residues. *Commun. Soil Sci. Plant Anal.* 28 (15&16):1359-1373.

King, A.W., Post, W.M. and. Wullschleger, S.D. 1996. "The potential response of terrestrial carbon storage to changes in climate and atmospheric CO_2." Climate Change 35: 199–228

King J.S., Allen, H.L., Dougherty, P. and Strain, B.R. 1997. Decomposition of roots in loblolly pine: effects of nutrient and water availability and root size class on mass loss and nutrient dynamics. Plant Soil 195:171–84.

Kononova, M.M. 1966. Soil organic matter, its nature, its role in soil formation and in soil fertility. Pergammon Press, Oxford. pp 252.

Kowanlenko, C.G. 1978. Organic Nitrogen, Phosphorus and Sulfur in Soils pp 95-136. In: Soil Organic Matter.M.Schnitzer and S.U Khan-(eds). Elsevier Scientific Publishing Co., Newyork,NY

Kreger, C. 2004. Exploring the environment global climate change.Wheeling Jesuit University NASA. 1-12p

Kuo, S., Sainju, U.M. and Jellum, E.J. 1997. Winter cover cropping influence on nitrogen in soil. Soil Sci. Soc. Am. J. 61:1392–1399.

Kuo, S. and Sainju, U.M. 1998. Nitrogen mineralization and availability of mixed leguminous and non-leguminous cover crop residues in soil. Biol. Fertil. Soils 22:310–317.

Lal, R. 1997. Residue management, conservation tillage and soil restoration for mitigating greenhouse effect by CO_2- enrichment. Soil & Tillage Research. 43, 81-107.

Lal, R., Kimble, J.M., Follett, R. F. and Cole, C.V. 1999. Managing U.S. cropland to sequester carbon in soil. Journal of Soil Water Conservation. 54: 374-381.

Larson, W.E., Clapp, C.E., Pierre, W.H. and Morachan, Y.B., 1972. Effects of increasing amounts of organic residues on continous corn. II. Organic carbon, nitrogen, phosporus and Sulphur. Agron.J. 64: 204-208

Liski, J., Ilvesniemi, H., Makela, A. and Westman, C.J. 1999. CO_2 emissions from soil in response to climatic warming are overestimated - The decomposition of old soil organic matter is tolerant of temperature. Ambio 28: 171-174

Magill, A.H.and Aber, J.D. 1998. Long-term effects of experimental nitrogen additions on foliar litter decay and humus formation in forest ecosystems. Plant Soil 203:301–11

Mahendrappa, M.K. 1978. Changes in the organic layers under a black spruce stand fertilized with urea and tripple superphosphate. Can. J .For .Res.8, 237-242.

Maikslenien, S. 2000. Possibilities of primary tillage reduction in clay loam soil. The results of long- term field experiments in Battic States, pp106-113.Jelgava.In Lauringson ,E.,

Talgre,L., Roostatu ,H. and Vooper,H. Agronomy Research 2 (ii) ,63-70,2004.The effect of tillge and crop rotation on the content of available nitrogen, phosphorus and potassium.

Manu, A., Bationo, A. and Geiger, S.C. 1991. Fertility status of selected millet producing soils of West Africa with emphasis on phosphorus. Soil Sci. 152:315-320.

Martin, J.P. and Haider, K 1986. Influence of mineral colloids on turnover rates of soil organic carbon. p. 283- 304. In P.M.H.;M. Schnitzer (ed.) Interactions of Soil Minerals with Natural Organics and Microbes. SSSA Spec. Publ. 17. Soil Sci. Soc. Amer., Madison, WI.

McBridge, M.B.1994. Environmental chemistry of soils. Oxford University Press, Oxford,UK. p336

McGill, W.B. 1996. "Review and classification of ten soil organic matter (SOM) models." In D.S.Powlson, P.Smith, and J.U. Smith (eds), Evaluation of soil organic matter models using existing, long-term datasets. NATO ASI Series, Global Environmental Change, Vol. 38, pp. 111–132, Springer Verlag, Berlin.

McGill, W.B., Hunt, H.W., Woodmansee, R.G. and Reuss, J.O. 1981. PHOENIX: A model of the dynamics of carbon and nitrogen in grassland soils. Ecol. Bull., 33: 49-115.

McKenney, D.J., Wang, S.W., Drury, C.F. and Findlay, W.I. 1995. Denitrification, immobilization, and mineralization in nitrate limited and non limited residue-amended soil. Soil Sci. Soc. Am. J. 59:118–124.

McNelly, J. 1999. National Technological Composting systems. Inc, http;//www; composter. com.thus sep.2:13, 43:12

Melillo, J.M., Aber, J.D. and Muratore, J.F.1982. Nitrogen and lignin control of hardwood leaf litter decomposition dynamics. Ecology 63:621–6.

Merritts, D., Dewet, A. and Menking, K. 1998. Environmental Geology. An Earth System Science Approach. W.H. Freeman and company, New York, NY.1-179

Miller, P., Engel, R.and Brinklemyer, R. 2004. Soil carbon Sequestration: Farm Management Practices Can Affect Greenhouse Gases. MontGuide Fact Sheet #200404/Agriculture from the Montana State University Extension Service Issued April 2004

Müller, M.M., Sundman, V.., Soininvaara, O. and Meriläinen, A.. 1988. Effect of chemical composition on the release of nitrogen from agricultural plant materials decomposing in soil under field conditions. Biol. Fertil. Soils 6:78–83.

Mulungoy, K. and Merckx, R. 1991. Soil Organic Matter Dynamics and Sustainability of Tropical Agriculture.John Wiley and Sons, Chichester-New York, Toronto. pp7-10

Murphy, J. and Riley, J.P. 1962. A modified single method for the determination of phosphate in natural waters. Anal. Chim. Acta. pp. 27: 31-31.

Nichols, 1984. Relation of organic carbon to soil properties and climate in the Southern Great Plains. Soil Sci.Soc. Amer, J.pp 1382-1384.

Nziguheba, G. 2001. Improving phosphorus availiablility and maize production through organic and inorganic amendments in phosphorus deficient soils in Western Kenya. PhD Thesis Katholieke University Leuven, Belgium .pp .462

O'Connell, A.M. 1994. Decomposition and nutrient content of-litter in a fertilized eucalypt forest. Biol Fertil Soils 17:159–66.

Oglesby, K.A.and Fownes, J.H. 1992. Effects of chemical composition on nitrogen mineralization from green manures of seven tropical leguminous trees. Plant Soil 143(1):127–132.

Olson, J.S.1963. Energy Storage and the balance of producers and decomposers in ecological systems. Ecology 44:322-330.

Parker, D.T., 1962. Decomposition in the field of buried and surface cornstalk residue. Soil Sci. Soc.Am.Proc., 26: 559-562.

Parr, J.F. and Papendick, R.I. 1978. Factors affecting the decomposition of crop residues by microorganisms. p.101–129. In: W.R. Oschwld (ed.) Crop residue management systems. ASA Spec. Publ. 31. ASA, CSSA, SSSA, Madison, WI.

Parton, W.J., Anderson, D.W., Cole, C.V. and Stewart, J.W.B. 1983. Simulation of soil organic matter formation and mineralization in somi-arid agroecosystems. pp. 533-550 In R.R. Lowrance et al. (ed) Nutrient Cycling in Agricultural Ecosystems. University of Georgia special Publications, vol. 23.

Parton, W.J., Schimel, D.S., Cole, C.V. and. Ojima,D.S. 1987. Analysis of factors controlling organic matter levels in Great Plains grasslands. Soil Sci. Soc. Am. J. 51:1173–1179.

Parton, W.J., Ojima, D.S., Cole, C.C. and Schimel, D.M. 1994. A general model for soil organic matter dynamics: Sensitivity to litter chemistry, texture and management. p. 147–167. *In* R.B. Bryant and R.W. Arnold (ed.) Quantitative modeling of soil forming processes. SSSA Spec. Publ. 39. SSSA, Madison, WI

Pastor, J., Stillwell, M.A. and Tilman, D. 1987. Little blue stem litter dynamics in Minnesota old fields. Oecologia 72:327–30.

Paul, E.A. and Clark F.E. 1989. Soil Microbiology and Biochemistry. 2nd ed. San Diego: Academic Press. p 11 -234

Paul, E.A., Paustian, E., Elliott, T.and Cole, C.V. 1997. Soil organic matter in temperate agroecosystems: Long-term Experiments in North America. CRC Press, Boca Raton, FL.224 p

Paustian, K. 1998. Opportunities for Agriculture to mitigate greenhouse gases: A Grass Roots Approach. Agricultural Outlook forum. Feb.23, 10:30-12:15

Paustian, K., Conant, R., Ogle, S., Paul, E. and Six, J. 2002. Environmental and Management drivers of soil organic carbon stock changes. University of California-Davis 15-18 October Plenary Session 2 OECD Expert Meeting on Soil Organic Carbon Indicators for Agricultural Land. Ottawa, Canada

Phogat, V.K., Agrawal, R.P., Oswal, M.C. and Kuhad, M.S.1999. Laoratory Manual for Soil Physical Analysis.Dept. of Soil Sci., CCS Haryana Agricultural University, India, Pp 16-18.

Prescott, C.E. 1995. Does nitrogen availability control rates of litter decomposition in forests? Plant Soil Vol.168–9:83–8.

Prescott ,C,E., Corbin, J.P. and Parkinson, D. 1992. Immobilization and availability of N and P in the forest floors of fertilized Rocky Mountain coniferous forests. Plant Soil 143:1–10

Purves, W. K. and Orians, G.H. and Helle, H.C.1995. Life. The Science of Biology. Fouth Edition, Sinaver Associates, Inc., Sunderland. M.A. pp. 305

Quemada, M. and Cabrera., M.L. 1995.Carbon and nitrogen mineralization from leaves and stems of four cover crops.Soil Sci.Soc.Am.J.59:471-477.

Recous, S., Robin, D., Darwis, D. and Mary, B. 1995. Soil inorganic N availability: effect on maize residue decomposition. Soil Biol. Biochem.; 27:1529-1538

Reinertsen, S.A., Elliott, F.F., Cochran, V.L. and Campbell, G.S. 1984. Role of available carbon and nitrogen in determining the rate of wheat straw decomposition. Soil Biol. Biochem. 16:459–464.

Rice, C.W. and Angle J.S. 2004. Applications of biotechnology to mitigation of greenhouse warming: A role for genetically modified organisms in soil carbon sequestration. P. 61-78. In: Proceeding of the St. Michael's II Workshop, April 2003.

Rice, C.W., White, P. M., Fabrizzi, K. P. and Wilson, G. W. T. 2004. Managing the microbial community for soil carbon management.In: Sighn B (ed) Supersoil. 2004 3rd Australian New Zealand Soil Conference, 5-9 Dec. 2004. University of Sydney. Australia

Ruffo, M .L. and Bollero., G. A. 2003 .Residue Decomposition and Prediction of Carbon and Nitrogen Release Rates Based on Biochemical Fractions Using Principal-Component Regression In Modeling rye and hairy vetch residue decomposition as a function of degree-days and decomposition-days. Agron. J. 95:1034-1040.

SAS Institute. 1991. SAS user's guide: Version 6.03 ed. SAS Inst.Statistics. SAS Inst., Cary, NC.

Schimel, D.S., Stillwell, M.A. and R.G.Woodmansee, 1985. Biochemistry of C, N and P in a soil catena of the short grass steppe. Ecol.66:276-282.

Schlesinger, W.H. 1997. Biogeochemistry. An analysis of global change. 2nd ed. San Diego. Academic Press.p.156

Science Daily Magazine 1995—2003.Editor@Science daily.com

Shields, J. A and Paul, E.A. 1973. Decomposition of 4 C qabeled plant mat under field conditions. Can.J.Soil Sci.., 53: 297-306.

Smith, J.L., Papendick, R.I., Bezdicek, D.F. and. Lynch, J.M. 1992. Soil organic matter dynamics and crop residue management. p. 65–94. In: B. Metting (ed.) Soil microbial ecology. Marcel Dekker, New York.

Smith, P., Smith, J.U., Powlson, D.S., McGill, W.B., Arah, J.R.M., Chertov, O.G., Coleman, K., Franko, U., Frolking, S., Jenkinson, D.S., Jensen, L.S., Kelly, R.H., Klein Gunnewiek, H., Komarov, A.S., Li, C., Molina, J.A.E., Mueller, T., Parton, W.J., Thornley, J.H.M., Whitmore, A.P. and. Elliott, E.T. 1997. A comparison of the performance of nine soil organic matter models using datasets from seven long-term experiments. Geoderma 81:153–225.

Spain, A.1990 .Influence of environmental conditions and some soil chemical properties on the carbon and nitrogen of some Australian rainforest soils.Aust.J.Soil Res 28:828-839.

Staaf, H. 1980. Influence of chemical composition, addition of raspberry leaves and nitrogen supply on decomposition rates and dynamics of nitrogen and phosphorus in beech (Fagus sylvatica) leaf litter. Oikos 35:55–62

Steiner, J.L., Schomberg, H.H., Unger, P.W. and Cresap. J. 1999. Crop residue decomposition in no-tillage small-grain fields. Soil Sci. Soc. Am. J. 63:1817–1824]

Stroo, H.F., Bristow, K.L., Elliott, L.F., Papendick, R.I. and Campbell, G.S. 1989. Predicting rates of wheat residue decomposition. Soil Sci. Soc. Am. J. 53:91–99.

Stout, J.D., Goh, K.M. and Rafter,T. A. 1981. Chemistry and turnover of naturally occurring resistant organic compounds in soil. In: Soil Biochemistry (E.A. Paul and J.N.Ladd eds) Vol.5. pp.1-73. Dekker, New York.

Swift, M.J., Heal, O.W and Anderson, J.M. 1979.The influence of resources quality on decomposition processes, In: decomposition in Terrestrial Ecosystems, Oxford, UK, Blackwel.

Taylor, B.R., Parkinson, D. and Parsons, W.F.J. 1989. Nitrogen and lignin as predictors of litter decay rates: a microcosm test. Ecology. 70:97–104.

Theodorou, C. and Bowen, G.D. 1990. Effects of fertilizer on litterfall and nitrogen and phosphorus release from decomposing litter in a Pinus radiata plantation. For Ecol Manage 32:87–102.

Thien, S.J. and Graveel, J.G. 1997. Laboratory Manual for Soil Science, 7th ed. Agricultural and Environmental Principles. pp. 167-183.

Thomas, R.J. and Asakawa., N. M. 1993. Decomposition of leaf litter from tropical forage grasses and legumes. Soil Biol. Biochem. 25:1351–1361

Thompson, L.M. 1957. Soils and Soil Fertility. 2nd edition. Mcgraw-willly book Company.New York.Toronto London .. pg. 85-102.

Thompson, L.M. and Troeh, F.R. 1978. Soils and Soil Fertility.4th edition. Mcgraw –Hill book Company.5:120-122.

Tisdale, S.L. and Nelson, W.L. 1966. Soil Fertility and Fertilizers. 2nd edition. Macmillan Company, New York, Colliner-macmillan limited. London. 5:133-137.

Titus, B.D. and Malcolm, D.C. 1987. The effect of fertilization on litter decomposition in clearfelled spruce stands. Plant Soil 100: 297–322.

Trinsoutrot, I., Recous, S., Bentz, B., Lineres, M., Chèneby, D. and Nicolardot, B. 2000. Biochemical quality of crop residues and carbon and nitrogen mineralization kinetics under non-limiting nitrogen conditions. Soil Sci. Soc. Am. J. 64:918–926.

Trumbore,S.E., Davidson,E.A, de Camargo, P.B, Nepstad, D.C,Martinelli, L.A. 1995. Belowground cycling of carbon in forests and pastures of Eastern Amazonia.Global Biogeochemical Cycles 9, 515-528.

Turner, J. and Lambert, M. 2000. Change in organic carbon in forest plantation soils in eastern Australia. *Forest Ecology and Management* 133, 231-247.

Vallis, I. and Jones, R. 1973. Net mineralization of nitrogen in leaves and leaf litter of *Desmodium intortum* and *Phaseolus atropurpureus* mixed with soil. Soil Biol.Biochem.5:391-398.

Van Faasen, H.G. and Smilde, K.W, 1985. Organic matter and nitrogen turnover in soils. In: Proceedings of Symposium, Nitrogen Management in Farming Systems in Humid and Subhumid Tropics. Kang, B.T. and Van der Heide, J. (eds) International Institute of Tropical Agriculture, Ibadan, Nigeria and Institute of Soil Fertility, Haren, The Netherlands. pp.39-55.

Vanlauwe, B., Nwoke, O.C., Sanginga, N. and Merckx, R. 1996.Impact of residues quality on the C and N mineralization of leaf and root residues of three agroforestry species. Plant Soil; 183:221-231.

Van Veen, J.A. and Paul, E.A. 1981 : Organic carbon dynamics in grassland soils.1. Background information and computer simulation. Can. J. Soil Science (61), P. 185-201

Van Vuuren, M.M.I. and Van der Eerden, L.J. 1992. Effects of three rates of atmospheric nitrogen deposition enriched with decomposition in a heathland. Soil Biol Biochem 24:527–32.

Vigil, M.F. and. Kissel. D.E 1991. Equations for estimating the amount of nitrogen mineralized from crop residues. Soil Sci. Soc. Am. J. 55:757–761

Vigil, M.F., and Kissel. D.E. 1995. Rate of nitrogen mineralized from incorporated crop residues as influenced by temperature. Soil Sci. Soc. Am. J. 59:1636–1644

Vigil, M.F. and Sparks, D.L. 2002. Factors affecting the Rate of Crop Residue Decomposition under field conditions. Conservation tillage Fact Sheet No.3-95. Published by USDA-ARS, USDA-NRCS, Alamosa, Co. p 1-5

Vine, H. 1953. Experimentation on the maintenance of soil fertility at Ibadan, Nigeria, 1922-51. The Empire. J.of Exp. Agric .21:65-85.

White, P.M. Jr., Rice, C.W. and Tuinstra, M.R. 2003. Potential for enhanced soil carbon sequestration using lodging-resistant grain sorghum varieties. In: 2003 Agronomy Abstracts [CD-ROM]. ASA, Madison, WI.

White, R.E .1979. Introduction to the Principles and Practices of Soil Science. Blackwell-Scienticfic publications. Oxford, London Edinburgh Melbourne. 3: 24-37.

Wielopolski, L.', Mitra, S., Hendrey, G., Orion, I., Prior, S.A., Rogers Jr, H.H., Runion, G.B., Torbert III, H.A. 2004. Non-Destructive Soil Carbon Analyzer (Nd.sa). Technical Report, Terrestrial Carbon Processess (Tcp).Program of the office of science, Biological and

Environmental Research. (Ber), US. Department of Energy, Germantown, Md.89 Brookhawn National labourtory report 72, 2000 -2004.

Wilson, J.M. and Griffin, D.M. 1975. Water potential and the respiration of microorganisms' science in the soil .pp 141-163. In Soil Biol.Biochem.7, 199-204.

Wise, R. and Cacho, O. 1999. A Bioeconomic Analysis of Soil Carbon Sequestration in Agroforests.Working paper. CCO2. ACIAR project ASEM. 1999/093.

Woomer, P.L., Martin, A., Albrechit, A.R. and Scharpenseel, H. W.1994. The importance of management of soil organic matter in the tropics. 74-80 In: Biological Management of Tropical Soil Fertility.PL Woomer andMJ Swift (eds) John Wiley Chichester, UK.

World Reference Bases (WRB-Reference Groups (FAO/ISICI/ISSS, 1998). Food and Agriculture Organization of the United Nations Europeon Commission-Joint Research Centre.International Soil Reference and Information Centre.

Yoshida, T.1975. Microbial metabolism of flooded soils. In: Soil Biochemistry (E.A.Paul and A.D. McLaren, eds), Vol.3. pp 83-112. Dekker, New York.

APPENDIX 1

Analysis of variance for half-life at Water treatment W1

Source of variation	d.f.	s.s.	m.s.	v.r.	F pr.
treatments	4	1032.00	258.00	14.02	0.001**
Rep	2	110.80	55.40	3.01	0.106
Residual	8	147.20	18.40		
Total	14	1290.00			

Tables of means

Residue treatments	1.00	2.00	3.00	4.00	5.00	L.S.D.
	58.0	38.0	40.0	34.0	40.0	8.08

APPENDIX 2

Analysis of variance for Half –life of residue dry weight (Days) for W2 water treatments

Source of variation	d.f.	s.s.	m.s.	v.r.	F pr.
Treatments	4	585.60	146.40	2.63	0.114
Rep	2	12.40	6.20	0.11	0.896
Residual	8	445.60	55.70		
Total	14	1043.60			

Tables of means

Treatment	1.00	2.00	3.00	4.00	5.00	L.S.D	S.E
	62.0	48.0	50.0	50.0	62.0	14.05	4.31

S.E. –standard error of the means

APPENDIX 3

Analysis of variance for Half –life of residue dry weight (Days) for W3 water treatments

Source of variation	d.f.	s.s.	m.s.	v.r.	F pr.
Residue treatments	4	1394.400	348.600	108.94	<.001**
Rep	2	176.400	88.200	27.56	<.001**
Residual	8	25.600	3.200		
Total	14	1596.400			

Tables of means

Residue treatments	1.00	2.00	3.00	4.00	5.00	L.S.D	S.E
	80.00	85.00	84.00	76.00	104.00	3.368	1.033

APPENDIX 4

Analysis of variance for the k -value for residue RT1

Source of variation	d.f.	s.s.	m.s.	v.r.	F pr.
Water level	2	0.00002400	0.00001200	0.54	0.621
k-reps	2	0.00001067	0.00000533	0.24	0.798
Residual	4	0.00008933	0.00002233		
Total	8	0.00012400			

Tables of means

Water levels	1.00	2.00	3.00	L.S.D	S.E.
	-0.0130	-0.0110	-0.0090	0.01071	0.00273
kreps	1.00	2.00	3.00		
	-0.0110	-0.0097	-0.0123	0.01071	0.00273

APPENDIX 5

Analysis of variance for the k-value for residue treatment RT2

Source of variation	d.f.	s.s.	m.s.	v.r.	F pr.
Water levels	2	0.000218	0.00010900	6.67	0.053*
k-reps	2	0.00004267	0.00002133	1.31	0.366
Residual	4	0.00006533	0.00001633		
Total	8	0.00032600			

Tables of means

Water levels	1.00	2.00	3.00	L.S.D	S.E
	-0.0200	-0.0150	-0.0080	0.0092	0.00233

APPENDIX 6

Analysis of variance of the k-value for residue treatment RT3

Source of variation	d.f.	s.s.	m.s.	v.r.	F pr.
Water level	2	0.152E-03	0.760E-04	14.25	0.015*
k-reps	2	0.107E-04	0.533E-05	1.00	0.444
Residual	4	0.213E-04	0.533E-05		
Total	8	0.184E-03			

Tables of means

Water levels	1.00	2.00	3.00	L.S.D	S.E
	-0.01800	-0.01400	-0.00800	0.0052	0.0013

k-reps	1.00	2.00	3.00
	-0.01333	-0.01467	-0.01200

APPENDIX 7

Analysis of variance for the k-value for residue treatment RT4

Source of variation	d.f.	s.s.	m.s.	v.r.	F pr.
Water levels	2	0.258E	0.129E-03	96.75	<.001**
k-reps	2	0.267E-05	0.133E-05	1.00	0.444
Residual	4	0.533E-05	0.133E-05		
Total	8	0.266E-03			

Tables of means

Water level	1.00	2.00	3.00	L.S.D.	S.E.
	-0.02200	-0.01400	-0.00900	0.002618	0.00067

kreps	1.00	2.00	3.00
	-0.01500	-0.01433	-0.01567

APPENDIX 8

Analysis of variance for the k-value for residue treatment RT5

Source of variation	d.f.	s.s.	m.s.	v.r.	F pr.
Water levels	2	0.0002	0.00013	0.89	0.115
k-reps	2	0.00005	0.00002711	1.00	0.444
Residual	4	0.000108	0.00002711		
Total	8	0.00037356			

Tables of means

Water level	1.00	2.00	3.00	L.S.D	S.E
	-0.0187	-0.0110	-0.0070	0.01180	0.00301
k-reps	1.00	2.00	3.00		
	-0.0120	-0.0153	-0.0093		