

**Evaluating soil organic carbon, particulate
organic carbon and enzyme activities of soil
under different land use systems**

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By

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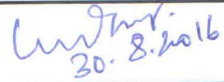
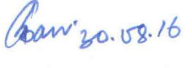
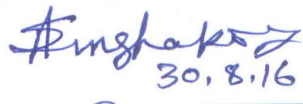
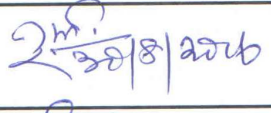
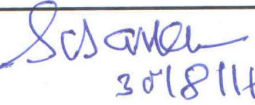
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Dedicated to my

**BELOVED
GRANDFATHER
AND
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ABSTRACT

As land use and agronomic management strategies influence the soil organic carbon (SOC) pools, this present study explored SOC, particulate organic matter (POM) of soil from four different land use systems. The difference in land use practices resulted in variable SOC contents where social forest exhibited highest average SOC (1.30%) resulting from higher plant litter deposition. Further, soil aggregate analyses indicated that cultivated land yielded significantly lower water stable soil macro-aggregates. Notwithstanding seasonal addition of manures and fertilizers, cultivated soils showed less C and N, probably resulting from continuous crop uptake, tillage practices, and periodical perturbation, ultimately resulting in enhanced organic matter turnover rate. Soil aggregates were destroyed from continuous cultivation, resulting in low physically protected C within aggregates in agricultural soils. Results showed lowest ratios of macro-aggregated C and N to micro-aggregated C and N in agricultural soils while highest ratios were found in social forest or agroforestry, confirming the relatively less aggregation as well as less aggregate associated C and N in cultivated soils. Further, this study explored the soil enzyme activities involved in C, N and P transformations. Averaging across all the crop rotations, cultivated land produced higher dehydrogenase activity ($16.25 \mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$) than fallow grass land ($11.40 \mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$) and social forest ($5.25 \mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$) in bulk samples, possibly as a result of inhibition from agrochemicals. Initial microbial respirations from fine and coarse POM mixed sand were higher in conservational agricultural practices than cultivated land as conservational agricultural managements significantly promote soil respiration and enzyme activities. The mean temporal N mineralization from both fine and coarse POM incubated at 40°C was higher in cultivated land than fallow grassland and relatively non-cultivated orchard. Future studies are warranted for complete biochemical characterization of POM extracted from different land use systems to explain the observed trends. Summarily, this study indicates the effect of variable land use systems on relatively unexplored biogeochemical dynamics of North Bengal soils.

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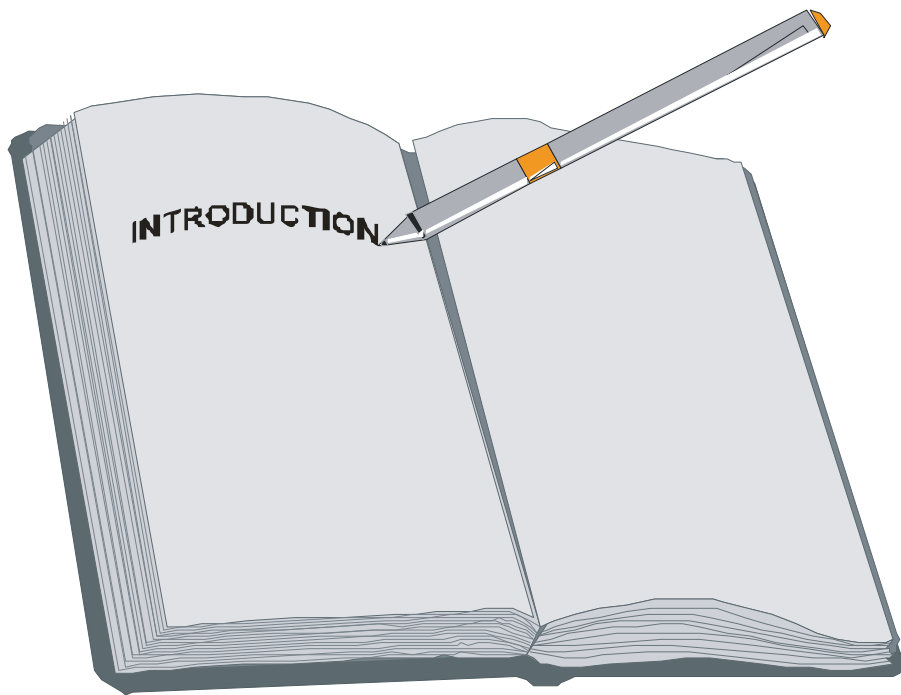
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CHAPTER-I



Introduction

Chapter -I

Introduction

Population growth and intensive cultivation aiming at maximizing productivity from limited land resources has caused serious natural resource degradation in India. This has resulted in low productivity of agricultural soils because of loss of nutrient through crop removal, water erosion and especially decline of soil organic matter (SOM) content. The SOM compasses plant, animal and microbial bodies in all stages of decay and a diversity of heterogeneous organic substances intimately associated with inorganic soil components (Christensen, 1992). Soil fertility and nutrient availability are closely related to SOM content and its mineralization (Zechand and Kogl-Kanaber, 1996). The SOM plays immense role in global carbon (C) cycle, both in serving as pool in sequestering C and also as a flux in releasing C. Land use and crop and soil management have drastic effects on the level of the soil organic carbon (SOC) pool.

Soil organic matter is an important indicator of soil quality and productivity (Larson and Pierce, 1991) and sustaining SOM quality and quantity is critical to ensuring long-term soil fertility (Tisdale *et al.*, 1993). The type of land use system is an important factor controlling organic matter contents of soils since it affects the amount and quality of litter input, decomposition rates and the processes of organic matter stabilization in soils (Six *et al.*, 1999). Changes of land use systems and management practices influence the amount and rate of SOM losses (Guggenberger *et al.*, 1995), thereby having its impact on agricultural productivity.

Dynamics SOC is gaining increasing importance for its impact on climate change and crop production. Organic C in soil has the slowest turnover rates in global ecosystems (Trumbore, 1997). Therefore C sequestration in soils has the capacity to mitigate CO₂ loading in atmosphere (Paustaian *et al.*, 1997). Soil is a significant long

term reservoir of organic C and plays a significant role in the global C cycle (Lal, 2003). The global carbon cycle is one of the most important biochemical cycles and soils are the largest earthly C sink, harboring approximately two-third of the C in ecosystems (Schimel *et al.*, 1994). Soils reserves organic C up to 615 Gt C (1 Gt =1 gigaton = 10^9 g) in the top 0.2 m soil depth and 2,344 Gt C at 3 m soil depth which is more than biomass and atmospheric CO₂ combined (Fontaine *et al.*, 2007).

Soil organic C storage is controlled by the soil-forming factors (Liang *et al.*, 2007). The roles of these factors as local and global biogeochemical process determine the C sequestration capacity of a soil. Among these factors, climate (temperature and precipitation) is the most important factor regulating soil organic C (Alveraze and Lavado, 1998). The stock of SOC is governed by the balance between C input and C losses, mainly through decomposition (Paustian *et al.*, 2000) and this C stock may increase or decrease depending on numerous factors including climate, vegetation type, nutrient availability, disturbance, land use, and management practices (Lal and Kimble, 1997; Carter, 2002).

The SOC stock is comprised of labile or actively cycling pool and stable, resistant/recalcitrant pools with varying residence time (Deb *et al.*, 2015). Labile C pool is the fraction of SOC with rapid turnover rates. It is important as energy source for the soil food web and thus influences nutrient cycling for maintaining soil quality and its productivity (Chan *et al.*, 2001). Some of the most important labile pools of SOC used as indicators of soil quality. These are microbial biomass C, mineralizable C, and organic C (OC) fractions. The first two pools of SOC generally signify the microbial dynamics through their population and activities in soil respectively. Changes in microbial dynamics in soil occur from the interactions of soil moisture, temperature, aeration, tillage and substrate availability (Feng *et al.*, 2003).

Changes in labile pools of SOC due to different soil management practices have been studied mainly in cooler and temperate regions of the world (Wu *et al.*, 2003; Sherrod *et al.*, 2005), but such studies in tropical and subtropical regions of the world are very few (Majumder *et al.*, 2008) although these studies are of special importance in this region since the organic C stock of soils here is inherently low. This low C stock poses a

serious threat to soil health and thus long-term sustainable crop production. Realization of the sink capacity of soils requires adoption of appropriate soil management practices that increase as well as stabilize SOC pools.

Soil C sequestration through enhanced aggregation is an important strategy of judicious soil management to mitigate the increasing concentration of atmospheric CO₂ (Shrestha *et al.*, 2004). Soil organic C associated with aggregates is an important reservoir of C, protected from mineralization because it is less subjected to physical, microbial and enzymatic degradation (Bajracharya *et al.*, 1998). Climate, soil type and mineralogy can also strongly influence both the degree of soil aggregation and SOC storage in soil (Six *et al.*, 2002; Zotarelli *et al.*, 2005); however, the mechanism of SOC protection within aggregates is poorly understood.

Aggregate stability of a soil defines the resistance of soil structure against mechanical or physico-chemical destructive forces (Duniway *et al.*, 2009), and soil structure is one of the factors controlling plant growth by its influence on root penetration, soil temperature and gas diffusion, water transport and seedling emergence and therefore it is an important soil characteristic for farmers. Soil structure defines the combination or arrangement of primary soil particles into compound elements, which are separated from adjoining structural elements by surfaces of weakness. Thus soil texture, soil structure, and the type of clay mineral, organic matter content and type, cementing agents as well as cropping history of a particular land collectively influence the aggregate stability (Goldberg *et al.*, 1988).

Soil aggregate stability is a fundamental property that determines its productivity through influencing a wide range of soil properties, including carbon stabilization, soil porosity, water infiltration, aeration, water retention, hydraulic conductivity, resistance to erosion and degradation (Six *et al.*, 2000). Maintaining high stability of soil aggregate is essential for preserving soil productivity, minimizing soil erosion and degradation and thus minimizing environmental pollution as well. This parameter is nowadays recognized as important soil quality measurements.

Soil aggregate stability is a product of interactions between soil environment, management practices, and land use patterns (Zhang *et al.*, 2008). Aggregate stability is dependent on soil type and texture class, on the content of SOM, biological activity of soil (Oades, 2005), fertilizer application (Annabi *et al.*, 2007), and also on soil tillage practices and vegetative cover. The differences in aggregate stability under different land use patterns are mainly due to the intensity of human disturbance and cultivation. Improper land use patterns will lead to the breakdown of unstable aggregates and production of finer and more-easily transportable particles and micro-aggregates (Zhang *et al.*, 2008).

Organic matter influences soil structure and stability by binding soil mineral particles, reducing aggregate wet ability, and influencing the mechanical strength of soil aggregates (Tisdall and Oades, 1982; Onweremadu *et al.*, 2007). A close relationship between SOC and aggregate stability has been established for temperate and for tropical soils and the retention of organic matter in soil depends on the quantity as well as quality of organics supply through plant residues or from manures (Six *et al.*, 2002; Bandyopadhyay *et al.*, 2010). However in the prevalent hot and humid climate, clay mineralogy and acidity hinders the aggregate binding agents like organic C and polyvalent cations and thus may decrease the aggregate stability in the study area. Hence taking up aggregation as the subject matter of study in the hot and humid region of Eastern India is of prime importance.

The available literature on land use and management effects on SOC dynamics mostly focus on changes in SOC pool. However, SOC pool is heterogeneously distributed among primary particles and aggregates and these SOC fractions respond differently to soil disturbances and management. Handling bulk SOC as one homogeneous pool ignores the variation in relative abundances of differently stabilized SOC fractions in response to change in environmental and climatic factors (Von Lutzow and Kogel-Knabner 2009). On the other hand, comprehensive evaluation of the effect of land use on the structural levels of SOC pools on similar soils has rarely been conducted. However, bulk SOM represents a heterogeneous mixture of operationally defined fractions that are characterized by differing compositions, microbial degradability and turnover time (von Lutzow *et al.*, 2006). The separation and

identification of labile SOM fractions from bulk organic matter is of particular importance for a mechanistic understanding of microbial decomposition processes and for predicting the response of SOM to changes in land use, management, and climate.

A desirable crop management practice would not only support sustained high production but also exhibit simultaneous potential of annual accretion in SOC stocks and thereby improving nutrient availability and soil health (Yadav *et al.*, 2000; Sleutel *et al.*, 2006). Identification and adoption of such practices assume great significance in subtropical and tropical regions, where SOC levels are inherently low (Mandal *et al.*, 2007). With the large-scale adoption of intensive cropping, the long-term balance between additions of organic C from different sources and losses through different pathways is disrupted, since on one hand more and more of the C is subjected to oxidative losses due to continued cultivation; while on the other hand, it leads to addition of C to the soil through crop residues resulting either a net build-up or depletion of SOC stock (Kong *et al.*, 2005). Although the intensity of land use and management has definite impact on SOC stocks, yet small changes are difficult to detect because of high background levels and natural soil variability (Elliot 1986).

Microbial decomposition of SOM plays a crucial role in the ecosystem nutrient cycling, in the development of soil structure, i.e., aggregation, and for the potential of soils to contribute to the greenhouse effect, among others. It is possible to assess microbial populations in the soil but this process is tedious and perhaps not very accurate because of the huge diversity of organisms and vast number that still have not been described (Morton, 1999).

Soil is a dynamic living system where all biochemical processes are taking place through enzymatic activities produced by microorganisms (Tabatabai, 1994). Some of the enzymes in the soil are associated with living cells (biotic) while others are abiotic (Skujins, 1976), or exclusive of live cells. There are several sources for the abiotic enzymes. Some are extracellular enzymes excreted from living cells into the soil solution to hydrolyze polymers that are too large or insoluble to be taken up directly by cells. Others are intracellular enzymes that leak into the soil solution from dead and lysed cells. These abiotic enzymes can be adsorbed and stabilized on the humic colloids and clay surfaces in the soil, remain catalytic, and reflect the cumulative effects of soil management on soil biology.

To carry out cycling and decomposition activities, soil enzymes are the mediators and catalysts of different organic matter decomposition, accumulation, mineralization and plant nutrient release process (Fansler *et al.*, 2005). Since enzyme activities show a rapid response to change in management practices, they have also been suggested as early indicators of changes in soil properties (Ekenler and Tabatabai, 2004). Soil enzymes produced by microorganisms play a key role in the process of SOC mineralization. They catalyze the decomposition of different organic substrates, release plant nutrients and influence whether SOC is depleted or sequestered (Fansler *et al.*, 2005). The distributions of SOC and microbial activities in soil aggregate fractions are important for determining how agricultural practices influence soil nutrient release and SOC balance (Kandeler *et al.*, 1999; Lagomarsino *et al.*, 2009).

The term “particulate organic matter” (POM) is sometimes used to describe organic particles isolated by size alone or by a combination of size and density. Gregorich *et al.* (2006) defined POM as the organic particle fraction isolated by size. It has been proposed that heterotrophic microorganisms in aerated soils gain most of their energy from organic particles (POC) and water-soluble OM (Gregorich *et al.*, 2003; Wander, 2004). However, some evidence exist that organic particles, as well as water-soluble SOM in soils, are not labile in total but instead consist of sub-fractions with various degrees of lability. One reason for this seems to be that organic particles as well as water-soluble organic components (e.g., polysaccharides) may act as binding agents during the formation of both macro- and micro-aggregates in soils (Golchin *et al.*, 1994; Six *et al.*, 2000). It can be hypothesized that sub-fractions of organic particles which are either physically uncomplexed or occluded in macro-aggregates and micro-aggregates, differ in lability and accessibility to microbial decomposition.

In the Eastern part of India, a number of land uses like orchard, agro forestry, forest, cropland, dry fallow etc. are practiced; however, there is little or no information on the impact of different land uses on the aggregate stability and soil C storage. Hence, the determination of the SOC stock and distribution in different aggregate sizes is important for determining the stability as well as the quantity of soil that is easily lost by erosion processes. There is also a hypothesis that minerals and aggregates could enhance

C stability by physically impeding the enzymatic degradation of C compounds. Soil aggregation could interfere with enzymatic decomposition by preventing microbes and enzymes from making contact with substrates inside aggregates (Sollins *et al.*, 1996). By investigating the soil enzyme activities involved in C, N, and P transformations, our objective would be to test whether the different land use pattern modified the microbial activities associated with the SOC balance at the soil aggregates. With the given background, the present study entitled “**Evaluating soil organic carbon, particulate organic carbon and enzyme activities of soil under different land use systems**” was undertaken with the following objectives:

OBJECTIVES:

- a. To elucidate the effect of different land use pattern on soil aggregates stability and the distribution of organic carbon in different aggregate fractions.
- b. To study the soil enzymes activities in different soil aggregate fractions.
- c. To study the microbial respiration on particulate organic matter of soils as influenced by land use pattern.
- d. To study the nitrogen and phosphorus supplying capacity of particulate organic matter.

CHAPTER-II



———— Review of Literature

Chapter -II

REVIEW OF LITERATURE

Pedosphere plays an important role in the global C cycle and a substantial part of the current atmospheric C pool comes from the global ecosystems (soil, freshwater sediments) (Cole *et al.*, 2007). The wide-reaching expansion of agriculture in the last decades has been accompanied by a rapid oxidation of organic carbon from soils while burning of fossil fuels from industries and automobiles has resulted in elevated levels of atmospheric CO₂ (four times pre-industrial level) (Vellinga and Wood, 2008). The global soil C pool of approximate 2500 Gt includes about 1550 Gt of SOC and 950 Gt of C (inorganic) (Lal, 2004a; Fontaine *et al.*, 2007). The soil C pool is 3.3 times of the size of the atmospheric C pool (760 Gt) and 4.5 times of the size of biotic C pool (560 Gt) (Lal, 2004a). Conversion of natural agricultural ecosystems causes depletion of SOC stock by as much as 60% in soil of temperate regions and 75% or more in cultivated soils of the tropics.

Severe depletion of SOC pool degrades soil quality and health, reduces biomass productivity, adversely impact water quality and the depletion may be exacerbated by projected global warming (Lal, 2004a). Soil organic C also imports desirable physical environments to soils by favourably affecting soil structure expressed through soil porosity, aggregation and bulk density and soil water storage (Benbi *et al.*, 1998; Chenu *et al.*, 2000). Organic C also exerts a significant influence on chemical properties of soils (Duxbury *et al.*, 1989; Rowsell *et al.*, 2004; Singh *et al.*, 2010), nutrient availability (Benbi and Biswas, 1997; Nieder and Richter, 2000), cation exchange capacity (Van Dijk, 1966) and retention and mobilization of metals (Stevenson, 1986). On the other hand, labile organic carbon is the soil nutrient reservoir and is closely related with diversified soil biology.

Since soils are the largest global C sink and there is a growing need to increase our understanding of the dynamics of C stock in soils and the role that soils may play in the long-term accumulation and sequestration of atmospheric C. The present research work is based on these ideas and literature pertaining it has been reviewed under the following heads:

2.1. Soil C sequestration

2.2. Pools of soil C

2.2.1 Soil organic C

2.2.2 C pools related to soil micro-organisms

2.2.3 Particulate organic carbon

2.2.3.1 Management effects on particulate organic matter

2.2.4 Soil aggregate associated C

2.2.5 Effect of land use on aggregate stability

2.2.5.1 Factors influencing soil aggregate stability

2.3. Land use on carbon storage

2.4. Factors influencing soil C budget

2.4.1 Climate

2.4.2 Soil properties

2.4.3 Land use and management

2.4.4 Cropping System

2.5 Soil enzymes

2.5.1. Role of Soil enzyme

2.5.2 Dehydrogenase

2.5.3 β -Glucosidase

2.5.4 Protease

2.5.5 N-acetyl- β -glucosaminidase

2.5.6 Acid- and alkaline-phosphatase enzyme

2.6 Soil Carbon Mineralization

2.6.1. Carbon fractions and soil respiration

2.6.2 Temperature and carbon mineralization

2.6.3 Land use and C-mineralization

2.6.4 Effect of moisture on C mineralization

2.7. Nitrogen Mineralization

2.7.1 Model describing soil nitrogen mineralization potential

2.7.2 Factor affecting soil nitrogen mineralization

2.7.2.1 Soil moisture and temperature

2.7.2.2 Wet/Dry cycles

2.7.2.3 Total soil N and soil organic C

2.7.2.4 Soil Texture

2.7.2.5 Soil rhizosphere and soil microbes

2.8. Phosphorus Mineralization

2.1. Soil C sequestration

Carbon sequestration can be defined as the capture and secure storage of C that would otherwise be emitted to or remain in the atmosphere i.e. transferring atmospheric CO₂ into long-lived pools and storing it securely so it is not immediately reemitted to the atmosphere. The C saturation implies that once the capacity for a soil to stabilize C is reached, additional C inputs will not be stabilized as SOC (Paustian *et al.*, 1997; Six *et al.*, 2002). Therefore, determining the C status of a soil relative to C saturation is important to gauging the potential for C sequestration.

Agricultural practices can render a soil either a sink or a source of the atmospheric CO₂, with direct influence on the greenhouse effect (Piao *et al.*, 2010). However, before soil can be credited as a sink of atmospheric CO₂, there are needs to understand the C sequestration process in the soil and develop appropriate methodology for determining how much and for how long C is sequestered in the soil. This means the methods must be able to measure the specific proportion of SOC (or the specific SOC pools) involved in C sequestration before they are useful for global C budgeting purposes. Soil C can be increased to new higher equilibrium with sustainable management practices. Long-term fertility experiments (LTFE) are the primary source of information to determine the effects of cropping systems, soil management, fertilizer use and residue utilization on the quantitative and mechanistic changes in SOC pools (Grace *et al.*, 2006). The LTFEs in both Europe and North America indicate that SOM and soil C are lost during intensive cultivation, typically showing exponential decline after the first cultivation of virgin soils, but with continuing steady loss over many years (Rasmussen *et al.*, 1998; Tilman, 1998; Robert *et al.*, 2001; Ogle *et al.*, 2003; Shi *et al.*, 2010). In Indian context., Katyal *et al.* (2001) had proven that treatment and practice that

supported SOM buildup, also favored sustainable productivity in 30 years old LTFE in tropical soil. A wide range of long-term comparative studies show that organic and sustainable systems improve soils through accumulation of soil organic matter (SOM) and soil C, with accompanying increases in microbial activity (Kong *et al.*, 2005; Lal, 2006; Manna *et al.*, 2006; Rudrappa *et al.*, 2006). Observed rates of C sequestration range from 0-150 kg C ha⁻¹ in humid and cool in dry and warm regions (Armstrong *et al.*, 2003) and 100 to 1000 kg C ha⁻¹ y⁻¹ in humid and cool climate (Sa *et al.*, 2001). The rate of C (inorganic) sequestration as secondary carbonates is low (5 to 150 kg C ha⁻¹ y⁻¹) and is accentuated by biogenic processes and leaching of carbonates into ground water, especially in soils irrigated with water containing higher carbonates. However, to understand the soil C sequestration it is indeed important to understand the pools of soil C.

2.2. Pools of soil C

2.2.1 Soil organic C

The SOC stock is comprised of active or labile pool and stable, passive or recalcitrant pool (Majumder *et al.*, 2008a). This active or labile pool of SOC is one of the most important components contributing in soil health, sustainability and crop production (Lal, 2004; Bationo *et al.*, 2007; Ghosh *et al.*, 2012) as well as the food and energy source for soil micro-organisms (Steenwerth *et al.*, 2002) whereas microbial activities regulate decomposition, mineralization, immobilization, oxidation-reduction, transformation of SOM, soil aggregation, nutrient cycling (Motavalli *et al.*, 2004; Kallenbach and Grandy, 2011).

Venkanna *et al.*, (2014) studied and the result shows that, Vertisols and associated soils contained greater total C stocks, followed by Inceptisols and Alfisols. The soil organic carbon (SOC) stock was highest in Alfisols (52.84 Mg ha⁻¹) followed by Inceptisols (51.26 Mg ha⁻¹) and Vertisols and associated soils (49.33 Mg ha⁻¹), whereas soil inorganic carbon (SIC) stock was highest in Vertisols and associated soil (22.9 Mg ha⁻¹) followed by Inceptisols (17.5 Mg ha⁻¹) and Alfisols (12.4 Mg ha⁻¹). Among the different land-use systems, total C stock was highest in forest soils followed by fodder system, paddy, maize, cotton, redgram, intercrop, chilli, permanent fallow and lowest in castor system.

Gupta *et al.*, (2014) 1141 samples were collected from different land uses viz. forests, block plantations, horticulture, agro-forestry and agriculture in different districts of southern region of Haryana then, Soil organic carbon (SOC) was estimated. The maximum SOC stock was under forests (37.61 t ha⁻¹), followed by horticulture (27.26 t ha⁻¹), plantation (27.96 t ha⁻¹), agriculture (17.72 t ha⁻¹) and agro-forestry land use (10.84 t ha⁻¹). The mitigation potential calculation revealed that soils under forests can sequester three times and plantations and horticulture land uses can hold more than double organic carbon as compared to agro forestry land use.

Quantitative data on SOC pools and fluxes at scales ranging from pedon or soil-scape to ecosystem and regional, national and global scales, are needed to assess the magnitude of soil C pool in relation to the biotic and atmospheric pool and to evaluate the contribution (flux) of the soil C pool to the atmospheric pool (Chan, 1997). On the other hand, stable, passive or recalcitrant pool of SOC is biochemically protected (Parton *et al.*, 1987), altered very slowly by microbial activities (Weil *et al.*, 2003). These pools are comprised of a range of resistant and non resistant aromatic and long chain aliphatic compounds because of the continuous nature of addition, stabilization, and decomposition of SOM. The most labile SOC components were considered sensitive indicators of change in the soil quality (Maia *et al.*, 2007). Labile SOC contains microbial biomass and is closely associated with nutrient mineralization and reflects organic input management over the past years (Barrios *et al.*, 1996). The labile or active C pools, typically including recent leaf, needle and root litter and microbial biomass, have been emphasised as an important source for heterotrophic respiration (Gaudinski *et al.*, 2000) and are known to control over nutrient availability in soils. In this context, dissolved organic matter (DOM) represents a small but very labile C pool (<10-20 g C m⁻² down to 1 m depth) (Ellert and Gregorich, 1995). In turn, a small part of the DOM (<10%) consists of identifiable compounds of low molecular weight such as organic acids, amino acids and simple sugars. These compounds are quickly assimilated by the soil microbial biomass, and are recognised to fuel microbial activity in the rhizosphere due to their presence in root exudates (Dakora and Philips, 2002).

2.2.2 C pools related to soil micro-organisms

Micro-organisms are indispensable for key soil functions like decomposition of SOM, degradation of xenobiotics, and formation of soil aggregates (Stenberg, 1999).

They also act as a source and sink for plant nutrients (Salinas-Garcia *et al.*, 1997). The microbial biomass denotes the living component of SUM (Jenkinson and Ladd, 1981). Microbial biomass in soil is composed of bacteria, fungi, algae, actinomycetes, protozoa and some nematodes. Soil microbial biomass serve as: (i) a labile source or an immediate sink of available nutrients and organic substrates on a short term basis, (ii) a driving force of nutrient transformation from stable organic forms to available mineral form over longer periods, (iii) an agent of polysaccharide secretion, a cementing agent of soil aggregates (Kennedy and Papendick, 1995) and (iv) an agent for releasing and containing enzymes which are responsible for nutrient cycling (Srivastava and Singh, 1991).

Soil respiration (CO_2 -C release) is an index of total soil biological activity including soil micro-organisms, macro-fauna and plant roots. It reflects the overall activity or energy spent by the indigenous soil microbial pool (Anderson and Domsch, 1990). Measurement of C mineralisation yields an index of total C availability. As noted by Sparling (1997), microbial respiration is highly variable and can show wide variation depending on substrate variability, moisture and temperature. Rate of soil respiration depends on the substrate availability, moisture and temperature of the soil (Brookes, 1995). The most straightforward soil microbial index used in the literature is the soil metabolic or respiratory quotient ($q\text{CO}_2$) (soil respiration to microbial biomass ratio), widely used to evaluate ecosystem development, disturbance or system maturity (Bastida *et al.*, 2008).

2.2.3 Particulate organic carbon

Particulate organic matter (POM) is defined as this fraction associated with sand-sized particles, and separated from the soil by sieving. Cambardella and Elliot (1992) defined particulate organic matter (POM) as soil organic matter (SOM) particles less than 2 mm and greater than 0.053 mm in size. Generally, it consists mainly of fine root fragments and other organic debris. The POM can account for well over 10% of the soil C (Carter *et al.*, 1994; Gregorich *et al.*, 1994).

POM is biologically and chemically active and is part of the labile (easily decomposable) pool of soil organic matter (SOM). This pool is significant to SOM turnover because it serves as a readily decomposable substrate for soil microorganisms

and as short-term reservoir for plant nutrients. This fraction has been suggested as a sensitive indicator of changes of SOM because of its responsiveness to management practices (Gregorich and Carter, 1997). Vieira *et al.* (2007) showed that particulate organic matter ($>53\ \mu\text{m}$) can be used to determine the index that measures the lability of organic matter. The particulate fraction acts as a cementing agent, to stabilize macro-aggregates and as intra-aggregate protection of the proper organic matter (Six *et al.*, 2002b). Costa *et al.* (2004) found differential contributions of total organic and particulate organic carbon (more labile) in the formation of soil aggregates.

2.2.3.1 Management effects on particulate organic matter

Reduced tillage and no-till help to sequester more POM in the soil than cultivated systems (Angers *et al.*, 1993). Mrabet *et al.* (2001) showed that higher aggregation, carbon sequestration, pH decline and particulate organic matter (POM) build-up were major changes associated with shift from conventional to NT system. Better stability of aggregates was demonstrated by a significantly greater mean weight diameter under NT (3.8 mm) than CT system (3.2 mm) at the soil surface.

Sherrod, *et al.* (2002) evaluated the effect of no-till cropping systems of wheat fellow (WF), wheat-com-fallow (WCF), wheat-corn-millet-fallow (WCMF), opportunity cropping (OPP), and native grass species (G) on particulate organic matter (POM-C) and mineral associated C in the silt and clay fractions. Soil organic carbon (SOC) was increased with increasing cropping intensity after 12 years, of no-till management. Particulate organic matter C accounted for 38 to 52 % of the total SOC in the 0-2.5 cm, 24 to 48 % of the total SOC in the 2.5-5 cm depth in the 5-10 cm depth, POM-C accounted for 22-35 % of the total SOC and in the 10-20 cm depth, POM-C accounted for only 0 to 12 % of the total SOC depending on cropping intensity. In all depth increments however, G had the highest POM-C followed by OPP. Overall, the increase in total SOC in surface soils with increasing cropping intensity was due to increased C in the POM-C fraction.

Franzluebbers and Stuedemann (2002) determined the depth distribution of particulate organic C (POC), non-particulate organic C (NPOC), particulate-to-total organic C (POC-to-TOC) ratio, and particulate organic C-to-N (POC-to-N) ratio under pastures near Watkinsville, GA, USA. POC was highly related with total organic C

(TOC), but became an increasingly larger portion of TOC near the soil surface, where both pools were greatest. POC and NPOC were (i) greater under pasture than under conservation-tillage cropland, (ii) greater when pasture was grazed than when hayed, (iii) marginally greater with higher fertilization of pasture, (iv) greater with higher frequency of endophyte infection of tall fescue, and (v) greater under increasing stand age of grass.

2.2.4 Soil aggregate associated C

Aggregation is an important component of soil structure, controlling water storage, aeration, crop growth and biological activity, as well as soil erosion (Grandy and Robertson, 2006). Soil C sequestration through enhanced aggregation is an important strategy of judicious soil management to mitigate the increasing concentration of atmospheric CO₂ (Bronick and Lal, 2005; Shrestha *et al.*, 2007). The complex dynamics of aggregation are the result of the interaction of many factors including the environment, soil management factors, plant influences and soil properties such as mineral composition, texture, SOC concentration, pedogenic processes, microbial activities, exchangeable ions, nutrient reserves, and moisture availability (Kay, 1998). Aggregates occur in a variety of manners and sizes. These are often grouped by size: macroaggregates (>0.25 mm) and microaggregates (< 0.25 mm) with these groups being further divided by size (Tisdall and Oades, 1982). Micro aggregates are formed from organic molecules (OM) attached to clay (Cl) and polyvalent cations (P) to form compound particles (Cl-P-OM), which are joined with other particles (Cl-P-OM) to form macro aggregates [(Cl-P-OM)_x]_y (Edwards and Bremner, 1967; Tisdall, 1996). Alternatively, macro aggregates can form around particulate organic matter (POM). As POM is decomposed and microbial exudates are released, the macro aggregate becomes more stable, the C: N ratio decreases, and micro aggregates form inside. The internally formed micro aggregates contain more recalcitrant SOC pool (Plante and McGill, 2002; Beare *et al.*, 1994).

The SOC associated with aggregates is an important reservoir of C, protected from mineralization, because it is less subjected to physical, microbial and enzymatic degradation (Shrestha *et al.*, 2007). Terrestrial C sequestration partly is achieved through SOC stabilization by physical occlusion within aggregates (Six *et al.*, 2000). In natural soils, the activity of micro-organism and plant roots and the formation of organo-mineral complexes lead to aggregation of soil particles which influences soil C storage and

dynamics (Oades and Waters, 1991). Thus fresh plant- or root-derived material enhances aggregate formation during and following its decomposition by micro-organisms because it serves as a C source for microbial activity and production of microbial-derived aggregate binding agents (Puget *et al.*, 1995; Jastrow, 1996). The storage processes in soil aggregates occurred mainly in soils where SOM is considered a major binding agent that stabilizes soil aggregates (Tisdall and Oades, 1982). A hierarchical model suggested that three different classes of organic matter, persistent, transient, and temporary, are associated with three different physical soil fractions, that is, > 0.25 mm macro-aggregates, 0.05-0.25 mm micro-aggregates, and < 0.05 mm silt and clay, respectively and indicated that decomposability of SOM decreases with decreasing particle size owing to physical protection (Tisdall and Oades, 1982; Balesdent *et al.*, 2000). According to this model, transient forms of SOM could act as binding agent causing micro-aggregates (0.5-0.25 mm) to form stable macro-aggregates (> 0.25 mm) (Olchin *et al.*, 2008). Increased SOM input can lead to increased aggregate formation (Kong *et al.*, 2005) which in turn enhances C sequestration by physical protection of SOM inside aggregates. The macro-aggregate occluded micro-aggregate associated C fraction may be a highly accurate and general diagnostic fraction for changes in total SOC in response to changes in tillage and potentially other management practices (Gillabel *et al.*, 2007). Aggregate protected C pools are more labile than unprotected pools, because protected pools are less exposed to microbial decay (Beare *et al.*, 1994).

The quality and amount of SOC associated with soil aggregates and particles vary with the size of these structural units. It is often observed that C in micro-aggregates is older and less labile than C macro-aggregates (Jastrow, 1996). Microbial derived C is believed to bind micro-aggregates into macro-aggregates (Tisdall and Oades, 1982). Cambardella and Elliott (1992) suggested that microbial-derived C with an intermediate turnover would be sequestered into micro-aggregates within macro-aggregates. Studies indicated that the macro-aggregate structure exerts some physical protection to SOM (Beare *et al.*, 1994), whereas SOM is mostly protected in free micro-aggregates (Six *et al.*, 1998; Lichter *et al.*, 2008) and in micro-aggregates within the macro-aggregates (Denef *et al.*, 2001; Bossuyt *et al.*, 2002; Lichter *et al.*, 2008). A study on the impact of continuous application of rice straw and farm-yard manure (FYM) either alone or in

combination in rice-wheat cropping system showed that macro-aggregates constituted ~32-54% of the total water-stable aggregates and were linearly related to SOC under sandy loam soils of northwest India (Benbi and Senapati, 2009). Under similar cropping system in sub-humid tropical India, Bandyopadhyay *et al.* (2010) postulated that structural stability was higher with organically amended soils and green manure (GM) was less efficient in increasing aggregate stability. In both natural and agricultural soils, mineral sorption and soil aggregation are important mechanisms leading to C stabilization (Elliott, 1986). Sorption by soil minerals can protect organic material from microbial attack, thereby increasing the mean residence time of mineral-stabilized C (Kaiser and Guggenberger, 2003). Aggregation also physically protects soil C, with smaller aggregates generally containing relatively older C (Six and Jastrow, 2002). Roots and mycorrhizal fungal hyphae are important drivers of the relationship between aggregate stability and C storage because they serve as binding agents and a source of organic material to soil aggregates (Guggenberger *et al.*, 1999) and create conditions favoring the formation of micro-aggregates within macro-aggregates (Miller and Jastrow, 1992; Six *et al.*, 2000). When soils are converted for agriculture, C inputs from plants and fungi decline, and cultivation breaks up existing soil aggregates, leaving C within aggregates more vulnerable to decomposition (Cambardella and Elliott, 1993; Six *et al.*, 1998). Therefore, loss of soil aggregation is probably both a result and a cause of C loss (Beare *et al.*, 1997). As soils recover from agricultural disturbance, restoration of perennial root systems and aggregate structure lead to increases in C stocks, which then feed back to further enhance aggregate stability (Jastrow and Miller, 1998).

2.2.5 Effect of land use on Aggregate stability

Vegetation cover is one of the key factors influencing soil erodibility. This is due to the positive feedback of the vegetation on soil quality due to the organic matter contribution by means of the litter. In addition, vegetation increases weathering, enhances rainwater infiltration and favours a less contrasted microclimate beneath plants due to the shadow. These conditions should generate a more active fauna and flora and as a consequence, stronger aggregates Oades, 1993; Zhang, 1994. Micro-aggregates reflect the influence of parent material due to the influence of clays on aggregate formation, but macro aggregation mainly reflects the influence of plants organic matter on the aggregate formation (Duchafour, 1975). This is why this research has been

focused on the changes of the plant cover, organic matter and the stability of macro-aggregates. Under the vegetation type with greater shade more stable soils are developed. Beneath plants, fluctuations in temperature and soil moisture are less marked and this will result in more stable aggregates. The influence of climate has been found very important by other authors on Mediterranean climatic conditions (Boix *et al.*, 1995). Another influence of vegetation is the litter and organic matter production. The greater the organic matter incorporated into the soil and the less contrast in climate beneath the plant, the more stable the aggregates. The more developed is the vegetation floristic succession the greater is the aggregate stability. This explains the influence of vegetation type on runoff and erosion such as other researchers found in Central Spain (Williams *et al.*, 1995).

2.2.5.1 Factor Influencing Soil aggregate stability

Several factors influence aggregate stability. The clay minerals calcium, iron, magnesium and aluminum form organic matter-clay bridges, stabilizing aggregates. Conversely, high levels of exchangeable sodium cause soil particles to disperse, disrupting aggregate stability. Soils containing higher clay tend to have higher aggregate stability than coarser soils. Increasing organic matter promotes biological activity in the soil increasing aggregate stability. This can be achieved through the addition of compost or by planting cover crops. Compost stimulates microbial activity that enhances hydrophobicity and inter-particle cohesion, increasing aggregate stability by decreasing the rate of aggregate wetting (Lynch and Bragg, 1985).

2.3. Land use on carbon storage

Forests that cover about 30% of the land surface, store about 45% of total terrestrial carbon. In undisturbed natural forest ecosystems, the additions and losses of carbon are balanced over time and soil carbon stock reaches a stable equilibrium. Agro-ecosystems are manmade goal oriented systems and derived generally from forest. Agro-ecosystems by their nature are perturbed ecosystems and can be defined as ecosystems containing a significant component of anthropogenic inputs and outputs necessary for its regulation, with some fraction of the system harvested periodically (Odum, 1984). Agro-ecosystems are manipulated by frequent, marked anthropogenic modifications of the biotic and abiotic environments (Coleman and Hendrix, 1988). Land use change may

lead to changes in soil physical and chemical properties through their influence on various ecological processes (Chen *et al.*, 2010).

Land use changes have caused of ~20% greenhouse gases (GHGs) globally that leads to shrinkage of carbon (C) storage in the potential areas. Soil carbon pool change rapidly in response to land use change. Conversely, in-depth understanding of carbon dynamics is needed with respect to eco-systems variability. Sustainable land use systems can help in sequestering C and reduce GHGs adverse effect (Datta *et al.*, 2015).

Highest soil organic carbon concentration in natural forest found in this study was might be due to the regular addition of plant litter including above and below ground plant parts, and limited disturbances like grazing, logging etc. (Steinbeiss *et al.*, 2008). The contribution of roots in accumulation of soil organic carbon in soil was considered to be more compared to above ground biomass (Puget and Drinkwater, 2001). Elaborated root systems of perennials trees and ground flora of natural forest attributed largely to the storage and stabilization of soil organic carbon in aggregates (Singh *et al.*, 2009). The disturbances associated with deforestation might have lead to loss of vegetation which in turn have resulted in land degradation, erosion and subsequently the considerable losses of soil organic carbon and nutrients (Tripathi and Singh, 2009). Changes in land use pattern through changes in type and diversity of plants are reported to exert major influence on the transfer and accumulation of carbon in soil (Tilman *et al.*, 2006). Intensive tillage might have accelerated native soil organic matter oxidation by destructing soil aggregates and exposing newer sites to microbial attack, which in turn have resulted in loss of soil organic carbon (Singh and Ghoshal, 2006). The organic inputs to the soil either through exogenous inputs or through crop plants were little and this might be a reason for reduction in soil organic carbon content in soil (Singh and Ghoshal, 2011).

Reduction in the level of soil organic carbon from the natural forest due to cultivation was also reported by Jenkinson and Rayner (1977), Paul *et al.* (1997), Saha *et al.* (2010). Vesterdal and Leifeld (2010) illustrated that native lands typically stored higher amounts of soil organic carbon than croplands under similar site conditions because of higher residue inputs and reduced turnover. Gol (2009) also reported higher soil organic carbon content in natural forest than tilled croplands. Vegetation cover is one of the key factors influencing soil erodibility. This is due to the positive

feedback of the vegetation on soil quality due to the organic matter contribution by means of the litter. Also, vegetation increases weathering, enhances rainwater infiltration and favours a less contrasted microclimate beneath plants due to the shadow. These conditions should generate a more active fauna and flora and, as a consequence, stronger aggregates (Oades, 1993). Generally, it is accepted that conversion to crop production practices has caused a decline in SOC compared with the original grassland levels throughout the Great Plains (Allmaras *et al.*, 2000). Tillage has caused soil carbon losses from 28 to 77% depending on geographic location (climate) and soil type (Pikul *et al.*, 2007). Changes in agricultural management from conventional tillage to no tillage and increased crop-rotation diversity can increase accumulation of SOC (West and Post, 2002). Tillage disrupts aggregates mechanically, changes the soil climate (temperature, moisture, aeration) and accelerates organic matter decomposition (Balesdent *et al.*, 2000), reducing the proportion of stable aggregates > 0.25 mm (Six *et al.*, 1999; Carter, 2002).

Chenu *et al.*, (2000) found that SOC associated with clay minerals gave increased hydrophobicity, and that greater water stable aggregate (WSA) could be ascribed to resistance of aggregates to slaking. Under forest soils, the presence of litter recycles continuously organic carbon storage in top-soil horizons (Garcia-Pausas *et al.*, 2004). The possible decrease of the SOC stock from croplands contrasts with the prevailing theory which assumes the equilibrium between C inputs and outputs in the Ap horizon, i.e. the ploughed layer of agricultural soil (Ellmer *et al.*, 2000). The loss of SOC is attributable to the intensification of agricultural practices in soils cultivated for thousands of years (Gardi and Sconosciuto 2007) and could be reduced by taking into account some of the different mitigation options such as reduced tillage, improved management or the use of animal manure (Freibauer *et al.*, 2004; Triberti *et al.*, 2008). Factors such as crop type (Scott *et al.*, 1994), tillage (Pagliai *et al.*, 1995), and application of organic residues (Rasse *et al.*, 2000) can affect soil physical properties. Effects of cropping systems on soil physical properties are often related to increases in soil organic matter (SOM) (Dormaar, 1983; Haynes, 2000).

Soil aggregate stability (AS) is a dynamic property of soil that changes over time (Coote *et al.*, 1988). The stability of soil aggregates often decreases for soil under annual crops (Angers *et al.*, 1999). Kay *et al.*, (1994) reported that continuous cotton production

reduced soil wet AS compared to that of pasture. Gantzer *et al.*, (1987) reported that residue quantity had a greater effect on splash detachment, shear strength, and aggregate stability than did residue type. Continuous tillage and production may cause a reduction of soil AS.

Six *et al.*, (2000) reported that cultivation reduces soil C content and changes the distribution and stability of soil aggregates. Breakdown of macro aggregates results in the release of labile SOM accompanied by increased availability for microbial decomposition. The increased microbial activity depletes SOM, which reduces microbial derived binding agents and causes aggregate losses (Jastrow, 1996; Six *et al.*, 1998).

2.4. Factors influencing soil C budget

2.4.1. Climate

Soil organic C storage is controlled by the basic soil-forming and several other factors (Singh *et al.*, 2007; Liang *et al.*, 2007). Among these factors, temperature and precipitation (climate) are most important factors regulating SOC (Alvarez and Lavado, 1998). Climatic conditions with high precipitation, low temperature and frozen soil in winter favour the high accumulation of organic matter in soil. These conditions slower the breakdown of added SOM through plant residues, roots, and FYM and hence the C sequestration in soil may be expected to be larger than warmer and drier climates. Reducing soil temperature through the use of surface mulches and no-till practices is important for maintaining SOM stocks especially in tropical soils (Lal, 1989). The expected climate changes with higher mean temperatures could greatly enhance the breakdown of OC in Nordic soils. Post *et al.* (1982) reported that forests in cooler climates often contain nearly twice as much soil C as warm tropical forests. Desjardins *et al.* (1991) found that the distribution of SOC is related to the temperature, showing a faster decrease with increasing depth. In Indian context Bhattacharyya *et al.* (2000) showed that, prevailing cold climate and high rainfall resulted slow turnover rate and thus high SOC stock in some sub-Himalayan soils. On the other hand, the potential for sequestering SOC with crop rotations was greater in the sub-humid than in the drier climates and it is established that tropical soil contains the least C in a stable pool (Tiessen *et al.*, 1998). Again, the C sequestration potential is higher in humid temperate areas ($0.1-0.5 \text{ t C ha}^{-1} \text{ y}^{-1}$) than in semi-arid and tropical areas ($0.05-0.20 \text{ t C ha}^{-1} \text{ y}^{-1}$)

(Pretty and Ball, 2001). It is the high annual temperature which consequences the lowest average SOC stock in Indian soils among the Asian countries (Kyuma, 1988).

2.4.2 Soil properties

The interaction of SOC with the soil primary particles and its distribution among the soil structural units are dominant controllers of SOC accumulation and decomposition (Oades, 1998). Linear correlations were found between soil texture and the SOC and the gains in soil C under reduced tillage were related to soil clay content (Campbell *et al.*, 1996). According to Plante *et al.* (2006), for a given level of C inputs, the relationship between mineral surface area and SOM varies with soil texture for physically and biochemically protected C fractions. Guggenberger *et al.* (1994) found that SOC content increased with diminishing particle size, whereas the C/N ratio decreased. Particulate organic matter or organic matter associated with particle-size fractions is a good indicator of soil quality and responds rapidly and selectively to changes in land use and soil management (Kong *et al.*, 2005). Saggar *et al.* (1996) noted that, in general, more than the half of the total SOC is associated with the clay (<0.002 mm) fraction, probably due to the protection effect of clay on SOC mineralization. Further, the degree of protection provided by clay appears to be dependent on the type of clays, e.g. higher porous (allophane), expanding (montmorillonite) type of clay mineral offers more protection to SOC than kaolinite (Franzluebbers *et al.*, 1996; Zech *et al.*, 1997). In temperate soils, silt-associated SOM is thought to be the most recalcitrant fraction, while the turnover rate of clay-associated SOM is considered intermediate (Christensen, 1992). Considering exceptions (Bhattacharyya *et al.*, 2006), in general, it can be said that, due to different mineralogy, temperatures and precipitation, SUM contents and composition in tropical soils vary considerably from temperate soils.

2.4.3 Land use and management

Land use and land cover changes are widely recognized as key drivers of global C dynamics (Houghton *et al.*, 1999). Nature of land cover viz. forest, pasture, plantation, fallow or annual agricultural crops has significant influences on the C stock as well as the content of different pools of C in soils (Grace *et al.*, 2006; Kurganova *et al.*, 2010; Banger *et al.*, 2010). Reconstructions of global land use change have contributed as much as half of the increases in CO₂ emissions from human activity in the past two centuries (Post *et al.*, 1990). Conversion of natural ecosystems to agricultural practices

causes depletion of OC pool by as much as 60% in soil of temperate regions and 75% or more in the tropics. Some soils have lost as much as 20 to 80 t C ha⁻¹, mostly emitted into the atmosphere (Lal, 2004a). Deforestation and change in land use can lead to the release of C from soil to the atmosphere. Lal and Logan (1995) estimated that total C emission from agricultural activities in the tropics is about 0.6 Pg yr⁻¹.

Forestry is known as an important land use system for C sequestration in soils. Galdo *et al.* (2003) experimented with a mixed deciduous forest, planted on continuous maize field in North Eastern Italy and demonstrated that forest-derived C contribution was 43% and 31% to the total soil C storage in the afforested systems in the 0-10 and 10-30 cm depths; furthermore, afforestation resulted in significant sequestration of new C and stabilization of old C in physically protected SOM fractions, associated with micro-aggregates (0.05-0.25 mm) and silt and clay (< 0.05 mm). On the contrary, the role of grassland has only recently received attention as a substantial potential C sink (Sampson *et al.*, 2000; Conant *et al.*, 2001). A research concluded that a variety of management practices including irrigation, fertilization, sowing legume species and improved grazing management all promote C sequestration in grasslands (Conant *et al.*, 2001). Grazed lands often have higher SOM contents than cropland soils due to larger root biomass, return of residues, and cycling of manures (Lal, 2002). Grazing stimulates aboveground production of forages, promotes nutrient cycling (Schuman *et al.*, 2002), alters plant allocation of C by promoting tillering and rhizome production and possibly stimulating root respiration and exudation (Schuman *et al.*, 1990).

Conversion of these natural systems into agricultural use resulted into the change in soil C equilibrium and thus its turnover rates (Batjes, 2004). Naitam and Bhattacharyya (2004) showed that, in the soils with similar substrate, the quasi-equilibrium value of SOC was lowest under agricultural crop system in comparison to soils under horticultural and forestry eco-system. Mandal *et al.* (2007) have shown that because of cultivation there is a significant decline in native C content in soils. The magnitude of decline also varied depending upon the nature and intensity of cropping. In agro-ecosystem, the crop species that are cultivated and the soil environment in which they are grown also play an important role in maintaining the SOC stock because both the quantity and quality of their residues that are returned to the soils vary greatly affecting their turnover or residence time in soils (Mandal *et al.*, 2008). Crop rotation can

modify microbial C dynamics, which determine the SOC sequestration potential (Balota *et al.*, 2004).

2.4.4 Cropping System

Cropping systems also has a prominent mark onto the soil Carbon stock and pools. A number of researches (Franzlubbers *et al.*, 1994; Lal *et al.*, 1998; Studdert and Echeverria, 2000; Halvorson *et al.*, 2002) reported that cropping system or crop rotation with or without fallow period in LTFE could affect the size and distribution of SOC pools. Inclusion of legumes and cover crops (Kuo *et al.*, 1997), the addition of manure and fertilizers (Hartwig and Ammon, 2002) and the reduction in fallow frequency (Rasmussen *et al.*, 1980) linearly increase SOC levels. Rice-upland crop rotations such as rice-wheat or rice-maize generally have lower SOC than strictly rice system (Nambiar, 2002). Legume-based cropping systems have positive impact to reduce C and N losses. Majumder *et al.*, (2008 a, b), working with a few LTFE in sub-tropical India, showed that inclusion of legume in cropping sequence (e.g., rice- barseem) checked the loss of applied C in the form of FYM significantly vis-à-vis other sequences (e.g. rice-wheat-fallow, rice-wheat- jute) where no such legumes were included. The inclusion were perennial forages in rotation increases SOM level relative to the once with annual crop alone (Nilsson, 1986). Yang and key (2001) in an experiment in Typic Hapludalf in Southern Ontario, Canada, observed that the rotation of Soybean + winter wheat or berley + berley (under seeded with red clover) followed by two years of corn had 2 to 9 Mg ha⁻¹ more C than the other corn based rotation. According to Lal *et al.* (1995), increased monoculture with plough based system of seed bed preparation can lead to depletion of SOC and emission of active gasses into the atmosphere. Most of the results presented above are related to ‘upland crops’ grown under aerobic conditions. Lowland rice systems differ markedly from upland crop system in C cycling, since it remains under submergence for a certain period of time. Transformation of Carbon in soil under submergence in different form well aerobic soils and therefore, carbon sequestration and dynamics of its different pools are expected to be different. This was reflected by more SOC in stable fractions under lowland rice-rice ecology due to slow decomposition of organic substances under prolong water-logging (Mandal *et al.*, 2008).

2.5 Soil enzymes

Soil is a dynamic living system where all biochemical processes are taking place through enzymatic activities produced by microorganisms (Tabatabai, 1994). Decomposition of soil C depends on the microbial production of extracellular enzymes that convert complex compounds into smaller products (Nannipieri *et al.*, 2002). For example, root and leaf litter contain large amounts of cellulose and hemicellulose, and microbes must produce cellulose-degrading enzymes to access these substrates (Beguin, 1990). Plant material is also rich in lignin, which is degraded by polyphenol oxidase and peroxidase enzymes that are produced primarily by fungi (Blanchette, 1991). Other microbes may produce extracellular enzymes to degrade chitin, an insoluble polymer of amino sugars (Keyhani and Roseman, 1999) that is a major component of the cell walls of fungal mycelia. Carbon-degrading microbes must also produce nutrient-acquiring enzymes, such as phosphatases and proteases, to obtain nutrients for growth and enzyme synthesis (Asmar *et al.*, 1994). Over time, soil microbes and abiotic reactions reprocess plant inputs into even more complex (and often insoluble) materials, such as humic compounds (Tate, 1987) that require oxidative enzymes for their degradation (Freeman *et al.*, 2001).

It is possible to assess microbial populations in the soil but this process is tedious and perhaps not very accurate because of the huge diversity of organisms and vast number that still have not been described (Morton, 1999). To carry out cycling and decomposition activities, soil enzymes are the mediators and catalysts of different OM decomposition, accumulation, mineralization and plant nutrient release process (Fansler *et al.*, 2005). Since enzyme activities show a rapid response to change in management practices, they have also been suggested as early indicators of changes in soil properties (Ekenler and Tabatabai, 2004). Correlation between N mineralization and enzyme activities has been reported in many studies (Tabatabai *et al.*, 2010).

Some of the enzymes in the soil are associated with living cells (biotic) while others are abiotic (Skujins, 1976), or exclusive of live cells. There are several sources for the abiotic enzymes. Some are extracellular enzymes excreted from living cells into the soil solution to hydrolyze polymers that are too large or insoluble to be taken up directly by cells. Others are intracellular enzymes that leak into the soil solution from dead and lysed cells. These abiotic enzymes can be adsorbed and stabilized on the

humic colloids and clay surfaces in the soil, remain catalytic, and reflect the cumulative effects of soil management on soil biology. Soil aggregation could interfere with enzymatic decomposition by preventing microbes and enzymes from making contact with substrates inside aggregates (Sollins *et al.*, 1996). Such restriction might cause C to build up within aggregates. Soil with a higher percentage of macro-aggregates will maintain and stabilize soil enzyme activity by increasing the amount of potential habitat for the microbial biomass, and offer greater potential to protect abiotic enzymes (Miller and Dick, 1995).

2.5.1 Role of Soil enzymes

A better understanding of the role of these soil enzymes activities in the ecosystem will potentially provide a unique opportunity for an integrated biological assessment of soils due to their crucial role in several soil biological activities, their simplicity of measurement, and their rapid response to changes in soil management practices (Dick, 1994; Dick, 1997; Bandick and Dick, (1999) Studies point out that high enzyme activity signals mineral element limitation in the ecosystem (Sinsabaugh *et al.*, 1993; Ndakidemi, 2006). Although there have been extensive studies on soil enzymes (Lizararo *et al.*, 2005; Mungai *et al.*, 2005; Wirth and Wolf, 1992; Ross, 1976; Perucci *et al.*, 1984), little has been reported on their roles in agricultural development. To better understand the roles of these enzymes in carbon and nitrogen cycles, nine enzymes in soils were reviewed for agricultural development. Enzymes excreted by microorganisms into the soil solution could have three main roles: (i) hydrolysis of substrates that are too large or insoluble to be taken up directly by cells; (ii) detoxification of the surrounding environment; and (iii) creation of a favourable environment for the survival of the organisms (Dick, 1997).

Soil enzymes are of ecological importance because they can be used as an index of soil fertility and indicators of soil pollution. They may be useful to assess ecosystem status and the impact of land use. For example, Metting (1993) and Dick and Tabatabai (1993) noted a close relationship between nutrient availability, soil fertility and enzyme activities. Thus, enzymatic approaches can play a significant role in reclamation management (Klein, *et al.*, 1985) and are considered as sensitive indicators of management-induced changes in soil properties (Bergstrom *et al.*, 1998). Enzyme measurements answer qualitative questions about specific metabolic processes and in

combination with other measurements (CO₂ evolution, etc.), may increase understanding of the effect of agrochemicals, cultivation practices, and environmental and climatic factors on the microbiological /activity of soil (Alef and Nannipieri, 1995). Soil enzyme activities can be measured to assess ecosystem function (Kulinska *et al*, 1982; Bergstrom *et al.*, 1998). Enzymes have been useful in determining the impact of severe perturbations (e.g. open cast mining) on soil health and to evaluate the success of remediation activities (Pankhurst *et al.*, 1997). They have also been used to assess the effects caused by acid rain, heavy metals, pesticides and other industrial and agricultural chemicals (Dick and Tabatabai, 1993; Dick, 1997). Dick (1997) considered soil enzyme activities as sensitive indicators of soil health. Many researchers have used enzyme activities to study the dynamics of the microbial population in various ecosystems and to assess the effects of land management on microbial activity (Haynes, 1999; Acosta-Martinez and Tabatabai, 2000).

2.5.2 Dehydrogenase

Dehydrogenase is an important enzyme in the carbon cycle, and is present in all viable organisms. For this reason the assays of dehydrogenase activity are considered an accurate measure of the soil microbial oxidative capacity (Dick, 1994). Therefore dehydrogenase activity in soil has been used to measure for overall microbial activity (Alef and Nannipieri, 1995). Dehydrogenase enzyme activity determination is attractive due to the fact they are an integral part of microorganisms and are involved in organic matter oxidation; nevertheless, this activity is not consistently correlated with other properties of biological systems such as O₂ consumption, CO₂ production or microbial biomass (Dick, 1996). However, it has been considered as a soil quality indicator, because it is involved in electron transport systems of oxygen metabolism and requires an intracellular environment (viable cells) to express its activity (Kandeler and Dick, 2007).

Consistently, the activity of this enzyme is not present in extracellular form as hydrolases (β - Glucosidase, urease, phosphatase), which suggests, that it is not an enzyme that can be used to evaluate the processes of soil degradation, since its activity fluctuates as microbial activity does, in response to management practices and/or climatic effects (Kandeler and Dick, 2007). Nevertheless, dehydrogenase activity is bound to living and active cells, but it depends on the presence of interferences with

heavy metals, catalysis of the assay procedure by extracellular phenol oxidase and other alternative electron acceptors as nitrate and humic substances (Tate, 2002; Speir and Ross, 2002). Studies by Speir and Ross (2002) and Kandeler and Dick (2007) suggest that Cu presence could affect enzyme activity when being assessed.

The dehydrogenase enzyme activity is commonly used as an indicator of biological activity in soils (Burns, 1978). This enzyme is considered to exist as an integral part of intact cells but does not accumulate extracellularly in the soil. Dehydrogenase enzyme is known to oxidise soil organic matter by transferring protons and electrons from substrates to acceptors. These processes are part of respiration pathways of soil micro-organisms and are closely related to the type of soil and soil air-water conditions (Doelman and Haanstra, 1979; Kandeler *et al.*, 1996; Glinski and Stepniewski, 1985). Since these processes are part of respiration pathways of soil microorganisms, studies on the activities of dehydrogenase enzyme in the soil is very important as it may give indications of the potential of the soil to support biochemical processes which are essential for maintaining soil fertility.

With regard to soil air-water relationships, studies have shown that dehydrogenase enzyme was greater in flooded compared to non-flooded soil (Dkhar and Mishra, 1983; Baruah and Mishra, 1984; Benckiser *et al.*, 1984; Tiwari *et al.*, 1989). The increase in this enzyme after flooding was also related to decreased redox potential (Okazaki *et al.*, 1983; Pedrazzini and McKee, 1984). A study by Brzezinska *et al.* (1998) suggested that soil water content and temperature influence dehydrogenase activity indirectly by affecting the soil redox status.

After flooding the soil, oxygen present is rapidly exhausted so that a shift of the activity from aerobic to anaerobic micro-organisms takes place. Such redox transformations are closely connected with respiration activity of soil micro-organisms. They may serve as indicators of the microbiological redox systems in soils and can be considered a possible measure of microbial oxidative activity (Glinski and Stepniewski, 1985; Gunnison, *et al.*, 1985; Skujins, 1973; Casida, 1977; Tabatabai, 1982; Trevors, 1984). The relationship between dehydrogenase activity and redox potential (Eh) as well as Fe^{2+} content may also be used to illustrate the reactions of soil microorganisms to the changes in soil environment. For instance, lack of oxygen may trigger facultative anaerobes to initiate metabolic processes involving dehydrogenase activities and the use

of Fe (III) forms as terminal electron acceptors (Bromfield, 1954, Galstian and Awungian, 1974), a process that may affect iron availability to plants in the ecosystem (Benckiser *et al.*, 1984). Some studies have shown that reducing conditions in the soil were associated with high Fe^{2+} concentration in the soil solution and a significant increase of extra plasmatic Fe in roots of maize due to intense stimulation of microbial growth and dehydrogenase activities in the ecosystem (Fiedler *et al.*, 2004).

Additionally, dehydrogenase enzyme is often used as a measure of any disruption caused by pesticides, trace elements or management practices to the soil (Reddy and Faza, 1989; Wilke, 1991; Frank and Malkomes, 1993), as well as a direct measure of soil microbial activity (Skujins, 1978; Trevors, 1984; Garcia and Hernandez, 1997). It can also indicate the type and significance of pollution in soils. For example, dehydrogenase enzyme is high in soils polluted with pulp and paper mill effluents (McCarthy *et al.*, 1994) but low in soils polluted with fly ash (Pitchel and Hayes, 1990). Similarly, higher activities of dehydrogenases have been reported at low doses of pesticides, and, lower activities of the enzyme at higher doses of pesticides (Baruah and Mishra, 1986).

2.5.3 β -Glucosidase

It is widely distributed in the environment, and its activity has been detected in soil, fungi and plants. It has been used as a key soil quality indicator due to its importance in catalytic reactions on cellulose degradation, releasing glucose as a source of energy to maintain metabolically active microbial biomass in soil (Dick, 1996; Sotres *et al.*, 2005). At the same time, it plays an important role in energy availability in the soil which is directly related to labile C content and with the ability to stabilize soil organic matter, showing low seasonal variability (Knight and Dick, 2004). On the other hand, it has been reported that enzyme activity could be inhibited by the presence of heavy metals like Cu and Cd (Makoi and Ndakidemi, 2008).

β -glucosidase is a predominant enzyme and has the lowest concentration in the soil, but possesses the highest activity and catalyzing efficiency (Klose and Tabatabai, 2002). It is involved in important reactions of degradation of cellulose to glucose which is an important energy source for soil microorganisms. This enzyme activity is very sensitive to soil management practices. Miller and Dick (1995) found higher activity with increased OM content. β -glucosidase activity also significantly correlated with SOC

content and was negatively related with soil pH in typical paddy soils (Wang and Lu, 2006).

Monkiedje *et al.* (2006) investigated the effects of land use on soil health indicators in peri-urban agriculture in the humid forest zone of southern Cameroon. They found that the potential β -glucosidase activity were higher in the peri-urban samples, and concluded that the land use types and associated practices were responsible for the higher potential β -glucosidase activity. Hall *et al.* (2009) investigated the effects of urbanization on soil microbial functioning in the Sonaran desert. Their investigation included β -glucosidase assays. They found that potential β -glucosidase activity was higher under plants than in the inter-plants spaces in outlying deserts. They concluded that patch type was more important in the urban environment in explaining the enzyme activity.

As a free enzyme in soil solution, it normally has a short – lived activity, because they can be rapidly degraded, denatured or irreversibly inhibited. However, a certain proportion of these free enzymes may lose stabilization because of adsorption on soil minerals or incorporation into humic material, which, despite affecting their catalytic potential it may enable enzyme activity to persist in soil (Marx *et al.*, 2005; Burns, 1982).

β -glucosidase is characteristically useful as a soil quality indicator, and may give a reflection of past biological activity, the capacity of soil to stabilise the soil organic matter, and can be used to detect management effect on soils (Bandick and Dick, 1999; Ndiaye *et al.*, 2000). This has greatly facilitated its adoption for soil quality testing (Bandick and Dick, 1999). Generally, β -glucosidase activities can provide advanced evidence of changes in organic carbon long before it can be accurately measured by other routine methods (Dick, 1994; Dick *et al.*, 1996; Wick *et al.*, 1998). Several researchers have however also reported its phytopathological effects in the ecosystem (Davis *et al.*, 1953; Sherrod and Domsch, 1970; Melouk and Horner, 1973). For example, some of the glycons are known to be the precursors of the toxic substances which cause soil sickness where plants are grown as mono-crops (Patrick, 1955; Borner, 1958).

β -glucosidase enzyme is very sensitive to changes in pH, and soil management practices (Dick *et al.*, 1996; Acosta-Martinez and Tabatabai, 2000; Kuperman and Carreiro, 1997; Bergstrom *et al.*, 1998; Leiros *et al.*, 1999; Bandick and Dick, 1999;

Madejon *et al.*, 2001). Acosta-Martinez and Tabatabai (2000) reported β -glucosidase as sensitive to pH changes. This property can be used as a good biochemical indicator for measuring ecological changes resulting from soil acidification in situations involving activities of this enzyme. β -glucosidase enzyme is also known to be inhibited by heavy metal contamination such as Cu and several others (Haanstra and Doelman, 1991; Deng and Tabatabai, 1995; Wenzel *et al.*, 1995). For instance, studies have shown that plant debris did not decomposed or show β -glucosidase activities when exposed to heavy metal polluted soils (Watson *et al.*, 1976; Geiger *et al.*, 1993). Consequently, more understanding of the β -glucosidase enzyme activities and factors influencing them in the ecosystem may contribute significantly to soil health studies.

2.5.4 Protease

Proteases in soil play a significant role in N mineralization (Ladd and Jackson, 1982), an important process regulating the amount of plant available N (Stevenson, 1986) and plant growth. This enzyme in the soil is generally associated with inorganic and organic colloids (Burns, 1982; Nannipieri *et al.*, 1996). Protease activities have been reported to occur partly in soil as a humocarbohydrate complex (Batistic *et al.*, 1980) from arable soil (Ladd, 1972; Hayano *et al.*, 1987); from solid municipal waste compost (Rad *et al.*, 1995), and from forest or permanent grassland soils (Nannipieri *et al.*, 1980, 1982, 1985). The amount of this extracellular enzyme activity may be indicative not only of the biological capacity of soil for the enzymatic conversion of the substrate, which is independent of the extent of microbial activity, but might also have an important role in the ecology of micro-organisms in the ecosystem (Burns, 1982).

Protease activities are affected by several biotic and abiotic factors. For example, low concentrations of neutralised soil humic acids (1-100 $\mu\text{g mL}^{-1}$) inhibit some and stimulate other protease activity by mechanisms involving primarily humic acid carboxyl groups (Ladd and Butler, 1969a, b; Butler and Ladd, 1969). The enzyme pronase is inhibited irrespective of the charge of the substrate hydrolysed, suggesting that decreased activity results from humic acid combining with enzyme rather than with substrate (Ladd and Butler, 1969b). Furthermore, quantitative considerations of the effects of humic acid and substrate concentrations on pronase hydrolysis of carbobenzoxy-glycyl leucine indicates that inhibition is not due to the combination of humic acid and substrate anions (Ladd and Butler, 1969a). There is a need to study the properties and factors affecting

naturally-occurring enzyme complexes such as those involving protease enzymes in the soil ecosystem as they may reveal some unknown role (s) in soil fertility management.

Langer and Mubarak (2007) revealed that, the activities of proteases a similar pattern like alkaline phosphatases, with significantly higher activities in the short-term cropping system of Rahad and the rain- fed systems of sorgham- sesame, continuous sorghum and the natural forest and low activities in Gezira, Managil and the Gezira permanent fallow. Statistically the increasing duration of cultivation in the irrigated Vertisols caused significantly reduces proteases activities.

2.5.5 N-acetyl- β -glucosaminidase

N-acetyl- β -glucosaminidase is the enzyme that hydrolyzes N-acetyl- β -glucosamine residues from the terminal non-reducing ends of chitooligosaccharides and plays an important role in both C and N cycling in soils. N-acetyl- β -glucosaminidase has an optimum pH value of around 5.5. Therefore, activities of N-acetyl- β -glucosaminidase might be of particular importance for N transformations in acidic soils, like the soils of this study, because most other enzymes known to be involved in N transformations in soil have pH optima in the alkaline pH range (Parham and Deng, 2000).

Study of β -glucosaminidase activity, another glycosidase, is important because very little information is available about chitin degradation in soils from tropical as well as temperate environments. This enzyme is also highly correlated to and may serve as a semi-quantitative indicator for soil fungal biomass (Miller *et al.*, 1998).

Jean *et al.*, (2012) reported that the enzymatic activities assayed, N-acetyl- β -glucosaminidase presented the lowest value. Nevertheless, the activity of this enzyme was significantly affected ($p < 0.0001$) by land-use types. N-acetyl- β -glucosaminidase was significant differences between land-use types in enzymatic activity levels. Indeed, cumulative enzymatic activities were higher in *Chromolaena odorata* fallow than in the other types of land use.

β -glucosaminidase is a key extracellular enzyme involved in N mineralization that hydrolyzes C-N bonds of organic sugars to free NH_4^+ that can be taken up by plants (Dick, 1997; Ekenler and Tabatabai, 2002). β -glucosaminidase is a key enzyme involved in the hydrolysis of N-acetyl- β -D-glucosamine (NAG) residues of chito-oligosaccharides (Parham and Deng, 2000). It is also involved in chitin degradation in soils and chitin is

an abundant biopolymer in soil serving as the important transient pool of OC and N (Wood *et al.*, 1994).

This hydrolysis is considered to be important in carbon and nitrogen cycling in soils. Amino sugars, the hydrolysis product of this enzyme comprises about 5-10% of organic mineralizable N in soils (Stevenson, 1994). β -glucosaminidase is one of the enzymes that play a major role in N mineralization in soils. The produced N-acetyl-D-glucosamine can be mineralized to ammonium which then nitrified under aerobic conditions (Tabatabai *et al.*, 2010). Ekenler and Tabatabai (2003) found significant correlation between β -glucosaminidase activity and N mineralization in soils of long term cropping systems. This is involved in the processes where chitin is converted to amino sugars, a major source of mineralizable carbon and nitrogen in soils (Stevenson, 1994; Ekenler and Tabatabai, 2002).

β -glucosaminidase activity has been correlated with the nitrogen mineralized in soils (Ekenler and Tabatabai, 2002), microbial biomass carbon, and fungal populations (Parham and Deng, 2000; Acosta-Martinez *et al.*, 2004). The substrates for this enzyme are chitobiose and glycoproteins. Chitin consists of NAG residues in β -1, 4 linkages and it is the second most abundant biopolymer on earth (Stryer, 1994).

In the soil N-acetyl- β -glucosaminidase (NAGase) activity among all of the size fractions within the soil is smaller than the aggregate size fractions in the remaining soils of the smallest size fraction in each soil had the highest specific activity of all the aggregate size fractions in that soil, which had no significant differences between the aggregate size fractions within soil.

For instance, β -glucosaminidase activities are involved in important reactions of the degradation of cellulose and chitin, respectively. Cellulose and hemicellulose constitute a large fraction of plant components whereas chitin is an important terrestrial biopolymer (Martens and Loeffelman 2002) and thus, they are important energy sources in soil.

David *et al.*, (2009) found that the higher β -glucosaminidase activity under both *Leucaena* and *Eucalyptus* forests results showing that, the old soil organic matter was less protected from microbial activity when land use was changed to *Eucalyptus*, as chitin is a more complex compound than cellulose. It was hypothesized that increased

enzyme activities would be in agreement with increased MBC and MBN due to enhanced substrate availability for microbes. Lower MBC and MBN in the agricultural soil were indeed in agreement with reduced activities of β -glucosaminidase. However, MBC was similar under both forests (Leucaena and Eucalyptus), whereas the activity of β -glucosidase tended to be lower and that of β -glucosaminidase was significantly lower under Leucaena compared to under Eucalyptus. These trends may be due to a lower fraction of stabilized extracellular β -glucosidase and β -glucosaminidase enzymes in soil under Leucaena compared to under Eucalyptus which is in agreement with the constant soil $\delta^{13}\text{C}$ in the last 10 years and our interpretations that mineralized C was predominantly from newly fresh vegetative residues in soil under Leucaena. Madeleine *et al.*, (2011) reported that the N-acetyl-glucosaminidase may have responded weakly to N because this enzyme releases N from organic matter in addition to C. Chitin is one of the dominant sources of organic N to soils, and N-acetyl-glucosaminidase releases small, N-containing amino sugars from chitin. This trend suggests that N addition may have resulted in more severe C limitation to microbes in Fernow soils, which contained less C, a lower C: N ratio, and more mineral-associated soil organic matter than the Bear Brook soils. Physical and biological differences between the sites contribute to the lower C stocks at Fernow, where the O-horizon is very shallow. As a result, the entire O-horizon and the upper 2.5 cm of the mineral horizon (A) were sampled for this study, whereas Bear Brook samples contained only O-horizon material, which is more C-rich.

2.5.6. Acid- and alkaline-phosphatase enzyme

Acid and alkaline phosphatases are both phosphomonoesterases. This class of enzymes has also been widely studied due to its importance in agriculture. Acid and alkaline phosphatases are the commonly measured enzymes in the soil (Tabatabai, 1994) and play an important role in plant nutrition because they catalyse the hydrolysis of organic phosphomonoester to inorganic phosphorus, which can be taken up by plants (Alef and Nannipieri, 1995). Alkaline and acidic phosphatases are so called because of the optimum pH range at which each enzyme functions. Phosphomonoesterases have been extensively studied because they catalyze the hydrolysis of organic phosphomonoester to inorganic phosphorous, which can be taken up by plants (Alef & Nannipieri, 1995). Phosphatase activity procedures used in this study are based on a similar principle as the β -glucosidase assay in that hydrolysis of *p*-nitrophenyl phosphate

releases *p*-nitrophenol, which is extracted quantitatively from soil and measured colorimetrically (Dick, 1996). Hall *et al.* (2009) investigated the effect of urbanization on soil microbial function in the Sonoran desert. They found that phosphatase activity were higher in urban soils samples than in all desert soil samples in their study. They concluded that microbial processes in arid soils are responsive to human activities through creation of managed urban landscapes, and through certain factors associated with the urban environment that increase carbon and nitrogen supply. Garcia-Ruiz *et al.* (2008) investigated the growing interest in the application of soil enzymes, including acid- and alkaline phosphatases, as early indicators of soil quality change. This was done under contrasting agricultural management practices. They found that organic management resulted in significantly higher potential enzyme activities, and that tillage intensity slowed down the improvement of soil quality in organic crops. These enzymes are usually extracellular (Kang and Freeman, 1999).

It has been proven that acid phosphatases are more prevalent in acid soil, and alkaline phosphatases are more prevalent in alkaline soils (Eivazi and Tabatabai, 1977). Alkaline phosphatase activity is completely derived from microorganisms as it does not exist in higher plants (Dick and Tabatabai, 1983), whereas acid phosphatases can come from both plants and microorganisms; plant roots excrete acid phosphatase as a mechanism for P uptake from soils. Legumes which have N-fixer associations excrete larger amounts of phosphatase, probably due to the increased requirements of P for the symbiosis. Phosphatase enzymes are a good indicator of soil fertility and soils with low amounts tend to show nutrient deficiency symptoms. Phosphomonoesterases hydrolyze many types of phosphomonoesters (Tabatabai, 1994).

Phosphatases have been correlated to P stress and plant growth, showing that they are good indicators of the P supply in a soil system. A major influence of phosphatase activity is soil pH. Exudation of this enzyme by plants and microorganisms is influenced by the need for orthophosphate, which is in turn influenced by soil pH. However, it is acid and not alkaline phosphatases, which correspond best to organic C and possibly other indicators in soil (Dodor and Tabatabai, 2003).

This study was conducted at the Oumé benchmark site (Jean *et al.*, 2012), and intended at measuring the enzymatic activities involved in the phosphorus (acid phosphatase and alkaline phosphatase), cycles. Results showed that the enzymatic

activity values were the highest in the fallow soil, whereas the maize crop displayed the lowest levels of enzymatic activity in soil.

Acid phosphatase belongs to the phosphomonoesterase hydrolases, which is the most studied phosphatase group (Turner and Haygarth, 2005). Phosphatases hydrolyse and mobilise inorganic phosphorous (P) from monoester soil organic phosphates, which have been estimated to account for 30-80% of total P in agricultural soils (Dakora and Phillips, 2002). Phosphatases have two pH optima; pH 6.5 (acid phosphatases) and pH 10-12 (alkaline phosphatases) (Speir and Ross, 1978). P-limited conditions (Olander and Vitousek, 2000) and organic P compounds (Gressel and McColl, 1997) can stimulate phosphatase activity, whereas orthophosphate inhibits the activity of phosphatases, either through inhibition of its activity or its synthesis (Colvan, Syers and O'Donnell, 2001).

2.6 Soil Carbon Mineralization

It is known that soil contains about 1600 Pg carbon ($1 \text{ Pg} = 10^{15} \text{ g}$) up to the depth of 1m, being the most important source of C amongst the terrestrial ecosystems. By comparison plants are considered to contain 670 Pg C while the atmosphere contains 750 PgC (Rustad *et al.*, 2000). Soil, the most important carbon source amongst the terrestrial ecosystems, is directly and indirectly connected to other pools carbon by different processes such as photosynthesis, carbon sequestration in biomass, respiration and decomposition of the dead organic matter. The carbon flux due to soil respiration is considered to be the second one, as greatest, in terrestrial ecosystem.

Biological oxidation of organic carbon in soil occupies a key position in the global carbon cycle and provides the principal means by which terrestrial fixed carbon is returned to the atmosphere. The term mineralization has been advanced by Alexander in 1977 and Paul and Clark in 1989 as the conversion of an element from an organic to inorganic form. Applied specifically to carbon, mineralization may be defined as the release of CO_2 from metabolizing organism. This concept is comparable to soil respiration. Defined as the sum of all soil metabolic activities that produce CO_2 (Lundegardh, 1927), or that result in the uptake of oxygen or release of CO_2 by metabolically active soil organism (Anderson, 1982). While oxygen uptake is a part of soil respiration, it is not accommodated by the general definition of mineralization.

Caution must be exercised when considering mineralization as an index of biodegradation. Witkamp (1969) and Dobbins and Pfaender (1988) recognized that respiration or mineralization measurements may not reflect those activities that do not result in complete oxidation of carbon, the uptake of metabolic intermediates or the loss of CO₂ by means other than volatilization. Under these circumstances, respiration measurements might not accurately reflect the actual degree of substrate degradation. However, respiration continues to be the most popular of methods to measure microbial activities and substrate decomposition in soils.

Hanson *et al.* (2000) defined soil respiration as the production of carbon dioxide by the soil organisms. Microorganisms, edaphic fauna and plant parts in the soil, such as roots and rhizomes, are the main organisms which produce CO₂ in soil.

CO₂ production in the soil is the result of many types of respiration processes, and therefore, in practice is difficult to interpret correctly the data. According to the data supplied by Ryan and Law (2005), up to half of the soil respiration is the result of the roots and associated mycorrhizae metabolic activity, representing *soil autotrophic respiration*. However, most of the studies published up to now concluded that the most part of the soil respiration (up to 95%) is due to the activity of heterotrophic microorganisms which use the organic matter recently introduced in the soil as an energetic source and which represent the *soil heterotrophic respiration* (Ryan and Law, 2005). A small part of the soil respiration (up to 10%) is the result of the decomposition of the recalcitrant organic carbon (Hanson *et al.*, 2000; Schlesinger and Andrews, 2000).

Fan *et al.*, (2015) revealed that the seasonal variation of the soil respiration rate was consistent with the seasonal variation of temperature and showed a single peak at all sites. The highest soil respiration rate was recorded in July/August, and the lowest in January. The annual cumulative respiration flux increased by 25.6% and 20.9% in the tea garden with high production (HP) and the vegetable field (VF), respectively, relative to woodland (WL). However, no significant differences were observed between tea gardens with medium production (MP), low production. Soil respiration rates were significantly and positively correlated with organic carbon, total nitrogen, and available phosphorous content. Each site displayed a significant exponential relationship between soil respiration and soil temperature measured at 5 cm depth, which explained 84–98% of the variation in soil respiration. The model with a combination of soil temperature and

moisture was better at predicting the temporal variation of soil respiration rate than the single temperature model for all sites.

The study was conducted in three prominent land use systems of East Siang district, Arunachal Pradesh to measure soil CO₂ released from the soils. Soil respiration rate varied significantly between different seasons as well as annually. Soil respiration rate was consistently the higher in the Forest area (296.54 mg CO₂ m⁻² h⁻¹), followed by the Paddy (291.42 mg CO₂ m⁻² h⁻¹) when both seasonal and annual means were compared. Soil respiration in all the sites showed strong seasonal patterns with higher value observed in the wet season compared to the dry season in most of the sites. Seasonal variations in the soil respiration rate are mostly influenced by the prevailing climatic conditions such as rainfall and temperature in the different land use patterns in north eastern India (Bhuyan *et al.*, 2014). Soil organic matter (SOM) or soil organic carbon (SOC) encompasses all organic constituents and fractions in the mineral soil, including plant and animal tissue in variable stages of decomposition, living biomass of microorganisms, root and microbial exudates and well-decomposed and highly stable organic material. Although, SOM consists of many C compounds, it is often divided conceptually into three pools of different magnitude and turnover times (Figure 1). The turnover time represents the time carbon resides in a certain pool and hence is a measure of the stability of carbon pools. The labile, active pool is composed of microbial biomass and easily decomposable compounds from leaf litter and root-derived material with short turnover times (from weeks to years), the slow (also called intermediate) pool (consisting of refractory components of litter, weakly sorbed carbon) has turnover times from 10 to more than 100 years and the passive (also called resistant or inert) pool (composed of highly humified organic compounds, often mineral-stabilized) has a turnover time on the order of 103 years (Parton *et al.*, 1987; Trumbore, 1997).

Mi *et al.*, (2009) found that the large pool of actively cycling carbon (C) held in soils is susceptible to release due to changes in land use, management, or climate. Yet, the amount and distribution of potentially mineralizable C present in soils of various types and the method by which this soil C fraction can best be quantified, are not well established.

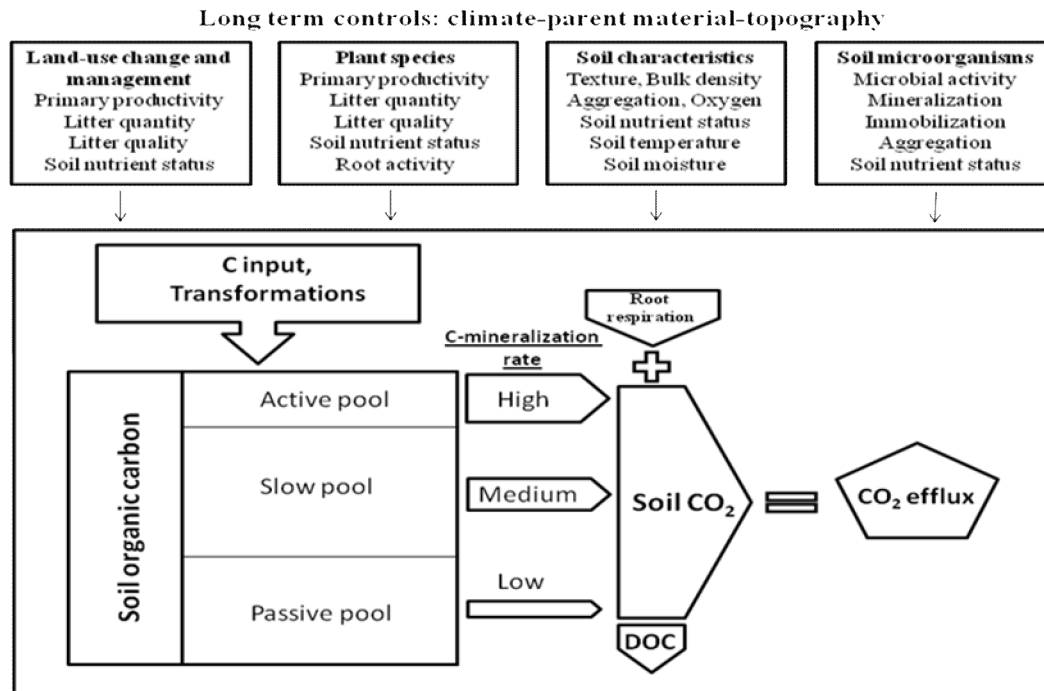


Fig.1. Simplified model on controls of carbon input, soil organic carbon formation and mineralization (Schwendenmann *et al.*, 2007)

2.6.1 Factors affecting soil carbon mineralization

Soil respiration is influenced mainly by soil temperature and moisture (Orchard and Cook, 1983; Howard and Howard, 1993) but also by vegetation type (Raich and Tufekcioglu, 2000) and substrate availability (Vasconcelos *et al.*, 2004). The turnover or mineralization rate of soil organic matter also depends on several factors such as quantity and quality of plant residues, water table, salinity, cations and those affecting microbial activity and soil properties (Bottner *et al.*, 1998; Baldock and Skjemstad, 2000; Blodau, 2002).

Aerobic microorganisms are believed to be capable of decomposing organic matter completely whereas mutualistic consortia of microorganisms accomplish anaerobic decomposition because no single type of anaerobic bacteria seems capable of complete mineralization (Kristensen and Holmer, 2001). Presence of submerged conditions for prolonged period of time leads to very slow organic matter mineralization in wetland soils as compared to upland soils. Previous work showed that under anoxic conditions degradation of organic matter was 5 to 10 times lower than that of oxic

conditions (Kristensen and Holmer, 2001). Anaerobic conditions for prolonged periods prevent complete microbial decay of organic matter (Stevenson, 1994). Following the depletion of oxygen, some respiratory microbes can utilize alternative electron acceptors and continue to mineralize the soil organic carbon. In anoxic conditions alternative electron acceptors such as SO_4^{2-} , NO_3^- , Fe^{3+} , and Mn^{4+} , likely play major role in the mineralization and transformation of organic carbon. Air and soil temperature, water table level, and the quality of organic substrates could be also the main local controls of CO_2 emissions from peat lands (Bridgham *et al.*, 1995; Mendelsohn, *et al.*, 1999).

2.6.2 Carbon fractions and soil respiration

Correct identification of the “mineralizable” soil C pool is essential as it is an important component in modeling soil C dynamics and ecosystem responses to changing environmental factors (Buchmann, 2000; IPCC, 1996; Schlesinger and Andrews, 2000; Stewart *et al.*, 2008). Various methods have been used to quantify mineralizable soil C (Bremer *et al.*, 1994). Two common methods are water extraction and acid hydrolysis. Some evidences do exist that these extractable C pools represent chemically labile C, whereas other evidence suggests C biological availability. The water-soluble C (SC) component of soil originates, in part, from microbial biomass and soluble carbohydrates (Ghani *et al.*, 2003; Sparling *et al.*, 1998). SC quantity has been correlated with the soil’s low-density fraction, microbial biomass, and total enzyme activity (Haynes, 2000; Landgraf *et al.*, 2003, 2005; Tirol-Padre *et al.*, 2005). SC has been used as a proxy for mineralizable C (Leinweber *et al.*, 1995) and, in specific circumstances, has been correlated with basal soil respiration (Tirol-Padre *et al.*, 2005), and has been found to be sensitive to soil management (Haynes, 2005). It has been proposed as an indicator of soil quality (Ghani *et al.*, 2003). Acid-hydrolyzable C (HC) has also been proposed as an indicator of mineralizable SOM (Stout *et al.*, 1981). For example, HC was used in forest litter decomposition models to represent the C fraction lost during early decomposition (Zhang *et al.*, 2007). HC has also been found to decrease during in vitro microbial incubations (Pare *et al.*, 1998). Soil HC quantity has been found to be positively correlated with microbial biomass, light fraction C, size of stable aggregates and 12-day soil organic C mineralization (C-min) rates (McLauchlan and Hobbie, 2004).

Soil incubations are a more direct approach to quantifying mineralizable soil C than various procedures using chemical extraction or organic compound class analysis.

Measured C_{min} rates have ranged from less than 0.007 to 35.6% of total soil C using varying incubation times (12–800 days) and soil temperature and moisture conditions (Alvarez and Alvarez, 2000; Collins *et al.*, 2000; Giardina *et al.*, 2001; Haile-Mariam *et al.*, 2000; McLauchlan and Hobbie, 2004). However, they are, by nature, short-term measurements, and must, therefore, measure only a subset of the total potential mineralizable soil C (Minderma and Vulto, 1973; Paul *et al.*, 2001, 2006). Further, laboratory incubation conditions are generally optimized for temperature and moisture. Thus in vitro C_{min} can only serve as a proxy for total potential mineralizable soil C.

Only a few studies have combined the measurement of extractable soil C pools such as SC and HC and laboratory incubations to better understand soil organic C dynamics (Paul *et al.*, 2006) and access the relative value of these techniques and data derived by these methods. The studies that exist in the literature have been limited to one or only a few land use or soil types and to a relatively small sample size (Alvarez and Alvarez, 2000; Collins *et al.*, 2000; Giardina *et al.*, 2001; Haile-Mariam *et al.*, 2000; McLauchlan and Hobbie, 2004). Thus, there is a lack of documentation that allows us to assess the relationship between soil C storage and C_{min} at the landscape scale, and across soil and land use types.

The amount and distribution of potentially mineralizable C present in soils of various types and the method, by which this soil C fraction can best be quantified, are not well established. The distribution of total organic C (TOC), extractable C pools (hot-water-extractable and acid hydrolyzable), and in vitro mineralizable C in 138 surface soils across a north Florida watershed was found to be quite heterogeneous (Ahn *et al.*, 2009). Thus, these C quality parameters could not statistically distinguish the eight land uses or four major soil orders represented. Only wetland and upland forest soils, with the largest and smallest C pool size, respectively, were consistently different from the soils of other land use types. Variations in potential C mineralization were best explained by TOC (62%) and hot-water extractable C (59%), whereas acid-hydrolyzable C (32%) and clay content (35%) were generally not adequate indicators of C bioavailability. Within certain land use and soil orders (Alfisol, Wetland and Rangeland, all with >3% clay content), however, C mineralization and clay content were directly linearly correlated, indicating a possible stimulatory effect of clay on microbial processing of C. Generally, the sandy nature of these surface soils imparted a lack of protection against C

mineralization and likely resulted in the lack of land use/soil order differences in the soil C pools. If a single parameter is to be chosen to quantify the potential for soil C mineralization in south eastern U.S. coastal plain soils, we recommend TOC as the most efficient soil variable to measure.

2.6.3 Temperature and carbon mineralization

Temperature effect influences every aspect of the respiratory processes. It is well known that during these processes numerous enzymes are involved, and their activity is influenced by temperature. Generally, respiration is agreed to increase once temperature goes up, reaching its maximum value to 45 – 50⁰C (Luo and Zhou, 2004). The influence of temperature over soil respiration depends on the component taken into consideration.

The temperature sensitivity of SOM decomposition is often described with a Q_{10} value, which is the factor by which the respiration rate (r) increases, when the temperature (T) increases by 10 °C:

$$Q_{10} = r(T+10)/r(T) \quad (1)$$

According to current knowledge the following factors affect the temperature sensitivity of SOM decomposition:

(i) SOM stability, (ii) substrate availability, which is determined by the balance between input of organic matter (e.g. leaf and root litter, root exudates), decomposition and stabilization of SOM, (iii) the physiology, substrate utilization efficiency and temperature optima of soil microbes, (iv) physicochemical controls of destabilization and stabilization processes (von Lützow and Kögel-Knabner, 2009). Because of the complexity of the soil system and the large number of factors possibly affecting the temperature sensitivity, there is no general theory that could be used to describe the temperature sensitivity of organic matter decomposition (Kirschbaum, 2006).

Zengin *et al.* (2008) studied the effects of different temperatures (23⁰C and 28⁰C) and humidity [field capacity (FC), 80% and 60% of their field capacity] conditions in the laboratory. C mineralization of all soils was determined using CO₂ respiration method. The microbial activity increased together with temperature increases. The microbial activity in the soils humidified at the field capacity and 60% of it was statistically lower compared to the soils humidified with 80% of field capacity at both 23⁰C and 28⁰C ($P < 0.000$). The rate (%) of C mineralization of 80% of field capacity at 28⁰C was

significantly higher than field capacity ($P = 0.009$) and 60% of field capacity ($P = 0.006$) at 23°C and 60% of field capacity ($P = 0.009$) at 28°C.

2.6.4 Land use and C-mineralization

The global carbon cycle is being altered in response to human interference; for instance land-use changes in the tropics are estimated to contribute about 23% to human-induced CO₂ emissions (Houghton, 2003). Carbon input, magnitude of soil organic carbon pools and finally carbon mineralization depend on many factors. Changing patterns of land-use and land-use management practices can have significant direct and indirect effects on soil organic pools, due to changes in plant species, primary productivity, litter quantity and quality and soil structure. However, the impact of land-use changes on organic carbon pools in the mineral soil depends also on long-term site-specific factors (e.g. climate, topography and parent material) and is often overridden by the high spatial heterogeneity of soil organic carbon. Consequently, effects on SOC pools are evident only with the most intensive practices. For example, forest clear-cuttings for pasture in the humid tropics combined with slash removal and/or burning cause significant losses of SOC (Brown and Lugo, 1990, Veldkamp, 1994). However, pasture establishment in the tropics may maintain or even increase the soil organic carbon content especially when grass species with high percentages of below-ground biomass production are used (Feigl *et al.*, 1995). A recent meta-analysis using data from temperate and tropical regions indicates that soil organic carbon stocks increased by 8% after converting native forests into pastures (Guo and Gifford, 2002). In contrast, no clear trend in soil C change following the conversion from forest to pasture was shown in a data compilation for the tropics by Lugo and Brown (1993). Murty *et al.* (2002) also found no trend in soil C after conversion from forest to pasture in temperate and tropical regions.

In recent years reforestation of degraded and abandoned tropical pastures has been proposed as a measure to mitigate increasing atmospheric CO₂ levels (Montagnini and Porras 1998, Silver *et al.* 2000). However, the effects of reforestation can result in losses or gains of soil C pools (Rhoades *et al.* 2000, de Koning *et al.* 2003, Silver *et al.* 2004). Land-use change affects carbon stocks and also soil respiration (also called soil CO₂ efflux) rates (Trumbore *et al.* 1995). To characterize the carbon exchange in ecosystems, an assessment of the magnitude and dynamics of soil CO₂ efflux is

important, considering that soil respiration is a major CO₂ flux in the carbon cycle, second in magnitude to gross canopy photosynthesis (Raich and Schlesinger 1992).

2.6.5. Effect of moisture on C mineralization

Soil moisture is another important factor influencing soil respiration. Soil CO₂ efflux is usually low under dry conditions due to low root and microbial activities, and is increasing with soil moisture till some limit. In very high soil moisture condition, soil CO₂ efflux is reduced due to limitation of diffusion of oxygen and suppression of CO₂ emissions. This relationship is sometimes described by a quadratic equation (Bunnel, *et al.*, 1977; Linn, *et al.*, 1984; Mielnik and Dugas, 2000). Other equations are also used to describe soil respiration-moisture relationship: linear, exponential and hyperbolic equations (Norman, *et al.*, 1992; Davidson, 1998; Liu, *et al.*, 2001; Schlentner and Van cleve, 1985; Carlyle and Than, 1988). Despite this, the relationship between soil respiration and moisture is usually scattered, and our understanding of this relationship the mechanisms underlying the relationship is still limited, compared to that of respiration/temperature relationship. This is partly due to the fact that most of the studies on this relationship are field studies based on observations of seasonal and spatial variations of soil respiration. Thus the effect of soil moisture is confounded with other factors, such as temperature, root and microbial activities.

The effect soil water content/potential on C mineralization from soil and decomposing plant residues has been studied for some time. Greaves and Carter (1920) noted that soils differing in relative saturation produced dramatically different levels of nitrate after 21 days of incubation. Russell *et al.* (1925) found insignificant levels of NO₃-N in soils that were at the “hygroscopic coefficient”, but increasing amounts at the highest level of moisture they studied, which was 1.25 times the “moisture equivalent” or field capacity. Quemada and Cabrera (1997) showed that as soil matric potential (ψ) increased from -5.0 to -0.003 MPa the total CO₂ evolved from unamended soil increased linearly with $\ln(-\psi)$, but CO₂ evolved from decomposing clover residue increased exponentially with ψ .

Cook and Allan (1992a) studied dissolved organic carbon (DOC) over 210 days in soils maintained at a constant moisture, and found asymptotic exponential response curves described positive associations between DOC and CO₂-C mineralization rates at

early (14 and 35d) incubation times, but not later. The results reflected decreased DOC utilization relative to supply, and suggested this could be caused by the accumulation of recalcitrant DOC. In a related paper (Cook and Allan, 1992b) they determined, using the Leenheer DOC fractionation scheme, that although they could not directly determine DOC utilization, their results demonstrated that soil DOC was altered during the decomposition of soil organic matter.

2.7. Nitrogen Mineralization

Nitrogen mineralization (N_{min}) is the conversion of organically-bound nitrogen in the soil and organic matter into inorganic forms that can be used by plants, and is usually expressed as the rate of kilograms of nitrogen mineralized per hectare per year or some other appropriate unit when calculating additions to bulk nutrient pools or annual fluxes. The mineralization process is driven by microbial activity that utilizes carbon in organic matter as chemical energy and is influenced by moisture, temperature, and chemical composition of the organic matter (Edmonds, 1980). Nitrogen is immobilized from the available pool as microbial biomass increases until carbon becomes limited. When this happens the microbial biomass begins to die off producing NH_4^+-N via ammonification. Nitrification occurs when NH_4^+-N is further converted to nitrite (NO_2^-) and then NO_3^- N in the presence of oxygen, with this chemical reaction producing 2 H^+ ions that can acidify a soil.

Since nitrification is a secondary process, it is dependent upon the total amount of NH_4^+-N produced and available following assimilatory processes (i.e., plant uptake or microbial immobilization). Aber and Mellilo (1991) indicate that nitrification is sensitive to changes in pH with less acidic soils having higher rates compared to acidic soils, but soils with low pH can still have high amounts of available NO_3^- N if NH_4^+-N levels exceed vegetation requirements.

In submerged or wetland soils, because of the absence or lack of molecular O_2 , N mineralization, i.e., the conversion of organic N stops at NH_4^+-N production (also termed ammonification). Unlike in submerged soils, in upland soils the NH_4^+ formed is readily oxidized to NO_3^- via NO_2^- , termed as nitrification (Sahrawat, 2009). Mineralization can occur in either aerobic or anaerobic conditions. In submerged soils, mineralization is carried out by a different group of heterotrophs, mainly bacteria (Kyuma, 2004) and the

process is slower under anaerobic conditions due to less efficient and incomplete decomposition (Sahrawat, 2004a).

As a result of mineralization of organic N in submerged soils, part of NH_4^+ produced becomes a part of soil solution and the rest remains in the adsorbed phase on the soil exchange complex; and there is a dynamic equilibrium between NH_4^+ in solution and in the adsorbed state (White and Reddy, 2001). In some submerged rice soils, Ponnampetuma (1972) found a considerable portion of NH_4^+ in the solution phase, depending especially on the OM content and texture of the soil. He also observed that the N in the soil solution is a readily source of N to the growing rice plant and forms an important part of the available N pools in wetland rice soils. But sometimes, very large amounts of NH_4^+ -N in soil solution can be toxic to the young growing rice plant in wetland rice soils rich in decomposable OM (Ponnampetuma, 1972).

The total flux of inorganic nitrogen is referred to as gross mineralization Net mineralization is considered the difference between gross mineralization and gross immobilization with gross mineralization also defined as the net mineralized plus the microbial uptake of NO_3^- -N and NH_4^+ -N and any leaching or atmospheric losses (Hart *et al.*, 1994). The balance between these two processes will determine plant available nitrogen (PAN).

2.7.1. Model Describing Soil N Mineralization Potential

Incubation of soils in the laboratory under controlled environmental conditions is a technique that is often used as a measure for mineralizable or potentially mineralizable SON. Mostly, aerobic incubation techniques are used, *i.e.*, soil is incubated under free oxygen exchange with the air. Both leaching methods as well as destructive sampling are used. In leaching methods small samples of soil are mixed thoroughly with quantities of pure quartz sand in tubes and then incubated under well specified conditions of temperature and moisture (Stanford and Smith, 1972).

Models describing soil N mineralization potential typically are based on the results from incubation studies. Nitrogen mineralization is generally considered to be a first order reaction (Stanford and Smith, 1972; Cabrera and Kissel, 1988; Rasiah, 1995). The one-pool model proposed by Stanford and Smith (1972) is shown as Follows:

$$\text{Nm} = \text{N}_0 - \text{N}_0 e^{-kt}$$

$$\text{or, Nm} = \text{N}_0 (1 - e^{-kt})$$

Where, N_m is amount of N mineralized at time t (cumulative N), a value derived through the incubation of a soil sample; N_0 is the mineralization potential or potentially mineralizable N (Rasiah, 1995) or the pool of mineralizable N (Cabrera and Kissel, 1988); and k is the rate constant of N mineralization. The amount of N mineralized is the difference between N_0 at time 0 and N_0 at time t (Campbell *et al.*, 1993).

N_m is often measured directly in the lab as the amount of N accumulated from successive extractions of a soil sample over the duration of incubation (cumulative N), (Campbell *et al.*, 1993). The incubation is terminated when the amount of N mineralized, in the period between each extraction, approaches zero (Campbell *et al.*, 1993). According to Campbell *et al.* (1993), the calculation of N_0 is only valid if incubation occurs under ideal conditions; i.e., 35°C and field capacity. Moreover, N_0 values for a particular soil sample are generally 50% greater than the cumulative N value for that same soil sample (Campbell *et al.*, 1993). The procedure assumes there is only one active N pool. In reality there is more than one pool, but Campbell *et al.* (1993) suggested that separating these N pools is not likely to be worth the effort in terms of improving estimates of potentially mineralizable N.

Stanford and Smith (1972) suggested that a common value for k could be derived that would apply to a wide range of soils. They estimated k to be $0.054 \pm 0.009 \text{ wk}^{-1}$ which means that 5.4% of the mineralizable N pool is available per wk, based on the amount of mineralizable N remaining at the end of each wk. This estimate was based on pooled regressions for 29 soils of varied types. Subsequent work has shown that k is not constant and may have to be calculated from field to field (Rasiah, 1995; Mahmoudjafari *et al.*, 1997). Campbell *et al.* (1993) and Jalil *et al.* (1996) used non-linear regression (least squares) to calculate N_0 and k , assuming first order kinetics, and found that k ranged from 0.05 to 0.20 wk^{-1} . Initial values of k and N_0 , to start off the iterative process of the non-linear regression, can be obtained by estimating k at 0.10 wk^{-1} and N_0 as 50 % greater than accumulated mineralized N (Campbell *et al.*, 1993).

2.7.2 Factor affecting soil nitrogen mineralization

2.7.2.1 Soil moisture and temperature:

As temperature and moisture availability regulate many ecosystem functions, including microbial activity and nutrient storage, they are considered the most critical driving forces in ecosystems, and have the greatest control over decomposition and

mineralization rates (Murphy *et al.*, 1998; Holmes and Zak, 1994). In temperate forests of the North America there are strong seasonal trends in N mineralization. Higher rates occur in spring and summer when soil moisture is high and temperatures warm and negligible activity occurs in the winter (Knoepp and Swank, 1998; Zak *et al.*, 1993). Temperature and soil moisture were used to explain more than 90% of the variation in laboratory N mineralization measurements made by Gonsalves and Carlyle (1994).

Quemada and Cabrera (1997) state that the effect of increased water potential on N mineralization was enhanced as temperature increased, indicating an interaction effect between both factors. They also reported that volatilization losses in the form of N₂O and NH₃ increased with moisture at every temperature (Quemada and Cabrera, 1997). Relative nitrification also seems to be a linear function of moisture content except at higher moisture contents when net mineralization and nitrification become independent of moisture (Tietema *et al.*, 1992).

Goncalves and Carlyle (1994) found that the quantity of mineral N produced increased linearly with time for the majority of moisture and temperature combinations. The size of the mineralizable pool appeared to increase with increasing soil temperature, possibly due to the positive response of the microbial community to temperature (MacDonald *et al.*, 1995).

Within a single growing season, water and temperature will have more influence on the rate and amount of N mineralized than will the size of the N pool itself (Goncalves and Carlyle, 1994). Herlihy (1979) observed higher mineralization values under fluctuating moisture contents or where soil was allowed to go through wet and dry cycles. Ellert and Bettany (1992) concluded that mineralizable pools changed as temperature changed. This may be due to its affect on microorganisms, substrate composition, or transport processes.

2.7.2.2 *Wet/Dry cycles*

Cabrera (1993) felt that the method of Stanford and Smith (1972) did not accurately reflect N mineralization that occurs in soils that undergo wet-dry cycles typical in semi-arid conditions. This is because mineralization, upon the rewetting of a 28 dry soil, occurs as a flush. Campbell *et al.* (1988) considered wet/dry cycles to be important to N mineralization in the field. According to Cabrera (1993) wet/dry cycles are a mechanism by which N is made available from less readily available pools by

means of desorption of organic substrates from soil surfaces. During incubation, the flush is represented by that portion of cumulative mineral N obtained from the first extraction, which also tends to be the largest fraction (Cabrera, 1993). Clay appears to isolate organic matter from the microbial community, but upon drying and rewetting, this protection is broken down and this organic matter becomes available (Strong *et al.*, 1999). Soils with clays that fix organic matter will experience a greater flush of mineral N than soils with lower clay content or clays which are less able to fix organic matter (Strong *et al.*, 1999). In contrast, Magid *et al.* (1999) found that mineralization did not increase due to increased organic matter decomposition, but probably due to the availability of microbial debris from death of microorganisms.

Samples which are not subject to the wet/dry cycles experience N mineralization that follows zero order kinetics, whereas samples which undergo wet-dry cycles demonstrate first order kinetics (Cabrera, 1993). Flushes appear to be short-term, and subside after only two or three days (Murphy *et al.*, 1998; Franzluebbers, 1999). Murphy *et al.* (1998) found that changes in gross N mineralization were correlated to the soil water content in the top 2.5 cm of soil. Changes in water content in the top 2.5 cm of soil may heavily influence N mineralization because of its influence on the microbial activity concentrated in this layer (Murphy *et al.*, 1998). Thus, minor rain events can have a disproportionate effect on mineralization. The microbial population appears to respond rapidly to small rainfall, even with rapid drying, by mineralizing a significant amount of N (Murphy *et al.*, 1998), suggesting that the microbial population is adapted to the wet/dry cycling.

2.7.2.3 Total soil N and Soil organic C

Total soil N has been found to be positively correlated with N mineralization potential (Rasiah, 1995). Selles *et al.* (1999) found total soil N and soil N availability to be weakly related according to an analysis of their semi-variance. However, total soil N does not appear directly related to how much or how fast the N is mineralized (Stanford, 1982). Thicke *et al.* (1993) observed that unless total soil N is very low, N mineralization is unrelated to total soil N. Total soil N has been found to correlate with N released by hot KCl and phosphate-borate extractants (Curtin *et al.*, 1998). In some studies, total soil N was correlated to crop yields, such as the study by Stein *et al.* (1997) in which a good r^2 of 0.407 was achieved.

Soil organic C is intimately related with total soil N (Rasmussen *et al.*, 1998) and is reported in much the same way as is total soil N. A positive relationship is always reported between N mineralization potential and soil organic C (Cabrera and Kissel, 1988; Rasiah, 1995; Selles *et al.*, 1999). Bonmati *et al.* (1991) found soil organic C to be significantly and positively correlated with total soil N. Keeney (1982) viewed organic matter as an index for N mineralization and reported that organic matter indices for N recommendations are used in several states in America. MacDonald *et al.* (1995), however, found mineralized N was not well correlated with soil organic C.

2.7.2.4 Soil Texture

The influence of soil texture on N mineralization is primarily related to clay content and, to some degree, clay mineralogy (Breland and Hansen, 1996; Franzluebbers *et al.*, 1996; Scott *et al.*, 1996). Soil texture has a major influence on the soil microenvironment and on soil processes such as decomposition (Scott *et al.*, 1996). A simple example is the positive correlation between soil organic matter and soil clay content across the Great Plains (Burke *et al.*, 1990). It is generally agreed that coarse textured soils have a more active microbial population and organic matter is more available for mineralization than soils of finer texture (Breland and Hansen, 1996; Franzluebbers *et al.*, 1996; Scott *et al.*, 1996). Franzluebbers *et al.* (1996) found that the rate of basal soil respiration per unit of soil microbial biomass carbon was greatest in soils of coarse texture; as clay content increased, basal soil respiration decreased. Mineralizable C to N ratio also decreased with increasing clay content (Franzluebbers *et al.*, 1996).

Soil organic matter in coarse textured soils appears to be more labile, whereas in finer textured soils, the complex soil structure provides greater protection to the SOM, thus reducing the turnover of C and N (Franzluebbers *et al.*, 1996). As clay content increases, soil surface area and organic matter stabilization potential increases (van Veen *et al.*, 1985). Tisdale and Oades (1982) noted that SOM tended to increase with clay content because of the increase in surface area and charge leading to more chemical and physical binding with the organic matter. Rasiah (1995) reported that mineralization was negatively correlated with clay content and cation exchange capacity (CEC), which further suggests a decrease in mineralization with increasing protection of substrate.

Clays with low surface charge, such as a kaolinite appear to have much less ability to protect adsorbed organic matter (Franzluebbers, 1999).

Cabrera and Kissel (1988) suggested that the ratio of clay to total soil N can be used as an index of the degree of protection that clays provide to organic matter from microbial attack. A pretreated or disturbed soil of low clay content, which has a reduced ability to protect organic matter, may have mineralization values closely correlated with an undisturbed sample of the same soil. In such cases, disturbed samples could provide an accurate N mineralization index for that type of soil. For higher clay soils with good aggregation, this correlation of N mineralization between disturbed and undisturbed likely would be poor.

2.7.2.5 Soil Rhizosphere and Soil Microbes

The total rhizosphere environment is determined by interactions between soil, plant and organisms associated with the roots (Gobran *et al.*, 1998). The rhizosphere is an important zone in the soil where mineralization rates are often double that of the bulk soil (Jaeger *et al.*, 1999). Due to root turnover, sloughed cells, exudates and secretions, the rhizosphere strongly influences heterotrophic microbes that depend on C as an energy source (Hobbie, 1992). The rhizosphere tends to support higher microbial biomass, a more active population of microbes, and a higher ratio of bacteria to fungi than the bulk soil (Hobbie, 1992). An enhanced C availability in the rhizosphere has been suggested to increase N immobilization by heterotrophic soil microbes, attract microbial grazers that in turn increase N mineralization rates (Hobbie, 1992). During the Breakdown of substrates rich in N, microbes tend to immobilize N until sufficient C has been respired to a point where C or some element other than N is limiting catabolism (Mitter *et al.*, 1984). Microbes are the most important labile C pool since they are vital for soil organic C dynamics and nutrient cycling (Hu *et al.*, 1997).

Rates of gross N mineralization are correlated with rates of C mineralization, but a very substantial proportion of the mineralized N is re-immobilized into microbial biomass (Prescott *et al.* 2000). Results by Zak *et al.* (1993) also show mineralized N to be well correlated with respired C among stands.

The concept that soil fauna regulates the decomposition process has become increasingly popular in spite of the fact that the amount of soil metabolism (CO₂ production) that can be attributed to all soil animals is 10 % or less of the total amount

(Mitter *et al.*, 1984). Fungi and bacteria are directly responsible for most of the organic matter breakdown, but a diverse assemblage of protozoans, nematodes, annelids, and arthropods greatly influence the functioning of the decomposer flora as a direct and indirect result of their feeding activities (Hunt *et al.*, 1987; Coleman *et al.*, 1983). Mycorrhizal fungi are also important as they allow plants access to organic nitrogen forms nutrients, bypassing the mineralization step (Chalot and Brun, 1998; Abuzinadah and Read, 1986). They also represent a sizable nutrient reservoir themselves and are important in translocating nutrient resources through the soil column (Mitter *et al.*, 1984).

2.8 Phosphorus Mineralization

Net P mineralization rates and extractable P are also important to assess in relation to organic matter management since pine plantations in the south eastern U.S. coastal plain are also known to be P limited. Additionally, these soils are strongly weathered and leached and therefore little P is released from the weathering of the primary P-bearing minerals. Phosphorus availability is largely a function of the mineralization of organic P and is determined by competition between biological and geochemical sinks for P anions (PO_4^{3-}). Increased rates of P mineralization from soil organic matter in response to fertilizer application have been documented and these increases were linked to critical organic matter substrate C: P ratios and metabolic satiation of microbial populations (Polglase *et al.*, 1992; Saggar *et al.*, 1998). Currently little is known about the effects of organic matter additions on mineral soil P mineralization. It has been documented that mineralization and immobilization of P are the dominant processes determining the concentration of P in soil solution in coastal plain sites limited by sorptive surfaces in the forms of Al and Fe and organic complexes (Fox, 1990).

Residue incorporation studies in New Zealand proposed that management techniques of mixing litter into the soil increased P availability due to the increase in local concentrations of organic acids (Comerford and Dyck, 1988; Comerford and Skinner, 1989). Hence, the importance of forest floor and slash after harvest needs to be investigated in the early years of pine plantation establishment in order to understand the

nutrient cycling (N and P) response to different amounts and types of post-harvest residues.

Biochar is a carbonaceous substance produced as soil additive for agricultural and environmental management (Guo and Rockstraw, 2007). Increasing wastes disposal, global warming, and food insecurity call for the use of biochar (produced from agroindustrial wastes) in agriculture. This is essential because biochar improves soil fertility, crop productivity, soil water retention, and soil carbon sequestration (Galinato, 2011). Compost produced from pineapple leaves and biochar could be used to minimize P fixation in acid soils (Chng *et al.*, 2014).

Tisdale, (2008) reported that the mineralizable P was not affected by treatment. The overall mean for mineralizable P was $1.4 \mu\text{g g}^{-1}$. This pool of P, unlike microbial biomass P, was influenced by collection time. The mineralizable P content mean for all treatments was 1.8 kg ha^{-1} . Compared to mineralizable P findings, extractable P showed no treatment differences due to large variation within each treatment. The treatment mean for extractable P was $2.8 \mu\text{g g}^{-1}$. Content data for microbial biomass P, mineralizable P, and extractable P were statistically similar across treatments and averages were 23, 1.3 and 3.2 kg ha^{-1} , respectively.

Phosphorus mineralization, averaged across rates and amendments, was similar for the two soils, except on Days 28 and 70 when it was higher in the Dark Brown Chernozem. On average, for the non-amended soils, the Black Chernozem had higher P_{min} than the Dark Brown Chernozem throughout the incubation period. Increasing separated solids (SS) rate resulted in the highest increase in P_{min} (30 mg P kg^{-1}) while increasing manure and pelletized separated solids rates caused lower mineralized P increases of 18 and 6 mg P kg^{-1} , respectively (Waraidzo, 2011).

In forest soils where a large fraction of total phosphorus (P) is in organic forms, soil micro-organisms play a major role in the P cycle and plant availability since they mediate organic P transformations. However, the correct assessment of organic P mineralization is usually a challenging task because mineralized P is rapidly sorbed and most mineralization fluxes are very weak. The objectives of the present work were to quantify in five forest Spodosols at soil depths of 0–15 cm net mineralization of total organic P and the resulting increase in plant available inorganic P and to verify whether net or gross P mineralization could be estimated using the C or N mineralization rates.

Net mineralization of total organic P was derived from the net changes in microbial P and gross mineralization of P in dead soil organic matter (David *et al.*, 2010).

Abbasi *et al.*, (2015) revealed that, the P mineralization tendency of soil amended with soluble P fertilizers + insoluble rock phosphate did not show any increasing effect. However, rock phosphate when combined with poultry manure in 1/2 RP + 1/2 PM released significantly higher P compared to full RP treatment (80 %) and equivalent to that recorded under full PM treatment. Effect of PSB on P release capacity of different P amendments was significant ($P \leq 0.05$). Phosphorus release capacity of soil amended with RP varied between 6.0-11.5 mg kg⁻¹ significantly ($P \leq 0.05$) higher than the control but lower than the remaining treatments. Application of phosphorus solubilizing bacteria with rock phosphate in RP + PSB did not show any remarkable effect on P solubilization or P mineralization except that a significant increase was noticed on 25 and 60 days of incubation. Soil amended with the soluble P fertilizers *i.e.* SSP and DAP displayed the highest P mineralization of 73.3 and 68.5mg kg⁻¹ immediately after their application at day 0.

Fritz *et al.*, (2004) reported that the soil organic P (P_o) mineralization plays an important role in soil P cycling. Quantitative information on the release of available inorganic P (P_i) by this process is difficult to obtain because any mineralized P_i gets rapidly sorbed. Basal P_o mineralization rates in non-irradiated samples were between 1.4 and 2.5 mg P kg⁻¹ day⁻¹. This is an amount lower, approximately equivalent to, or higher than water soluble P_i of MIN, ORG and DYN soils, respectively, but in every soil was less than 10% of the amount of P isotopically exchangeable during one day. Organic P mineralization rates were higher, and differences between soils were more pronounced in γ -irradiated than in non-irradiated soils, with mineralization rates ranging from 2.2 to 4.6 mg P kg⁻¹ day⁻¹. These rates of hydrolysis, however, cannot be compared to those in non-sterile soils as they are affected by the release of cellular compounds.

Sinaj *et al.*, (2002) studied in the sandy acidic soil and suggest, that white clover initially used a fraction of the rapidly exchangeable compost P, while at a later stage plant roots enhanced the mineralisation of compost organic P and took up a fraction of the mineralized P. These relations were not observed in the clayey calcareous soil probably because of its high sorbing capacity for P. In the sandy acidic soil, composts application increased the uptake of soil P by the plant from 31.4 mg P kg⁻¹ soil in the

control without P to values ranging between 37.9 to 42.7 mg P kg⁻¹ soil in the presence of composts.

Lopez and Nino, (1993) the dynamics of phosphorus in soils derived from different textured parent materials were studied and found that, the microbial P and P_o mineralization rates were significantly affected by parent materials. The high rates of mineralization for the fine-textured soils correspond well with their higher content in substrates able to mineralize e.g. total carbon, P_o and microbial P_i. The residual P (e.g. more stable P_i and P_o forms) is the principal P fraction in the analyzed Mollisols.

Murungu, *et al.*, (2011) studies was done at the University of Fort Hare Research Farm (32°46'S, 26°50'E) which has a warm temperate climate and mean monthly temperature ranges from 19 to 23° C during the summer season (October to April). In this study net P mineralization was in the order grazing vetch > forage peas = oats with grazing vetch mineralizing about 3.6 mg P/kg. Whether net mineralization of P occurs depends on the P content of the material. Maximum net mineralized P was higher for grazing vetch (3.6 mg P/kg) than for forage peas (2.7 mg P/ha) and oats (2.8 mg P/kg).

Rita *et al.*, (2013) revealed that, the distribution of P fractions (P_i and P_o) in the soil size fractions revealed the distinctive ability of vegetation covers to recycle soil P. As a result, the mineralization potential of P_o in the soil varied with the soil size fraction and the vegetation cover. Because P_o was the predominant form in the total labile P composition, P availability to plants was closely related to the mineralization of labile P_o. Zibilske, and Bradford, (2003) suggeste that, there may be a difference in substrate composition caused by tillage, and consequent differences in microbial respiratory activity indicate that biological controls of phosphorus mineralization are becoming better established, although absolute changes in soil organic C and N concentrations were modest.

Spohn and Kuzyakov (2013). Reported that despite the importance of phosphorus (P) mineralization to maintain soil fertility, little is known about the mechanisms that regulate microbial P mineralization. They tested the hypothesis that microbial P mineralization can be driven by microbial need for carbon (C). For this purpose, net microbial uptake kinetics of ¹⁴C and ³³P from glucose-6-phosphate were studied in a Leptosol depending on availability of C, nitrogen (N), and P. After 60 h of incubation, 16.4% of the ¹⁴C from glucose-6-phosphate was recovered in the microbial biomass,

while ^{33}P incorporation into the microbial biomass was a third less. The higher net uptake of ^{14}C than of ^{33}P from the glucose-6-phosphate indicates that soil microorganisms use the organic moiety of phosphorylated organic compounds as a C source, but only use a small proportion of the phosphorus.

CHAPTER-III



== **Materials and Methods**

Chapter -III

Materials and Methods

3.1. Materials:

3.1.1. Study location and climate:

The study was conducted in university farm of Uttar Banga Krishi Viswavidyalaya, Cooch Behar district of West Bengal, India. The university farm lies at approximately 26°23'48.4" N latitude and 089°23'05.3" E longitude.

The area has a typical humid climate. The area receives 3000 mm annual rainfall. About 98 percent of the annual precipitation falls between April to September. The mean maximum and minimum temperatures (31-years average) are 29.55°C and 19.09°C, respectively.

3.1.2. Land-use practices:

Following four common land uses practices were targeted for this study i.e. orchard, social forest, cultivated land, and fallow grass land (Table 1). All the land-use practices were located within the university farm. All sites were located on the same physiographical units. These sites were either adjacent to one another or across the farm roads, with maximum distance of 500 m.

Table 1: Land use practices used in the study.

No	Land Use	Vegetation/ Cropping systems
1.	Cultivated land	Jute - Rice- Fallow Jute -Maize-Potato Fallow- Maize-Wheat Brinjal-Pumpkin-Cabbage
2.	Orchard	Mango, Litchi, Guava and Safeda
3.	Social Forest/Agro-forest	Sissoo, Teak, Gamar and Rubber
4.	Fallow grass land	<i>Cyperus sp.</i> , <i>Polygonum sp.</i> , <i>Cynodon dactylon</i> and <i>Panicum sp.</i>

3.1.3. Soil sampling procedure:

Soil samples were collected from 0 to 15 cm. The soil sampling design involved selection of four sites from each land use pattern. These represent four replications for each land use pattern. Following collection, soil samples were sealed in plastic bags and transported to the laboratory.

3.1.4. Soil processing:

Field-moist soils were processed within 2 days of collection, passing the soil through an 8-mm sieve. Root pieces and organic debris (not incorporated into aggregates and longer than 8 mm) that passed through the sieve were removed. After thorough mixing, a subsample was dried at 105⁰C for gravimetric determination of the water content. Another subsample of the bulk soil was stored at 4 ⁰C for enzymatic analysis.

3.1.5. Equipments:

- a) **Electrical balance:** All solutions used for the investigation were prepared by using an electronic balance (METTLER TOLEDO-PL-202-S/03).
- b) **Spectrophotometer:** The spectroscopic estimation of different compounds of the experimental soils and organic matters was done by Systronics make digital Spectrophotometer (Model: Systronics, 104).
- c) **Flame photometer:** Available K of the soil and organic matters was measured by a flame photometer (Model: Systronics, 128).
- d) **pH meter:** pH of the soils, organics and adjustments of solutions to defined pH was measured by using a digital glass electrode pH meter (Model: Systronics, 335).
- e) **Centrifuge:** Supernatant solutions were collected by using a research centrifuge (Model: Sigma-3k30).
- f) **BOD Incubator:** The incubation study and kinetics experiments were conducted in a BOD Incubator (Make: Remi).

- g) Hot Water Bath:** For the estimation of dehydrogenase activity, anaerobic mineralization index and for heating of samples hot water bath was used (Make:YOMA).
- h) Kjeltex-2300:** For the estimation of available nitrogen Kjeltex-2300 was used.
- i) CHN-Analyser:** Total organic carbon and sulphur were analysed by CHN-Analyser (Elementer, Vario EL, Germany).
- j) Yoder Apparatus:** Soil Aggregate analysis was determined by Yoder Apparatus (LABLINE APPLIANCES) Kolkata-700089

3.1.6. Chemicals:

All the chemicals used in the study were AR and GPR quality and were purchased from Hi Media (Mumbai, India), Merck (Mumbai, India) and Sigma-Aldrich (Germany) .

3.2. Methods:

3.2.1. Physico-chemical properties of soil:

Important Physico-Chemical properties of collected soils (pH, EC, Maximum water holding capacity, Organic carbon, Textural classes, Available-N, Available-P and Available-K, Cation Exchange Capacity, Exchangeable Ca^{+2} and Mg^{+2} , total N, total organic carbon and bulk density) were determined following the standard methodology.

- a) Soil texture:** Sand, silt and clay content was estimated following Bouyous hydrometer method as described by Gee and Bauder (1986).
- b) Maximum water holding capacity:** for determination of moisture holding capacity of the soil, Keen Rackzowski method as described by Baruah and Barthakur (1997).
- c) pH:** pH of the soils was determined by using soil suspension in water in the ratio of 1:2.5 (Jackson, 1973).
- d) Organic carbon:** Oxidisable organic carbon of soil samples was estimated by the method of Walkley and Black (Nelson and Sommers, 1982).

- e) **Electrical Conductivity:** Electrical Conductivity of the suspension (soil: water:: 1:2.5) was determined at room temperature by using a direct reading conductivity meter (Jackson, 1973).
- f) **Available-N:** Available-N was determined by the method of Kenny and Bremner (1964) as described by Page *et al.* (1982) using 2 M KCl and titrimetrically determining the ammonia liberated after steam distillation with magnesium oxide and Devardas alloy.
- g) **Available-P:** Available-P content of the experimental soil was determined by extracting 2.5 gm of each soil with a mixture of 25ml of 0.03M NH_4F and 0.025 M HCl for 5 minutes (Bray and Kurtz, 1945), followed by colorimetric measurement of P by spectrophotometer.
- h) **Available-K:** Available K was extracted by shaking the soil with neutral normal ammonium acetate (1N NH_4OAc) in 1:5 ratio followed by filtration as described by Hanway and Heidel (1952).
- i) **Total-N:** Total N was determined by modified kjeldhal digestion method. Soil was digested with sulphuric acid, catalyst mixture. The digested material was then steam distilled with concentrated (40%) NaOH. The amount of liberated ammonia was absorbed in boric acid and titrated with standard HCl (Bremner and Mulvaney, 1982).
- j) **Cation Exchange Capacity (CEC):** This was determined by ammonium saturation method (Jackson, 1973). In this method, experimental soils were treated with neutral normal ammonium acetate solution, which saturated the soil with NH_4^+ ions, followed by the removal of excess salts using alcohol. The adsorbed NH_4^+ ions were finally determined by distilling the soils with magnesium oxide (MgO) and collecting the evolved ammonia gas in a known volume of mixed indicator (Bromocresol green + Methyl red +Boric acid) followed by titrating the solution with a H_2SO_4 acid of known strength
- k) **Total-P:** Total-P was determined per Sommers and Nelson (1972).

I) Total N and total organic C: Total nitrogen and total carbon of soil were directly analysed by CHNS elemental analyser. For total carbon soil was treated with 6M sulphuric acid to remove the calcium carbonate.

3.2.2. Aggregate analysis and structural indices:

3.2.2.1. Aggregates Separation:

The aggregate separation was done according to Six *et al.* (1998) by wet sieving air-dried soil through a series of six sieves (2000, 1000, 500, 250, 100 and 53 μ m). The air dried soil was quickly submerged in deionised water on top of the 2000 μ m sieve, resulting in slaking of the soil. Slaking disrupts aggregates due to the build of internal air pressure during the rapid wetting of the soil. The aggregates were oven dried at 50°C, weighed and stored at room temperature. Subsequently, seven aggregate fraction were obtained: (i) >2000 μ m, (ii) 2000 to 1000 μ m, (iii) 1000 to 500 μ m, (iv) 500 to 250 μ m, (v) 250 to 100 μ m, (vi) 100 to 53 μ m, (vii) < 53 μ m. The following indices were calculated subsequently:

Analysis of aggregates, its stability and structural indices were done using an instrument based on the principle of Yoder wet sieving apparatus (Kemper and Rosenau, 1986). After removing visible pieces of crop residue and roots from the field moist soil samples aggregates ranging in diameter from 2 to 5 mm were obtained from the air dried bulk soil of each land use that had been broken apart by hand before air drying for the wet sieving procedure. Before sieving, 50 g subsamples (2.0–5.0 mm) in duplicate were slaked by submerging it in deionized water on top 2.0 mm sieve of the seven nested sieves viz. 2.0, 1.0, 0.5, 0.25, 0.1, 0.053 and <0.053 mm aggregate size class for 5 min at room temperature. One of the samples was kept for determination of aggregate analysis and other was used for primary particles. The drum of the sieve shaker was filled with tap water to a level slightly below that of the top screen. Another samples for determination of primary particles, was first used for dispersion by mechanical stirrer with 0.5% (w/v) sodium hexa metaphosphate (1: 3: soil: solution) for 15 minutes and then placed on the top sieve (2.0 mm) of another nest of sieves. The nests of sieves of such prepared soil samples were immersed in water and vertically shaken for 30 min with a frequency of 30-35 cycles min⁻¹ through a stroke length of 3.8 cm with an electric

motor, taking care that the samples on the top sieve remain immersed throughout the full stroke. After this period, the sieve sets were removed from water and was kept for 5 minutes to drain the water from the sieves. Aggregates and primary particles retained on sieves were backwashed separately into filter paper on Buckner funnel, attached to a suction instrument. Aggregate fractions retained on each sieve were transferred into a preweighed container, dried at 60°C in an oven and weighed. The weights were recorded for correction of primary particles. With the data of soil aggregates and primary particles of the first five nested sieves (expected 0.053 and <0.053 mm), the following soil aggregate indices were calculated.

3.2.2.2. Water stable aggregates:

From the weight of the soil particles (Aggregates + primary particles) in each size group, its proportion to the total sample weight was determined. Water stable aggregate (WSA) was the mass of stable aggregates divided by the total aggregate (stable + primary particles) mass calculated as [Eq. 1]:

$$WSA (\%) = \left[\frac{(Weight\ of\ soil + Sand)_i - (Weight\ of\ sand)_i}{Weight\ of\ sample} \right] \times 100 \quad [1]$$

Where, *i* denotes the size of the sieve. In this way, aggregates were then fractioned into macro (> 2.0 mm) and micro aggregate (< 0.25 mm).

3.2.2.3. Aggregate ratio:

Aggregate ratio (AR) was calculated as [Eq. 2]:

$$AR = \left[\frac{Percentage\ of\ water\ stable\ macroaggregate\ > 0.25\ mm}{Percentage\ of\ water\ stable\ microaggregate\ < 0.25\ mm} \right] \quad [2]$$

3.2.2.4. Percent aggregate stability:

The percent aggregate stability or degree of aggregation (AS) of soil was calculated as [Eq. 3]:

$$\%AS = \left[\frac{\% \text{ soil particle } > 0.25 \text{ mm} - \% \text{ primary particle } > 0.25 \text{ mm}}{\% \text{ primary particle } < 0.25 \text{ mm}} \right] \quad [3]$$

3.2.2.5. Proportion of Micro-Aggregate within Macro-Aggregate:

Proportion of micro-aggregate (0.05-0.25 mm) weight within macro-aggregates (0.25-2.0 mm) was calculated as (Six *et al.*, 2000) [Eq. 4]:

$$\text{Micro-aggregates within macro-aggregate} = \left[\frac{\text{Micro-aggregate wt.} - \text{Wt. of 0.05-0.25 mm sand}}{\text{Macro-aggregate wt.} - \text{Wt. of 0.25-2.0 mm sand}} \right] \quad [4]$$

3.2.2.6. Structural coefficient:

Structural coefficient (SC) was calculated according [Eq. 5]:

$$SC = (D-S)/S \quad [5]$$

Where, D is the % of particles <0.25 mm as determined by mechanical analysis and S is the % of aggregates <0.25 mm as determined by wet sieving analysis. Higher the value of SC, the better is the structure.

3.2.3. Carbon mineralization of soils (Plante *et al.*, 2010):

Inoculum was created by incubating 20 g of untreated (non-hydrolysed) soil for 7 days following the addition of 6.5 g mixture consisting of 1 part Cellobiose dissolved in water (1 mg g⁻¹) and one part Hoaglands solution. The inoculum was extracted from the soil by adding 200 ml of water and shaking the soil for 30 min. The soil was allowed to settle and the liquid portion was extracted. This solution was added to bring the samples to 60% of water holding capacity. Determination of the C concentration in the inoculum and subsequent correction, calculation showed that C added in the inoculum did not contribute significantly to the CO₂ respired during incubation. 20 g soil (oven dry basis) was weighted and water was added to raise the moisture to 50% of water holding capacity and taken into 500ml conical flasks. 10 ml of 1.0 N NaOH was taken in small glass container. The glass container was hanged with the help of thread inside the conical flask without touching the soil. The flasks were made airtight by rubber stopper and incubated at 28°C and 35°C appropriately in BOD incubator. A blank was maintained only with NaOH. The flask was opened periodically and removed the NaOH

for CO₂ determination. The flask was allowed to remain open when the titration procedure was done. This was done to replenish O₂ in the jar for the next incubation period. Each flask along with soil was weighed and moisture was added if required. Carbon dioxide free water (25ml) and an excess amount of 3N BaCl₂ was added to NaOH. Phenolphthalein indicator (3 or 4 drops) was added and titrated with 0.5N H₂SO₄. NaOH from the blank was titrated in the same manner. The mineralization experiment was continued for 91 days with three replications.

3.2.4. Enzyme activities of soils:

3.2.4.1. β -glucosidase activities (Dick *et al.* 1996):

β -glucosidase activities of soil was measured using method described by Dick *et al.* (1996). Briefly, one gram of fresh soil was taken in a 50ml Erlenmeyer flask, treated with 0.25ml toluene, 4ml Modified Universal Buffer (MUB) (pH 6) and 1 ml p-nitrophenyl β -D-glucoside (PNG) solution (made in the same buffer). After covering the mouth of flask with aluminium foil, the contents was gently mixed and incubated at 37⁰C for 1 h. After incubation, 1 ml CaCl₂ solution and 4ml tris buffer (pH 12) were added to the mixture. The content was mixed and then filtered through whatman No. 42 filter paper. The absorbance of the filtrate was measured at 440 nm. To perform control, 1 ml p-nitrophenyl β -D-glucoside (PNG) solution was added to the soil suspension after the addition of CaCl₂ and Tris solution. Rest of the procedure was same as done for the sample [Eq. 6]:

Modified Universal Buffer (MUB) stock solution: 3.025g Tris buffer, 2.9 g maleic acid, 3.5g citric acid and 1.575g boric acid were dissolved in about 125 ml (0.5M) NaOH and dilute to 250 ml with distilled water.

Modified Universal Buffer (MUB) pH 6: 50ml of MUB stock solution to be titrated to pH with 0.1 (M) HCl and dilute to 250ml by distilled water.

p-Nitrophenyl β -D-glucoside (PNG) solution: 0.377g of PNG to be dissolved in 40ml of MUB buffer (pH 6) and dilute to 50ml with same buffer.

Calculation:

$$\text{Glucosidase activity } (\mu\text{g pnp released/g oven dry soil/h at } 37^{\circ}\text{C}) = \frac{(C \times V)}{(dw \times sw \times t)} \quad [6]$$

Where,

C= Measured concentration of PNP in sample (μg /ml) - Measured concentration of PNP in control (μg /mL).

V= Total volume (ml) of soil suspension.

dw = Oven dry of weight of 1 g soil.

sw = The weight of soil sample taken (1 g)

t= the incubation time (1h).

3.2.4.2. N-acetyl-β-glucosaminidase assay:

Soil β-glucosaminidase activity was assayed by placing 1.0 g of soil into a 50 ml Erlenmeyer flask, and then adding 4 ml of 0.1 M acetate buffer (pH 5.5) and 1 ml of 10 mM p-nitrophenyl-N-acetyl- β -D-glucosaminide solution. The slurries were mixed thoroughly, stoppered, and placed in an incubator at 37°C. After 1 h of incubation, 1 ml of 0.5 M CaCl₂ and 4 ml of 0.5 M NaOH were added to stop the reaction. The samples were swirled and filtered through Whatman no. 2 filter paper. The colour intensity of the filtrate was measured at 405 nm with a spectrophotometer. Controls were performed with the substrate being added after the reactions were stopped. Additional controls were performed by following the procedure described but without addition of soil to the reaction mixtures. The controls were designed so that they allowed for subtraction of the soil background colour and any trace amount of p-nitrophenol produced by chemical hydrolysis of pNNAG during the incubation. The p-nitrophenol contents of the filtrates were then calculated by comparing the results to a standard curve for p-nitrophenol developed as described by Tabatabai and Bremner (1969).

Reagents:**1. Acetate buffer (100 mM, pH 5.5)**

Prepared by dissolving 13.6 g sodium acetate trihydrate in about 800 ml of distilled water. The solution was titrated to pH 5.5 with 99% glacial acetic acid, and the volume was adjusted to one liter.

2. Modified universal buffer (MUB, 5X stock solution):

Prepared by dissolving 12.1 g tris (hydroxymethyl) aminomethane (THAM), 11.6 g maleic acid, 14.0 g citric acid, and 6.3 g boric acid (H_3BO_3) in 488 ml of 1 M sodium hydroxide and adjusting to a final volume of one liter with distilled water. The 5X stock solution was stored at 4°C. This solution was titrated to the desired pH and diluted five times by volume with distilled water before use.

3. ρ -nitrophenyl-N-acetyl- β -D-glucosaminide (ρ NNAG, 10 mM)

Prepared by dissolving 0.342 g ρ -nitrophenyl-N-acetyl- β -D-glucosaminide (N-9376, Sigma Chemicals, St. Louis, MO.) in 100 ml of acetate buffer (0.1 M, pH 5.5). The solution was stored at 4°C.

4. Calcium chloride (CaCl_2 , 0.5 M)

Prepared by dissolving 36.75 g $\text{CaCl}_2 \cdot 2\text{H}_2\text{O}$ in about 400 ml of distilled water, and adjusting to a final volume of 500 ml with distilled water.

5. Sodium hydroxide (NaOH, 0.5 M)

Prepared by dissolving 10 g of NaOH in about 400 ml of distilled water, and adjusting to a final volume of 500 ml with distilled water.

6. Standard ρ -nitrophenol solution

Prepared by dissolving 1.0 g of ρ -nitrophenol (Sigma Chemicals, St. Louis, MO.) in ca. 800 ml of distilled water, and adjusting the final volume to one liter with distilled water. The solution was stored at 4°C.

Standard Curve

Solutions of 0-5 μg ρ -nitrophenol (15 mM ρ -nitrophenol in MUB, pH 6) were made to develop a standard curve for β -glucosidase and NAGase assays (Tabatabai and Bremner 1969). The solutions were incubated for 1 hour at 37°C. After incubation, the yellow PNP color was developed by adding 1 mL of 0.5 M CaCl_2 and 4 mL of Tris buffer (pH 10). The standard solutions were measured by absorption on a spectrophotometer at a wavelength of 405 nm and a standard curve developed by plotting absorption vs concentration.

3.2.4.3. Protease activity (Ladd and Butler, 1972):

Protease activity was determined with sodium caseinate as the substrate. One g of moist, sieved soil (2mm) was placed in a centrifuge tube, 5 ml Tris buffer and 5 ml sodium caseinate solution were added. The tube was stoppered, contents were mixed and

incubated for two hrs at 50°C on a shaking water bath. At the time end of incubation, 5 ml of TCA solution was added followed by mixing the content thoroughly. To perform the controls, add 5ml sodium caseinate solution at the end of the incubation and immediately before adding the TCA solution. Centrifuge (10,000-12,000 rev min⁻¹, 10 min). pipette 5 ml of the clear supernatant into tubes, mix with 7.5 ml of the alkaline reagent, and incubate for 15 min at room temperature. After adding 5 ml of the Folin reagent, filter the mixture through Whatman 42 filter paper into glass tubes and measure the absorbance after exactly 1 h at 700 nm.

1. **Tris buffer (50mM, pH 8.1):** Dissolve 6.05 g Tris (hydroxyl methyl) amino methane in 700 ml distilled water, adjust the pH to 8.1 with HCl and dilute to 1000 ml with distilled water.
2. **Sodium caseinate (2%):** Suspend 10 g sodium caseinate in warm distilled water (50°C) and bring up with distilled water to 500 ml (use a stirrer). If only casein is available, dissolve 10 g in Tris buffer, adjust the pH to 8.1 with NaOH solution and bring upto 500 ml with Tris buffer (50mM, pH 8.1).
3. **Trichloro-acetic acid (15%):** Dissolve 75 g trichloro-acetic acid (TCA) in about 300 ml distilled water and dilute to 500 ml with distilled water.
4. **Alkaline reagent:** Dilute 60 ml of NaOH (1M) with distilled water before dissolving 50 g Na₂CO₃ (water free) in the solution and bring up to 1000 ml with distilled water.
 - a. Dissolve 0.5 g CuSO₄. 5H₂O in distilled water and dilute to 100 ml with distilled water.
 - b. Dissolve 1 g potassium sodium tartrate (C₄H₄KNaO₆. 4H₂O) in distilled water and dilute to 100 ml with distilled water.
 - c. Mix 1000 ml of NaOH/Na₂CO₃ solution with 20 ml of CuSO₄ solution and 20 ml of potassium sodium tartrate solution.
5. **Folin-Ciocalteu reagent (33%):** Dilute 167 ml of Folin reagent to 500 ml with distilled water.

- a. Tyrosine standard solution (500 µg ml⁻¹): Dissolve 50 g tyrosine in tris buffer and dilute to 100 ml with tris buffer.

Calculation: Correct the measured absorbance for controls and calculate as follows [Eq. 7]:

$$\text{Protease activity } (\mu\text{g tyrosine g}^{-1} \text{ dwt} 2\text{h}^{-1}) = \frac{C \times 15}{\text{dwt}} \quad [7]$$

Where,

dwt is the dry weight of 1 g of moist soil, 15 is the final volume solutions, added to the soil in the assay and C is the measured tyrosine concentration (µg ml⁻¹ supernatant or filtrate).

3.2.4.4. Assay of acid and alkaline phosphatase:

The assay of both acid (EC 3.1.3.2) and alkaline (EC 3.1.3.1) phosphatases were done using acetate buffer (pH 5.0) and borate-NaOH buffer (pH 10.0), respectively (Tabatabai and Bremner, 1969; Eivazi and Tabatabai, 1977). For this assay 4 ml acetate buffer (pH 5.0) for acid phosphatase or 4 ml borate buffer (pH 10.0) for alkaline phosphatase was added to 0.5 ml sample extract in a test tube. One (1.0) ml substrate i.e. disodium salt of *p*- nitrophenyl phosphate (PNPP) was mixed with culture filtrate and the test tubes, after sealing with parafilm, were incubated in an incubator at 37°C for one hour. After incubation, parafilm sealing was removed and 4 ml NaOH (0.5 M) was added to stop the reaction. The flasks were swirled for few seconds to mix the contents. A control for the assay was maintained by adding substrate (without culture filtrate) after the addition of NaOH. The amount of released PNP (intensity of yellow colour) was measured at 430 nm using a blue filter.

One unit of phosphatase activity was defined as the amount of enzyme per ml releasing 1.0 nmol of PNP per minute under the assay condition employed.

Preparation of reagents:

1. **Borate- NaOH buffer (pH 10.0):** 12.404 g boric acid was dissolved in 700 ml distilled water, pH was adjusted to 10.0 with NaOH and volume made to 1 liter.
2. **Acetate buffer (pH 5.0):** 82.03 g of sodium acetate anhydrous was dissolved in 700 ml distilled water, pH was adjusted to 5 with acetic acid and volume was made to 1 liter.
3. ***P*-nitrophenyl phosphate, disodium salt (100mM):** 0.92 g was dissolved in 25 ml of distilled water.
4. **NaOH, 0.5 M:** 20g NaOH was dissolved in 1 liter of distilled water.
5. **Standard *p*-nitrophenol (1000ppm):** 1.0 g *p*-nitrophenol was dissolved in 700ml of distilled water and dilutes the solution to 1L.

Preparation of standard curve:

Standard *p*-nitrophenol (1000 ppm) was diluted to 10ppm in a volumetric flask. 0, 1, 2, 3, 4 and 5 ml aliquots were taken in test tubes, volume was adjusted to 6ml and 4 ml of 0.5 M NaOH solution was added to the substrate before measuring the yellow colour intensity.

3.2.4.5. Dehydrogenase activities (Tabatabai, 1994):

Dehydrogenase enzyme activity was determined as described by Tabatabai (1994). Six grams of moist soil sample were used in this analysis. Soil was incubated with 2, 3, 5-triphenyltetrazolium chloride substrate at 37°C for 24 h. A previously developed standard curve was used to calculate the concentration of triphenyl formazan (TPF) product colorimetrically at 485 nm. The enzymatic activity was expressed in μg TPF released g^{-1} dry soil.

3.3. N and P mineralization under anaerobic condition (Waring and Bremner, 1964; Keeny, 1982):

The anaerobic incubation procedure (waterlogged) used in the study was previously described by Waring and Bremner (1964) and modified and developed by Keeney (1982) and is strongly recommended for the assessment of the mineralizable N and P during an incubation period. The method involves the incubation of a soil sample under waterlogged conditions in an enclosed test tube with as little headspace as possible in the tube. Distilled water of about 12 ml was placed in a 16 ×150 mm screw cap test

tube, and then 5 gm of oven dry equivalent soil was added to the tube. The test tubes were capped and placed in a constant temperature water bath maintained at $40^0 \pm 1^0\text{C}$. Three replicates of soils were maintained for three sets. At the end of 7, 14 and 21 days, the tubes were removed from the water bath and shaken briefly to mix the content. The soil water mixture of each tube was quantitatively transferred to a 150-ml distillation flask. About 50 ml of 2M KCl was used to extract the mineral N and P. The mineral nitrogen was estimated by distillation method while P was estimated spectrophotometrically at 780 nm.

3.4. Statistical analysis:

Pearson correlation coefficients (r) were calculated among the C, N-mineralization and selected parameters, which may be potential indices for N-mineralization. Significance of correlation coefficients was tested from Fisher's and Yates' statistical table. For this experiment, the critical ' r ' value for 37 d.f. was found 0.3165 and 0.4082 at 5 and 1% level significance of respectively. Linear regression analyses (r^2) were also done. All statistical tests were implemented in SPSS v. 16 and Indostat softwares. Comparisons among treatment means were performed using the Least significant difference (LSD). Through Duncan's multiple range test (DMRT), calculated at 5% level of significance.

CHAPTER-IV



Results and Discussion

Chapter -IV

Results and Discussion

4.1 Summary of Soil Physicochemical Properties

Tables 2 and 3 summarized soil physical and chemical properties of four different land use systems, respectively. Soils from all four land use systems exhibited acidic reaction (Table 2), conforming earlier findings (Maji *et al.*, 2012). While soils from all land use systems showed high organic carbon content, as expected, none of them exhibited soil salinity problem (Table 2).

The difference in management practices resulted in difference organic carbon contents. Social forest exhibited highest average organic carbon content (1.30%), probably due to high plant litter deposition, followed by orchard (1.16%), fallow grass land (1.08 %) and cultivated land (1.05%). By contrast, the fallow grass land soils exhibited slightly higher range of organic carbon (0.6-1.51 %) than social forest (0.7-1.66%). Exchangeable Ca^{2+} contents were higher than exchangeable Mg^{2+} . Particle size analysis followed by the use of USDA textural triangle revealed that most soils were sandy loam while maximum water holding capacity ranged between 37.67-62.44 % (Table 3), indicating optimal moisture condition for soil microbial activity. Consequently, these light textured soils yielded higher bulk density ranging from 1.30-1.83 g cm⁻³.

Low available- P_2O_5 ($< 45 \text{ kg ha}^{-1}$) in all soils probably indicated P fixation in acid soils. Furthermore, on average, deficiency in available N was evident in cultivated land (209 kg ha^{-1}), indicating the need for N fertilizer applications (Table 2). On the contrary, higher range of available N ($245\text{-}267 \text{ kg ha}^{-1}$) was exhibited by social forest soils. No linear correlation was found between available N and organic carbon ($\rho=0.14$) while total N exhibited significant correlation with the former ($\rho=0.7$).

Table 2: Chemical properties of soils under different land use systems.

Land Uses		pH	EC (dS m ⁻¹)	OC (%)	CEC Cmol (P ⁺)Kg ⁻¹	Available-N (kg ha ⁻¹)	Total-N (kg ha ⁻¹)	Available- P ₂ O ₅ (kg ha ⁻¹)	Available- K ₂ O (kg ha ⁻¹)	Exchangeable-Ca ⁺² & Mg ⁺² (meq 100gm ⁻¹)	
										Ca ⁺²	Mg ⁺²
Cultivated land	Jute - Rice- Fallow	5.42	0.04	1.01	5.10	209.90	568.66	26.576	110.69	4.59	1.47
	Jute -Maize-Potato	5.52	0.01	1.38	6.53	206.98	896.90	19.742	238.00	5.07	1.12
	Fallow- Maize-Wheat	5.23	0.04	0.96	6.69	204.47	752.64	18.477	130.03	5.24	1.27
	Brinjal-Pumpkin-Cabbage	5.33	0.04	0.86	4.91	197.57	926.17	6.075	158.59	4.60	1.40
Orchard land	Mango	5.54	0.02	1.51	7.47	163.70	756.82	38.725	134.89	2.70	1.67
	Litchi	5.57	0.02	1.04	6.38	165.79	792.36	7.087	99.38	3.26	2.08
	Guava	5.57	0.02	0.99	6.12	175.83	715.01	10.124	193.87	2.43	1.73
	Safeda	5.47	0.01	1.12	6.35	169.76	815.36	17.971	183.64	3.40	2.39
Social forest / Agro Forest land	Sissoo	5.14	0.02	1.66	6.96	257.78	1124.78	27.336	158.78	3.12	1.20
	Gamar	5.33	0.02	1.15	6.70	245.86	825.81	35.941	247.03	3.49	1.33
	Teak	5.59	0.03	1.62	4.93	251.30	848.81	6.834	88.93	2.80	1.03
	Rubber	5.49	0.02	0.76	6.25	267.19	646.02	8.859	80.57	3.81	1.15
Fallow grass land	<i>Cyperus sp.</i>	5.29	0.03	1.21	7.37	98.47	767.27	7.593	106.51	3.58	1.26
	<i>Polygonum sp.</i>	5.36	0.05	1.03	4.26	102.65	721.28	12.908	121.15	3.68	1.42
	<i>Cynodon dactylon</i>	5.46	0.03	0.60	4.00	110.60	579.11	4.556	114.87	2.67	1.27
	<i>Panicum sp.</i>	5.22	0.02	1.51	5.33	95.75	1024.43	3.037	95.83	4.15	1.32

Table 3: Physical properties of soils under different land use systems

Land Uses		Bulk density (g cm ⁻³)	M.W.H.C (%)	Mechanical analysis			Textural Classes
				Sand (%)	Silt (%)	Clay (%)	
Cultivated Land	Jute - Rice- Fallow	1.42	48.59	52.64	37.01	10.35	Sandy Loam
	Jute -Maize-Potato	1.35	46.64	55.64	32.01	12.35	Sandy Loam
	Fallow- Maize-Wheat	1.30	47.67	52.42	30.4	17.18	Sandy Loam
	Brinjal-Pumpkin-Cabbage	1.34	46.57	37.6	51.3	11.1	Silt Loam
Orchard Land	Mango	1.52	45.73	53.86	31.3	14.84	Sandy Loam
	Litchi	1.56	42.76	52.64	32.2	15.16	Sandy Loam
	Guava	1.51	41.60	33.8	51.56	14.64	Silt Loam
	Safeda	1.56	43.76	53.14	32.6	14.26	Sandy Loam
Social Forest/Agro-Forest	Sissoo	1.65	62.44	38.76	50	11.24	Silt Loam
	Gamar	1.61	58.76	54.56	30.18	15.26	Sandy Loam
	Teak	1.61	57.77	41.22	55.27	3.51	Silt Loam
	Rubber	1.59	60.45	36.64	46.01	17.35	Loam
Fallow Grass Land	<i>Cyperus sp.</i>	1.76	39.03	32.23	50.36	17.4	Silt Loam
	<i>Polygonum sp.</i>	1.71	37.67	40.91	52.36	6.73	Silt Loam
	<i>Cynodon dactylon</i>	1.77	41.74	36.58	53.36	10.06	Silt Loam
	<i>Panicum sp.</i>	1.83	43.51	49.94	43.84	6.22	Sandy Loam

4.2 Effects of Different Land Use Patterns on Soil Aggregates Stability

Aggregation is a soil physical phenomenon which has prominent impact onto soil structure, aeration, temperature, crop growth and biological activity (Deb *et al.*, 2015). The quality and amount of C associated with soil aggregates defines the physical stability of C in soil (Jastrow, 1996). Now C is the source of food for soil microbes and therefore, aggregation plays a vital role in soil microbial dynamics. Consequently, this study focused the impact of land use pattern on soil aggregate stability.

As per Table 4, cultivated land has significantly less water stable soil macro-aggregates. Within cultivated lands, lowest macro as well as micro-aggregate formation has been found under vegetables (Brinjal-Pumpkin-Cabbage). On the other hand, within orchard/ forestry/ fallow grassland, no individual tree/ grass species showed precise impact on soil aggregation. For better understanding, different soil aggregate indices has been calculated in this study (Table 5). Quantitatively, micro-aggregate formation within macro-aggregate (MicA within MacA) was low in orchard and agricultural soils. Comparison within cultivated lands inferred maximum MicA within MacA in Jute-Maize-Wheat soils. Soil under Sissoo plantation and *Cyperus sp.* showed highest MicA within MacA among social forest/agro-forestry and grasslands respectively. Percent aggregate stability, aggregate ratio as well as structural coefficient of soil aggregates showed lowest values under cultivated lands. Less value of these indices in cultivated soils point towards degradation of soil aggregation under long-term agriculture practices (Bronick and Lal, 2005).

Table 4: Water stable aggregate % in soils under different land use systems

		Water stable aggregate (%)					
		MacA				MicA	
Land Uses		> 2.0	2.0-1.0	1.0-0.5	0.5-0.25	0.25-0.1	0.1-0.05
Cultivated Land	Jute - Rice- Fallow	17.30 ^a	9.16 ^a	10.34 ^c	18.50 ^c	9.56 ^d	2.10 ^b
	Jute -Maize-Potato	16.86 ^b	6.64 ^c	12.48 ^a	20.28 ^b	13.88 ^a	2.92 ^a
	Fallow- Maize-Wheat	17.16 ^a	7.00 ^b	11.60 ^b	18.32 ^c	11.18 ^b	2.00 ^b
	Brinjal-Pumpkin-Cabbage	13.26 ^c	6.26 ^d	10.30 ^c	21.60 ^a	10.42 ^c	0.12 ^c
LSD ($P=0.05$)		0.34	0.10	0.09	0.12	0.09	0.18
Orchard Land	Mango	20.16 ^d	11.56 ^a	13.36 ^d	17.24 ^c	7.18 ^c	0.22 ^d
	Litchi	20.76 ^c	10.82 ^b	13.96 ^c	16.98 ^d	7.20 ^c	0.50 ^c
	Guava	21.46 ^b	10.76 ^b	14.40 ^b	17.50 ^b	7.66 ^a	1.96 ^b
	Safeda	23.76 ^a	9.46 ^c	14.64 ^a	18.80 ^a	7.46 ^b	2.28 ^a
LSD ($P=0.05$)		0.19	0.25	0.21	0.16	0.14	0.12
Social Forest/Agro-Forest	Sissoo	22.26 ^b	15.08 ^b	7.60 ^d	17.40 ^a	12.14 ^d	2.34 ^d
	Gamar	22.66 ^a	10.52 ^d	10.90 ^a	14.02 ^c	13.32 ^c	6.62 ^a
	Teak	21.86 ^c	12.86 ^c	9.12 ^b	11.78 ^d	16.60 ^a	5.42 ^b
	Rubber	21.82 ^c	15.26 ^a	8.68 ^c	14.70 ^b	14.98 ^b	4.54 ^c
LSD ($P=0.05$)		0.22	0.19	0.17	0.19	0.13	0.12
Fallow Grass Land	<i>Cyperus sp.</i>	22.26 ^a	11.56 ^c	8.52 ^d	17.10 ^a	13.20 ^c	4.54 ^b
	<i>Polygonum sp.</i>	22.58 ^a	9.40 ^d	8.86 ^c	13.54 ^d	15.72 ^a	5.72 ^a
	<i>Cynodon dactylon</i>	21.36 ^b	11.94 ^b	9.70 ^b	14.30 ^c	13.98 ^b	2.06 ^d
	<i>Panicum sp.</i>	19.32 ^c	14.20 ^a	10.16 ^a	16.14 ^b	9.80 ^d	4.10 ^c
LSD ($P=0.05$)		0.58	0.15	0.11	0.20	0.10	0.17

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 5: Different aggregate indices of soils under different land use systems

Land Uses		MicA within MacA	%Aggregate Stability	Aggregate Ratio	Structural Coefficient
Cultivated Land	Jute - Rice- Fallow	0.35 ^a	47.5 ^a	0.89	0.47
	Jute -Maize-Potato	0.30 ^c	46.6 ^b	0.94	0.48
	Fallow- Maize-Wheat	0.39 ^b	45.5 ^c	0.93	0.47
	Brinjal-Pumpkin-Cabbage	0.29 ^c	44.6 ^d	0.86	0.47
LSD ($P=0.05$)		0.01	0.60	ns	ns
Orchard Land	Mango	0.28 ^b	69.02 ^b	2.49 ^b	0.69
	Litchi	0.28 ^b	69.98 ^b	2.73 ^a	0.72
	Guava	0.33 ^a	70.35 ^b	2.85 ^a	0.73
	Safeda	0.33 ^a	74.25 ^a	2.94 ^a	0.74
LSD ($P=0.05$)		0.04	2.26	0.23	ns
Social Forest/Agro-Forest	Sissoo	0.65 ^a	68.27 ^a	2.18 ^a	0.66
	Gamar	0.51 ^c	65.25 ^b	1.78 ^c	0.63
	Teak	0.36 ^d	60.73 ^c	1.71 ^d	0.62
	Rubber	0.56 ^b	64.73 ^b	2.01 ^b	0.66
LSD ($P=0.05$)		0.04	0.98	0.07	ns
Fallow Grass Land	<i>Cyperus sp.</i>	0.67 ^a	64.41 ^a	2.19 ^a	0.68 ^a
	<i>Polygonum sp.</i>	0.48 ^b	62.89 ^b	1.99 ^b	0.66 ^{ab}
	<i>Cynodon dactylon</i>	0.34 ^c	59.51 ^c	1.63 ^d	0.61 ^c
	<i>Panicum sp.</i>	0.45 ^b	61.77 ^b	1.81 ^c	0.62 ^{bc}
LSD($P=0.05$)		0.03	1.63	0.05	0.04

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Presence of total C, N and P in bulk soils as well as in different aggregate size fractions were also analysed (Table 6, 7 and 8). Outcomes inferred that cultivated soils (bulk) have less C (Table 6) as well as N (Table 7) in spite of seasonal addition of manures and fertilizers. Besides continuous crop uptake, tillage practices as well as periodical perturbation might be the reason here as these increases the organic matter turnover rate (Balesdent *et al.*, 2000). Similar results were found by Saviozzi *et al.* (2001) earlier. Within cropping systems, lowest soil C was found under vegetable lands (Brinjal-Pumpkin-Cabbage). Soils under Gamar and Rubber and further Mango and Guava plantation had highest and lowest soil C among social forest/ agroforestry and orchard land respectively. Soil under *Cyperus sp.* and *Panicum sp.* showed highest and lowest C within fallow grasslands. Total P of bulk soil was highest under agricultural lands (Table 8).

Analysis of aggregate associated C, N and P shows that maximum macro-aggregated C (MacAC) was found in soils of social forest/ agroforestry. Highest micro-aggregated C (MicAC) was found in soils under fallow grassland and social forest/ agroforestry (Table 6). Lowest macro as well as micro-aggregate occluded C was found under agricultural fields. This finding was similar to the trend of soil aggregation. It infers destruction of soil aggregation under continuous cultivation and subsequent low physically protected C within aggregates in agricultural soils (Balesdent *et al.*, 2000; Saviozzi *et al.*, 2001). Highest macro and micro-aggregate associated N was found in soils under social forest/ agroforestry (Table 7) while soils of orchard area had highest macro and micro-aggregated P (Table 8).

Table 6: Total C (g kg⁻¹, mean value) in bulk soil and associated with different aggregate size fractions (mm) of soils under different cropping systems

		Aggregate associated C (g kg ⁻¹)							
		Bulk Soil	MacAC				MicAC		
Land Uses			>2.0	2.0-1.0	1.0-0.5	0.5-0.25	CMicAC		Silt+ Clay
							0.25-0.1	0.1-0.05	<0.05
Cultivated Land	Jute - Rice- Fallow	19.48 ^a	17.36 ^a	14.29 ^a	14.78	9.19 ^a	7.56 ^a	5.57 ^{ab}	7.96 ^a
	Jute -Maize-Potato	16.46 ^b	11.76 ^b	16.11 ^a	12.72	10.94 ^a	9.01a	7.70a	8.42 ^a
	Fallow- Maize-Wheat	11.13 ^c	10.94 ^b	12.65 ^a	9.43	8.85 ^a	7.83 ^a	7.09 ^{ab}	7.00 ^a
	Brinjal-Pumpkin-Cabbage	8.99 ^d	10.39 ^b	10.28 ^a	5.79	7.16 ^a	6.59 ^a	4.55 ^b	7.77 ^a
LSD(P=0.05)		2.23	2.25	1.37	ns	0.69	0.56	0.63	0.32
Orchard Land	Mango	33.76 ^a	17.22 ^a	15.64 ^b	13.54 ^a	8.14 ^c	10.48 ^a	8.89 ^a	15.20 ^a
	Litchi	26.65 ^b	17.09 ^a	22.37 ^a	14.09 ^a	10.19 ^a	8.27 ^b	5.34 ^c	8.40 ^b
	Guava	9.80 ^d	15.71 ^b	12.08 ^c	10.38 ^b	9.08 ^b	7.37 ^c	4.17 ^d	6.46 ^c
	Safeda	18.16 ^c	10.52 ^c	11.85 ^c	8.81 ^c	8.10 ^c	8.40 ^b	7.17 ^b	8.97 ^b
LSD(P=0.05)		2.68	0.92	2.50	0.58	0.63	0.51	0.17	0.71
Social Forest/Agro-Forest	Sissoo	29.27 ^b	22.15 ^b	16.90 ^c	11.96 ^{bc}	9.03 ^c	10.42 ^a	6.52 ^b	10.94 ^b
	Gamar	37.31 ^a	27.82 ^a	22.25 ^a	20.50 ^a	11.14 ^b	7.49 ^c	4.36 ^d	10.07 ^c
	Teak	24.51 ^c	23.60 ^b	19.18 ^b	14.74 ^b	12.25 ^a	11.74 ^b	8.97 ^a	14.83 ^a
	Rubber	8.37 ^d	7.98 ^c	7.45 ^d	7.79 ^c	7.21 ^d	4.19 ^d	5.71 ^c	7.30 ^d
LSD(P=0.05)		4.23	1.62	0.63	4.23	0.84	0.73	0.45	0.34
Fallow Grass Land	<i>Cyperus sp.</i>	32.35 ^a	13.66 ^b	13.38 ^b	12.88 ^b	11.35 ^b	8.58 ^c	9.94 ^b	11.46 ^c
	<i>Polygonum sp.</i>	28.91 ^b	12.19 ^b	10.98 ^c	12.52 ^b	10.66 ^c	9.46 ^b	8.53 ^c	7.70 ^d
	<i>Cynodon dactylon</i>	18.34 ^c	19.16 ^a	23.79 ^a	20.62 ^a	18.57 ^a	15.86 ^a	13.48 ^a	13.16 ^b
	<i>Panicum sp.</i>	16.59 ^d	12.40 ^b	9.38 ^d	6.78 ^c	4.07 ^d	5.59 ^d	6.06 ^d	14.98 ^a
LSD(P=0.05)		1.54	2.09	0.74	0.81	0.61	0.08	0.65	1.53

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 7: Total N (g kg⁻¹, mean value) in bulk soil and associated with different aggregate size fractions (mm) of soils under different cropping systems

		Aggregate associated N (g kg ⁻¹)							
		Bulk soil	MacAN				MicAN		
Land Uses			>2.0	2.0-1.0	1.0-0.5	0.5-0.25	NMicAN		Silt+ Clay
							0.25-0.1	0.1-0.05	<0.05
Cultivated Land	Jute - Rice- Fallow	9.63 ^a	1.73 ^a	1.57 ^a	1.64 ^a	1.20 ^a	1.00 ^a	0.76 ^a	1.08 ^a
	Jute -Maize-Potato	8.56 ^b	1.32 ^a	1.68 ^a	1.38 ^a	1.20 ^a	1.00 ^a	0.87 ^a	1.04 ^a
	Fallow- Maize-Wheat	7.52 ^c	1.27 ^a	1.28 ^a	1.16 ^a	0.99 ^a	0.97 ^a	0.92 ^a	1.01 ^a
	Brinjal-Pumpkin-Cabbage	7.63 ^c	1.17 ^a	1.12 ^a	1.13 ^a	0.99 ^a	1.03 ^a	0.81 ^a	1.23 ^a
LSD(P=0.05)		0.60	0.18	0.17	0.42	0.27	0.20	0.21	0.32
Orchard Land	Mango	11.65 ^a	1.81 ^a	1.73 ^a	1.57 ^a	1.08	1.39 ^a	1.27 ^a	1.97 ^a
	Litchi	11.13 ^a	1.73 ^a	2.07 ^a	1.59 ^a	1.34	1.22 ^a	0.84 ^b	1.32 ^b
	Guava	7.13 ^b	1.50 ^{ab}	1.28 ^b	1.07 ^b	0.93	0.80 ^b	0.53 ^c	0.91 ^b
	Safeda	7.54 ^b	1.16 ^c	1.14 ^b	0.99 ^b	0.93	1.10 ^{ab}	0.89 ^b	1.29 ^b
LSD(P=0.05)		0.70	0.31	0.51	0.35	ns	0.32	0.27	0.36
Social Forest/Agro-Forest	Sissoo	11.09 ^b	2.49 ^a	1.91 ^a	1.42 ^{ab}	1.00 ^{ab}	1.28 ^a	0.79 ^a	1.42 ^a
	Gamar	12.60 ^a	2.52 ^a	2.10 ^a	1.98 ^a	1.36 ^a	1.03 ^{ab}	0.64 ^a	1.45 ^a
	Teak	13.61 ^a	2.51 ^a	2.11 ^a	1.70 ^a	1.58 ^a	1.45 ^a	1.09 ^a	1.68 ^a
	Rubber	5.19 ^c	0.84 ^b	0.75 ^b	0.81 ^c	0.64 ^b	0.43 ^b	0.60 ^a	0.87 ^b
LSD(P=0.05)		1.31	0.26	0.12	0.32	0.19	0.33	0.22	0.30
Fallow Grass Land	<i>Cyperus sp.</i>	9.13 ^b	1.31	1.49 ^b	1.43 ^{ab}	1.28 ^{ab}	1.05 ^b	1.13 ^b	1.41 ^b
	<i>Polygonum sp.</i>	8.47 ^b	1.47	1.24 ^b	1.47 ^{ab}	1.23 ^b	1.09 ^b	1.05 ^b	0.88 ^c
	<i>Cynodon dactylon</i>	14.14 ^a	1.72	1.99 ^a	1.92 ^a	1.77 ^a	1.89 ^a	1.69 ^a	1.76 ^a
	<i>Panicum sp.</i>	8.78 ^b	1.55	1.19 ^b	0.98 ^b	0.70 ^c	0.94 ^b	1.04 ^b	2.08 ^a
LSD(P=0.05)		1.55	ns	0.32	0.22	0.19	0.25	0.38	0.28

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 8: Total P (g kg⁻¹, mean value) in bulk soil and associated with different aggregate size fractions (mm) of soils under different cropping systems

			Aggregate associated P (g kg ⁻¹)						
			Bulk Soil	MacAP				MicAP	
								PMicAP	Silt+ Clay
Land Uses			>2.0	2.0-1.0	1.0-0.5	0.5-0.25	0.25-0.1	0.1-0.05	<0.05
Cultivated Land	Jute - Rice- Fallow	4.02 ^a	0.88 ^a	0.67 ^c	1.27 ^a	1.02 ^a	1.28 ^a	0.98 ^b	0.65 ^b
	Jute -Maize-Potato	2.99 ^d	0.69 ^c	0.79 ^a	0.72 ^c	0.82 ^c	0.88 ^c	1.00 ^b	0.59 ^c
	Fallow- Maize-Wheat	3.55 ^c	0.88 ^a	0.70 ^b	0.97 ^b	0.91 ^b	0.95 ^b	1.33 ^a	0.67 ^a
	Brinjal-Pumpkin-Cabbage	3.74 ^b	0.71 ^b	0.61 ^d	0.59 ^d	0.54 ^d	0.61 ^d	0.73 ^c	0.65 ^b
LSD(P=0.05)		0.07	0.01	0.04	0.03	0.02	0.03	0.19	0.01
Orchard Land	Mango	3.34 ^a	0.92 ^a	0.90 ^b	0.97 ^a	0.67 ^b	1.05 ^a	1.00 ^a	1.42 ^a
	Litchi	2.89 ^b	0.87 ^b	0.89 ^b	0.79 ^b	0.85 ^a	0.94 ^b	0.93 ^b	0.55 ^d
	Guava	2.74 ^c	0.79 ^c	0.81 ^c	0.99 ^a	0.66 ^b	0.74 ^d	0.76 ^c	0.75 ^c
	Safeda	1.40 ^d	0.85 ^b	0.95 ^a	0.37 ^c	0.36 ^c	0.87 ^c	0.70 ^d	0.88 ^b
LSD(P=0.05)		0.07	0.03	0.02	0.03	0.03	0.01	0.03	0.02
Social Forest/Agro-Forest	Sissoo	1.35 ^c	0.67 ^c	0.63 ^d	0.57 ^d	0.48 ^b	1.31 ^a	0.62 ^d	0.58 ^d
	Gamar	2.28 ^a	0.79 ^a	0.92 ^a	0.88 ^a	0.46 ^c	0.78 ^c	0.97 ^a	0.86 ^a
	Teak	2.15 ^a	0.74 ^b	0.85 ^b	0.75 ^c	0.46 ^c	0.89 ^b	0.79 ^b	0.74 ^c
	Rubber	1.88 ^b	0.69 ^c	0.80 ^c	0.79 ^b	0.70 ^a	0.89 ^b	0.71 ^c	0.80 ^b
LSD(P=0.05)		0.17	0.02	0.02	0.02	0.02	0.02	0.02	0.02
Fallow Grass Land	<i>Cyperus sp.</i>	2.18 ^b	0.69 ^b	0.71 ^b	0.91 ^b	0.95 ^a	0.78 ^b	1.11 ^a	0.56 ^c
	<i>Polygonum sp.</i>	0.39 ^d	0.57 ^c	0.56 ^c	0.59 ^d	0.58 ^c	0.55 ^c	0.55 ^d	0.56 ^c
	<i>Cynodon dactylon</i>	0.75 ^c	0.66 ^b	0.55 ^c	0.66 ^c	0.55 ^d	0.56 ^c	0.66 ^c	0.66 ^b
	<i>Panicum sp.</i>	3.19 ^a	0.85 ^a	1.02 ^a	0.97 ^a	0.74 ^b	0.99 ^a	0.94 ^b	1.01 ^a
LSD(P=0.05)		0.09	0.01	0.02	0.02	0.02	0.02	0.04	0.02

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Now, micro-aggregates are the most physically protected home of C and N in soil (Lichter *et al.*, 2008). Therefore, estimation of micro-aggregated C and N has been conducted. Separate evaluation of total C and N associated with soil micro-aggregate size fraction 0.25-0.1 mm and 0.1-0.05 has been done. Outcomes reveals lowest presence of micro-aggregated C in soils under cultivated land (for both fractions) and highest micro-aggregate occluded C under fallow grass land (for both fractions) (Fig 2 & 3). The trend was just similar for soil micro-aggregate associated N (Fig 4 & 5).

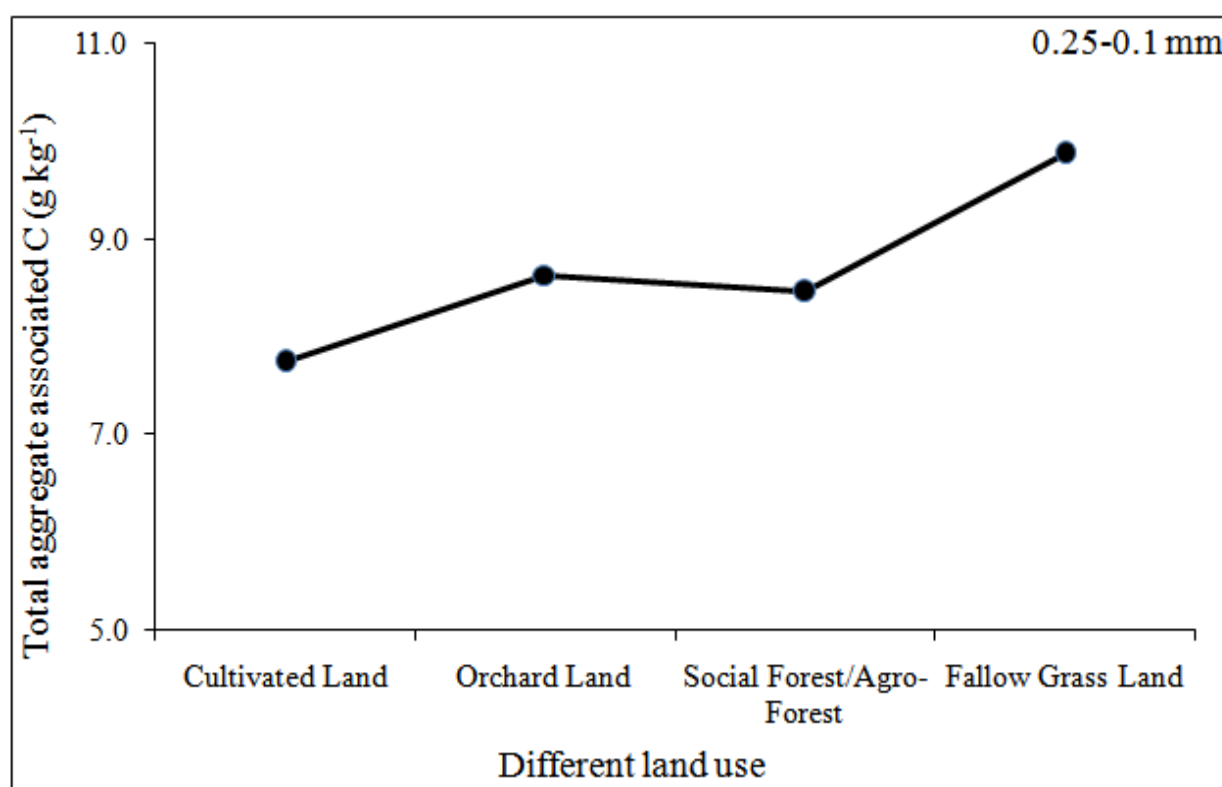


Fig. 2. Total C present in the aggregate size 0.25-0.1mm under different land use systems

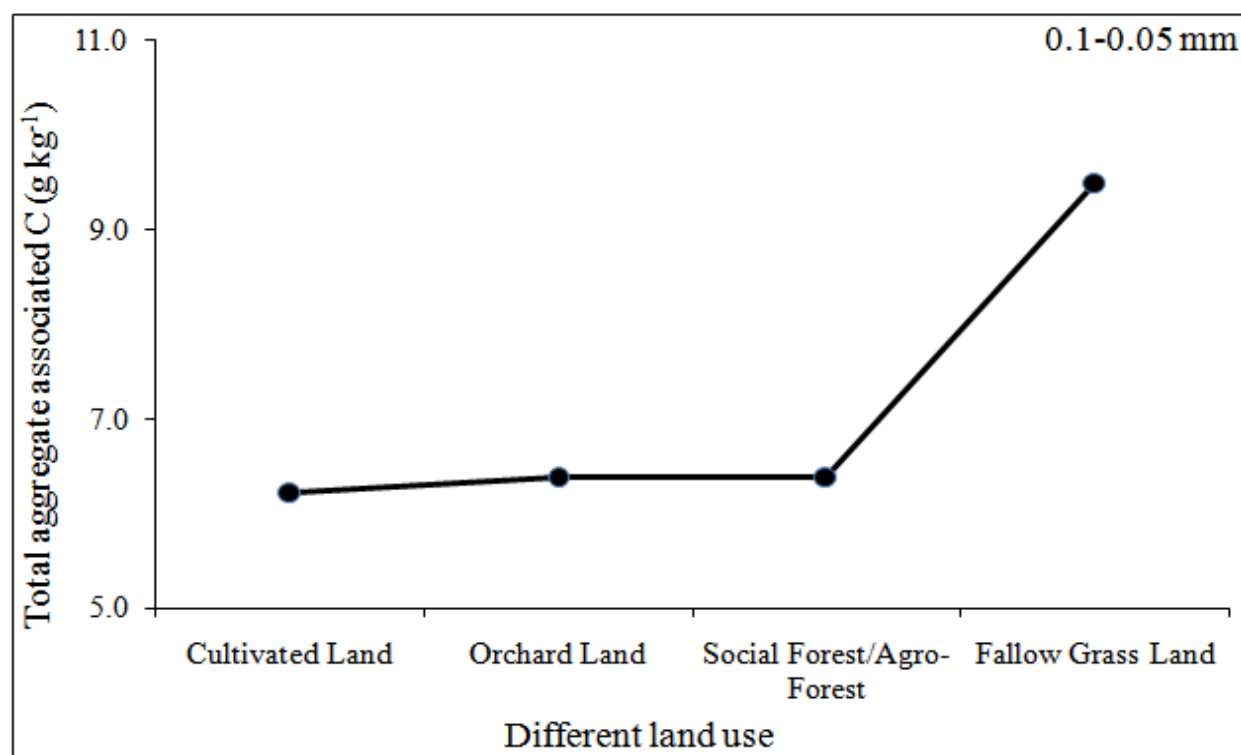


Fig. 3. Total C present in the aggregate size 0.1-0.05mm under different land use systems

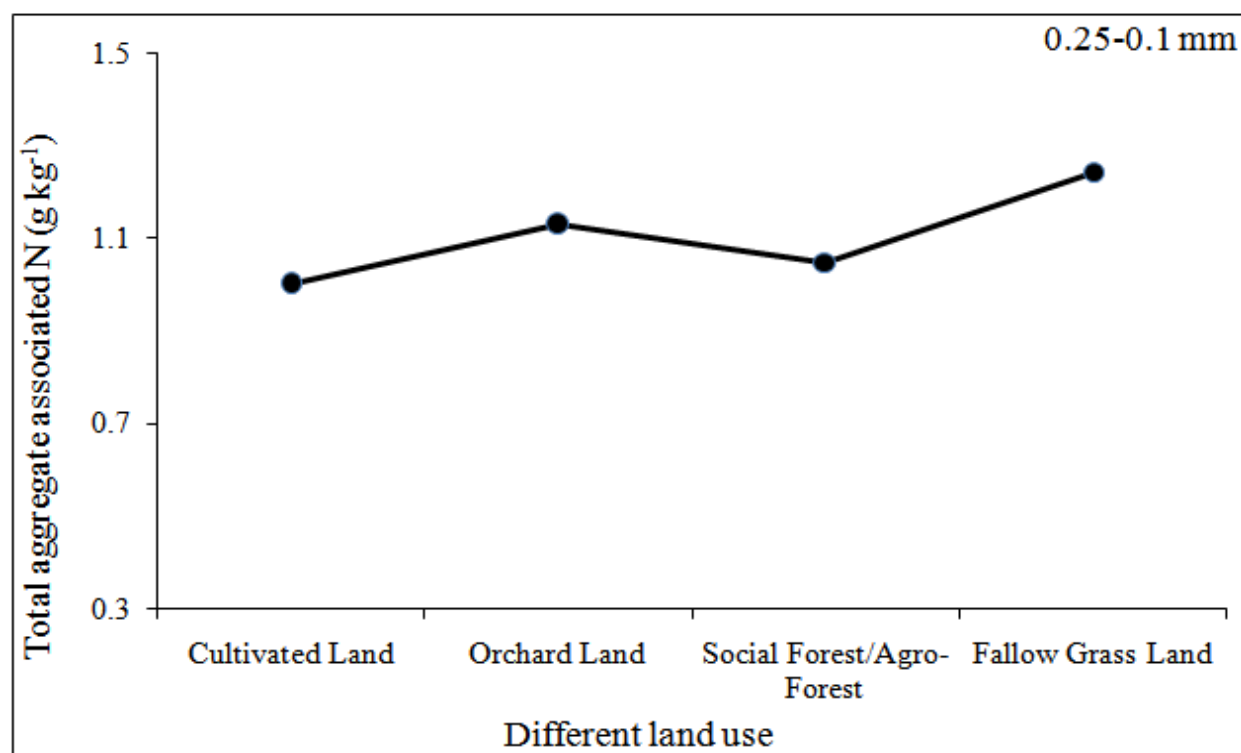


Fig. 4. Total N present in the aggregate size 0.25-0.1mm under different land use systems

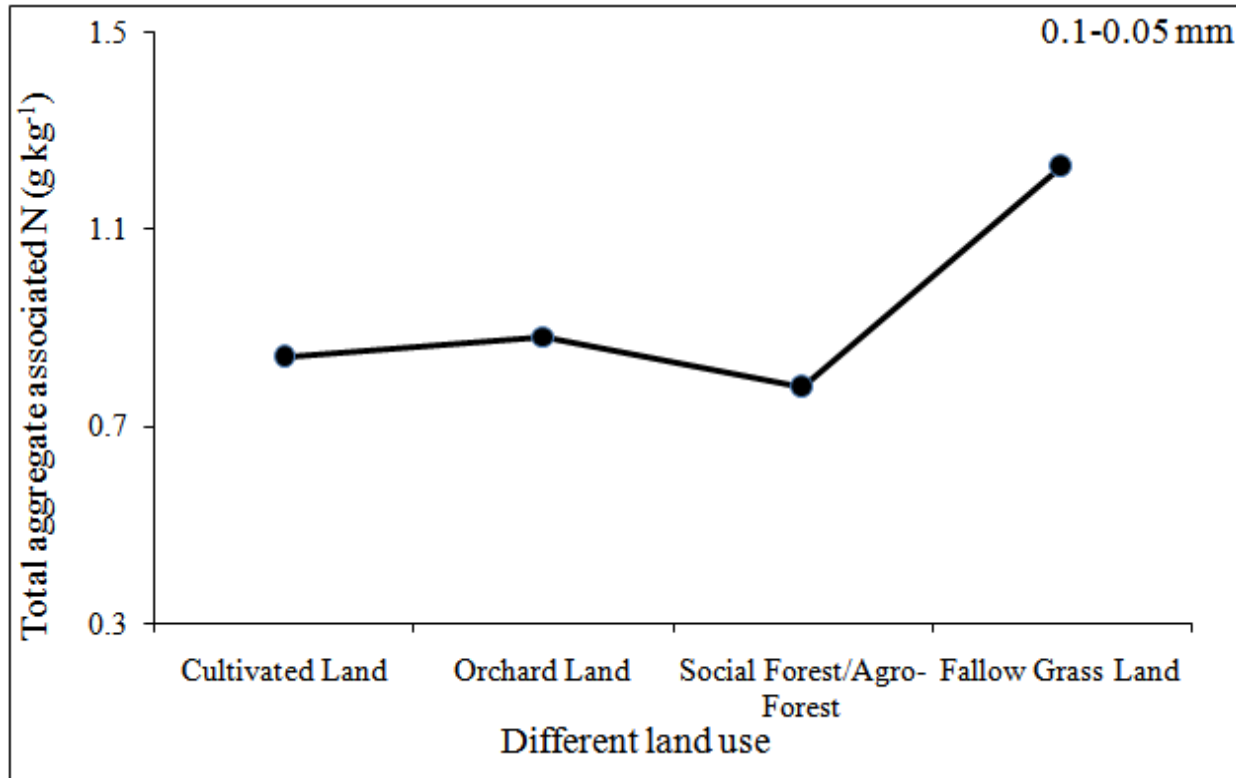


Fig. 5. Total N present in the aggregate size 0.1-0.05mm under different land use systems

Again, soil organic matter (SOM) is highly protected inside the micro-aggregates within macro-aggregates (Denef *et al.*, 2001; Bossuyt *et al.*, 2002). As C and N are the integral part SOM, knowledge of the % presence of micro-aggregated C and N within macro-aggregated C and N is necessary in this regard. Following this, present study tried to understand the proportion of C and N associated with macro-over the micro-aggregates (MacAC/ MicAC and MacAN/ MicAN). Results shows lowest MacAC/ MicAC and MacAN/ MicAN in agricultural soils while highest was under social forest/ agroforestry. These data confirms the less aggregation as well as less aggregate associated C and N in cultivated soils in comparison to soils under social forest/ agroforestry and fallow grass land. Therefore, it seems that C and N available in cultivated soils are physically more available to soil micro-organisms (Fig. 6 and 8).

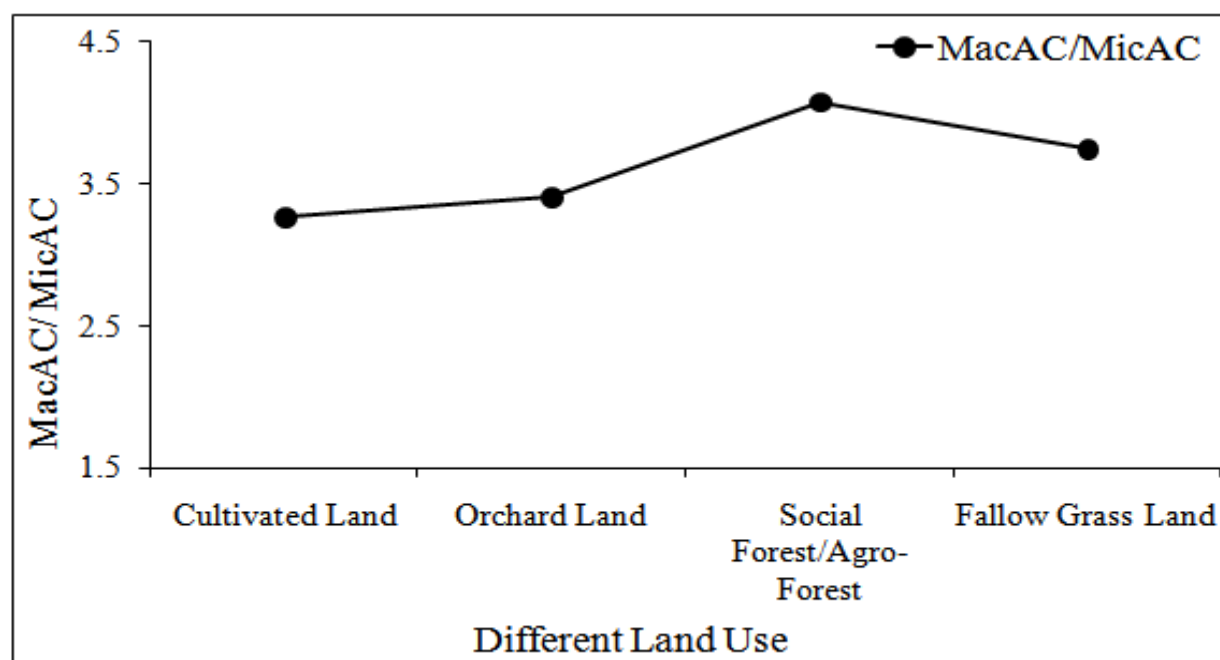


Fig. 6. Proportion of macro-aggregated C (MacAC) and micro- aggregated C (MicAC) under different land use systems

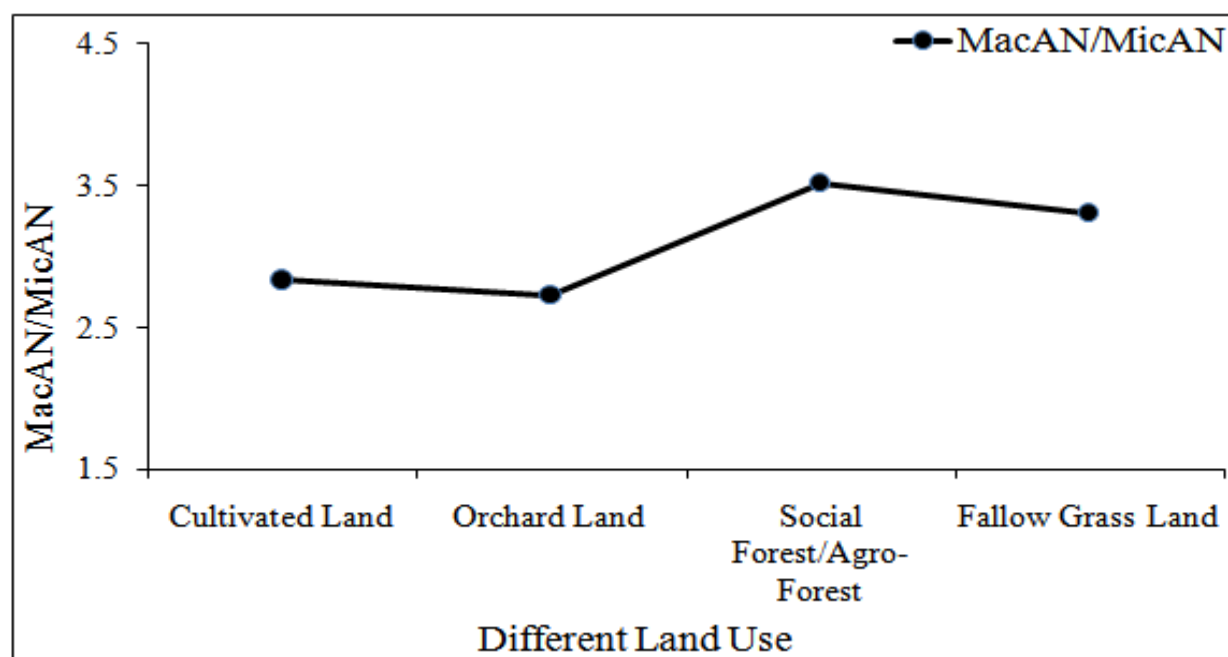


Fig. 7. Proportion of macro-aggregated N (MacAN) and micro- aggregated N (MicAN) under different land use systems

4.3 Enzymatic Activities in Different Soil Aggregate Fractions

Soil enzymes produced by microbes play a crucial role in the process of SOC mineralization by catalyzing the decomposition of different organic substrates, releasing plant nutrients and influencing whether SOC is depleted or sequestered (Fansler *et al.*, 2005; Ahn *et al.*, 2009). In this study, potential activities of dehydrogenase, glucosaminidase, β -Glucosidase, protease, acid phosphatase and alkaline phosphate were measured in three aggregate size fractions (<0.05, 0.25–0.05 and 0.25–2 mm) obtained from soils of four different land use systems (cultivated land, orchard, social forest and fallow grass land). Enzyme activities were within the range of values usually found in soil (Nannipieri *et al.*, 2002).

In case of dehydrogenase, significant effects of different crop rotations were apparent where Jute-Rice-Fallow rotation produced significant higher enzymatic activity in bulk soil (22.69 $\mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$) than other three rotations (Table 9). Conversely, Jute-Maize-Potato rotation produced the least enzymatic activity in bulk soil (5.83 $\mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$). Averaging across all the crop rotations, an interesting trend appeared where cultivated land produced higher enzymatic activity (16.25 $\mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$) than fallow grass land (11.40 $\mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$) and social forest (5.25 $\mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$) (Fig. 8a). Similar trend for bulk soils were found in β -glucosidase (Fig. 8b) and protease (Fig. 8c) where enzymatic activity reduced from cultivated to fallow grass lands. Notably, β -glucosidase activity under the conventional cultivated systems was significantly greater (~2 times) than under the fallow grass land system. This trend can be attributed to the disruption of soil aggregates from conventional to cultivated system and subsequent exposure of organic matter, leading towards enhanced microbial activity along with higher enzymatic activities.

Notably, the enzymatic activities of uncultivated orchard were higher than cultivated land in case of dehydrogenase (21.52 and 16.25 $\mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$ for orchard and cultivated land, respectively) and protease (53.13 and 38.14 $\mu\text{g tyrosine g}^{-1} \text{ soil h}^{-1}$ for orchard and cultivated land, respectively). Conversely, a trend for enzymatic activity was found for acid phosphates, alkaline phosphates and glucosaminidase where bulk soils from fallow grass land showed subtle increase in enzymatic activities than cultivated land.

On average, increasing trend in enzymatic activity from silt- plus clay -size particles (<0.05 mm) to macro-aggregates (0.25-2 mm) were evident across all four land use systems for all enzymes except dehydrogenase which showed elevated enzymatic activity in intermediate micro-aggregates (0.25-0.05 mm) for cultivated land, orchard and fallow grass land (Table 9-14, and Fig. 9a). For instance, the average β -glucosidase activity was significantly greater (~46%) in macro-aggregates (0.25-2 mm) than micro-aggregates (0.25-0.05 mm) for cultivated land (Table 10). Following the same trend, the average protease activity was almost 2-fold higher in macro-aggregates (0.25-2 mm) than silt- plus clay -size particles (<0.05 mm) for cultivated land (Table 11). The higher enzymatic activities in macro-aggregates were expected since disruption of macro-aggregates to micro-aggregates perhaps resulted in concomitant loss of organic matter. Conversely, the increased potential dehydrogenase activity observed in intermediate aggregate size fractions may be associated with the production of enhanced enzyme levels by the active microbial communities that decompose greater amounts of labile organic matter (microbial biomass C). However, the greater relative abundance of soil fungal community in the micro-aggregates needs to be confirmed by phospholipid fatty acid (PLFA) analysis before making any stronger conclusion.

High magnitudes of acid phosphates (Fig. 9d), alkaline phosphates (Fig. 9e) and glucosaminidase (Fig. 9f) activities were found in aggregates under fallow grass land than conventional crop land since long-term tillage disrupts soil aggregates and increases the losses of organic matter, resulting in a faster turnover rate of macro-aggregates. In no-till systems, however, soil organic matter is protected from degradation by the formation and stabilization of micro-aggregates within macro-aggregates, accompanied by a slower turnover rate of macro-aggregates (Six *et al.*, 1999). As a result, increased organic matter in macro-aggregate size fractions also promotes a greater level of microbial activity in comparison with micro-aggregates (Gupta and Germida, 1988; Franzluebbers and Arshad, 1997; Mendes *et al.*, 1999). The crop residues under conservational agricultural practices generally have a high availability to soil microorganisms and consequently can be easily incorporated into macro-aggregate fractions during their decomposition (Stemmer *et al.*, 1998), contributing to the high enzymatic activities. In addition to influencing the composition of microbial communities, pore space associated with different aggregate size fractions could also regulate the movement of substrates and soil enzymes (Sollins *et al.*, 1996).

Table 9: Dehydrogenase activity in bulk soil and within different aggregate size fractions under different cropping systems

Land Uses		Dehydrogenase ($\mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$)			
		Bulk soil	Aggregate size (mm)		
			2.0-0.25	0.25-0.05	<0.05
Cultivated Land	Jute - Rice- Fallow	22.69 ^a	9.44 ^a	10.66 ^a	1.19 ^c
	Jute -Maize-Potato	5.83 ^d	1.44 ^d	2.80 ^b	0.68 ^d
	Fallow- Maize-Wheat	20.53 ^b	5.52 ^c	10.75 ^a	3.39 ^a
	Brinjal-Pumpkin-Cabbage	15.93 ^c	7.23 ^b	4.01 ^b	2.83 ^b
Mean		16.25	5.91	7.06	2.02
LSD($P=0.05$)		0.27	0.71	5.91	0.18
Orchard Land	Mango	30.95 ^b	11.79 ^b	13.93 ^b	4.24 ^a
	Litchi	9.81 ^c	5.57 ^c	3.87 ^c	0.43 ^c
	Guava	6.71 ^d	1.08 ^d	1.15 ^d	4.38 ^a
	Safeda	38.59 ^a	16.82 ^a	19.48 ^a	1.86 ^b
Mean		21.52	8.82	9.61	2.73
LSD($P=0.05$)		1.67	0.44	0.75	0.79
Social Forest/Agro- Forest	Sissoo	5.03 ^b	1.55 ^c	3.01 ^a	0.53 ^c
	Gamar	7.24 ^a	3.62 ^a	1.39 ^a	1.02 ^b
	Teak	4.11 ^c	2.30 ^b	0.73 ^a	0.53 ^c
	Rubber	4.61 ^{bc}	1.04 ^d	1.39 ^a	2.00 ^a
Mean		5.25	2.13	1.63	1.02
LSD($P=0.05$)		0.76	0.37	3.73	0.15
Fallow Grass Land	<i>Cyperus sp.</i>	4.21 ^d	2.18 ^d	1.75 ^d	0.31 ^d
	<i>Polygonum sp.</i>	7.18 ^c	2.59 ^c	2.46 ^c	1.58 ^a
	<i>Cynodon dactylon</i>	12.09 ^b	5.35 ^b	6.27 ^b	0.91 ^c
	<i>Panicum sp.</i>	22.10 ^a	9.52 ^a	10.71 ^a	1.34 ^b
Mean		11.40	4.91	5.30	1.04
LSD($P=0.05$)		0.38	0.19	0.40	0.19

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

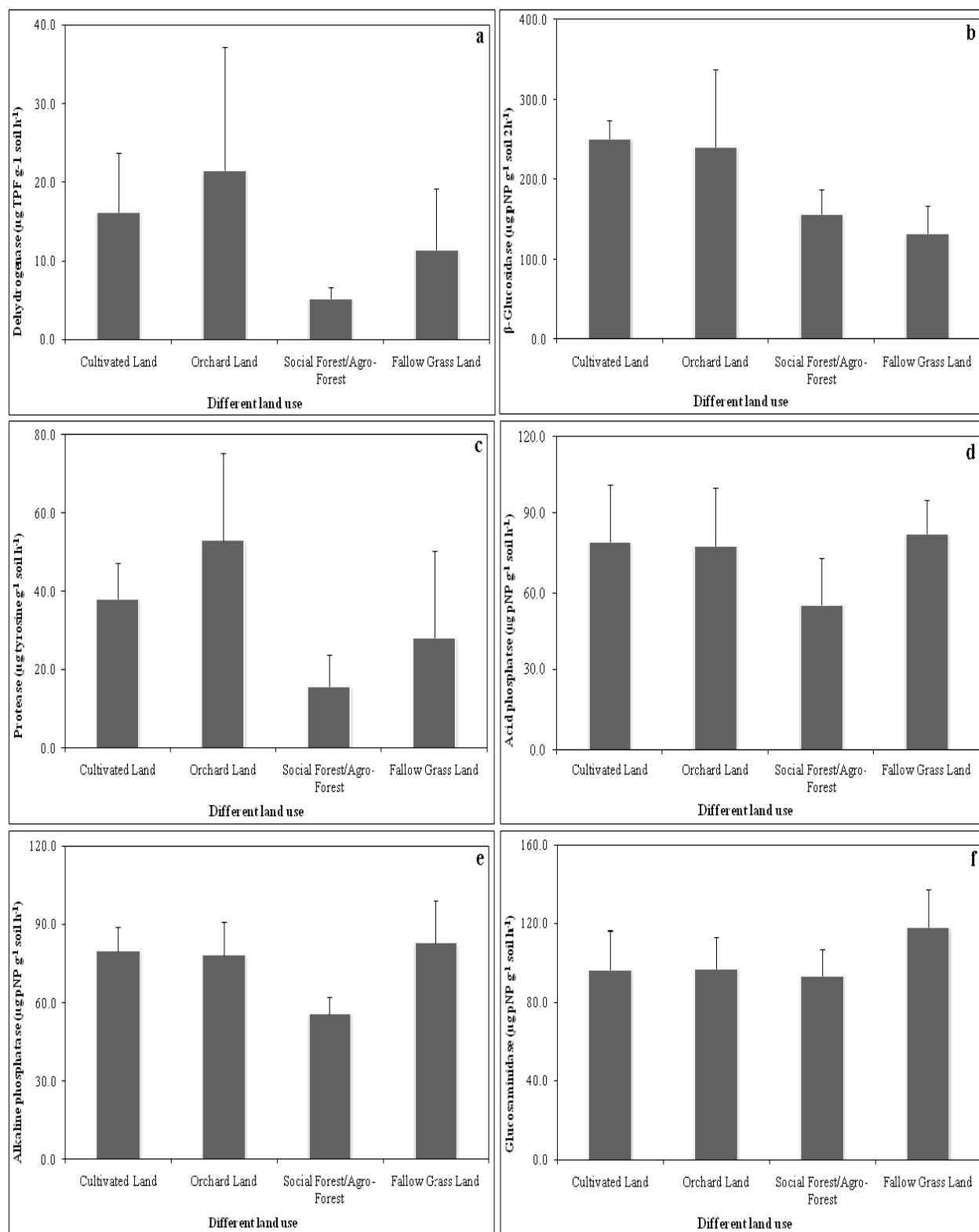


Fig. 8. Plots showing a) Dehydrogenase, b) β -Glucosidase, c) Protease, d) Acid phosphates, e) Alkaline phosphates and f) Glucosaminidase activities in bulk soil under different land use systems. Range bars indicate the standard deviations of the means.

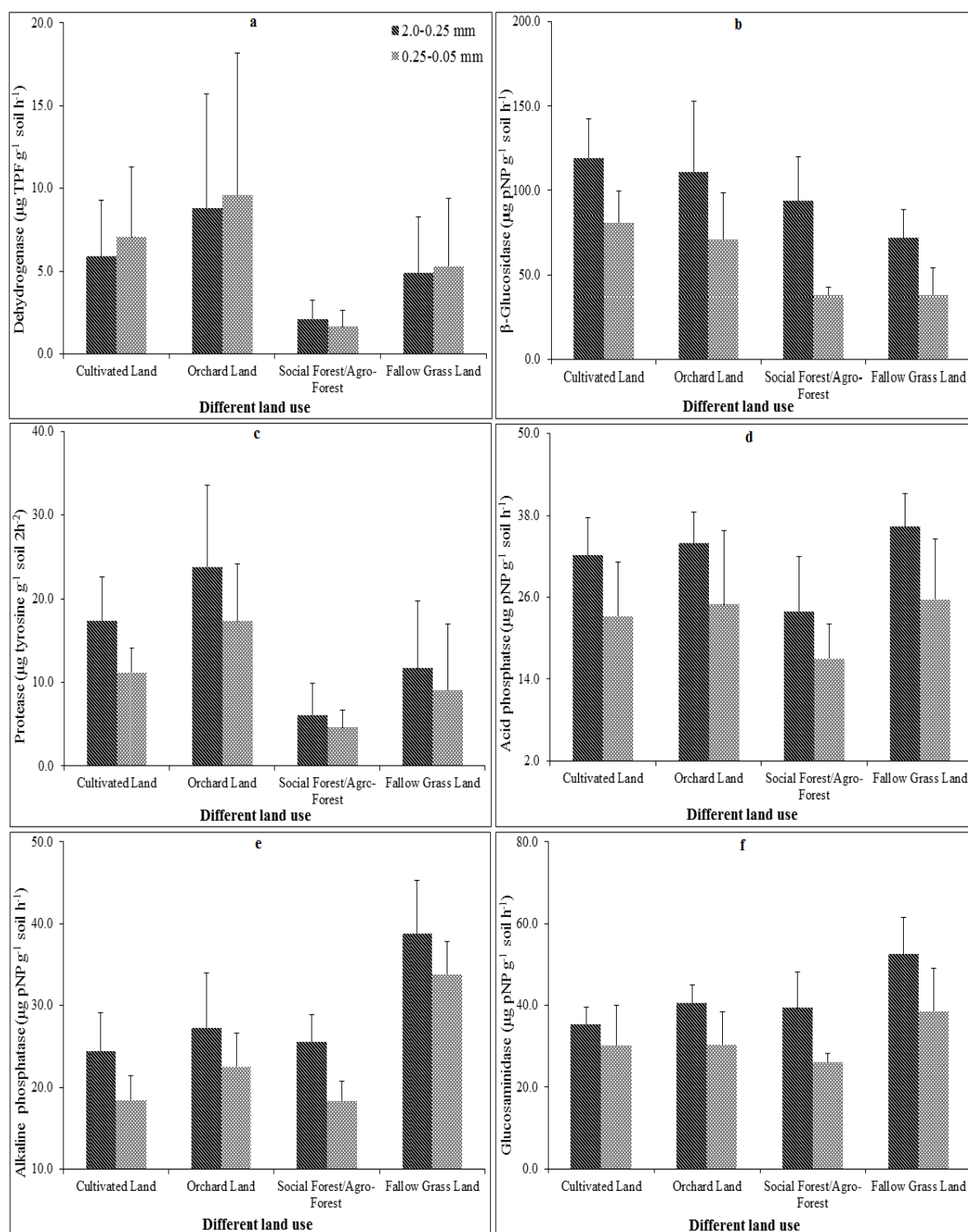


Fig. 9. Plots showing a) Dehydrogenase, b) β -Glucosidase, c) Protease, d) Acid phosphates, e) Alkaline phosphates and f) Glucosaminidase activities in macro and micro- aggregates under different land use systems. Range bars indicate the standard deviations.

Table 10: β -Glucosidase activity in bulk soil and within different aggregate size fractions under different cropping systems

Land Uses		β -Glucosidase ($\mu\text{g pNP g}^{-1} \text{ soil h}^{-1}$)			
		Bulk soil	Aggregate size (mm)		
			2.0-0.25	0.25-0.05	<0.05
Cultivated Land	Jute - Rice- Fallow	265.06 ^a	139.91 ^a	69.15 ^c	41.70 ^b
	Jute -Maize-Potato	220.72 ^c	109.56 ^b	64.23 ^c	33.41 ^c
	Fallow- Maize-Wheat	273.76 ^a	136.75 ^a	88.32 ^b	52.32 ^a
	Brinjal-Pumpkin-Cabbage	243.72 ^b	91.58 ^c	103.60 ^a	42.65 ^b
Mean		250.82	119.45	81.33	42.52
LSD(P=0.05)		19.16	12.45	8.49	7.03
Orchard Land	Mango	307.28 ^b	156.28 ^a	84.18 ^b	65.53 ^b
	Litchi	190.44 ^c	85.47 ^c	60.87 ^c	37.56 ^c
	Guava	130.07 ^d	67.08 ^d	38.98 ^d	17.35 ^d
	Safeda	333.28 ^a	135.98 ^b	101.79 ^a	84.06 ^a
Mean		240.27	111.20	71.46	51.13
LSD(P=0.05)		10.78	10.49	7.51	4.41
Social Forest/Agro- Forest	Sissoo	167.83 ^b	97.90 ^b	32.38 ^b	29.79 ^a
	Gamar	192.40 ^a	129.76 ^a	38.72 ^{ab}	25.12 ^b
	Teak	125.78 ^d	74.07 ^c	40.15 ^a	9.07 ^c
	Rubber	142.94 ^c	73.78 ^c	42.22 ^a	27.48 ^{ab}
Mean		157.24	93.88	38.37	22.87
LSD(P=0.05)		13.11	3.72	3.39	4.43
Fallow Grass Land	<i>Cyperus sp.</i>	152.29 ^b	91.95 ^a	50.51 ^a	10.88 ^c
	<i>Polygonum sp.</i>	93.01 ^d	52.84 ^c	25.12 ^b	16.42 ^{bc}
	<i>Cynodon dactylon</i>	169.00 ^a	72.89 ^b	53.95 ^a	37.62 ^a
	<i>Panicum sp.</i>	116.45 ^c	73.31 ^b	23.34 ^b	19.55 ^b
Mean		132.39	72.75	38.23	21.12
LSD(P=0.05)		4.75	2.28	5.67	8.60

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 11: Protease activity in bulk soil and within different aggregate size fractions under different cropping systems

Land Uses		Protease ($\mu\text{g tyrosine g}^{-1} \text{ soil h}^{-1}$)			
		Bulk soil	Aggregate size (mm)		
			2.0-0.25	0.25-0.05	<0.05
Cultivated Land	Jute - Rice- Fallow	29.18 ^d	14.03 ^b	6.78 ^b	5.05 ^b
	Jute -Maize-Potato	33.79 ^c	14.59 ^b	12.07 ^a	6.45 ^b
	Fallow- Maize-Wheat	50.33 ^a	25.25 ^a	13.19 ^a	10.10 ^a
	Brinjal-Pumpkin-Cabbage	39.25 ^b	15.31 ^b	12.63 ^a	11.05 ^a
Mean		38.14	17.30	11.17	8.16
LSD($P=0.05$)		2.59	2.26	4.95	1.88
Orchard Land	Mango	49.25 ^c	21.89 ^b	17.47 ^c	9.26 ^c
	Litchi	66.24 ^b	26.09 ^b	20.48 ^b	18.24 ^a
	Guava	23.61 ^d	11.78 ^c	7.65 ^d	3.65 ^d
	Safeda	73.43 ^a	35.35 ^a	23.57 ^a	14.32 ^b
Mean		53.13	23.78	17.29	11.37
LSD($P=0.05$)		1.61	4.43	1.25	2.31
Social Forest/Agro- Forest	Sissoo	25.25 ^a	11.31 ^a	6.73 ^a	5.33 ^a
	Gamar	18.46 ^b	6.17 ^b	5.61 ^a	4.72 ^a
	Teak	12.07 ^c	4.54 ^b	3.93 ^b	2.53 ^b
	Rubber	6.47 ^d	2.24 ^b	1.96 ^c	1.69 ^b
Mean		15.56	6.07	4.56	3.57
LSD($P=0.05$)		2.88	3.87	1.26	1.79
Fallow Grass Land	<i>Cyperus sp.</i>	60.72 ^a	22.68 ^a	20.62 ^a	16.72 ^a
	<i>Polygonum sp.</i>	23.76 ^b	11.34 ^b	7.37 ^b	5.05 ^b
	<i>Cynodon dactylon</i>	10.42 ^d	3.65 ^d	3.37 ^c	2.81 ^c
	<i>Panicum sp.</i>	17.34 ^c	8.98 ^c	4.82 ^c	3.09 ^{bc}
Mean		28.06	11.66	9.05	6.92
LSD($P=0.05$)		5.06	2.52	2.11	2.05

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 12: Acid phosphates activity in bulk soil and within different aggregate size fractions under different cropping systems

Land Uses		Acid phosphatase ($\mu\text{g pNP g}^{-1} \text{ soil h}^{-1}$)			
		Bulk soil	Aggregate size (mm)		
			2.0-0.25	0.25-0.05	<0.05
Cultivated Land	Jute - Rice- Fallow	74.19 ^b	30.66 ^c	23.44 ^b	14.55 ^b
	Jute -Maize-Potato	57.19 ^c	25.83 ^d	17.25 ^c	11.01 ^d
	Fallow- Maize-Wheat	109.91 ^a	38.88 ^a	34.41 ^a	22.67 ^a
	Brinjal-Pumpkin-Cabbage	77.80 ^b	33.55 ^b	17.86 ^c	11.93 ^c
Mean		79.77	32.23	23.24	15.04
LSD($P=0.05$)		6.11	0.53	1.14	0.45
Orchard Land	Mango	99.92 ^a	39.53 ^a	31.75 ^b	26.27 ^a
	Litchi	91.52 ^b	35.82 ^b	34.96 ^a	12.40 ^c
	Guava	52.25 ^c	30.78 ^c	10.80 ^d	8.81 ^d
	Safeda	68.83 ^{bc}	29.83 ^d	22.39 ^c	15.76 ^b
Mean		78.13	33.99	24.98	15.81
LSD($P=0.05$)		9.16	0.37	0.11	0.48
Social Forest/Agro- Forest	Sissoo	36.49 ^d	15.28 ^d	12.16 ^d	8.08 ^c
	Gamar	66.60 ^b	27.18 ^b	18.25 ^b	18.98 ^a
	Teak	44.16 ^c	20.14 ^c	14.19 ^c	7.46 ^d
	Rubber	74.85 ^a	33.40 ^a	23.49 ^a	15.50 ^b
Mean		55.53	24.00	17.02	12.51
LSD($P=0.05$)		7.45	0.80	0.59	0.30
Fallow Grass Land	<i>Cyperus sp.</i>	76.02 ^b	35.33 ^b	18.64 ^c	21.21 ^a
	<i>Polygonum sp.</i>	70.10 ^b	30.09 ^c	18.53 ^c	18.91 ^c
	<i>Cynodon dactylon</i>	87.38 ^b	40.34 ^a	29.34 ^b	14.49 ^d
	<i>Panicum sp.</i>	97.72 ^a	40.03 ^a	36.55 ^a	19.54 ^b
Mean		82.81	36.45	25.77	18.54
LSD($P=0.05$)		10.52	0.50	0.47	0.34

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 13: Alkaline phosphates activity in bulk soil and within different aggregate size fractions under different cropping systems

Land Uses		Alkaline phosphatase ($\mu\text{g pNP g}^{-1} \text{ soil h}^{-1}$)			
		Bulk soil	Aggregate size (mm)		
			2.0-0.25	0.25-0.05	<0.05
Cultivated Land	Jute - Rice- Fallow	67.10 ^b	26.15 ^b	20.75 ^a	15.87 ^b
	Jute -Maize-Potato	72.91 ^a	30.30 ^a	20.00 ^a	16.56 ^a
	Fallow- Maize-Wheat	58.12 ^b	21.03 ^c	18.97 ^b	15.19 ^c
	Brinjal-Pumpkin-Cabbage	52.05 ^c	20.44 ^c	14.33 ^c	13.76 ^d
Mean		79.77	32.23	23.24	15.04
LSD($P=0.05$)		1.42	0.58	0.83	0.77
Orchard Land	Mango	68.20 ^c	26.15 ^b	23.09 ^{ab}	14.96 ^c
	Litchi	85.14 ^a	36.92 ^a	27.31 ^a	16.15 ^b
	Guava	54.32 ^d	21.75 ^d	17.35 ^b	14.29 ^d
	Safeda	69.78 ^b	24.43 ^c	22.40 ^{ab}	20.97 ^a
Mean		78.13	33.99	24.98	15.81
LSD($P=0.05$)		1.39	0.55	5.88	0.54
Social Forest/Agro-Forest	Sissoo	52.77 ^d	22.40 ^d	15.22 ^c	11.61 ^d
	Gamar	67.24 ^a	29.24 ^a	21.24 ^a	14.05 ^c
	Teak	65.14 ^b	27.59 ^b	18.24 ^b	17.59 ^a
	Rubber	59.57 ^c	22.89 ^c	18.59 ^b	15.63 ^b
Mean		55.53	24.00	17.02	12.51
LSD($P=0.05$)		0.88	0.48	0.60	0.80
Fallow Grass Land	<i>Cyperus sp.</i>	80.07 ^d	30.03 ^d	28.07 ^c	20.89 ^d
	<i>Polygonum sp.</i>	111.54 ^b	44.42 ^a	35.03 ^b	29.34 ^c
	<i>Cynodon dactylon</i>	115.76 ^a	42.69 ^b	37.38 ^a	30.86 ^b
	<i>Panicum sp.</i>	107.72 ^c	38.28 ^c	34.87 ^b	32.04 ^a
Mean		82.81	36.45	25.77	18.54
LSD($P=0.05$)		7.16	0.55	0.75	0.93

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 14: Glucosaminidase activity in bulk soil and within different aggregate size fractions under different cropping systems

Land Uses		Glucosaminidase ($\mu\text{g pNP g}^{-1} \text{ soil h}^{-1}$)			
		Bulk soil	Aggregate size (mm)		
			2.0-0.25	0.25-0.05	<0.05
Cultivated Land	Jute - Rice- Fallow	119.73 ^a	52.92 ^a	42.95 ^a	21.33 ^a
	Jute -Maize-Potato	105.37 ^b	40.30 ^b	33.05 ^b	17.00 ^a
	Fallow- Maize-Wheat	85.26 ^c	33.24 ^c	23.35 ^c	18.69 ^a
	Brinjal-Pumpkin-Cabbage	75.71 ^d	32.86 ^c	21.75 ^d	19.02 ^a
Mean		96.52	39.83	30.28	19.01
LSD($P=0.05$)		0.52	0.63	0.85	13.12
Orchard Land	Mango	105.13 ^b	45.62 ^a	34.32 ^b	23.31 ^b
	Litchi	115.30 ^a	42.75 ^{ab}	38.65 ^a	32.20 ^a
	Guava	78.25 ^d	36.44 ^b	20.43 ^d	19.44 ^d
	Safeda	89.45 ^c	37.38 ^b	28.63 ^c	20.20 ^c
Mean		97.03	40.55	30.51	23.79
LSD($P=0.05$)		0.44	8.02	0.54	0.37
Social Forest/Agro-Forest	Sissoo	83.90 ^c	33.99 ^b	27.35 ^a	20.53 ^b
	Gamar	105.86 ^a	48.16 ^a	26.08 ^a	23.35 ^a
	Teak	104.19 ^b	45.24 ^a	27.92 ^a	23.68 ^a
	Rubber	79.80 ^d	30.23 ^b	23.31 ^a	19.73 ^b
Mean		93.44	39.41	26.17	21.82
LSD($P=0.05$)		1.08	8.08	8.15	1.06
Fallow Grass Land	<i>Cyperus sp.</i>	94.96 ^d	44.26 ^c	24.72 ^d	17.61 ^d
	<i>Polygonum sp.</i>	141.76 ^a	65.25 ^a	47.41 ^a	26.79 ^a
	<i>Cynodon dactylon</i>	114.83 ^c	50.56 ^b	36.30 ^c	25.71 ^b
	<i>Panicum sp.</i>	119.44 ^b	50.09 ^b	46.05 ^b	20.90 ^c
Mean		117.75	52.54	38.62	22.75
LSD($P=0.05$)		1.40	0.68	0.74	0.52

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

4.4 Microbial Respiration on Particulate Organic Matter (POM) of Soils as Influenced by Land Use Pattern

Besides the carbon input, the SOC decomposition rate is another important factor influencing SOC balance. Microbial respiration directly indicates the SOC decomposition rate. As noted before elsewhere (Qin *et al.*, 2010), microbial respiration at 0 -day from fine and coarse POM mixed sand incubated at 28°C and 35°C were higher in conservational agricultural practices like social forest and fallow grass land than cultivated land, followed by a steep slope of decline for CO₂ release (Figs 10 and 11). Given that conservational agricultural managements significantly promotes soil respiration and enzyme activities, this trend can be justified by the fact that higher enzymatic activities for acid phosphates, alkaline phosphates and glucosaminidase were found in bulk soils from fallow grass land than cultivated land (Fig. 8). Allison and Jastrow (2006) concluded that enzymes involved in soil C cycling, such as β -glucosidase and glucosaminidase can provide the information on SOC decomposition potential since β -glucosidase catalyzes the final step of cellulose degradation and glucosaminidase hydrolyzes the glucosamine (Reboreda and Cacador, 2008). In this study, while the activity of β -glucosidase did not produce expected trend, glucosaminidase tended to decrease from fallow grass land to cultivated land, corroborating the higher decomposition potential and CO₂ release from conservational agricultural practices. On the contrary, the trend of CO₂ release between 7-91 days were non-significantly affected by management modifications and reached a plateau (Figs. 10 and 11). Only CO₂ release from coarse POM at 28°C showed higher rate of respiration in social forest and fallow grass land than cultivated land from 0-7 days which further enhanced to surpass CO₂ release of cultivated land from 7-14 days before reaching the plateau at 21 days (Fig. 11).

A critical analysis is required for the higher rate of CO₂ flux from POM of fallow grassland than that of social forest (Figs. 10 and 11). Much of contemporary understanding regarding the cycling of plant derived C-compounds has been developed from studies that examine leaf litter decomposition (Harmon *et al.*, 2009). Nevertheless, leaf litter is not the only plant-derived input of C to terrestrial systems. Of growing interest is the role that rhizodeposits play in ecosystem level C-processes (Hogberg *et al.*, 2008; Phillips *et al.*, 2008). Rhizodeposits are in part composed of low molecular weight organic C compounds, such as sugars (mono- and disaccharides), amino acids, and organic acids (Dakora and Phillips, 2002). These compounds

represent a highly labile, rapidly cycling, form of C which is immediately available to the soil microbial community for growth, maintenance and respiration and so their impact on C dynamics may be large (Hogberg *et al.*, 2008). Research suggests that communities which derive the bulk of their C from low molecular weight organic C compounds may have greater mineralization rates than communities deriving their C from another source (Saggar *et al.*, 2000; Stevenson *et al.*, 2004). Based on this possibility, the mineralization rates of low molecular weight organic C compounds may be dependent on the plant community and factors such as land-use/land-cover, season (van Hees *et al.*, 2005). In this study, in fallow grass land where the dominant plant cover is grass, inputs of low molecular weight organic C compounds were perhaps more important C source than in social forests, explaining higher initial CO₂ flux in the latter (Stevenson *et al.*, 2004). However, there is a need for complete biochemical characterization of POM extracted from different land use systems before making any strong conclusion.

Significant effects of different crop rotations were apparent for cultivated land during the 0-91 days incubation period. While incubated at 35°C, fine POM extracted from Jute-Maize-Potato and Fallow-Maize-Wheat rotations released significantly higher CO₂ than Jute-Rice-Fallow and Brinjal-Pumpkin-Cabbage rotations initially (Table 15). One possible explanation for this observation is differential enzymatic activity arising from qualitative variation of organic matter in different land use systems, leading to variable CO₂ release. However, this trend was reversed at the end of incubation period (91 days). Conversely, while incubated at 28°C, CO₂ release from fine POM extracted from different crop rotations under cultivated land system did not show significant variation initially (Table 16). However, the CO₂ release trend at the end of incubation period matched with that observed during 35°C incubation. Overall, a steep decline in CO₂ release from 0-7 days followed by reaching a plateau from 7-91 days was observed for both fine and coarse POM extracted from all land use systems (Tables 15, 16, 17 and 18). No significant effect of plant cover on CO₂ release was observed in case of POM extracted from different plantations under orchard and social forest (Tables 15 and 17).

Table 15: Soil respiration (at 35°C) from the mixture of fine particulate organic matter and sand under different cropping systems

		Soil respiration rate ($\mu\text{g CO}_2\text{-C release 1g soil}^{-1} \text{ day}^{-1}$)													
Land Uses		0-2	2-7	7-14	14-21	21-28	28-35	35-42	42-49	49-56	56-63	63-70	70-77	77-84	84-91
Cultivated Land	Jute - Rice- Fallow	7.97 ^b	1.14 ^a	0.77 ^b	0.71 ^b	0.79 ^a	0.66	0.69 ^b	0.70 ^a	0.60 ^a	0.74 ^a	0.59 ^b	0.57 ^{ab}	0.55 ^b	0.69 ^a
	Jute -Maize-Potato	8.44 ^a	0.98 ^{ab}	0.82 ^b	0.62 ^{bc}	0.81 ^a	0.64 ^a	0.83 ^a	0.55 ^b	0.49 ^c	0.55 ^b	0.57 ^b	0.74 ^a	0.67 ^a	0.57 ^b
	Fallow- Maize-Wheat	8.73 ^a	0.92 ^b	0.92 ^a	0.86 ^a	0.78 ^a	0.58 ^a	0.66 ^c	0.55 ^b	0.56 ^{ab}	0.53 ^b	0.52 ^b	0.56 ^b	0.61 ^b	0.54 ^b
	Brinjal-Pumpkin-Cabbage	7.97 ^b	0.89 ^b	0.72 ^b	0.60 ^c	0.65 ^a	0.58 ^a	0.62 ^c	0.58 ^b	0.55 ^b	0.68 ^a	0.69 ^a	0.69 ^a	0.71 ^a	0.66 ^a
Mean		8.28	0.98	0.81	0.70	0.76	0.62	0.70	0.60	0.55	0.63	0.59	0.64	0.64	0.62
LSD($P=0.05$)		0.26	0.17	0.05	0.11	0.05	0.05	0.04	0.09	0.09	0.03	0.05	0.13	0.05	0.08
Orchard Land	Mango	15.10 ^a	1.47 ^c	1.58 ^a	1.31 ^a	1.11 ^a	1.45 ^a	1.06 ^b	1.09 ^{ab}	1.02 ^a	1.15 ^a	0.97 ^a	1.01 ^a	1.03 ^{ab}	1.03 ^a
	Litchi	13.93 ^a	1.61 ^b	1.54 ^{ab}	1.12 ^a	1.22 ^a	1.34 ^a	1.28 ^a	1.12 ^a	1.02 ^a	0.93 ^a	0.92 ^a	1.19 ^a	1.07 ^a	1.11 ^a
	Guava	13.12 ^a	1.92 ^a	1.36 ^b	0.99 ^a	1.20 ^a	1.09 ^a	1.16 ^b	1.01 ^b	0.99 ^a	0.87 ^a	0.92 ^a	0.95 ^a	0.96 ^b	1.33 ^a
	Safeda	13.84 ^a	1.58 ^{bc}	1.57 ^{ab}	0.97 ^a	1.45 ^a	1.48 ^a	1.19 ^b	1.07 ^b	0.88 ^b	0.84 ^a	0.95 ^a	1.07 ^a	1.01 ^{ab}	0.98 ^a
Mean		14.00	1.65	1.51	1.10	1.25	1.34	1.17	1.07	0.98	0.95	0.94	1.06	1.02	1.11
LSD($P=0.05$)		0.96	0.14	0.28	0.24	0.21	0.45	0.10	0.12	0.04	0.19	0.14	0.11	0.09	0.27
Social Forest/Agro-Forest	Sissoo	13.35 ^b	2.01 ^a	1.31 ^a	1.08 ^{ab}	1.25 ^b	1.27 ^a	1.29 ^{bc}	1.01 ^c	1.04 ^b	0.92 ^b	1.05 ^b	1.11 ^a	1.04 ^b	1.05 ^c
	Gamar	13.71 ^b	1.47 ^b	1.21 ^a	1.04 ^b	1.32 ^{ab}	1.21 ^a	1.31 ^b	1.13 ^{bc}	1.01 ^b	0.96 ^b	1.10 ^{ab}	1.20 ^a	1.16 ^{ab}	1.03 ^c
	Teak	13.71 ^b	1.61 ^{ab}	1.38 ^a	1.19 ^a	1.52 ^a	1.30 ^a	1.55 ^a	1.41 ^a	1.34 ^a	1.42 ^a	1.22 ^a	1.29 ^a	1.28 ^a	1.24 ^a
	Rubber	14.34 ^a	1.45 ^b	1.18 ^a	1.04 ^b	1.13 ^b	1.22 ^a	1.08 ^c	1.31 ^{ab}	1.05 ^b	0.95 ^b	1.02 ^b	1.01 ^a	1.00 ^b	1.12 ^b
Mean		13.78	1.64	1.27	1.09	1.31	1.25	1.31	1.22	1.11	1.06	1.10	1.15	1.12	1.11
LSD($P=0.05$)		0.045	0.28	0.15	0.09	0.11	0.22	0.15	0.19	0.23	0.17	0.18	0.06	0.11	0.06
Fallow Grass Land	<i>Cyperus sp.</i>	10.25 ^a	1.16 ^b	0.93 ^b	0.76 ^b	0.86 ^{bc}	0.76 ^a	0.80 ^b	0.71 ^c	0.74 ^c	0.66 ^c	0.76 ^c	0.89 ^{ab}	0.73 ^b	0.79 ^a
	<i>Polygonum sp.</i>	11.36 ^a	1.48 ^a	1.04 ^b	0.94 ^a	1.01 ^{ab}	0.87 ^a	1.01 ^a	0.91 ^b	0.88 ^b	0.76 ^{bc}	0.88 ^{bc}	1.10 ^a	1.00 ^a	0.95 ^a
	<i>Cynodon dactylon</i>	11.26 ^a	1.22 ^b	0.89 ^c	0.75 ^b	0.84 ^c	0.73 ^a	0.79 ^b	0.68 ^c	0.65 ^d	0.85 ^{ab}	0.95 ^{ab}	0.76 ^b	0.73 ^b	0.78 ^a
	<i>Panicum sp.</i>	11.09 ^a	1.52 ^a	1.19 ^a	0.89 ^a	1.24 ^a	0.93 ^a	1.08 ^a	1.00 ^a	0.99 ^a	0.95 ^a	1.04 ^a	1.00 ^{ab}	1.00 ^a	0.83 ^a
Mean		10.99	1.35	1.01	0.84	0.99	0.82	0.92	0.83	0.82	0.81	0.91	0.94	0.87	0.84
LSD($P=0.05$)		1.15	0.08	0.06	0.07	0.08	0.03	0.10	0.06	0.05	0.10	0.08	0.09	0.12	0.05

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 16: Soil respiration (at 28°C) from the mixture of fine particulate organic matter and sand under different cropping systems

		Soil respiration rate ($\mu\text{g CO}_2\text{-C release 1g soil}^{-1} \text{ day}^{-1}$)													
Land Uses		0-2	2-7	7-14	14-21	21-28	28-35	35-42	42-49	49-56	56-63	63-70	70-77	77-84	84-91
Cultivated Land	Jute - Rice- Fallow	2.92 ^b	0.95 ^a	0.70 ^a	0.70 ^a	0.62 ^{ab}	0.59 ^a	0.54 ^b	0.60 ^c	0.54 ^b	0.59 ^b	0.72 ^a	0.53 ^c	0.49 ^b	0.69 ^a
	Jute -Maize-Potato	3.07 ^a	0.97 ^a	0.64 ^b	0.61 ^b	0.67 ^a	0.68 ^a	0.58 ^b	0.65 ^b	0.57 ^{ab}	0.65 ^a	0.71 ^b	0.60 ^b	0.52 ^b	0.55 ^b
	Fallow- Maize-Wheat	2.93 ^b	0.97 ^a	0.69 ^a	0.66 ^{ab}	0.60 ^b	0.61 ^a	0.71 ^a	0.75 ^a	0.62 ^a	0.68 ^a	0.63 ^b	0.64 ^b	0.66 ^a	0.68 ^a
	Brinjal-Pumpkin-Cabbage	2.46 ^c	0.91 ^a	0.59 ^c	0.57 ^c	0.54 ^c	0.60 ^a	0.53 ^b	0.65 ^b	0.53 ^b	0.57 ^c	0.54 ^c	0.93 ^a	0.49 ^b	0.50 ^b
Mean		2.85	0.95	0.66	0.64	0.61	0.62	0.59	0.66	0.57	0.62	0.65	0.68	0.54	0.61
LSD($P=0.05$)		0.11	0.04	0.04	0.07	0.03	0.06	0.02	0.04	0.03	0.02	0.07	0.04	0.06	0.07
Orchard Land	Mango	4.45 ^b	1.49 ^b	1.21 ^b	1.30 ^a	1.01 ^b	1.05 ^a	0.88 ^b	1.08 ^b	1.12 ^a	1.26 ^a	1.17 ^a	1.02 ^{ab}	0.92 ^a	1.05 ^a
	Litchi	4.36 ^{bc}	1.60 ^a	1.30 ^a	1.09 ^a	1.00 ^b	1.01 ^{ab}	0.99 ^{ab}	1.60 ^a	0.89 ^b	0.93 ^d	1.15 ^a	1.14 ^a	0.81 ^a	1.01 ^a
	Guava	4.27 ^c	1.43 ^b	1.12 ^c	1.13 ^a	1.02 ^b	0.96 ^b	1.02 ^{ab}	1.09 ^b	0.86 ^b	1.11 ^c	0.93 ^b	0.91 ^b	0.85 ^a	0.85 ^b
	Safeda	5.14 ^a	1.46 ^b	1.37 ^a	1.09 ^a	1.24 ^a	1.03 ^a	1.11 ^a	1.16 ^b	1.00 ^{ab}	1.15 ^b	1.08 ^a	1.19 ^a	0.84 ^a	1.03 ^a
Mean		4.56	1.50	1.25	1.15	1.07	1.01	1.00	1.23	0.97	1.11	1.08	1.07	0.86	0.99
LSD($P=0.05$)		0.17	0.08	0.09	0.19	0.14	0.05	0.25	0.05	0.17	0.05	0.13	0.19	0.20	0.17
Social Forest/Agro-Forest	Sissoo	5.48 ^a	1.43 ^a	1.09 ^{ab}	1.07 ^a	1.19 ^a	1.28 ^a	0.92 ^a	1.13 ^b	0.99 ^{ab}	1.29 ^a	1.17 ^a	1.25 ^a	0.89 ^b	0.99 ^a
	Gamar	4.30 ^a	1.43 ^a	1.03 ^b	1.01 ^a	0.94 ^b	1.02 ^b	0.89 ^a	1.01 ^c	0.87 ^b	0.96 ^b	0.93 ^c	1.11 ^b	0.83 ^b	0.85 ^a
	Teak	4.18 ^a	1.52 ^a	1.19 ^a	0.87 ^b	0.91 ^b	0.94 ^b	0.91 ^a	1.03 ^c	1.00 ^{ab}	1.03 ^b	1.07 ^b	1.08 ^b	0.82 ^b	0.99 ^a
	Rubber	4.33 ^a	1.53 ^a	1.18 ^a	1.00 ^a	1.09 ^a	1.00 ^b	0.95 ^a	1.22 ^a	1.06 ^a	1.12 ^{ab}	1.10 ^b	1.06 ^b	1.04 ^a	0.98 ^a
Mean		4.57	1.48	1.12	0.99	1.03	1.06	0.92	1.10	0.98	1.10	1.07	1.13	0.90	0.95
LSD($P=0.05$)		2.09	0.10	0.04	0.12	0.10	0.07	0.10	0.05	0.03	0.22	0.04	0.05	0.05	0.27
Fallow Grass Land	<i>Cyperus sp.</i>	3.46 ^{ab}	1.17 ^a	0.78 ^a	0.72 ^d	0.91 ^a	0.72 ^b	0.76 ^a	0.85 ^b	0.93 ^a	0.95 ^b	0.89 ^b	0.78 ^a	0.67 ^b	0.63 ^c
	<i>Polygonum sp.</i>	4.55 ^a	1.67 ^a	1.06 ^a	0.92 ^b	0.85 ^a	0.93 ^a	0.80 ^a	0.84 ^b	0.99 ^a	1.25 ^a	1.17 ^a	0.76 ^a	0.65 ^b	0.95 ^a
	<i>Cynodon dactylon</i>	3.09 ^b	1.15 ^a	1.04 ^a	0.81 ^c	0.97 ^a	0.71 ^b	0.74 ^a	0.81 ^c	0.77 ^b	0.72 ^d	0.76 ^c	0.74 ^a	0.72 ^b	0.78 ^b
	<i>Panicum sp.</i>	4.44 ^a	1.42 ^a	0.90 ^a	1.02 ^a	0.93 ^a	0.88 ^a	0.82 ^a	0.95 ^a	0.74 ^b	0.82 ^c	0.96 ^b	0.88 ^a	0.84 ^a	1.00 ^a
Mean		3.89	1.35	0.95	0.87	0.92	0.81	0.78	0.86	0.86	0.94	0.95	0.79	0.72	0.84
LSD($P=0.05$)		1.17	0.62	0.51	0.05	0.14	0.09	0.10	0.03	0.04	0.05	0.11	0.15	0.04	0.12

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 17: Soil respiration (at 35°C) from the mixture of coarse particulate organic matter and sand under different cropping systems

		Soil respiration rate ($\mu\text{g CO}_2\text{-C release 1g soil}^{-1} \text{ day}^{-1}$)													
Land Uses		0-2	2-7	7-14	14-21	21-28	28-35	35-42	42-49	49-56	56-63	63-70	70-77	77-84	84-91
Cultivated Land	Jute - Rice- Fallow	22.13 ^b	2.59 ^c	2.45 ^{bc}	1.77 ^b	1.60 ^b	2.16 ^a	2.41 ^a	1.76 ^a	2.00 ^b	2.35 ^a	1.84 ^a	1.68 ^a	1.66 ^a	1.80 ^a
	Jute -Maize-Potato	24.98 ^a	3.39 ^b	2.75 ^{ab}	1.81 ^{ab}	2.04 ^a	2.05 ^a	2.63 ^a	1.58 ^a	2.33 ^a	1.98 ^{ab}	1.90 ^a	1.54 ^a	1.88 ^a	1.96 ^a
	Fallow- Maize-Wheat	28.04 ^a	3.33 ^b	2.85 ^a	1.88 ^a	2.21 ^a	2.11 ^a	2.27 ^a	2.07 ^a	2.09 ^b	2.11 ^{ab}	1.98 ^a	1.83 ^a	2.02 ^a	1.76 ^a
	Brinjal-Pumpkin-Cabbage	22.13 ^b	4.07 ^a	2.28 ^c	1.94 ^a	1.52 ^b	1.61 ^b	2.08 ^a	1.46 ^a	1.58 ^c	1.64 ^c	2.09 ^a	2.11 ^a	1.92 ^a	1.78 ^a
Mean		24.32	3.35	2.58	1.85	1.84	1.98	2.35	1.72	2.00	2.02	1.95	1.79	1.87	1.83
LSD(P=0.05)		2.47	0.41	0.24	0.12	0.14	0.20	0.55	0.41	0.12	0.56	0.46	0.46	0.26	0.91
Orchard Land	Mango	21.99 ^a	2.50 ^b	1.79 ^c	1.63 ^b	1.54 ^b	1.50 ^a	1.72 ^b	1.40 ^c	1.39 ^a	1.52 ^a	1.56 ^a	1.36 ^a	1.70 ^a	1.37 ^a
	Litchi	27.26 ^a	2.39 ^b	2.22 ^a	2.71 ^a	1.47 ^b	1.46 ^a	2.37 ^a	4.26 ^a	1.58 ^a	1.56 ^a	1.68 ^a	1.40 ^a	1.68 ^a	1.35 ^a
	Guava	22.35 ^a	2.56 ^{ab}	2.34 ^a	1.57 ^b	2.15 ^a	1.44 ^a	1.66 ^b	1.54 ^{bc}	1.50 ^a	1.43 ^a	1.60 ^a	1.54 ^a	1.51 ^b	1.29 ^a
	Safeda	22.13 ^a	2.87 ^a	2.04 ^{ab}	1.67 ^b	1.64 ^b	1.65 ^a	1.80 ^b	1.68 ^b	1.43 ^a	1.54 ^a	1.58 ^a	1.50 ^a	1.82 ^a	1.51 ^a
Mean		23.43	2.58	2.10	1.90	1.70	1.51	1.89	2.22	1.48	1.51	1.61	1.45	1.68	1.38
LSD(P=0.05)		2.67	0.25	0.21	0.09	0.15	0.22	0.13	0.09	0.09	0.09	0.17	0.10	0.13	0.18
Social Forest/Agro-Forest	Sissoo	21.85 ^c	3.64 ^a	1.79 ^a	0.88 ^a	1.54 ^a	1.50 ^a	1.66 ^b	1.40 ^b	1.48 ^{ab}	1.39 ^a	1.66 ^a	1.48 ^a	1.80 ^{ab}	1.37 ^a
	Gamar	22.42 ^b	3.73 ^a	1.31 ^a	1.57 ^a	1.60 ^a	1.71 ^a	1.74 ^b	1.52 ^b	0.97 ^b	1.54 ^a	1.66 ^a	1.54 ^a	1.74 ^{ab}	1.33 ^a
	Teak	22.92 ^a	2.76 ^b	2.14 ^a	1.59 ^a	1.72 ^a	1.85 ^a	2.00 ^a	1.90 ^a	1.68 ^{ab}	1.58 ^a	1.78 ^a	1.68 ^a	2.12 ^a	1.71 ^a
	Rubber	21.42 ^d	2.30 ^b	1.75 ^a	1.57 ^a	1.47 ^a	1.44 ^a	1.76 ^b	1.31 ^b	1.64 ^a	1.54 ^a	1.58 ^a	1.34 ^a	1.51 ^b	1.37 ^a
Mean		22.15	3.11	1.75	1.40	1.58	1.63	1.79	1.53	1.44	1.51	1.67	1.51	1.79	1.45
LSD(P=0.05)		0.39	0.80	0.56	0.44	0.23	0.24	0.21	0.07	0.33	0.15	0.23	0.24	0.53	0.19
Fallow Grass Land	<i>Cyperus sp.</i>	22.49 ^{ab}	2.76 ^b	1.94 ^a	1.71 ^a	1.72 ^a	1.67 ^b	2.02 ^{ab}	1.68 ^a	1.66 ^a	1.64 ^a	1.84 ^{ab}	1.68 ^a	1.72 ^b	1.29 ^a
	<i>Polygonum sp.</i>	23.41 ^a	2.45 ^{bc}	1.84 ^{ab}	1.57 ^a	1.66 ^a	2.22 ^a	1.96 ^b	1.54 ^a	1.64 ^a	1.43 ^a	1.96 ^a	1.77 ^a	2.02 ^a	1.51 ^a
	<i>Cynodon dactylon</i>	21.71 ^b	2.30 ^c	1.73 ^b	1.51 ^a	1.54 ^a	1.73 ^b	1.80 ^b	1.58 ^a	1.58 ^a	1.48 ^a	1.64 ^b	1.44 ^a	1.37 ^c	1.37 ^a
	<i>Panicum sp.</i>	22.49 ^{ab}	3.10 ^a	2.04 ^a	1.51 ^a	1.74 ^a	1.71 ^b	2.33 ^a	1.70 ^a	1.86 ^a	1.70 ^a	1.88 ^{ab}	1.70 ^a	1.84 ^{ab}	1.59 ^a
Mean		22.53	2.65	1.89	1.58	1.67	1.83	2.03	1.63	1.69	1.56	1.83	1.65	1.74	1.44
LSD(P=0.05)		1.41	0.27	0.10	0.26	0.10	0.11	0.18	0.16	0.21	0.05	0.19	0.43	0.27	0.17

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 18: Soil respiration (at 28°C) from the mixture of coarse particulate organic matter and sand under different cropping systems

		Soil respiration rate ($\mu\text{g CO}_2\text{-C release 1g soil}^{-1} \text{ day}^{-1}$)													
Land Uses		0-2	2-7	7-14	14-21	21-28	28-35	35-42	42-49	49-56	56-63	63-70	70-77	77-84	84-91
Cultivated Land	Jute - Rice- Fallow	8.25 ^c	2.35 ^b	2.25 ^b	1.52 ^d	2.89 ^a	2.12 ^c	2.16 ^a	1.55 ^c	1.59 ^b	1.95 ^c	1.82 ^c	1.67 ^d	1.54 ^b	1.51 ^c
	Jute -Maize-Potato	8.46 ^b	6.30 ^a	3.53 ^a	2.45 ^a	2.75 ^a	2.53 ^b	2.51 ^a	2.34 ^a	1.99 ^a	2.23 ^a	2.10 ^b	2.41 ^a	2.15 ^a	1.86 ^b
	Fallow- Maize-Wheat	9.18 ^a	5.88 ^a	3.98 ^a	2.05 ^b	2.28 ^a	2.08 ^c	2.22 ^a	1.91 ^b	1.65 ^b	2.23 ^{ab}	2.39 ^a	2.16 ^b	1.94 ^{ab}	2.33 ^a
	Brinjal-Pumpkin-Cabbage	7.89 ^c	5.48 ^a	2.82 ^b	1.94 ^c	2.38 ^a	3.24 ^a	2.35 ^a	1.99 ^b	1.69 ^b	2.13 ^b	2.18 ^b	1.88 ^c	1.94 ^{ab}	1.59 ^{bc}
Mean		8.45	5.00	3.15	1.99	2.58	2.49	2.31	1.95	1.73	2.14	2.12	2.03	1.89	1.82
LSD($P=0.05$)		0.37	1.36	0.57	0.25	0.69	0.17	0.43	0.11	0.13	0.11	0.07	0.13	0.30	0.08
Orchard Land	Mango	7.54 ^{bc}	5.28 ^b	4.18 ^a	1.62 ^b	1.87 ^b	2.18 ^b	1.90 ^a	1.83 ^b	1.59 ^a	2.01 ^a	2.08 ^a	1.90 ^b	1.84 ^a	1.94 ^a
	Litchi	7.68 ^b	4.97 ^d	3.98 ^a	1.42 ^c	2.28 ^a	2.49 ^a	1.63 ^b	2.58 ^a	1.48 ^{ab}	2.03 ^a	1.57 ^b	1.57 ^d	1.42 ^a	1.61 ^b
	Guava	7.40 ^c	5.19 ^c	3.88 ^a	2.57 ^a	1.67 ^b	2.43 ^a	1.78 ^a	1.73 ^{bc}	1.44 ^b	1.99 ^a	1.47 ^b	1.71 ^c	1.72 ^a	1.74 ^b
	Safeda	8.53 ^a	5.88 ^a	4.10 ^a	1.60 ^b	2.04 ^a	1.92 ^c	1.69 ^b	1.71 ^c	1.48 ^b	2.03 ^a	1.53 ^b	2.08 ^a	1.66 ^a	1.57 ^b
Mean		7.79	5.33	4.04	1.80	1.97	2.26	1.75	1.96	1.50	2.02	1.66	1.82	1.66	1.72
LSD($P=0.05$)		0.28	0.07	0.21	0.13	0.12	0.06	0.09	0.69	0.03	0.07	0.07	0.07	0.34	0.09
Social Forest/Agro-Forest	Sissoo	8.11 ^a	5.19 ^a	4.18 ^a	1.92 ^a	2.36 ^a	1.84 ^a	1.74 ^a	1.79 ^b	1.48 ^a	1.97 ^a	1.68 ^a	1.98 ^a	2.01 ^a	1.92 ^a
	Gamar	7.40 ^{ab}	2.57 ^b	3.66 ^a	1.37 ^b	1.67 ^b	1.80 ^{ab}	1.57 ^a	1.69 ^b	1.46 ^a	1.91 ^a	2.59 ^a	1.47 ^d	1.74 ^a	1.61 ^b
	Teak	6.83 ^b	2.46 ^c	3.96 ^a	1.33 ^b	1.63 ^b	1.63 ^b	1.53 ^a	1.73 ^b	1.44 ^a	1.85 ^a	2.02 ^a	1.59 ^c	1.54 ^a	1.35 ^b
	Rubber	7.04 ^b	2.35 ^c	3.68 ^a	1.33 ^b	1.67 ^b	1.59 ^b	1.63 ^a	2.38 ^a	1.42 ^a	1.83 ^a	1.80 ^a	1.78 ^b	1.52 ^a	1.41 ^b
Mean		7.35	3.14	3.87	1.49	1.83	1.72	1.62	1.90	1.45	1.89	2.02	1.71	1.70	1.57
LSD($P=0.05$)		0.57	0.05	0.21	0.16	0.45	0.09	0.18	0.07	0.07	0.03	1.11	0.06	0.19	0.14
Fallow Grass Land	<i>Cyperus sp.</i>	7.25 ^c	2.32 ^c	3.66 ^b	1.82 ^b	2.06 ^c	2.08 ^a	2.31 ^a	1.93 ^{bc}	1.67 ^b	2.03 ^a	1.55 ^{bc}	1.82 ^b	1.92 ^{ab}	1.71 ^c
	<i>Polygonum sp.</i>	8.25 ^a	2.77 ^a	4.29 ^a	2.01 ^a	2.48 ^a	2.06 ^a	2.16 ^a	2.20 ^a	1.89 ^a	2.07 ^a	1.88 ^a	2.39 ^a	2.55 ^a	2.57 ^a
	<i>Cynodon dactylon</i>	7.68 ^b	2.49 ^b	3.57 ^b	1.37 ^c	2.28 ^{bc}	1.61 ^c	2.04 ^a	2.04 ^{ab}	1.71 ^b	1.95 ^a	1.43 ^c	2.08 ^b	1.54 ^c	1.51 ^b
	<i>Panicum sp.</i>	8.39 ^a	2.83 ^a	3.86 ^b	1.54 ^b	2.38 ^{ab}	1.98 ^b	2.24 ^a	1.83 ^c	1.63 ^b	1.85 ^a	1.68 ^{ab}	2.39 ^a	1.84 ^{ab}	1.80 ^{bc}
Mean		7.89	2.60	3.85	1.69	2.30	1.93	2.19	2.00	1.73	1.98	1.64	2.17	1.96	1.90
LSD($P=0.05$)		0.28	0.07	0.29	0.16	0.21	0.06	0.21	0.13	0.10	0.56	0.12	0.38	0.26	0.27

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

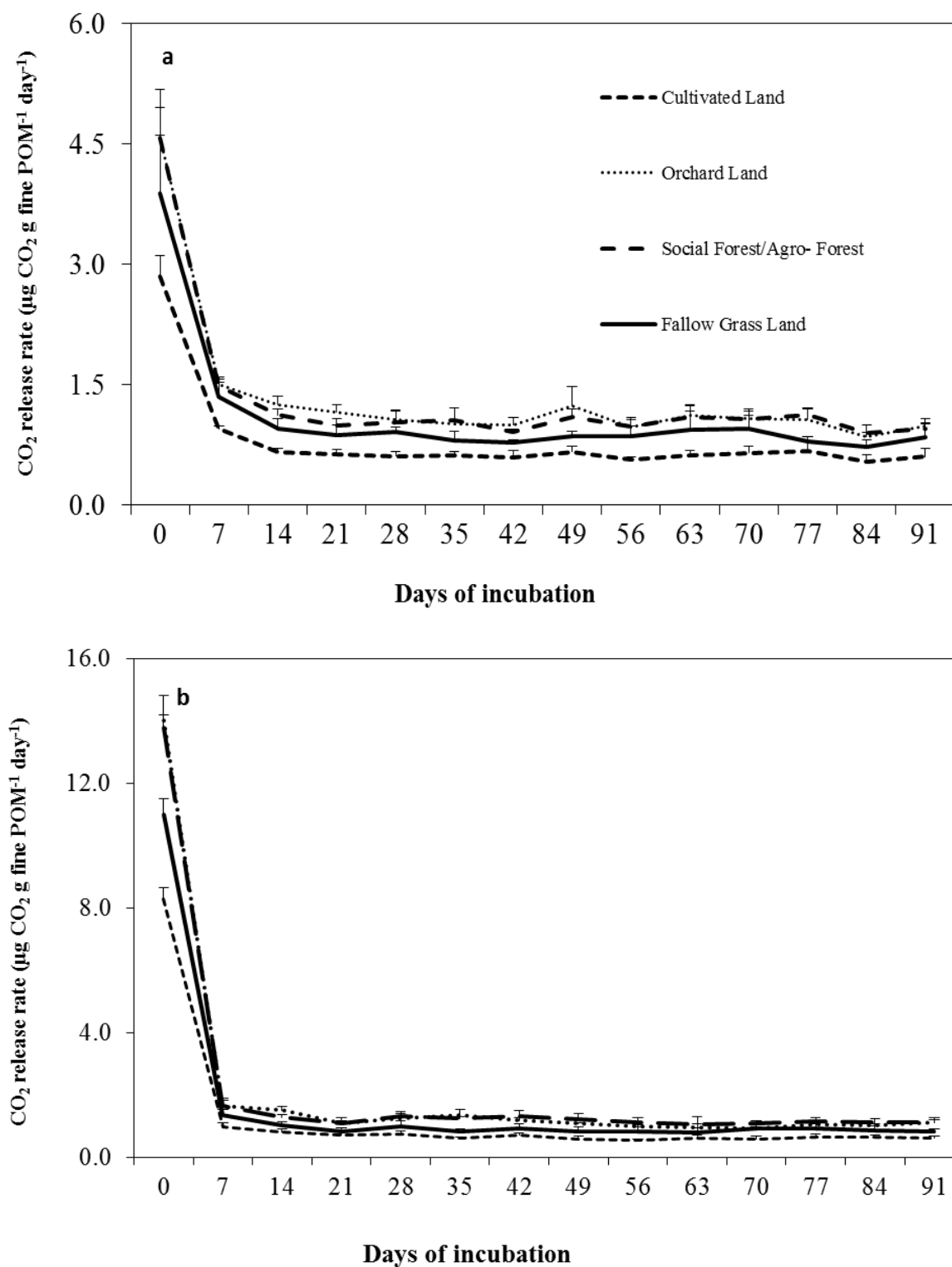


Fig.10. Average microbial respiration from fine POM incubated at a) 28°C and b) 35°C under different land use system. Range bars indicate the standard deviations of the means.

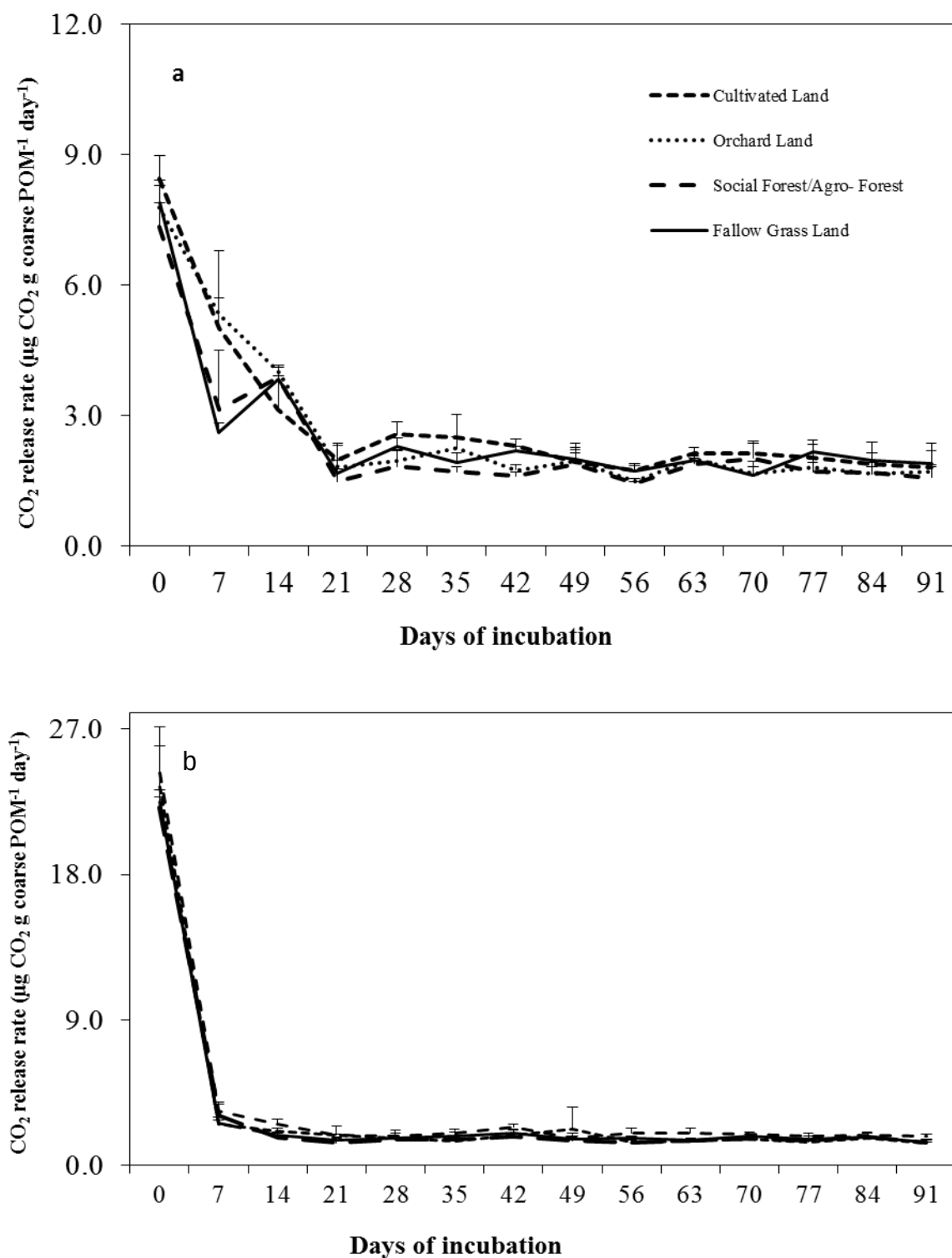


Fig.11. Average microbial respiration from coarse POM incubated at a) 28°C and b) 35°C under different land use system. Range bars indicate the standard deviations of the means.

4.5 Nitrogen and Phosphorus Supplying Capacity of Particulate Organic Matter

One of the objectives of this research was to evaluate the short-term (7-21 days) N and P supplying capacity of particulate organic matter (POM) extracted from surface soils under different land use systems. Contrary to the findings of Qin *et al.* (2010), mean temporal N mineralization from both fine and coarse POM incubated at 40°C was higher in cultivated land than fallow grassland and relatively non-cultivated orchard (Tables 19 and 20, Fig. 12). Fallow tillage management practices greatly influenced the distribution of N in soil. As illustrated in tables 19 and 20, mean N mineralization from coarse POM of surface soil with ploughing declined by ~30% in fallow grass land than cultivated land at both 7 and 21 days of incubation. The possible explanations for changes in N mineralization could be due to (i) biological oxidation of soil organic matter and (ii) variations in C and N inputs to soil due to changes in plant inputs due to crop and fallow management. Although temporal increase of N mineralization was evident in coarse POM from all land use systems from 7-21 days (Fig. 12a), a mixed trend was observed in case of fine POM (Fig. 12b).

Significant effects of different crop rotations were apparent for cultivated land during the incubation period. While incubated at 40°C, both coarse and fine POM extracted from Jute - Rice- Fallow rotation released significantly higher N (139.77 and 129.03 mg kg⁻¹ N for coarse and fine POM, respectively) than other three rotations initially (Tables 19 and 20). One possible explanation for this observation is differential enzymatic activity arising from qualitative variation of organic matter in different land use systems, leading to variable N mineralization. While this trend was maintained at 21 days of incubation in case of fine POM (Table 20), no significant effect of crop rotation was found in case of coarse POM (Table 19).

As shown in case of microbial respiration, P mineralized from both coarse and fine POM showed temporal decline (Tables 21 and 22, Fig. 13). During the first 7-14 days, the decrease in P activity proceeded at higher rate, as found in Spohn and Kuzyakov (2013). From both Fig. 10 and 11, the simultaneous temporal decrease in C and P activity was evident. Thus, the P mineralization seems to have been driven by the microorganisms' need for C. However, it cannot be excluded that the microorganisms were co-limited by C in combination with P, and that their C and P uptake was driven by the need for both elements.

It is possible that microorganisms also use phosphorylated organic compounds mainly as a C source. These findings are in accordance with studies showing that microorganisms in the sea use dissolved organic P compounds to satisfy their C demand (Hoppe and Sören, 1999; Nausch and Nausch, 2007; Steenbergh *et al.*, 2011). However, complete biochemical characterization of POM is warranted before drawing any strong conclusions.

As shown in N mineralization, significant effects of different crop rotations were apparent in P mineralization for cultivated land during the incubation period. While incubated at 40°C, both coarse and fine POM extracted from Jute - Rice- Fallow rotation released higher P (41.03 and 40.26 mg kg⁻¹ P for coarse and fine POM, respectively) than other three rotations initially (Tables 21 and 22) and maintained at 21 days after incubation. Moreover, significant effects of different vegetation were apparent in P mineralization for orchard, social forest and fallow grass land.

Table 19: Mineralized nitrogen (at 40°C) from coarse particulate organic matter under different cropping systems

Land Uses		Days of incubation		
		Mineralized nitrogen (N mg kg ⁻¹) soil		
		7 Days	14 Days	21 Days
Cultivated Land	Jute - Rice- Fallow	139.77 ^a	141.17 ^a	154.23 ^a
	Jute -Maize-Potato	82.37 ^b	94.73 ^b	108.03 ^a
	Fallow- Maize-Wheat	85.63 ^b	90.77 ^b	101.03 ^a
	Brinjal-Pumpkin-Cabbage	77.93 ^b	88.90 ^b	101.27 ^a
Mean		96.43	103.89	116.14
LSD(P=0.05)		23.31	38.37	75.25
Orchard Land	Mango	66.97 ^c	79.80 ^{ab}	82.83 ^a
	Litchi	77.47 ^a	88.67 ^a	103.37 ^a
	Guava	51.57 ^d	56.47 ^b	82.13 ^a
	Safeda	71.63 ^b	74.90 ^{ab}	85.63 ^a
Mean		66.91	74.96	88.49
LSD(P=0.05)		4.91	24.72	15.64
Social Forest/Agro- Forest	Sissoo	98.70 ^a	102.20 ^b	105.30 ^b
	Gamar	90.07 ^b	122.27 ^a	166.13 ^a
	Teak	50.63 ^d	58.57 ^d	63.23 ^c
	Rubber	59.27 ^c	68.83 ^c	71.17 ^c
Mean		74.67	87.97	101.46
LSD(P=0.05)		4.34	7.33	33.40
Fallow Grass Land	<i>Cyperus sp.</i>	70.70 ^b	72.10 ^b	84.00 ^a
	<i>Polygonum sp.</i>	54.13 ^c	60.67 ^c	65.57 ^a
	<i>Cynodon dactylon</i>	80.97 ^a	80.50 ^a	79.57 ^a
	<i>Panicum sp.</i>	65.57 ^b	81.90 ^a	88.90 ^a
Mean		67.84	73.79	79.51
LSD(P=0.05)		10.58	5.98	22.67

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 20: Mineralized nitrogen (at 40°C) from fine particulate organic matter under different cropping systems

Land Uses		Days of incubation		
		Mineralized nitrogen (N mg kg ⁻¹) soil		
		7 Days	14 Days	21 Days
Cultivated Land	Jute - Rice- Fallow	129.03 ^a	129.50 ^a	121.57 ^a
	Jute -Maize-Potato	58.57 ^c	82.60 ^b	97.53 ^b
	Fallow- Maize-Wheat	81.43 ^b	76.30 ^b	95.67 ^b
	Brinjal-Pumpkin-Cabbage	76.77 ^b	65.80 ^b	71.63 ^c
	Mean	86.45	88.55	96.60
	LSD(P=0.05)	19.59	22.01	18.99
Orchard Land	Mango	63.23 ^{ab}	63.93 ^a	73.03 ^a
	Litchi	74.43 ^a	65.80 ^a	91.70 ^a
	Guava	47.60 ^b	51.10 ^b	79.33 ^a
	Safeda	62.30 ^{ab}	54.83 ^{ab}	77.47 ^a
	Mean	61.89	58.92	80.38
	LSD(P=0.05)	16.18	10.84	18.76
Social Forest/Agro- Forest	Sissoo	89.60 ^a	84.23 ^{ab}	97.53 ^b
	Gamar	85.40 ^a	96.13 ^a	156.10 ^a
	Teak	48.07 ^b	41.53 ^c	50.17 ^c
	Rubber	51.80 ^b	51.80 ^{bc}	61.83 ^c
	Mean	68.72	68.43	91.41
	LSD(P=0.05)	32.66	39.86	14.00
Fallow Grass Land	<i>Cyperus sp.</i>	66.50 ^{ab}	61.83 ^b	73.97 ^a
	<i>Polygonum sp.</i>	47.60 ^c	51.33 ^c	55.07 ^a
	<i>Cynodon dactylon</i>	75.37 ^a	57.17 ^{bc}	76.07 ^a
	<i>Panicum sp.</i>	58.68 ^{bc}	69.30 ^a	78.63 ^a
	Mean	62.04	59.91	70.93
	LSD(P=0.05)	16.92	8.66	20.53

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 21: Mineralized phosphorus (at 40°C) from coarse particulate organic matter under different cropping systems

Land Uses		Days of incubation		
		Mineralized phosphorus (P mg kg ⁻¹) soil		
		7 Days	14 Days	21 Days
Cultivated Land	Jute - Rice- Fallow	41.03 ^a	36.41 ^a	34.87 ^a
	Jute -Maize-Potato	26.15 ^c	23.59 ^c	22.82 ^b
	Fallow- Maize-Wheat	38.97 ^a	30.77 ^b	26.41 ^b
	Brinjal-Pumpkin-Cabbage	33.33 ^b	29.49 ^b	24.62 ^b
Mean		34.87	30.06	27.18
LSD(P=0.05)		5.21	2.72	3.65
Orchard Land	Mango	17.18 ^b	15.13 ^b	14.62 ^b
	Litchi	24.10 ^a	20.77 ^a	19.74 ^a
	Guava	11.79 ^c	10.51 ^c	8.97 ^c
	Safeda	17.95 ^b	16.92 ^b	14.36 ^b
Mean		17.76	15.83	14.42
LSD(P=0.05)		3.87	1.68	2.84
Social Forest/Agro- Forest	Sissoo	18.85 ^a	18.21 ^{ab}	17.69 ^a
	Gamar	20.00 ^a	16.15 ^b	15.13 ^b
	Teak	21.54 ^a	18.97 ^a	17.44 ^{ab}
	Rubber	21.79 ^a	19.23 ^a	15.64 ^{ab}
Mean		20.54	18.14	16.47
LSD(P=0.05)		5.24	3.27	2.41
Fallow Grass Land	<i>Cyperus sp.</i>	10.77 ^a	9.00 ^a	7.95 ^a
	<i>Polygonum sp.</i>	4.62 ^b	4.00 ^b	3.59 ^b
	<i>Cynodon dactylon</i>	5.00 ^b	4.62 ^b	3.85 ^b
	<i>Panicum sp.</i>	10.51 ^a	8.21 ^a	8.21 ^a
Mean		7.72	6.46	5.90
LSD(P=0.05)		3.81	2.04	2.16

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

Table 22: Mineralized phosphorus (at 40°C) from fine particulate organic matter under different cropping systems

Land Uses		Days of incubation		
		Mineralized phosphorus (P mg kg ⁻¹) soil		
		7 Days	14 Days	21 Days
Cultivated Land	Jute - Rice- Fallow	40.26 ^a	32.31 ^a	34.10 ^a
	Jute -Maize-Potato	19.74 ^c	20.77 ^c	20.77 ^c
	Fallow- Maize-Wheat	36.15 ^a	26.41 ^b	26.15 ^b
	Brinjal-Pumpkin-Cabbage	26.92 ^b	26.67 ^b	25.13 ^b
Mean		30.77	26.54	26.54
LSD(P=0.05)		6.62	5.50	4.31
Orchard Land	Mango	14.36 ^b	12.56 ^c	13.59 ^b
	Litchi	23.59 ^a	20.51 ^a	19.74 ^a
	Guava	11.54 ^c	8.46 ^d	10.00 ^c
	Safeda	14.36 ^b	16.15 ^b	15.38 ^b
Mean		15.96	14.42	14.68
LSD(P=0.05)		2.90	2.58	1.56
Social Forest/Agro- Forest	Sissoo	15.38 ^a	17.18 ^{ab}	14.87 ^b
	Gamar	18.46 ^a	15.64 ^b	15.90 ^b
	Teak	18.97 ^a	18.21 ^a	16.67 ^{ab}
	Rubber	16.41 ^a	16.15 ^{ab}	18.72 ^a
Mean		17.31	16.79	16.54
LSD(P=0.05)		4.69	2.65	2.52
Fallow Grass Land	<i>Cyperus sp.</i>	8.97 ^a	8.21 ^a	8.72 ^a
	<i>Polygonum sp.</i>	3.59 ^a	3.59 ^b	3.33 ^b
	<i>Cynodon dactylon</i>	4.87 ^a	4.36 ^b	4.62 ^b
	<i>Panicum sp.</i>	10.26 ^a	7.18 ^a	7.95 ^a
Mean		6.92	5.83	6.15
LSD(P=0.05)		2.28	2.30	2.03

Different letters in the column indicate significant differences ($P < 0.05$, Duncan test of variation of mean).

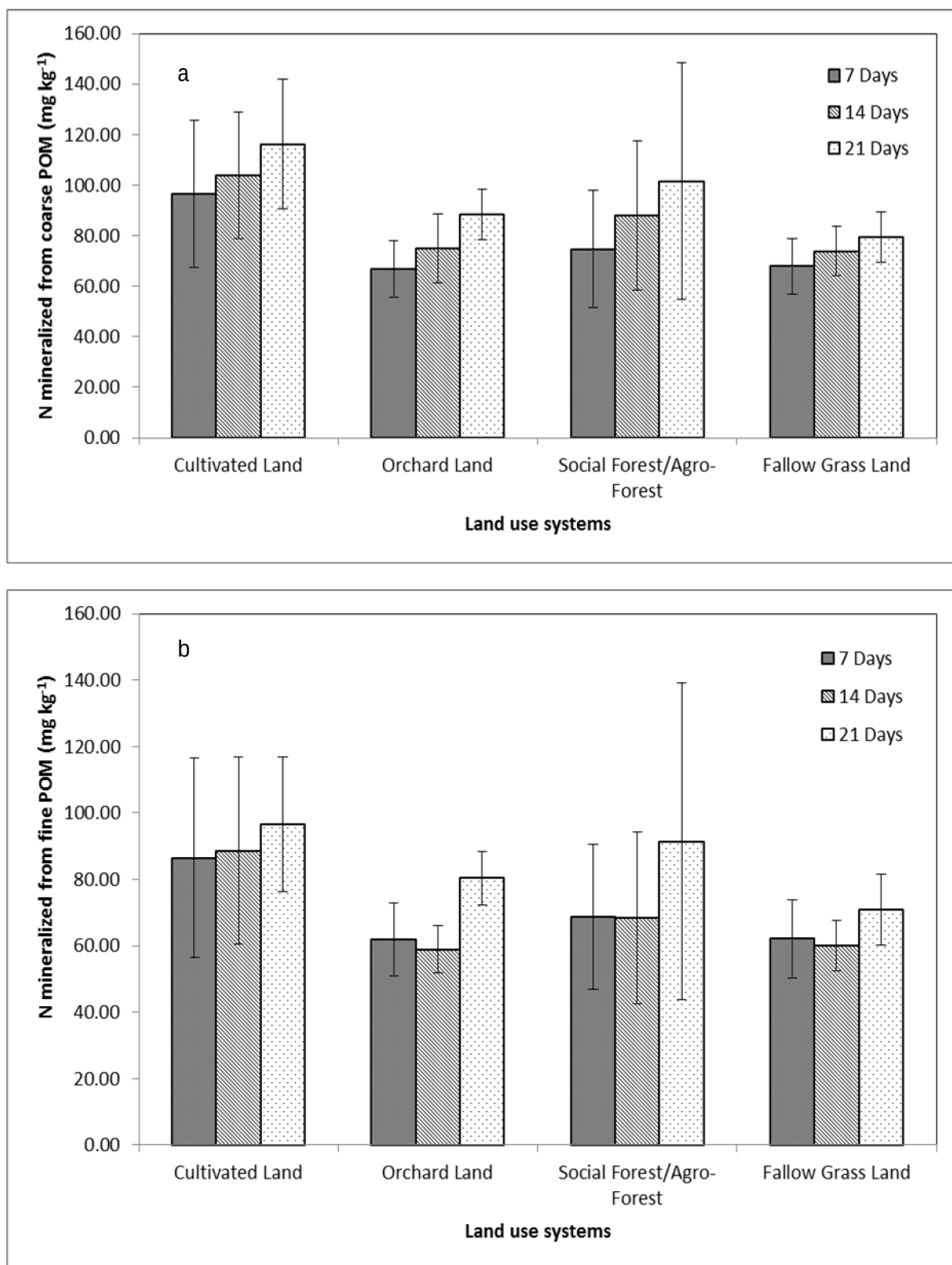


Fig.12. Averaged mineralized nitrogen (at 40°C) from a) coarse and b) fine POM under different land use systems. Range bars indicate the standard deviations of the means.

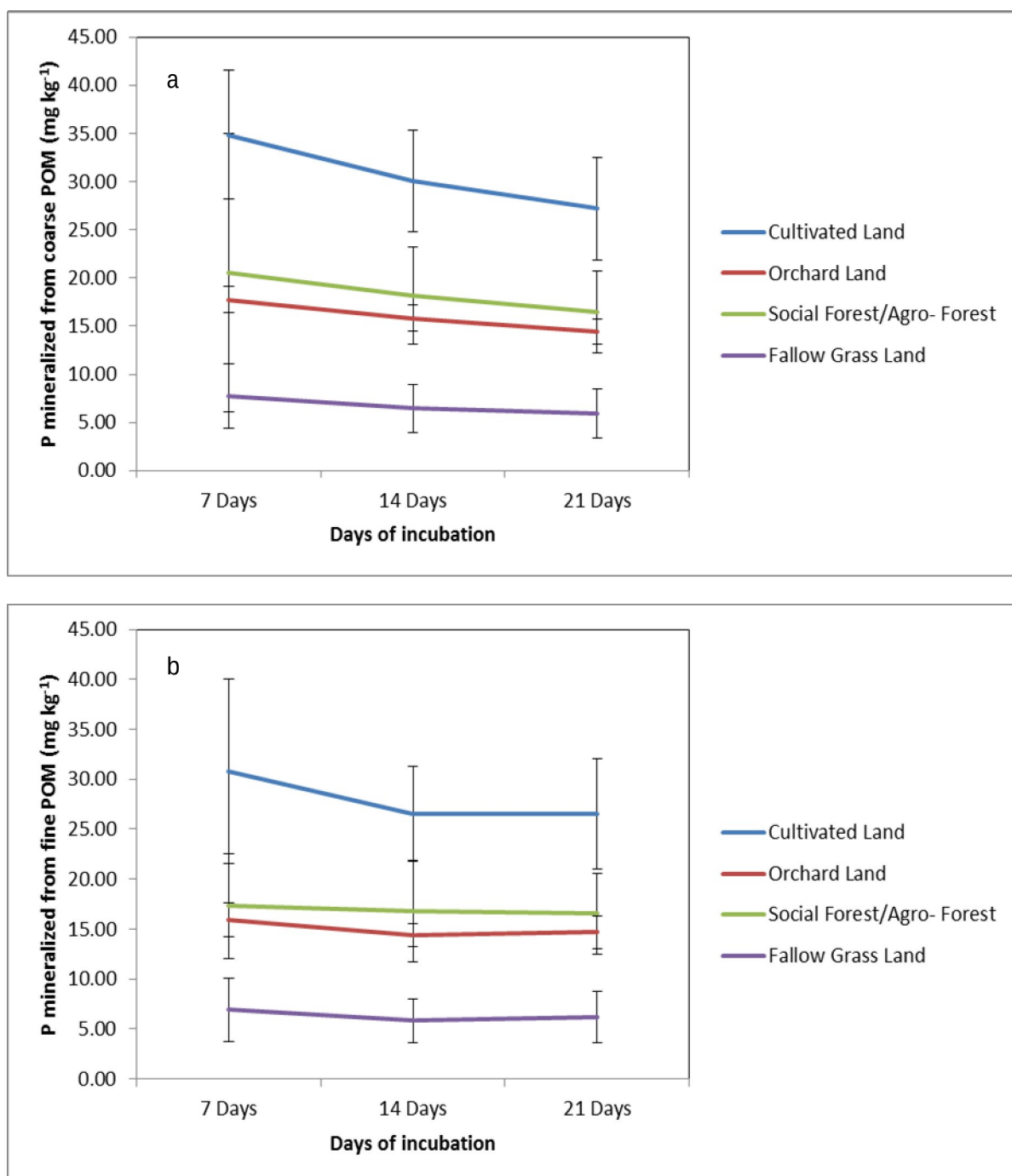


Fig.13. Averaged mineralized P (at 40°C) from a) coarse and b) fine POM under different land use systems. Range bars indicate the standard deviations of the means.

CHAPTER-V



===== **Summary and conclusions**

Chapter -V

Summary and Conclusions

This study investigated soil organic carbon, particulate organic carbon of soil under different land use systems since land use and agronomic management strategies have drastic effects on the level of the soil organic carbon (SOC) pools. Further, this study explored the soil enzyme activities involved in C, N and P transformations, in order to test whether the different land use pattern modified the microbial activities associated with the SOC balance at the soil aggregates. Results indicated that:

- i. The difference in management practices resulted in difference organic carbon contents where social forest exhibited highest average organic carbon content (1.30%), owing to high plant litter deposition.
- ii. Cultivated land showed significantly less water stable soil macro-aggregates while within cultivated lands, lowest macro as well as micro-aggregate formation has been found under vegetables (Brinjal-Pumpkin-Cabbage).
- iii. Lower value of aggregate indices in cultivated soils implied degradation of soil aggregation under long-term agriculture practices.
- iv. Cultivated soils (bulk) showed less C and N in spite of seasonal addition of manures and fertilizers, probably due to continuous crop uptake, tillage practices, and periodical perturbation, ultimately increasing the organic matter turnover rate.
- v. Lowest macro as well as micro-aggregate occluded C was found under agricultural fields. This finding was similar to the trend of soil aggregation. It

infers destruction of soil aggregation under continuous cultivation and subsequent low physically protected C within aggregates in agricultural soils.

- vi. Estimation of micro-aggregated C and N revealed lowest presence of micro-aggregated C in soils under cultivated land and highest micro-aggregate occluded C under fallow grass land.
- vii. Results showed lowest ratio of MacAC/ MicAC and MacAN/ MicAN in agricultural soils while highest was under social forest/ agroforestry, confirming the less aggregation as well as less aggregate associated C and N in cultivated soils in comparison to soils under social forest/ agroforestry and fallow grass land.
- viii. Averaging across all the crop rotations, an interesting trend appeared where cultivated land produced higher dehydrogenase activity ($16.25 \mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$) than fallow grass land ($11.40 \mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$) and social forest ($5.25 \mu\text{g TPF g}^{-1} \text{ soil h}^{-1}$), possibly as a result of agrochemicals that inhibit enzyme activities by masking catalytically active groups, having denaturing effects on the conformation of proteins, or competing with the metal ions involved in the formation of enzyme–substrate complexes. Similar trend for bulk soils were found in β -glucosidase and protease where enzymatic activity reduced from cultivated to fallow grass lands.
- ix. Microbial respiration at 0-day from fine and coarse POM mixed sand incubated at 28°C and 35°C were higher in conservational agricultural practices like social forest and fallow grass land than cultivated land since conservational agricultural managements significantly promotes soil respiration and enzyme activities.
- x. The mean temporal N mineralization from both fine and coarse POM incubated at 40°C was higher in cultivated land than fallow grassland and relatively non-cultivated orchard. Moreover, significant effects of different crop rotations were apparent in P mineralization for cultivated land during the incubation period.

CHAPTER-VI



===== **Future Scope of Research**

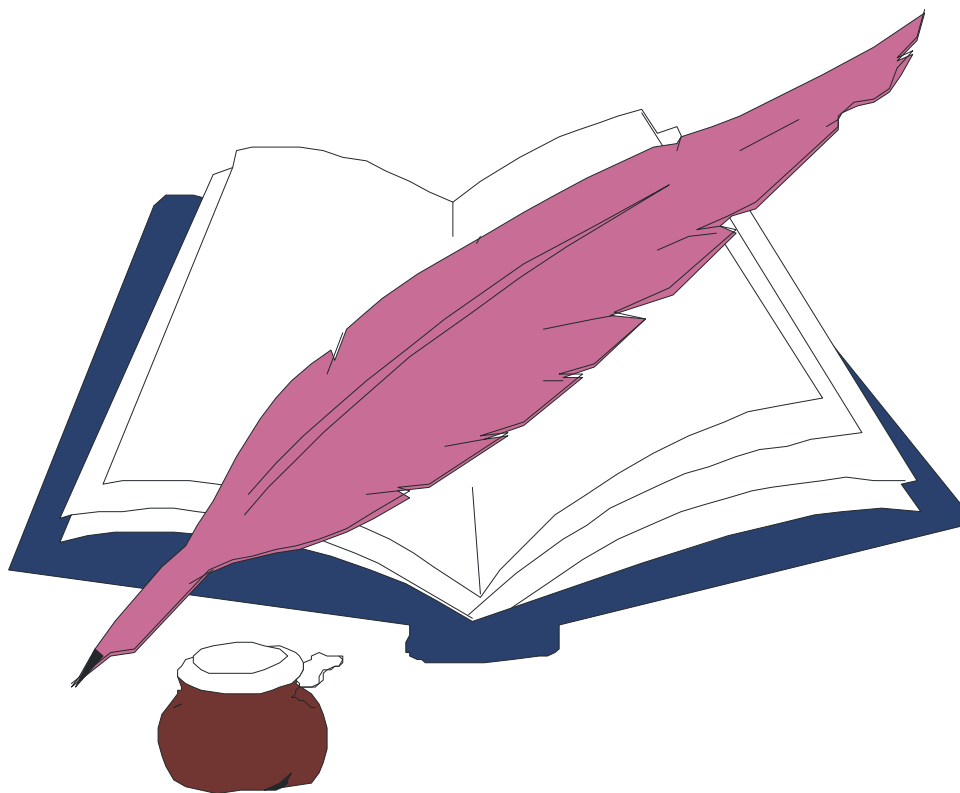
Chapter -VI

Future Scope of Research

This study identifies some anomalies in enzymatic dynamics resulting from different land use systems and requires the following investigations in future:

- i. Since dehydrogenase enzyme is often used as a measure of any disruption caused by agrochemicals, a detailed residual analysis of agrochemicals and heavy metals is required to test their masking effects on dehydrogenase activity.
- ii. The greater relative abundance of soil fungal community in the micro-aggregates needs to be confirmed by phospholipid fatty acid (PLFA) analysis before making any stronger conclusion about their influence on enzymatic activities.
- iii. There is a need for complete biochemical characterization of POM extracted from different land use systems before making any strong conclusion.

CHAPTER-VII



References

Chapter -VII

References

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