SOIL MICROBIAL DIVERSITY IN THE DISTURBED AND UNDISTURBED DECIDUOUS FORESTS OF MEGHALAYA

BY

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IN LOVING MEMORY

OF MY

BELOVED GRANDPARENTS

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I, **Melboreen Dkhar,** hereby, declare that the subject matter of this thesis entitled "Soil microbial diversity in the disturbed and undisturbed deciduous forests of Meghalaya" is the record of work done by me, that the contents of this thesis did not form basis of the award of any previous degree to me or to the best of my knowledge to anybody else, and that the thesis has not been submitted by me for any research degree in any other University/Institute.

This is being submitted to the North Eastern Hill University, Shillong for the award of the degree of Doctor of Philosophy in Botany.

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INTRODUCTION

Microbial diversity is an unseen national as well as international resource that deserves greater attention than it has been receiving. It encompasses the spectrum of variability among all types of microorganisms in the natural world and as altered by human intervention. Microbial diversity studies are important in order to understand the microbial ecology in soil and other ecosystems (Atlas, 1984 and Reid, 1994). It plays an important role in both natural and agroecosystems. The diversity of plants and animals in forests and agro-ecosystems receives a great deal of scientific attention, whereas the diversity of microorganisms is often ignored. Therefore, much more needs to be done to understand the role of microorganisms and inventory their diversity and to find ways to exploit them beneficially.

Diversity means state of being diverse, difference, unlikeness and variety. The total variability of life on earth means the variability among living organisms from all sources including interalia, terrestrial, marine and other aquatic ecosystems and the ecological complexes of which they are part of; this includes diversity within species and of ecosystems. Strictly speaking, the biodiversity refers to the quality, range or extent of differences between the biological entities in a given set. In total, it would thus be the diversity of all life and is a characterizing property of nature, not in entity or a resource. Biodiversity is the established norm of the nature. The term "diversity", as used today, stands from a molecular to a global level of biological organization. It can be applied to different ecosystems, populations, and even to different individuals. Biological diversity can be defined as "the variety of species in ecosystems as well as the genetic variability within each species" (Conservation, 1987). Diversity is, therefore, the range of

significantly different kinds of organisms and their relative abundance in natural assemblage and habitat. An estimate of microbial diversity is a prerequisite for understanding the functional activities of microorganisms in ecosystems (Garland and Mills, 1994; Zak *et al.*, 1994).

Studies on biodiversity and its relation to ecosystem structure and function have mainly focused on macroorganisms, and little attention has been directed towards microorganisms. Microorganisms, however, are important, especially due to the fact that life is dependent on microbial processes. The last decade has produced an extraordinary new awareness of microbial diversity (Dykhuisen, 1997). Microorganisms have an enormous impact and role in our daily lives, including everything from maintaining the biosphere to improving our life style (Hunter-Cevera, 1998).

Biodiversity is the variety of nature. It is manifested at several levels of biological organization and should be studied at the levels of individuals, populations and biocoenoses (plant, animals and microbial communities). In evolutionary terms, the ultimate cause of diversity exists at higher levels of genetic variation. Any change at one level can affect other levels, leading either to increased or decreased biodiversity (Romke *et al.*, 1996). Soil biodiversity should not only mean, however, to maximize the number of species in a certain ecosystem, more likely it is the ability to maintain current macro- and micro-flora and emphasizing the various species and processes so as to accomplish specific objectives. The term species diversity consists of two components; the first component is the total number of species richness. In other words, it refers to the quantitative variation among species. The second component is the distribution of individuals among these species, which is referred to as evenness or equitability. One

problem is that evenness is often unknown in bacterial assemblages because individuals very seldom are identifiable to the species level. The term "bacterial diversity" has been used to describe complexity and variability at different levels of biological organization, including genetic diversity within bacterial taxons (species), diversity of bacterial taxons in assemblages or habitats and ecological diversity including variability in community structure, complexity of interactions, number of trophic level and number of guilds (functional diversity). As the soil microbial community is a complex picture of interwoven relationships between organisms of different trophic levels, this will lead to many indirect effects. There are two important implications of microbial diversity in this regard. Firstly, a decrease in diversity will generally result in the risk that there is a decrease in ability of the biological system to respond to perturbations (Ekschmitt and Griffiths, 1998). Secondly, bacterial diversity reflects the state and history of influences on the microenvironment, the diversity itself gives an indication as to how stressed the ecosystem has been. Biodiversity is affected by both qualified and quantified factors: in less diverse habitats a relatively slight damage is able to cause a more completed destruction, where less type of species will be found. So, the general assessment should be based on the status of the species: conservation value of a given species is higher if it occurs exclusively in endangered habitats.

In an ecosystem, spatiotemporal complexity is of fundamental importance in maintaining species diversity (Tilman, 1994; White *et al.,* 1998). Although it is not possible to be sure that different species are using the same resource, the following well-documented patterns are suggestive of this: (1) seasonal variations in fungal populations following variations in temperature and moisture (Christensen 1969; Bisset and

Parkinson ,1979a and b; Widden and Arbitol, 1980; Widden, 1981,1986b; Eastburn and Butler, 1988; Cooke and Whipps, 1993); and (2) short-term rhythmic behavior, e.g. diurnal cycles of growth and reproduction which are often controlled by light, temperature or water (Cooke and Whipps, 1993). Microorganisms play an integral and often unique role in ecosystem functions, yet, little is known about dominant populations that presumably play vital roles in these functions, nor do we know much about how these populations differ with habitat. The greatest microbial diversity at small scales appears to reside in the soil. Soil microbial communities are among the most complex, diverse and important assemblages in the biosphere diversity (Zhou *et al.*, 2003).

Diversity can vary with a number of factors such as disturbances and stress, in addition to ecological interactions (predator-prey interactions). The major underlying principle of diversity studies is probably the assumption that interactions between populations in a habitat lead to an organized and stable community (Atlas, 1984).

Microbial diversity indices can function as bio-indicator to show community stability and describing the ecological dynamic of community (Atlas, 1984) and analysis of soil microbial diversity is important to evaluate the importance of perturbations in soil systems (Turco *et al.*, 1994). Microbial populations can also provide an early indication of changes in soil long before it can be measured by changes in organic matter (Powlson *et al.*, 1987). It has been generally hypothesized that reduction in soil microbial diversity will result in reduction in the functional capability of soil (Giller *et al.*, 1997). However, decline in soil microbial diversity does not consistently result in reductions in the functional diversity of microbial communities (Klein *et al.*, 1986; Atlas *et al.*, 1991).

The diversity of soil fauna and microorganisms is influenced by vegetation, soil factors, climate and management practices (Gupta and Malik, 1996). Soil microorganisms play a very important role in fertility of soil not only because of their ability to carry out biochemical transformation but also due to their importance as a source and sink of mineral nutrients (Jenkinson and Ladd, 1981). There have been studies on the distribution of microbial diversity in various environmental niches (Stolp, 1988). Limited work has been carried out on microbial diversity of various management practices (Wardle, 1995).

The knowledge of complex relationship between plant and microorganisms and also among different microorganisms is an important aspect to understand plant behavior. There is a growing interest in the relationships among ecosystem diversity, structure, function and a number of theories have been formulated concerning how species diversity is related to ecosystem function (Muller *et al.*, 2001). Enhanced species diversity is beneficial to ecosystem function (Naeem *et al.*, 1995). In contrast, other authors suggest that the properties of an ecosystem depend more upon the functional abilities of a particular species than on the total number of species (Tilman *et al.*, 1997 and Wardle *et al.*, 1997). The diversity and functional importance of soil organisms present an excellent opportunity to test currently tropical aspects of ecological theory, the hypotheses relating ecosystem diversity and function have mainly been developed by plant ecologists with only a few studies having concerned the soil community (Mikola and Setala, 1998; Griffiths *et al.,* 2000). Hypotheses from the aboveground communities may not easily be applied to belowground, since there are differences. The microbial diversity in soil is enormous (Torsvik *et al.,* 1990) and there may be substantial overlap in function between

microbial species (Chapin *et al.,* 1997). Furthermore, it is likely that microorganisms within a functional group differ in their response to the environment. As microorganisms are fast growing, they can quickly fill out empty niches occurring when the environment is changing (Giller *et al.,* 1998). These circumstances could create a high degree of stability, but it is unknown what level of diversity is necessary to maintain stability (Wardle and Giller, 1996).

Soil life is highly diverse and consists of interacting population of microorganisms and soil fauna whose activities influence physical, chemical and biological properties of the soil. Soil, the most important natural resource harbors a variety of microorganisms and is considered to be the most dynamic site of interactions in nature. Determination of the optimum diversities of soil microbial populations of both natural and agricultural systems for their sustainable management is very important.

Soil microorganisms are of great importance for soil ecosystems because they affect plant available nutrients and soil structural stability (Paul and Clark, 1989). The abundance, size and activity of the microbial populations depend on quantity and quality of organic matter, texture, and other environmental factors (e.g. soil type, nutrient status, pH, moisture) as well as plant factors e.g. species, age (Insam *et al.,* 1989; Kaiser *et al.,* 1992). Microbial growth in soil is carbon limited and therefore, the presence of organic matter has the greatest influence on microbial populations (Lynch and Whipps, 1990; Wardle, 1992).

The impact of burgeoning human populations has destroyed the soil physico-chemical environment and the soil's species through activities such as: inputs of chemicals from the atmosphere, disposal of waste products in soils, ground water

contamination and physical modification or removal of soil by cultivation and erosion. Soil among the vast unknown life on our planet, is a dark frontier, despite their critical importance to understanding ecosystem function. For example, thousands of species of microbes and invertebrates inhabit just a square meter of temperate grassland soil, organisms whose identities and contributions to sustaining our biosphere are largely undiscovered. The elucidation of species diversity of soils in conjunction with sustainability assessments of soil-mediated ecosystem processes must be a high priority in global biodiversity efforts.

It is also suggested that the total functional diversity of microorganisms in different soils may be similar and that a combination of environment and plant factors influence organisms which are active, become culturable (Colwell *et al.*, 1985) and proliferate under different conditions. Many fungi and more than half of the species of bacteria at present classified can be found in soil. Soil microorganisms are largely hidden underground population; frequently have greater mass than the plants and animals aboveground (Jenkinson and Ladd, 1981 and Sparling, 1985). Despite their comparable mass, we know much less about microorganisms than we do about higher plants and animals (Wardle and Giller, 1996). The analysis of the functional diversity of soil populations is a useful way to characterize and compare the microbial community. The high degree of functional redundancy in soil microbial communities may result in there being no effect of changes in microbial diversity on the function of the communities (Andren *et al.*, 1995; Giller *et al.*, 1997). This is likely because, in the case where a large number of species conduct similar functions, a reduction in any group of species has little effect on overall soil processes since other organisms fill the functional role (Lawton and

Brown, 1993; Andren *et al.*, 1995). However, broad functional diversity may be additionally important in influencing the resilience of soils (Elliot and Lynch, 1994; Pankhurst *et al.*, 1996; Giller *et al.*, 1997). Soil resilience principally concerns the capacity for soil to function after different disturbance treatments. However, an important element of soil resilience should also include the capacity for soils to continue functioning under a range of environmental conditions. It is, therefore, relevant that an assessment of the functional capability of microbial communities with reduced diversity be conducted under a range of environmental conditions.

Soil microbial communities remain some of the most difficult communities to characterize, because of their immense phenotypic and genotypic diversity. Activity and growth of microorganisms is restricted by soil environmental factors, e.g., temperature, moisture, pore size, distribution and nutrient availability and therefore, indirectly by cultivation practices.

The structure of soil microbial communities is determined by a hierarchical series of interacting variables. Climate and microclimate set the physiological limits for microbial activities. The quality and quantity of plant resource inputs influence the abundance of different fungal and bacterial species. At the micro-scale, physico-chemical environment and biotic interactions refine the composition and dynamics of specific species associations or unit communities. Vegetation cover directly and indirectly influences all these variables and often has a dominant effect on the spatial patterning of soil microbial communities. This is particularly evident for trees that modify the environment and underlying soil properties at scales that are amendable to systematic analysis. The application of geostatistical technique has shown that the spatial

distribution of microbial communities in forest is related to the location of different tree species and the associated ground flora (Pennanen *et al.*, 1999; Saetre, 1999).

Fungi and bacteria control many of the vital processes on which the very maintenance and survival of tropical forests depend (Hawksworth and Colwell, 1992). An overview of the role of microorganisms in ecosystem functioning as a whole has been presented by Allsopp *et al.* (1995). Their goal is to identify functional attributes of microorganisms in tropical forests and to identify those processes that are most likely to be sensitive to losses of diversity especially in the face of disturbance or broad environmental changes. In some cases, microbes may influence ecosystem processes indirectly by altering the diversity of other organisms.

Humans are altering global nutrient cycles as well as reducing biological diversity. It is of paramount importance to understand the effect of these global changes on ecosystem functions, especially those on which human life directly depends. Microbes are key players in ecosystem functions such as decomposition of organic matter and nitrogen cycling. It has been demonstrated that microbial community composition can influence certain ecosystem process rates. However, little is known about the factors that determine microbial community composition and/or functioning, including anthropogenic disturbance.

The structure and functional diversity of the microbial communities in the soil is tightly related to plant species composition above - ground, thus providing an important link between above and below ground processes in terrestrial ecosystems (Grayston and Campbel, 1996; Westover *et al.,* 1997; Priha *et al.*, 1999; Grayston *et al.*, 2001).

Soil management practices affect soil microbial communities, which mediate many processes essential to the productivity and sustainability of soil. Microbes also play a major role in the formation of good soil structure. Bacterial mucigel and hyphal threads produced by fungi and actinomycetes bind the soil particles together. Microbial activity helps to aggregate the soil, which reduces soil erosion, allows for good water infiltration, and maintains adequate aeration of soil. Soil microbes also affect the persistence of organic compounds applied to soil (Biro *et al.*, 1983).

The effect of disturbance on microbial community function depends on its duration and specificity. After a transient disturbance, system function may eventually return to its former state, whereas a permanent disturbance will result in a new altered state (Rykiel, 1985). Disturbance with a specific mode of action only alter a few groups of organisms, whereas, those that act non-specifically affect a wide range of organisms. Investigation of how different kinds of disturbances affect the system function will enhance our knowledge of the relationships among diversity, structure and function.

Recognition of the importance of soil microorganisms in the functioning of ecosystems has led to an increased interest in measuring the nutrients held in soil biomass (Brookes *et al.,* 1982; Martikainen and Palojarvi, 1990).The microbial biomass is a potential source of nutrients for plants especially of their ability to bind temporarily the nutrition to microbial cells. The microbial biomass and its activity are important indicators of changes in content and vertical distribution of the organic matter in a soil profile. Numerous studies on the measurement of microbial C, N and P in different natural and disturbed ecosystems have been shown that the soil microbial biomass is among the most labile pools of C and mineral nutrients which are liberated after the death

of microorganisms (Anderson and Domsch, 1980; Smith and Paul, 1990; Diaz-Ravina *et al.,* 1993). It serves as an important reservoir of plant nutrients such as N and P (Jenkinson and Ladd, 1981; Marumoto *et al.,* 1982; Van Veen *et al.,* 1987; Duxbury *et al.,* 1989; Jenkinson and Parry, 1989; Smith and Paul, 1990; Lodge, 1993).

Large annual fluctuations in the microbial biomass has been reported by Lynch and Panting (1980a and b) while others observed only small annual changes (Schnurer *et al.,* 1986; Patra *et al.,* 1990). Soil microbial biomass plays a key role in maintaining soil fertility because its activity is the primary driving force for cycling of elements such as carbon and nitrogen.

With increased study, more precise information has been gained on the influence of the microbial biomass on carbon and nutrient cycling, controlling turnover and mineralization of organic substrates (Sparling, 1985) and influencing forest productivity (McGill *et al.,* 1986). Estimation of soil microbial biomass is now frequently made because of the importance of soil organisms in nutrient cycling and their role as a source and sink of plant nutrients (Smith and Paul, 1990). Study of microbial communities and biomass in forest soil may give insight into the role of microbes in restoring soil fertility. Microbial biomass is the characteristic of microorganisms which participate in the biochemical cycles and are living part of the soil organic matter (Cengel, 1990; Srivastava, 1992).

The microbial biomass content of the soil depends on the quality and quantity of resource and its distribution of the C-input varies with time and depth (Kaiser and Heinemeyer, 1993). Generally, microbial biomass C comprises about 2-4 % of total organic C (Anderson and Domsch,1989) but variation within this range is influenced by

the quantity and quality of organic inputs to the soil (Wardle, 1992). A recent review by Nannipieri *et al.* (2002) has focused on the inter-relationship between the biomass, its diversity and function in soil. The microbial biomass is a sensitive indicator of changes resulting from agronomic practices and other perturbations of the soil ecosystem (Doran, 1987; Smith and Paul, 1990).

Though, microbial biomass is relatively small fraction of the total biomass in terrestrial ecosystem, the microbial activity is of paramount importance in the nutrient cycling and energy flow (Diaz- Ravina *et al.*,1993b). Usually, the microbial biomass of the tropical environments is sensitive to land use changes and even appears to be a sensitive indicator of both soil carbon and background nitrification. The changes in microbial biomass can provide an early indication for a slower and less detectable soil organic matter and soil fertility as well (Srivastava, 1992; Henrot and Robertson, 1994; Maithani *et al.*,1996; Taylor *et al.*,1999).

The physico-chemical characteristics of soil influence the level of biomass and activity of microorganisms. Values of microbial biomass can provide one of the most satisfactory estimates of the restoration of soil microbial populations (Maithani *et al.*, 1996). The close relationship between pH and the microbial biomass is indicative of complex feedback mechanisms which are activated by the fauna. Less acidic environments stimulate the microbial biomass as determined by substrate-induced respiration (Anderson and Domsch, 1993). The stimulation leads to higher enzymes production, as shown by the close correlation between microbial biomass and N turnover. Higher enzyme production, in turn, may raise the soil pH by releasing NH^{+}_{4}.

All the biochemical reactions in soil are carried out in the presence of enzymes. Degree of enzymes activity is usually the important indicator of soil bio-activity and fertility. Major biological processes such as mineralization, immobilization, nitrification, nitrate reduction etc. are the result of microbial activities and are catalyzed by enzymes. Chemical reaction in soil is highly influenced by enzymatic activity. The biological activity in soil provides better insight in the understanding of transformation of organic matter (Pietikainen and Fritze, 1995). Therefore, knowledge about enzymes activities and their temporal and seasonal variations in soil has considerable biological significance.

Enzymes in soils can persist for an extended period of time, because the organic fractions have a protective influence on soil enzyme activity (Skujins, 1976; Coyne, 1999). The decomposition of organic matter is a chain reaction in which enzymatic reactions control the supply of substrate and energy for microbial growth, while the latter controls the microbial processes. Soil enzymes have an essential role in integrating the effects of climate, cultivation, soil amendments and edaphic properties; therefore, they may be considered as important indicators of the total biological activity and also the fertility of soil. The activity of a particular enzyme in the soil is a composite of activities associated with various biotic and abiotic components (Burns, 1982). These soil enzymes have been considered as potential component of group of indices to assess soil quality as the level of soil enzymes can serve as an estimate of the ecosystem disruption (Tate, 1995). Measurements of enzymatic activities have been used as a measure of total microbiological activity and soil fertility levels (Stevenson, 1959; Tiwari *et al.,* 1988a and b; Chander and Brookes, 1991). There appears to be a direct relationship between number of microorganisms and the enzymatic activity.

Enzymes in soils originate not only from microbial sources but also from animals, plants and the resulting soil biological activity includes the metabolic processes of all these organisms (Ladd, 1978; McKay, 1991; Tabatabai, 1994; Schinner *et al.*, 1995; Sarapatka, 2003). During recent times, much emphasis is laid on the activity of microorganisms in the soil. Generally, the rates of microbial mediated biochemical reactions are used for this purpose as they provide an index of microbial activity (Skujins, 1978). Soil enzymes play a key role in releasing nutrients. It is useful to consider some of the soil properties that may influence phosphomonoesterase (PME) and phosphodiesterase (PDE) activity. Evaluation of soil organisms is closely related to soil enzyme activity. Soil enzymes have been suggested as potential indicators of soil quality because of their relationship to soil biology, ease of measurement and rapid response to changes in soil management (Dick, 1994; Dick *et al.,* 1996). Such an index would integrate chemical, physical and biological characteristics and be used to monitor the effects of soil management on long-term productivity (Doran and Parkin, 1994). The activity of soil microorganisms is strongly linked to the activity of enzymes and soil management (including crop rotations, fertilization, tillage and crop residue placement) strongly influences the activity of soil enzymes (Miller and Dick, 1995; Deng and Tabatabai 1996a and b, 1997; Klose *et al.,* 1999). Most of the enzymes are added to soils by decaying microbial tissues, plant residues and animal remains.

The physiological and metabolism of soil microorganisms are driven by enzymes and the microbial habitat in soil is affected by these soil enzymes. The enzymes whose activities are most widely studied are dehydrogenase (oxido-reductase), urease (hydrolases) and phosphatase due their importance in various management practices.

Dehydrogenase, urease and phosphatase are commonly used for this purpose. Dehydrogenase, being a respiratory enzyme, provides a measure of catabolic activity (Lenhard, 1956; Stevenson, 1959; Casida *et al.,* 1964). The dehydrogenase activity in the soil may be used as a measurement of overall microbial activity. Dehydrogenase enzymes are considered to play an essential role in the initial stage of the oxidation of soil organic matter by transferring hydrogen or electron from substrates to acceptors. This aspect has been studied by several workers since the introduction by Lenhard (1956). Being a respiratory enzyme, it provides a measure of catabolic activity of soil (Skujins, 1976) and is considered to be caused by broad group of endocellular enzymes (Skujins, 1978). It acts as an indicator of the microbiological system in soils and can be considered a good measure of microbial oxidative activity. It is a sufficiently reliable method of measuring metabolic activity of the soil microorganisms.

Urease and Phosphatase have been the most commonly studied soil enzymes along with dehydrogenase activity, which provide an index of total biological activity. Urease enzyme is responsible for the break-down of urea into carbon dioxide and ammonia. Due to this property it has got an applied importance in the N-economy of soil. Soil ureases are microbial products that can accumulate in cell free form in the soil because they are highly resistant to environmental degradation (McNaughton *et al.,* 1997). Urease acts as an intermediary enzyme in the transformation of organic nitrogen, while phosphatase provides an estimate of breakdown of organic phosphate compound and release of phosphate in the soil (Cosgrove, 1967). The term 'phosphatase' has been widely used to describe a group of enzymes that catalyze and hydrolyse both esters and anhydrides of phosphoric acid (Schmidt and Laksowski, 1961). The phosphatases are

involved in transformation of organic phosphorus compounds in soil. Their activity may play a significant role in release of phosphorus compounds (Rastin *et al.,* 1988). The phosphatase activity of soil has been shown to vary with the standing vegetation (Neal, 1973). Phosphatase has been found to be a good indicator of the organic matter in soil (Hattori, 1988). The estimation of the activity of these enzymes provides an assessment of three different microbe-mediated processes in soil and gives the most reliable measure of microbial activity.

Enzymes are markedly dependant on pH, ionic strength, inhibitors (or pollutants), moisture regimes, temperature and other environmental factors (Tabatabai and Dick, 1979; Frankenberger and Johanson, 1982; Dick and Tabatabai, 1983), but they may become stabilized in the soil by forming humus-enzyme or clay-enzyme complexes (Makboul and Ottow, 1979).

Soil microbes and plants roots are sources of extracellular enzymes, mainly through either secretion from living cells or from lysed cells (Burns, 1982). Once in the soil, enzyme may be protected from denaturation by being absorbed into organic or inorganic surfaces (Pang and Kolenko, 1986). In this absorbed state, extracellular enzymes developed stability to desiccation and heat and can remain active for several years (Miller and Dick, 1995). Under favourable conditions, microorganisms increase most of the enzyme activity. The effect of plants on soil enzymatic activity is due to changes in organic matter content and microbial populations, but is also formed by accumulated enzymes and by continuous release of extracellular and endocellular enzymes; all of which originate in the plant root. The overall enzyme activity of the soil

is derived from the activity of accumulated enzymes and from that of proliferating microorganisms (Burns, 1982; Tabatabai and Fu, 1992).

Changes in soil chemical and physical conditions influence microbial activity and population structure. On a primary basis, the natural environment provides long-term and seasonal fluctuations in temperature, moisture and plant growth to which microorganisms must response (Schimel, 1995). Microorganisms are intimately associated with their physical and chemical environment and it is, therefore, conceivable that the temporal dynamics observed will partly reflect adaptation to environmental variables rather than competition between different components of the microbial biomass. Many aspects of soil microbial communities are affected by prevailing conditions with respect to substrate quality and stresses (Soren *et al.*, 2002).

Although changes in land growth and allocation in response to changing supplies of light, water, or nutrients are widely recognized (Mooney, 1972; Chapin III, 1980; Tilman, 1985; Ingestad and Agren 1988; Rastetter and Shaver, 1992), the nature of environmental limitation of soil microbial activity is less clearly understood. Energy supply is often viewed as the major factor limiting soil microbial activity (Richards, 1987; Tate III, 1995) because of the heterogeneity nature of decomposition. However, most soils contain 30-100 times more dead organic carbon than live microbial C (Jenkinson and Ladd, 1981). Consequently other factors such as substrate quality, nitrogen (N) availability, physical environment (e.g. temperature and moisture) and physical protection by clays have been invoked as additional controls over microbial processing of soil organic matter (Swift *et al.*, 1979; Richards, 1987; Tate III, 1995; Mary *et al.*, 1996).

The physico-chemical properties of soils can directly influence the structure, spatial distribution and activity of microbial populations and enzymes in soils, which are potential early indicators of soil health and quality (Schnurer *et al.,* 1985; Dick, 1994). Each of the organic and microbial fractions in soil has special influence on enzyme activity (McLaren, 1975; Skujins, 1976). Soil microbial activity contributes to the regulation of soil carbon storage and ecosystem productivity (Bauhus *et al.,* 1998).

Most of the forest lands on the earth are under tremendous pressure of developmental activities such as urbanization and industrialization which cause undesirable changes in various physico-chemical, biological and biochemical characteristics leading to the problem of soil degradation globally. It has been reported that many of the world's ecosystems are in various state of decline as evident by erosion, low productivity and poor water quality caused by forest clearing, intensive agricultural production and continuous use of soil resources that are not sustainable (Kennedy and Smith, 1995). This includes loss of structure and an increase in soil compaction (Vazquez *et al.,* 1993) commonly associated with soil organic matter reduction and ash formation (Prieto-Fernandez *et al.,* 1993). In the past, the disturbance of natural forest was less and was confined to areas of some country only. But at present, the condition has been changing due to increase in human population, environmental pollution, decline in soil quality, lower fertility level and encroachment of public to protected areas, environmental effects such as global warming. The problem of soil degradation and disturbance becomes the subject of environmental debate which attracts attention from the scientific communities of the world (Sehgal and Abrol, 1994) for successful restoration and rehabilitation of the disturbed areas.

It is widely accepted that each type of vegetation community harbors characteristic soil mycoflora population, particularly the assemblage of the more predominant species. However, there is no evidence that the microfungi form discrete communities; rather they constitute a continuum, the species composition gradually changing with differences in the vegetational cover and soil characteristics. Several studies have shown that there is a shift in the species composition of the soil mycoflora which parallels pioneer vegetational succession (Brown, 1958; Cooke and Lawrence, 1959; Pugh, 1963; Wohlrab *et al.*, 1963).

Soil physical conditions play an important role in determining the environment in which biological processes take place (DeVos *et al.*,1994) while chemical characteristic determines maximum quality of a particular soil (Hassink,1997). Nowadays, much attention is paid to the study of biological processes in soil because of the reason that nutrient transformation processes make soil a dynamic part of the biosphere with the vital role of soil microorganisms and invertebrates (DeVos *et al.*, 1994). The biological and biochemicals are important indicators which can sensitively respond to anthropogenic and environmental stresses on soil as dynamic system (Filip, 1998). Therefore, research studies in the last two decades have revealed that soil quality may be assessed using selected indicators related to soil microorganisms (Staddon *et al.*, 1998). The important microbiological parameters consist of population dynamics, diversity, soil respiration, microbial biomass carbon and nitrogen and enzyme activities. These parameters are considered as bio-indicators of soil quality and use as group of indices as they are quickly responsive and sensitive to changes occurring in the soil environment and could illustrate the effects of anthropogenic activities and other disturbances in soil (Dick, 1994; Turco *et*

al., 1994; Filip, 1998; Trasar-Cepeda *et al.,* 1998; Bending *et al.,* 2000; Palma *et al.,* 2000). Of the environmental factors which define the fundamental niche of terrestrial fungi, water is probably the most important (Cooke and Whipps, 1993). Water potential affects fungal "individual" through both impact on germination, radial growth, sporulation (e.g. Griffin, 1994; Dix and Webster, 1995; Bruchl and Kaiser, 1996) and mycelial cord development (Donnelly and Boddy, 1997). Water potential also affects the distribution and abundance of fungal population (Shameemullah *et al.,* 1971; Dowding and Widden, 1974; Bissett and Parkinson, 1979a and b; Widden, 1986a; Eastburn and Butler, 1988; Cooke and Whipps, 1993; Domsch *et al.,* 1993; Dix and Webster, 1995) and the interactions between fungal species (Eastburn and Butler, 1991; Marin *et al.,* 1998a and b).

The microbial communities are influenced by soil moisture and temperature (Campbell and Biederbeck, 1976), physical disturbance of soil (Doran, 1987) and interaction with soil fauna (Beare *et al.,* 1992).

Forest harvesting alters the amount of soil organic matter (Mattson and Smith, 1993), soil temperature, soil moisture and pH (Bormann and Likens, 1979) all of which affect microbial activity (Harvey *et al.,* 1980; Hendrickson *et al.,* 1985; Entry *et al.,* 1986).

The accumulation of organic nitrogenous compounds is more evident in forest soils (Pritchett and Fisher, 1987) and nitrogen mineralization is markedly retarded in soils like those developed over granite due to the abundance of organo-aluminium compounds, which present a high resistance to microbial mineralization (Gonzalez-Prieto *et al.,* 1991,

1996). It is, therefore, important to know the composition, variability and bioavailability of the nitrogenated substrate of soils.

Inputs of acidifying substances to forest ecosystems have increased in recent decades (Fowler *et al.,* 1999) and have led to increased acidification processes in forest soils on sensitive sites (Blaser *et al.*, 1999). In particular, inputs of N can alter forest soil chemistry significantly and in addition to acidification, nitrate can leach into the ground water resulting in relative shortages of nutrient elements (Magill *et al.*, 1997; Emmett, 1999; Rennenberg and Gessler, 1999).

Organic C and N are associated in soil organic matter and their pattern of distribution down the soil profiles studies are similar and closely followed the texture and bulk density data. The depth wise distributions of C and N underline the importance of sampling at depth to characterize soil organic storage and also indicate that C and N accumulation down the profile can be subject to mathematical analysis and modelling once the pattern of vertical distribution is established (Arrousays and Pelissier, 1994).

Understanding of the variation in fungal population in time and space has paramount importance due to its relevance in biodiversity and role of fungi in regulating population of other organisms and ecosystem processes. Therefore, research on microbial diversity and activity may provide some advance evidence of ecosystem degradation. Little is known of the importance of microbial diversity in the functioning of soils (Klein *et al.*, 1986; Pankhurst *et al.*, 1996; Giller *et al*, 1997). The amount and diversity of microorganisms are extremely important for soil metabolic processes, because they act on the organic matter decomposition contributing for the soil fertility (Andrea *et al.*, 2000). It is important to determine optimum diversities of soil microbial populations of both

natural and agricultural systems for their sustainable management. In order to maximize the beneficial effects of microbial activity, there is a need for greater understanding of factors influencing microbial diversity and activity. The relationship between microbial diversity, microbial activity, plant quality, and ecosystem sustainability of disturbed and rangelands are still poorly understood.

North eastern region of India is unique in many respects such as, rich floristic composition, high annual precipitation, undulated topography and varying types of forest ecosystems. This region experiences high rainfall of a monsoonic type which is followed by a dry winter and a brief summer. It is known to possess diverse forest types from Tropical Evergreen forests to Moist Alpine Shrub Forest and occupy 59.9 % of its geographical area, which is much higher than all India average of 28 %. The state of Meghalaya, in particular, is endowed with rich natural vegetation, which ranges from tropical type to sub-tropical type of vegetation or evergreen to mixed deciduous types of forests. It lies between 25° 00' and 26°15' N latitude and 89° 45' and 90°47' E longitude.

Most of the original vegetation cover of Meghalaya have been interfered with and modified by man to a great extent and if the process continues unabated, very soon the state will be devoid of true natural vegetation. The result is that the entire ecosystem of the region would undergo changes and create a chaotic condition.

The information on microbial diversity from northeast India is sporadic, as compared to other parts of India and no comprehensive study has so far been made to explore and conserve the microbial diversity of northeast India as a whole particularly in disturbed and undisturbed forests of Meghalaya. It was, therefore, proposed to investigate

the microbial diversity and their activities of disturbed and undisturbed forest soil of Meghalaya. The present studies were carried out under the following heads:

1. Diversity of soil microorganisms (fungi and bacteria)
2. Soil microbial biomass carbon (C_{mic})
3. Soil enzymes activities (dehydrogenase, urease and phosphatase)
4. Physico-chemical characteristics of the soil viz., temperature, moisture content, pH, organic carbon, total nitrogen, avaiable phosphorus and exchangeable potassium

Study area, climate and vegetation

Meghalaya known as "the abode of clouds" is endowed with rich natural vegetation. It lies between 25°10' and 26°50' N latitude and 85°49' and 92°52' E longitude. Shillong the capital of Meghalaya is situated at an altitude of 1496 m above sea level. It has a total geographical area of 22,429 sq. km. The total estimated forest of the state is 8,514 sq. km of which only 722.36 sq. km are directly under the control of the State Forest Department. The remaining areas are managed by the respective District Councils of Khasi hills, Jaintia hills and Garo hills as per provisions of the Sixth scheduled of the Constitution of India. Except the reserved forest areas and protected forests in and around, Shillong being managed by the department in arrangement with the District Councils, the rest of the forest areas are subjected to the primitive agricultural practice of shifting cultivation or slash and burn method especially in Garo hills. The forest of Meghalaya can broadly be grouped under the tropical type and the temperate type, mainly based on the altitude, rainfall and dominant species composition.

Climate and geomorphic factors largely influence and determine the type of vegetation. Meghalaya is rich in forest resources. The richness and variety of vegetation ranging from sub-temperate to tropical is due to diverse topography, variation in rainfall and differential climate of the state. Natural vegetation plays an important part in the maintenance of ecological balance. Trees are important among preserving factors. The natural vegetation in Meghalaya does not only determine the ecosystem of the region but of the whole of northeast India and the Gangetic valley.

Study sites

Two sub-tropical forest stands were selected viz., disturbed and undisturbed forests of Law Lyngdoh located at Nongkrem, 20 km away from Shillong at an altitude of 1786 msl. The geographical position of the study site is between latitude 25°34′ N and longitude 91°46′ E. The undisturbed forest is a protected area with dense vegetation where rituals are performed every year according to religious belief.

Climate

The climate of Shillong is cool with winter temperature going down to 5.29° C in the month of January 2002 and 6.50° C in the month of December 2003. The lower temperature resulted in frost which could be seen sometime early in the morning during December to February. The maximum temperature in the first year was 24.85° C in the month of August and in the next year it was 23.90° C in the month of June and July. The average maximum temperature was 21.49° C and minimum temperature was 9.49° C in the first year. In the second year average maximum temperature was 21.05° C and minimum temperature was 11.05° C. In the first year, the rainfall had average maximum of 250.698 mm and average minimum rainfall of 4.089 mm. In the second year, the rainfall had an average maximum rainfall of 651.510 mm and an average minimum rainfall of 3.048 mm. The relative humidity ranged between 53.5 % to 90.27 % (Fig. 1).

Plate 1 Disturbed Forest

Plate 2 Disturbed Fallow Land

Plate 3 Disturbed Fallow Land

Plate 4 Undisturbed Forest

Plate 5 Undisturbed Forest

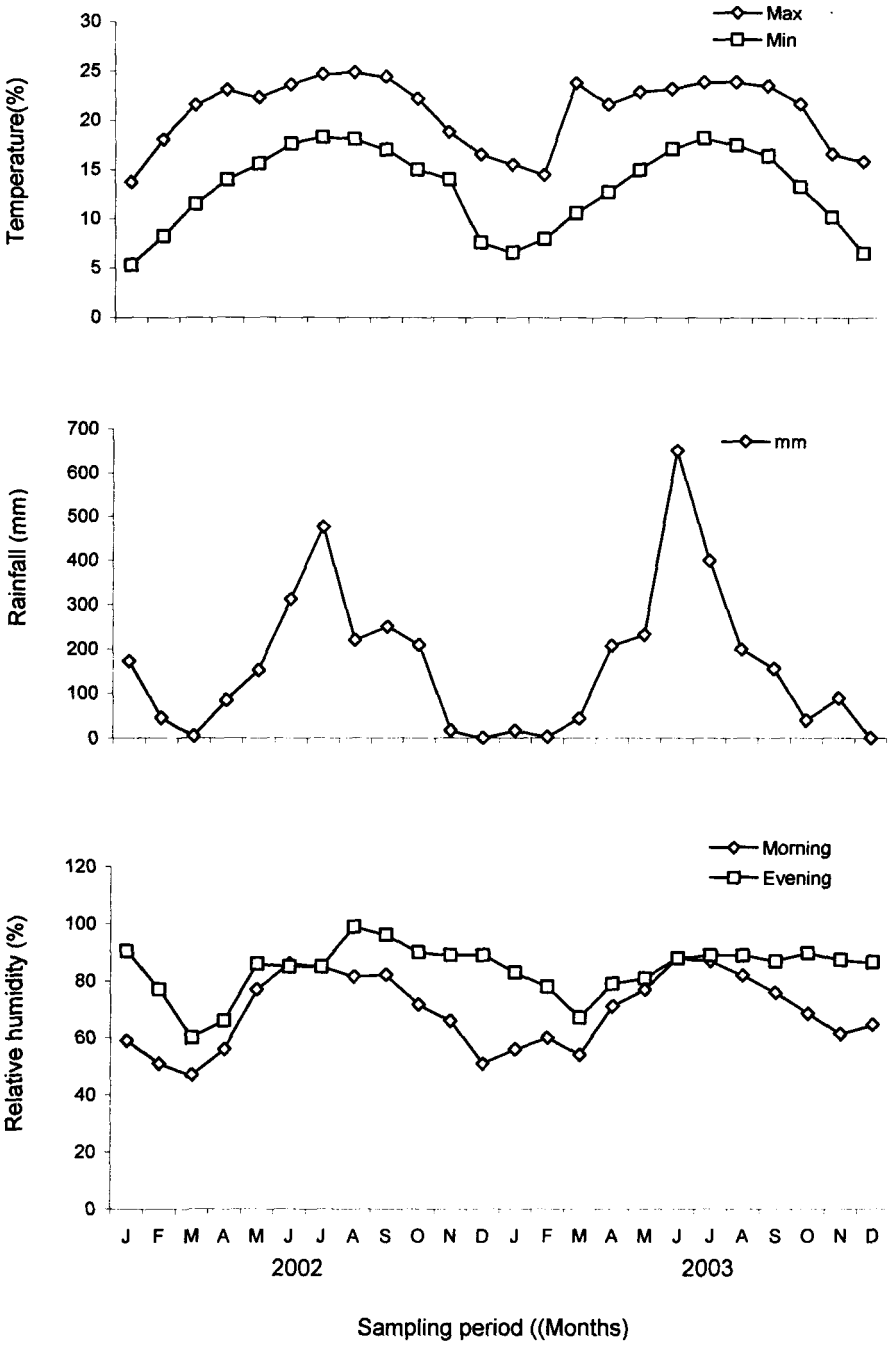

Fig. 1 Average temperature, rainfall and relative humidity during the study periods 2002-2003

The State of Meghalaya is directly influenced by the southwestern monsoon that originates in the Bay of Bengal. The typical summer season is not found in Meghalaya. However, based on the meteorological condition the year can be divided into the following seasons:

Spring season: The period from the middle of February up to middle of April covers the spring season which experiences very high wind velocity with less humidity and moderate temperature.

Rainy season: The rainy season extends from the middle of April to the middle of October. However, the early period of the rainy season is a bit warm representing summer, while the later period of the season is comparatively cool. Due to high rainfall the humidity is also high during rainy season.

Winter season: The winter season starts at the end of October and continue up to middle of February.

Vegetation

In the undisturbed forest the dominant tree species are *Cinnamomum glanduliferum, Elaeocarpus lancifolius, Eurya japonica, Lithocarpus dealbatus, Eleagnus pyriformis, Myrica esculenta, Pinus kesiya* and *Schima khasiana* together with few shrubs and herbs. In some places, ferns grow thickly and cover the ground area. On the other hand, the disturbed forest is thinly populated with few pine trees (*Pinus kesiya*) and species such as *Eleagnus pyriformis* and *Myrica esculenta* are found to be dominant one. The ground area is covered with grasses and herbs along with few shrubs. The

vegetation is not as dense as that of the undisturbed site. Besides the dominant species, other species such as *Symplocus crataezoides, Hedera helix, Pyrus fastia, Phyllanthus glaucus, Eleocarpus simplex, Symplocus javanica, Rhus acuminata, Morus australis, Viburnum foetidum, Polygala arillata, Perrea kingii, Docynia indica, Eurya japonica, Persea duthiei, Tetrastigma serrulutum, Symplocus specata, Ficus merifolia, Mangliitia insignis, Engelhardtia spicata, Prunus* sp., *Phlocazanthus eubiflorus, Photinia integerrima, Lindira mebstonaacia, Sarcococca pruniformis, Meliosma wallichi, Litsea elongate* and *Lindira nacusua* were present in the two study sites.

Soil texture

The soil texture in the study was found to consist mostly of sand and loam (Table 1).

Table 1 Site characteristics of disturbed and undisturbed forests at three different depths (0-10cm, 10-20cm and 20-30cm)

Soil Properties	Disturbed Forest			Undisturbed Forest		
	0-10cm	10-20cm	20-30cm	0-10cm	10-20cm	20-30cm
Clay (%)	5.40	3.23	3.30	13.70	5.33	11.40
Silt +Clay (%)	13.94	15.94	14.06	32.60	19.76	25.85
Silt (%)	8.54	12.71	10.76	18.90	14.54	14.48
Soil textural class	Sandy	Loamy sand	Loamy sand	Sandy loam	Loamy sand	Sandy loam

MATERIALS AND METHODS

Soil Samplings

Soil samplings were done from both the study sites at monthly intervals for a period of two years i.e., 2002 and 2003. The samples were collected randomly at three different depths i.e., 0-10cm, 10-20cm and 20-30cm from five places at each study site and mixed thoroughly to make a composite sample. This was done to minimize local variation in the microbial populations. Various estimations were carried out within 24 hours of collection. Collections were done in aseptic conditions and the samples were brought to the laboratory and stored at a temperature of 4^{o} C.

Isolation of Fungi and Bacteria

Serial dilution plate method (Johnson and Curl, 1972) was followed for the isolation of fungi and bacteria using rose bengal agar medium (Martin, 1950) and nutrient agar medium (Difco manual, 1953) respectively. One gram of soil sample was taken into 250 ml conical flask containing 100 ml of sterilized distilled water to give 1:100 dilutions. To prepare homogenous solution, the flask was swirled for 15 minutes. Then 10 ml of this solution was transferred to another flask containing 90 ml of sterilized distilled water with the help of sterilized pipette to get 1:1000 dilution. 10 ml of this solution was again transferred to another flask containing 90 ml of sterilized distilled water to get 1:10,000 dilution.

For the isolation of fungi, 1 ml of the soil dilution (1:1000) was transferred into a Petri dish containing rose bengal agar medium, which was then rotated gently to disperse

the suspension. Three replicates were maintained for each sample. The inoculated Petri dishes were incubated upside down at 25 ± 1^0 C for 5 days in a BOD incubator. Colony forming unit (CFU) of fungi was estimated by counting the number of fungal colonies. The CFU of fungi per gram soil was calculated on the dry weight basis.

For isolation of bacteria, 0.5 ml of the soil dilution (1: 10,000) was transferred to a Petri dish containing nutrient agar medium, which was then rotated gently to disperse the suspension. Three replicates were maintained for each sample. The inoculated Petri dishes were then incubated upside down at 30 ±1^0 C in bacteriological incubator. Colony forming unit (CFU) of bacteria was estimated by counting the number of bacterial colonies. The CFU of bacteria per gram soil was calculated on the dry weight basis. The whole process was carried out in aseptic conditions.

Estimation of Fungal Population

$$CFU\ of\ fungi\ /\ g\ D_w = \frac{Number\ of\ colonies \times dilution\ factor \times inoculum}{Dry\ weight\ of\ soil \quad (g)}$$

Where, D_w = Dry weight of the soil (g)

The fungal species were identified on the basis of their morphology and reproductive structures, consulting monographs of Subramaniam (1971), Barnet and Hunter (1972), Ellis (1972) and Domsch *et al.* (1980). The following formula was used for the determination of relative abundance of fungal species:

$$Relative\ abundance\ (\%) = \frac{Total\ number\ of\ the\ colonies\ of\ individual\ species}{Total\ number\ of\ colonies\ of\ all\ species} \times 100$$

Estimation of Bacterial Population

For estimation of bacteria population and relative abundance the above formulae were used.

Composition of the Media used for the Isolation of Fungi and Bacteria

Fungi

Rose Bengal Agar medium (Martin, 1950)

Agar	20 g
KH_2PO_4	10 g
$MgSO_4.7H_2O$	0.5 g
Peptone	5 g
Dextrose	10 g
Rose Bengal	3.3 ml
Streptomycin	30 mg
Distilled water	1000 ml

Bacteria

Nutrient agar medium (Difco manual, 1953)

Agar	15 g
Beef extract	3 g
Peptone	5 g
NaCl	8 g
Distilled water	1000 ml

(The final pH of the medium was adjusted to 7.3)

Statistical analysis

Using the data obtained, the following indices of fungal and bacterial species were assessed:

(1) Index of general diversity (H'); Shannon and Weaver (1949) cited in Odum (1971)

$$H' = -\sum (n_i / N \log_e n_i N)$$

Estimation of microbial biomass carbon (C_{mic})

The soil microbial biomass carbon was estimated by chloroform fumigation incubation (FI) method of Anderson and Ingram (1993). The soil was sieved through 0.2 mm mesh sieve to remove stones, coarse roots and all visible litters. 10 g of each sample was taken in a beaker and was placed in a vacuum desiccator containing 30 ml of alcohol free chloroform in shallow dish. The lid was disclosed and sealed and the vacuum was used till the last trace of chloroform evaporated and thereafter the desiccator was kept in the dark for 5 days at 25° C. After 5 days, the fumigated soil (ct_1) sample was extracted with 50 ml of 0.5MK_2SO_4 and was shaken for 30 minutes. Weight another 10 g of each sample for unfumigated extraction (ct_2). The sample was then kept in a water tight extraction bottle (125 ml) and extracted directly without fumigation with 50 ml of 0.5 M K_2SO_4 and was shaken for 30 minutes. The extracted soil was then filtered through Whatman filter paper No. 42. To a 4 ml filtrate, 1 ml of 0.0667 M potassium dichromate and 5 ml of concentrated sulphuric acid were added. The mixture sample was then preheated at 150° C for 30 minutes. Two blanks were prepared i.e. one preheated at 150° C for 30 minutes and the other without heating. The digested sample was then transferred to a 100 ml conical flask and to it 0.3 ml of indicator solution (O-phenanthroline

monohydrate) was added. The sample was then titrated with acidified ferrous ammonium sulphate solution. The end point was a color change from green/ violet to red. Three replicates were maintained in each case. For blank, 4 ml of 0.5 M K_2SO_4 solution was added in place of sample filtrate solution.

The microbial biomass C was calculated as follows:

Organic C (%) = {(A x M x 0.003)/g} X (E/S) X 100

where, M = Molarity of ferrous ammonium sulphate (=0.033M)

$A = (Ml_{HB} - Ml_{sample}) \times \{(Ml_{UB} - Ml_{HB})/Ml_{UB}\} + (Ml_{HB} - Ml_{sample})$

G = Dry soil mass (g)

E = Extraction volume (ml)

S = Digest sample volume (ml)

HB=Heated blank

UB=Unheated blank

Microbial biomass C = (Extracted ct_1 *- Extracted* ct_2 *) X 2.46*

Indicator solution:

1.485 g of O-phenenthroline monohydrate was mixed with 0.669 g of ferrous sulfate hexahydrate and to this 100 ml of distilled water was added.

Estimation of enzyme activities

Dehydrogenase activity

2-3-5- Triphenyl tetrazolium chloride (TTC) reduction technique (Casida, 1977) was used for the estimation of dehydrogenase activity in soil. One gram of fresh soil was

taken in a test tube. The soil was then mixed with 0.1 g of calcium carbonate ($CaCO_3$) and 1 ml of 1 % TTC solution. The mixture was then shaken and plugged with a rubber stopper and incubated at 30^0 C for 24 hours in an incubator. Three replicates were maintained in each case. The resulting slurry was transferred on Whatman filter paper No.1 and extracted with successive aliquots of concentrated methanol. The volume of the filtrate was made to 50 ml by adding methanol. The optical density of the filtrate was read at 485 nm in Hitachi Spectrophotometer (220), using methanol extract as a blank. The activity was representing in terms of concentration of Formazan, which was calculated by a standard curve of triphenyl formazan in methanol. Dehydrogenase activity per gram dry soil was expressed in terms of milligram formazan per gram dry soil per hour.

Urease activity

The urease activity was measured by the method of McGarity and Myers (1967). One gram of fresh soil was taken in a 100 ml volumetric flask and to it 1 ml of toluene was added. It was then allowed to stand for 15 minutes to permit the complete penetration of toluene into the soil. Thereafter, 10 ml of buffer (pH 7) solution and 5 ml of 10 % urea solution were added. The flask was shaken and incubated at 37^o C for 3 hrs in an incubator. Whereas in control set, 10 ml of distilled water was added instead of urea solution. After incubation, the volume was made up to 100 ml by adding distilled water. The content in the flask was mixed thoroughly and was filtered through Whatman filter paper No. 5. Indophenol blue method was adopted for the measurement of ammonia released as a result of urease activity. O.5 ml of the filtrate was taken in 25 ml volumetric flask and to it 5 ml of distilled water was added. The mixture in the flask was treated with

2 ml of phenolate solution and 1.5 ml of sodium hypochlorite solution containing 5 % of active chlorine. The final volume was made up to 25 ml by adding distilled water. The optical density was read in a Hitachi (220) spectrophotometer at 630 nm. The amount of NH_4^+-N released was calculated by a reference-calibrated curve and was expressed as NH_4^+-N mg per gram dry soil per three hours.

Preparation of phenolate solution

20 ml of phenol solution + 20 ml of caustic soda solution were diluted to 100 ml with distilled water.

Phenol solution

62.5 g of phenol was dissolved in minimum volume of methanol denaturated alcohol. To this 18.5 ml of acetone was added and the mixture was made up to 100 ml with alcohol.

Caustic soda solution

27 g of sodium hydroxide were dissolved in 100 ml of distilled water. Both the solutions were kept in a freeze.

Phosphatase activity

Phosphatase activity was measured by the method of Tabatabai and Bremner (1969). 0.1 g of air-dried soil was taken into a 50 ml conical flask. Then 4 ml of modified universal buffer (pH 6.5), 0.25 ml of toluene and 1 ml of 0.115 M p-nitrophenyl phosphate (PNP) solution were added to the flask. The flask was swirled for few seconds

and then incubated at 37° C for one hour. After incubation, 1 ml of 0.5 M calcium chloride and 4 ml of 0.5 M sodium hydroxide were added to the mixture. The soil suspension was filtered through Whatman filter paper No.1. The optical density of the filtrate was measured at 430 nm in Hitachi (220) spectrophotometer. Blank was maintained similarly without soil. The phosphatase activity in terms of concentration of p-nitrophenyl in each sample was calculated by a standard curve of p-nitrophenol in water and was expressed as mole of p-nitrophenol released per gram dry soil per hour.

Analysis of Physico-Chemical Characteristics of Soil

Soil Texture

Soil texture was determined by Boyuous soil hydrometer method on air dried samples. 100 g air dried (2 mm sieved) soil sample was taken into a 500 ml glass beaker (borosil) and saturated with distilled water. 10 ml of 5 % calgon solution (sodium hexa-metaphoshate) was added into the beaker and stirred for one minute with a clean glass rod. The soil suspension was then transferred into a soil cup (400 ml) and mixed thoroughly for two minutes by using a tissue homogenizer. The suspension was then transferred carefully into a measuring cylinder using distilled water and made the volume up to 1350 ml after inserting the soil hydrometer. The measuring cylinder was then turned upside down for at least ten times after closing the mouth with a rubber band. A few drops of amyl alcohol were added to diffuse the bubbles, then the soil hydrometer was inserted at 20 seconds and reading was taken at 40 seconds. A clinical thermometer was also inserted along with the hydrometer to record the temperature of the suspension. The cylinder was again turned upside down for ten times and kept undisturbed. After 2

hours, a few drops of amyl alcohol were added and the soil hydrometer reading was recorded along with temperature of the suspension. After temperature correction of the hydrometer readings, the percent sand, silt and clay contents were calculated as follows:

$$Sand\ (\%) = \frac{Hydrometer\ reading\ at\ 40\ seconds}{Weight\ of\ sample\ (100\ g\ for\ sandy\ soil)} \times 100$$

$$Clay\ (\%) = \frac{Hydrometer\ reading\ at\ 2\ hours}{Weight\ of\ sample\ (100\ g\ for\ sandy\ soil)} \times 100$$

$$Silt\ (\%) = 100 - (\%sand + \%\ clay)$$

Temperature

Soil temperature was measured by using soil thermometer. The soil temperature was taken at monthly intervals at different soil depths.

Moisture Content

The moisture content of the soil was determined by oven dry basis. 10 g of soil was dried in a hot air oven at 105°C for 24 hrs and the dry weight was taken. The percentage moisture content was calculated as follows:

$$Moisture\ content\ (\%) = \frac{W_1 - W_2}{W_1} \times 100$$

where, W_1 = *initial weight*

W_2 = *final weight*

Three replicates were maintained for each sample.

pH

10 g of soil was taken in a beaker containing 50 ml of distilled water and stirred for 15 minutes on a magnetic stirrer. The pH was read by using electronic digital pH meter.

Organic carbon

Soil organic carbon was estimated by the method of Anderson and Ingram (1993). One gram of sieved soil (through 0.2 mm sieve) was taken in a 100 ml conical flask (if the soil was dark, or was suspected to be high in organic matter use about 0.5 g). To this 10 ml of 5 % potassium dichromate solution was added and allowed to completely wet the soil or dissolved the standards. 20 ml of sulphuric acid was then added and swirled the mixture gently and then allowed to cool it. To this 50 ml of 0.4 % barium chloride was added, swirled the mixture thoroughly and then allowed to stand overnight, so as to leave a clear supernatant solution. The blank was run without soil. The supernatant was then transferred into a colorimetric cuvette and the optical density was measured at 600 nm using Hitachi (220) spectrophotometer.

$$\textit{Organic carbon}\ (\%) = \frac{(K \times 0.1)}{(W \times 0.74)}$$

where, W = Weight of the soil.

Total nitrogen

Kjeldahl's method was followed for the determination of total nitrogen (Jackson, 1973). One gram of air dried sieved soil (through 0.2 mm sieve) was taken in a kjeldahl digestion flask and 6 ml of sulphuric acid was added. To this one kjeltablet was added and then the soil sample was digested in a block digester for about half an hour (till the color turns green). The flask was allowed to cool and diluted with 50 ml of distilled water. The solution was then filtered with Whatman filter paper No. 1. After this, distillation was done in a kjeldahl distillation unit with 10 ml of sample solution and 10 ml of 40 % sodium hydroxide. The distilled sample was then collected in a beaker with 5 ml of boric acid indicator till the pink color turned greenish. It was then titrated against N/140 hydrochloric acid. The titration was stopped when the color turned pink.

$$N(\%) = \frac{(T - blank) \text{ x solution volume}}{10^2 \text{ x } aliquot\ vol. \text{ x } sample\ weight}$$

where, T = Burette reading

Preparation of boric acid indicator

10.0 g of boric acid was dissolved in 1000 ml of distilled water. To this 10 ml of bromocresol green (dissolved 0.1 g bromocresol green in 10 ml methanol) and 7 ml methyl red (dissolved 0.07 methyl red in 7 ml of methanol) were added.

Available phosphorous

The available phosphorous was measured by following molybdenum blue method (Allen *et al.*, 1974). Weighed 5 g of air-dried sieved soil in a polythene bottle. To this 100 ml of Olsen's reagent was added and it was shaken for 30 minutes on a rotatory shaker. The mixture was filtered through Whatman filter paper No. 44. 10 ml of sample was pipetted into 50 ml volumetric flask. The sample was diluted about two third of the flask. To this 2 ml of ammonium molybdate and 2 ml of stannous chloride were added and then the final volume was made up to 50 ml by adding distilled water. Control was maintained without soil sample. After 30 minutes the optical density was read in a Hitachi (220) spectrophotometer at 700 nm. The calibration curve was prepared from the standard and was used to determine mg P in the same aliquot.

$$P\,(\%) = \frac{C\ (mg) \times solution\ volume\ (ml)}{10^3 \times aliquot\ (ml) \times sample\ weight\ (g)}$$

Preparation of reagents

Olsen's reagent

Dissolved 210 g of sodium bicarbonate in water in aspirator and to this 100 ml of 1M sodium hydroxide was added. The final pH was adjusted to 8.5 ± 0.05.

Ammonium molybdate sulphuric reagent

Dissolved 25.0 g of ammonium molybdate [$(NH4)_5\ MO_7O_{24}\ 4H_2O$] in about 200 ml of water by warming slightly. 280 ml of concentrated sulphuric acid was added (with mixing and cooling) to about 400 ml of water and the solution was mixed thoroughly and

distilled water was added to make up to 1 litre when it was cool. The mixture was then stored in a dark place.

Preparation of stannous chloride

10 g of stannous chloride was added to 25 ml of conc. HCl. 1 ml of the stock solution was taken and 330 ml double distilled water was added to it.

Exchangeable potassium

Exchangeable potassium was measured by using the method of Allen *et al.* (1974). Weighed 10.0 g of air-dried sieved soil in a 500 ml conical flask. To this 250 ml of ammonium acetate solution was added. The mixture was then shaken in a rotatory shaker for 1 hour and was kept overnight. The mixture was then shaken for 5 minutes and was filtered through Whatman filter paper No. 44. The blank was run without soil sample. Potassium was then determined in a flame photometer.

$$K\,(\%) = \frac{C\,(ppm) \times solution\ volume\ (ml)}{10^4 \times sample\ weight\ (g)}$$

Preparation of reagent

Extractant (Ammonium acetate)

575 ml of glacial acetic acid was added to 200 ml of water in a 10 litre container (preferably polythene). To this 600 ml of 0.880 N of ammonia solution was added slowly by cooling. The mixture was then diluted to 10 litre water by adding distilled water. The final pH was adjusted to 7± 0.05 by adding either few drops of acetic acid or ammonia solution.

REVIEW OF LITERATURE

Microbial populations (Fungi and Bacteria)

Studies on soil fungi have received much attention since the problem of mycological investigation was probed in by Adametz (1886). The microorganisms in the soil have been subjected to intensive studies during the recent years. The microbial population in soil is of ecological importance because of the essential role that microorganisms play in the conservation and cycling of plant nutrients. Waksman (1916) reviewed the early literature on soil fungi and concluded that each type of soil habitat has a distinct fungal flora and distribution of these organisms is influenced by abundance and nature of organic matter in soil. Owing to this, the microbial populations in the soils have been studied by several workers (Waksman, 1927; Thakur and Morris, 1928; Warcup, 1951). However, during recent years various workers have attempted to correlate the microbial population with season, physico-chemical characteristics, depth of soil and surface vegetation.

Jensen (1931) found that *Penicillium* and *Trichoderma* species were common in acidic soil and *Mycogone niger* and *Cercospora agricola* were common in alkaline soils.

Feher (1933) studied the seasonal variation in bacterial numbers in several European forest soils. He observed that the majority of bacteria in the soil were either large cocci or coccobacilli, some of which resembled *Azotobacter*.

Warcup (1951) stated that acidity and temperature play important role in the distribution of fungal species. He isolated the genus *Trichoderma* repeatedly from the acidic soils.

Saksena (1955) did pioneer work in India on the ecological factors governing the distribution of fungi. He observed that there is a direct correlation between fungal population and phosphate and nitrate contents of the soil.

Saksena (1955) and Warcup (1957) found a marked decrease in viable propagules of soil in summer. They further observed that moisture, was favorable for the growth of fungal as long as there was no water logging.

Kendrick and Burges (1962) stated that fungal communities can be highly variable between adjacent pine needles as a consequence of the grazing activities of invertebrates.

Roy and Dwivedi (1962) studied the fungal flora of different grass species situated in the same field and found variation in the population of fungal flora which they thought was due to variation in the vegetation.

Though several workers have reported that there is a decrease in microbial population with the increase in depth of the soil, yet Apinis (1963) observed a relatively uniform distribution in microfungal population throughout the soil horizons.

Griffin (1963) reported that the soil texture had affect oxygen and CO_2 relationships as well as moisture content and nutrient availability, all of which have been suggested to influence the composition of microfungal community.

Mishra (1966a) studied the seasonal variation in fungal flora of grasslands of Varanasi (India) and recorded a seasonal effect on the prevalence of different fungal species. Mishra (1966a, b and c) in his studies on the seasonal distribution and variation in the fungal population suggested that this is due to the soil organic matter and moisture content. He pointed out that temperature does not play direct role except that it affects the

moisture status of the soil, thus regulating the population. Mishra (1967) observed a positive correlation between the fungal population and temperature and moisture content of the soils.

Latter *et al.* (1967) in their comparative study on four moorland soils in England observed that the marked differences in the fungal flora was due to pH, temperature, water logging and oxygenation conditions in these soils. In addition, they noticed that *Trichoderma* sp. and *Penicillium* sp. and unidentified white sporulating fungal species were isolated with greater frequency in summer than in other seasons.

Hattori (1967) found in a garden soil of Serndai, a high correlation in fungal flora at various points in November and February, although the flora at the same study site varied widely in November and June. He concluded that fungal flora at various study sites in a field are not uniform and always vary widely.

Mishutin and Mirzoyeva (1968) studied the soil types of some mountain ranges in the Caucasus and Middle Asia. They found that the effect of altitudinal zonality on the composition of soil microflora is the same as that of latitudinal zonality. At higher mountains, where the average annual soil temperature is low, the number of bacteria decreases. Some soil types have characteristics profiles and they possess specific pattern in distribution of organic matter. These differences affect the microbiological profile of different soils.

Campbell *et al.* (1971) found that the temperatures, which fluctuated daily, reduced microbial organisms in the soil especially the population of bacteria. A reduction in the aggregate microbial population had relatively little effect on N mineralization due

to sheer numbers of organisms responsible for mineralization. Soil moisture directly affects microbial activity and in turns affects soil nitrogen mineralization.

Prakash and Khan (1971) and Tyagi (1973) showed that soil moisture content, seasonal weather fluctuations and depth were important factors governing the frequency and population of soil microflora.

Mishra and Kanaujia (1972) analyzed the ecological aspects of soil fungi in relation to climatic condition, vegetation and soil physico-chemical characteristics and found that the organic matter, pH, soil depth and season played a critical role in the distribution of mycoflora.

Disturbance is known to affect soil fungal communities. Various authors have attempted to assess the effects of disturbance on fungal communities. Wicklow (1972), Zak (1992) and Frey *et al.* (1999) reported that intermediate disturbance has been shown to maximize diversity in a model microbial system.

Chen and Alexander (1973) demonstrated the existence of soil bacteria susceptible and tolerant to desiccation. However, Boylen (1973) tried to select drought-resistant strains for a single species of common soil bacteria (*Arthrobacter*), but could not obtain a completely resistant progeny. Factors other than soil characteristics that may determine decrease in biomass by drying are the inherent properties of the soil microbial populations.

Dowding and Widden (1974) investigated the relationship between fungi and their environment in different tundra regions and observed the ubiquitous presence of the genus *Cladosporium.*

Holding *et al.* (1974) noted that the viable counts of bacteria decreased with depth and was correlated with the decrease in moisture content, organic matter and inorganic nutrients. They further stated that the decrease in numbers with the increase in depth was less marked for the anaerobes than the aerobes.

Dadalauri (1975) pointed out that the distribution of soil fungi is determined by the combined effect of moisture, temperature and organic carbon. Wong (1975) reviewed the literature on soil fungi and concluded that distribution of soil fungi is based primarily upon various soil factors. According to him moisture and pH are most important factors.

Bacteria occupy a wide variety of habitats and perform many ecological functions. The bacteria produce a vast array of chemical metabolites, many of which affect other organisms (Lynch, 1976).

Jones and Richards (1977) reported that the vegetation type has no effect on the total number of bacterial and actinomycetes populations.

Alexander (1977) has shown that the number of bacteria found in soils is usually higher than those of other groups and is the principle agent for the cycling of nitrogen, sulphur and phosphorus.

Rehder and Schafer (1978) reported that the nutritional conditions and microclimate factors must have a decisive influence on the distribution of individual species. However, the distribution of the genus *Mortierella*, which was isolated at all their study sites, appeared to be almost independent of substrate or environmental conditions.

Martinez and Ramirez (1979) investigated the temporal and depth-wise distribution of microorganisms, ecological aspects of soil fungi in relation to varying

cover vegetation, climatic conditions, and physico-chemical characteristics in an acid beech forest. They observed that the fungal population was generally found to be highest in upper layer and with an increase in depth, the fungi per gram dry soil, organic matter content gradually decreased and season seemed to play an importance role in distribution of mycoflora.

Upadhyay and Rai (1979) reported that the genus *Trichoderma* was recorded frequently and in high population from forest and some other soil containing high organic matter and low pH. Similarly Baruah *et al.* (1980) recorded the *Trichoderma viride* from acidic soils.

Tate and Terry (1980) indicated that within the moisture ranges detected in the surface soils, increased moisture stimulated microbial activity, whereas moisture ranges reach saturation, increased moisture inhibited aerobic activities and stimulated anaerobic process.

Alexander (1980) showed that the activity of many common bacteria is inhibited or suppressed by strong acidic conditions in soils but the relative abundance of fungi rises at lower pH because of their greater tolerance to acidity and through reduced competition from other microorganisms.

Some workers (Baath, 1980; Pietikainen and Fritze, 1995; Siira- Pietikainen *et al.*, 2001b) have reported that in general microbial community structure changes and fungal biomass decreases after clear felling.

Moser (1981) observed that the basidiomycetes and ascomycetes mycoflora are the examples of the species selection and adaptation (limitation) in the transitional zone from sub-alpine forests to alpine tundra.

Many soil microorganisms are known to be intolerant to low soil moisture content (Harris, 1981; Paul and Clark, 1989) and changes in soil moisture status can result in rapid shifts in the magnitude of soil microbial biomass (Bottner, 1985; Schnurer *et al.*, 1986).

Some workers (Klironomos *et al.*, 1993; Klironomos, 1995) have suggested that heterogeneity of fungal communities in forests could be due to a wide array of soil and host plant characteristics.

Deka (1981) reported the depth-wise decrease in the fungal and bacterial populations and suggested that it might be due to variation in the nutrient status at different depths.

Jenkinson and Ladd (1981); Duxbury *et al.* (1989) and Singh *et al.* (1989) recognized that soil microorganisms play an essential role in the sustainability of indigenous forest ecosystems.

Kauri (1982) studied the seasonal fluctuations in numbers of bacteria in beech forest soil and observed two peaks in bacterial population; one in autumn after leaf fall and another in spring.

Soil disturbance and management practices such as clear-cutting of forests and tillage systems are reported to have a profound impact on the distribution and activities of soil microorganisms. Lundgren (1982) reported a strong detrimental effect of clear-cutting of forest on the bacterial population of A_0 horizon. He observed a pronounced increase in bacterial biomass at the clear-cut area as compared to the natural pine stand. However, there was rapid decrease in bacterial biomass in comparison to a reference

stand and the results from an old clear-cut area (ca.10 year) indicated long lasting detrimental effects on the size of the bacterial population.

Pati *et al.* (1983) pointed out the relative contribution of microbes and roots to the total soil metabolism in a tropical grassland soil. They observed that fungal and bacterial biomass ratio was 12:1 indicating dominance of fungal component. They further observed that contribution of bacteria to soil metabolism was 5% whereas fungal contribution was 57%.

Biro *et al.* (1983) hypothesized that microbes also play a major role in the formation of good soil structure. Bacterial mucigel and the hyphal threads produced by fungi bind the soil particles together. Microbial activity helps to aggregate the soil, which reduces soil erosion, allows for good water infiltration, and maintains adequate aeration of soil.

Behera and Mukerji (1985) studied the seasonal variation and distribution of microfungi in forest soils of Delhi and noted that surface soils harbored the highest population and species number which gradually declined with increase in depth.

Lynch and Bragg (1985) stated that among soil microorganisms, fungi can play an important role in the formation and stabilization of soil aggregates. The two main processes involved in the biological improvement of soil aggregate stability are the adhesive effects of microbial metabolic products and entanglement of soil particles by filamentous fungi.

Fungi and bacteria often occupy separate ecological niches in the soil environment and thus play different roles in nutrient cycling. Hence, it is important to

quantify the soil biomass in terms of distinct fungal and bacterial components (West, 1986).

Tiwari *et al.* (1986) reported higher bacterial population from the surface soil, which may be due to higher organic carbon content and favourable moisture level. The rise in temperature may be responsible for the increased bacterial population. Bacterial population was positively correlated with soil temperature and pH.

Soil microbial activity has been shown to be dependent on temperature (Anderson and Domsch, 1986; Inzam, 1990; Nicolardot *et al.*, 1994), moisture (Linn and Doran, 1984; Anderson and Domsch, 1986; Insam, 1990; Kessel *et al.*, 1993; Liebig *et al.*, 1995).

Tiwari *et al.* (1987a and b) have reported wide differences on distribution of microbial populations, their activities and biochemical transformations in soils of various moisture regimes. They found significant variation in fungal population with different moisture contents. They concluded that soil moisture status not only regulates the population and activity of microbes but also modifies the relationship between various parameters.

Tiwari *et al.* (1987 b) reported that higher fungal and bacterial populations were always recorded from surface (0-10cm) soils which decreased with increasing depth except during winter season. They also reported that microbial population was positively correlated with soil organic carbon, moisture and temperature.

Perry *et al.* (1989) suggested that a high diversity of fungi may be necessary to maintain the stability of the forest ecosystem; in changing environments, fungal species with different environmental tolerances may shift in abundance on a plant host.

Ecosystem stability and resilience to disturbance may be increased by high fungal species diversity. Generally fungal diversity may provide resilience, buffering changes in forest ecosystems caused by natural disturbances.

Tiwari *et al.* (1989) reported that soil moisture significantly alters the microbial population, its activity and relationship between parameters.

Climate varies between sites, influencing soil temperature and moisture regimes, which, in turn, influence the dynamics of soil microbial communities (Insam *et al.*, 1989; Wardle and Parkinson, 1990; Alvarez *et al.*, 1995 and Schimel and Clein, 1996).

Gamliel and Katan (1991) determined that bacterial and fungal populations decreased significantly after solarization. The reduction resulted from the mesophilic characteristics of these microorganisms. The effects of soil depth were examined with respect to anaerobic bacteria, and the highest values were obtained from the 3rd (5-10cm) and the lowest from 0-1 cm depth. Owing to the fact that, higher soil layers possess more oxygen, anaerobe bacteria number was found to be less because of the lethal and/or promoting effect of the oxygen.

Behera *et al.* (1991) while conducting an ecological investigation of some microfungi in a tropical forest soil of Orissa could isolate 36 fungal species and one sterile mycelia from the soil. They observed seasonal variations of fungal population to be more pronounced in upper soil layers. They also observed positive correlation between fungal population, soil moisture and organic matter content.

Gammack *et al.* (1992) found that most bacteria in soil are adsorbed or attached to particles, although this varies with soil type, pH, and other factors.

Jha *et al.* (1992) studied the microbial population and enzyme activities in relation to altitude and forest degradation in north east India. They reported that fungal and bacterial populations showed marked variations at different altitudes. Higher fungal and bacterial population numbers were recorded in less degraded forest than more degraded one at both higher and lower altitudes. In more degraded forest stand, at lower altitude, both the fungal and bacterial population showed a significant positive correlation with organic C, whereas, in less degraded forest stand, there was a significant correlation between fungal population and organic carbon. In higher altitudes, fungal population was significantly correlated with soil moisture and organic C. Further, they found greater enzyme activity in less degraded forest soils in comparison to more degraded forest stands.

Wardle (1992) suggested that the temporal dynamics of the soil microbial community are likely to be important in determining the mineralization and hence availability of nutrients for plant productivity.

In a study of soil fungi from northern hardwood forest in Rhode Island, Carreiro and Koske (1992) found that different fungal species have different optimum temperatures. The fungal species isolated from these soils differed depending on the temperature used for incubation in the laboratory.

Kaiser *et al.* (1992) found that the size and activity of the microbial population depends on the quantity and quality of soil organic matter, soil texture, soil pH and other properties of soil.

Bardgett *et al.* (1993, 1996, and 1997) and Graystone *et al.* (2000) reported a common finding in their studies of upland grasslands along soil fertility gradients. They

observed that the size and activity of soil microbial community were higher under low fertility conditions than under high fertility conditions maintained by regular nitrogen additions. They also reported that the high soil fertility and nutrient availability is favourable to bacterial community only.

Diaz-Ravina *et al.* (1993) studied microbial populations in Spain with a Mediterranean climate. They found that populations were lowest in the summer and winter, presumably due to moisture in summer and temperature in winter.

Zvyaginstev (1994) studied management in case of the forest and suggested that soil properties which change with soil depth control the composition of microbial populations.

Conyers *et al.* (1995) reported that the changes in temperature and water potential determine changes in microbiological activity.

Zak *et al.* (1994) and Bossio and Scow (1995) stated that soil microbial diversity measurements have been the most extensively used parameters to investigate differences between soil communities from widely different habitats.

Frostegard and Baath (1996) stated that the microbial communities in forest soils rich in organic matter may differ considerably from those found in grassland and in arable soils with lower organic matter content.

Zabinski *et al.* (1997) compared the functional diversity of soil microbial communities in heavily impacted sub-alpine campsites and adjacent undisturbed areas using the Biolog method of carbon utilization profiles. They reported that microbial communities differentiate in response to disturbance in the top 6 cm of soil, while below 6 cm there were no recognizable differences between disturbed and undisturbed soil

communities. Analysis of the factors that differentiate the upper microbial communities between disturbed and undisturbed sites revealed that the percent of total carbon sources utilized was significantly less in the disturbed (54 %) than in undisturbed areas (95 %). They also compared among the total culturable actinomycetes, bacteria, and fungi and revealed no difference in overall number of colony forming units (CFU) on disturbed and undisturbed sites, but a marked decrease in actinomycetes on disturbed sites.

Giller *et al.* (1997) hypothesized that reduction in soil microbial diversity result in reduction in the functional capability of soil.

Hawksworth and Rossman (1997) estimated 1.5 million fungal species in existence and only 5-10 % has been formally described.

Garlang (1997) observed that microbial communities have great potential for temporal or spatial change and thus represent power tool for understanding community dynamic variation in microbial community structure, which may have effects on ecosystem process.

Arunachalam *et al.* (1997) recorded maximum bacterial population during rainy season and minimum during the winter, while fungal population was maximum during autumn and minimum during winter in four forest re growths (7, 10,13, and 16 years old) in northeast India. There was increase in microbial population with increasing age of the forest re-growths as well as increase in depths.

Tiwari and Sharma (1998) reported that the fungal and the bacterial populations in highland soils increased with increase in altitude up to 1100 m, but thereafter, the populations declined sharply. They also observed positive correlation between fungal and bacterial populations with organic matter content of the soil.

Berg *et al.* (1998) investigated the abundance and micro stratification of bacteria and fungi inhabiting the organic layers of a Scots pine forest. They suggested that the abundance of bacteria was influenced by water and that of fungi by water and temperature.

Houston *et al.* (1998) examined decomposer fungi and microbial processes in harvested and adjacent unharvested areas of two mixed wood forests in northwestern Ontario. They found that soil microbial processes and fungal community structure were similar in harvested and unharvested stands. Although reductions were not significant, basal respiration, microbial biomass C, qCO_2, C_{mic}: C_{org}, and nitrogen mineralization tended to be lower in organic soil from the harvested sites than the unharvested sites, with the exception of qCO_2. Comparing organic and mineral soil layers, fungal richness, diversity, and community composition corresponded more closely in the harvested sites than unharvested sites.

Coyne (1999) stated that each microbial group functions over a range of temperature in which they grow best. As a rule, the largest microbial populations are found at moderate soil temperatures.

Hu *et al.* (1999) reported that bacterial response to alteration in C availability is important in understanding the microbial community structure and microbial interactions in soil ecosystem.

Petraltis *et al.* (1989) and Buckling *et al.* (2000) believed that fungal diversity can be increased by the patchiness of the environment and niche availability without imposing catastrophic species loss.

McLean and Huhta (2000) studied the effects of temporal and spatial fluctuations in moisture on the microfungal community in birch litter plus pine humus microcosms over 6 months. They reported that at 6 months, fungal species richness was higher and diversity was lower under moisture fluctuations than under uniform moisture. The number of fungal isolates and proportional diversity was significantly higher in the bottom layer in the fluctuating moisture treatment. Their experiment suggests that even without drying, fluctuations in moisture can affect fungal community structure. They also reported that changes in plant composition within a vegetation type may affect the microbial community.

Liu *et al.* (2000) stated soil moisture, soil temperature, and/or substrate availability as the most important factors that influence soil microbial growth and population density. Soil microbial diversity (as measured by substrate utilization) and activity were generally reported to decrease with disturbance.

Zeller *et al.* (2000) observed increase fungal biomass in pastures abandoned for 10 years.

Klose *et al.* (2004) suggested that variations in amounts and quality of the litter produced by each stand contributed to the responses of microbial and biochemical properties of the four studied forest sites.

Microbial biomass carbon

The microbial biomass is a potential source of nutrients from plants especially of their ability to bind temporally the nutrition to microbial cells. The microbial biomass and its activity are important indicators of changes in content and vertical distribution of the

organic matter in a soil profile. Soil microbial biomass acts as a pool of biologically active C, N, and S (Jenkinson and Powlson, 1976; Jenkinson and Ladd, 1981; Brookes *et al.*, 1985) and has been widely used in investigations of nutrients dynamics and transformation in soil.

Jenkinson and Powlson (1976), Ayanaba *et al.* (1976), Anderson and Domsch (1978) and Brookes *et al.* (1982) reported that recognition of the importance of soil microorganisms in the functioning of ecosystem has led to an increased interest in measuring the nutrients held in soil biomass.

Biomass C measurements using the fumigation-incubation (FI) methods (Jenkinson and Powlson, 1976) showed higher values in autumn and late spring and lower values in winter. Their estimates are unrealistically low. Gray (1976) observed that the activity of much of the soil biomass is severely limited by nutrient availability and many soil organisms have very low metabolic rates or spend most of their lifetime in dormant or resting phases.

Ayanaba *et al.* (1976) reported relatively constant relationships between biomass C and mineral N for some African soil. They also indicated that changes in soil management cause microbial biomass to increase or decrease much faster than the total amount of soil organic matter so that microbial biomass carbon as a percentage of total organic carbon can provide a sensitive indicator of less detectable trends in total soil organic carbon loss or accumulation. Oades and Jenkinson (1979) found a relationship between biomass C and ATP for some Australian soil.

Tate and Terry (1980) found that microbial biomass, dehydrogenase activity and aerobic bacterial population were correlated with moisture level of the soil.

Paul and Voroney (1980) and Zak *et al.* (1990) revealed that soil microbial biomass is closely linked to the primary productivity and functioning of an ecosystem.

Jenkinson and Ladd (1981) reported that since microbial biomass is considered to be a transformation agent of soil organic materials and a labile reservoir of nutrients, information on its amount, activity, and composition is essential for evaluating the function of soil ecosystems. The size and the turnover of the microbial biomass affect the quantity of available nutrients, particularly C, N, P, and S.

Hassink *et al.* (1991) investigated the contribution of microbial biomass to the pool of mobile plant nutrients in two soils having different amount of microbial biomass contents. They observed 55-57 % of the N mineralized in the soils was derived from the freshly killed biomass. They have suggested that the amount of microbial biomass might be used for estimating the mobile plant nutrient pool in soil. The microbial biomass has, therefore, been used as an index of soil fertility that depends on nutrients fluxes.

According to Brookes *et al.* (1985a), microbial biomass in soil is a relatively large and a labile component of organic matter containing important plant nutrients, especially N and P.

An increase in the size of the microbial biomass is considered essential for the improvement of soil fertility because the larger the amount of biomass, the greater will be the potential availability to higher plants (Stevenson, 1986).

McGill *et al.* (1986) and Anderson and Domsch (1986) mentioned that microbial biomass carbon reflects the long term amount of C input into a soil.

Jenkinson *et al.* (1987), McCaskill and Blair (1988) suggested that estimation of pool sizes of microbial C and N are therefore, required for many multicompartmental models of nutrients dynamics in different ecosystems.

Soil microbial biomass C to total organic C ratios has been suggested as useful measures for monitoring organic matter dynamics (Powlson *et al.*, 1987; Sparling, 1992).

Sarathchandra *et al.* (1988) observed an increase in biomass C over a short nine week period between winter and late spring.

Several studies on soil microbial biomass have established the importance of this component in the nutrient budget of an ecosystem and to predict the effects of perturbations (Jenkinson and Powlson, 1976; Diaz-Ravina *et al.*, 1988; Henrot and Robertson, 1994).

High organic matter input could be the driving factor resulting in higher microbial biomass in pasture because grasses allot as much as 40-85 % of photosynthetic products below ground through roots and mycorrhizae (Fogel, 1985; Whipps, 1990).

Microbial biomass carbon (C_{mic}) reflects the long-term amount of C input into a soil (McGill *et al.*, 1986; Anderson and Domsch, 1986). Apart from substrate quantity, also its quality and distribution determine the amounts of C_{mic} (Herman *et al.*, 1977; Holland and Coleman, 1987; Insam and Domsch, 1988).

Sparling and Ross (1988) showed a significant increase in microbial biomass and net nitrogen mineralization after air drying and subsequent rewetting of soils. They suggested the reason being the release of a large fraction of the increased N was derived from microbial cells killed by desiccation.

Diaz-Ravina *et al.* (1988) have shown that soils with a low C content will have presumably a less biomass and vice-versa. The attribute could be related to low MBC in the Alnus plantation and the 7-yr-old regrowth. The variations in MBC level of their study sites could be explained in terms of their positive correlation with soil moisture, organic-C and TKN.

The importance of soil microbial biomass in the cycling of C and N is well documented (Van Veen *et al.*, 1989). Anderson and Domsch (1989) stated that generally, microbial biomass comprises about 2-4 % of total organic C. The soil microbial biomass constitutes a labile fraction of the soil organic matter and serves as a source and sink of plant nutrients (Singh *et al.*, 1989).

Smith and Paul (1990) reported that the turnover time for N immobilized in the microbial biomass was found to be about ten times faster than that derived from plant material.

Insam (1990) reported that studies on intimate relationship between climatic conditions and the microbial soil C pool and respiratory C flux showed the best possible prediction of microbial biomass C and its ratio to total organic C by using climatic variables such as precipitation: evaporation ratio in soil with equilibrium C content . Further he found that longer dry period reduced microbial biomass and increased with increase in moisture content.

Nanipieri *et al.* (1990) hypothesized that since microorganisms are considered the primary source of enzymes of soils, enzyme activities are strongly associated with the active microbial biomass. Nevertheless, there is no direct correlation between the size of the microbial biomass and its metabolic state.

Cengel (1990) and Srivastava (1992) defined that microbial biomass is the characteristic of microorganisms which participate in the biochemical cycles and are the living part of the soil organic matter. It is not affected negatively by polarization.

Van Gestel *et al.* (1992) observed no significant differences between labeled and total biomass decline due to soil drying.

Wardle (1992) stated that most of biomass consists of bacteria and fungi with the balance consisting of soil microflora and algae. Amount of microbial biomass is influenced by soil texture and quality of soil organic matter (Wardle, 1992). Gupta (1992) observed that soil texture affected the size and turnover of soil microbial biomass.

Turner *et al.* (1993) and Bauhus *et al.* (1998) found that plant cover and especially the dominant tree species affect both the biological and chemical properties of the soil. Microbial biomass, activity and community structure have all been shown to be affected by different tree species.

Ravina *et al.* (1993) reported that in the forest soils, the soil microorganisms immobilize relatively high amounts of nutrients in their biomass; the contribution of the microbial biomass to soil concentrations of available plant nutrients was large for N, P and K.

A seasonal trend in microbial C is not well understood. Soil microbial biomass carbon (SMB-C) has been shown to peak in the summer months (Kaiser and Heinemeyer, 1993; Bardgett *et al.,* 1999). SMB-C is often closely related to organic matter input (He *et al.,* 1997).

A number of soil microbiological parameters notably microbial biomass carbon and basal respiration (Doran and Parkin, 1994; Sparling, 1997) have been suggested as a possible indicator of soil quality and have been employed in national and international monitoring programs. Soil microbial biomass can be an important pool of plant nutrients and is often highly correlated with the organic matter contents of soils (Pankhurst *et al.*, 1995).

Witter *et al.* (1993) and Bardgett *et al.* (1994) have shown that stress by heavy metal toxicity and low pH result in a reduction in the size of soil microbial biomass.

Henrot and Robertson (1994) mentioned that microbial biomass can provide an index of soil fertility because it represents an important labile pool of soil nutrients and plays an important role in preventing nutrient loss from the soil.

Brookes (1995) reported that the microbial biomass, which is a small fraction of soil's total organic matter content, is a source and sink of nutrients and controls soil organic matter mineralization. Disturbing the biological equilibrium with changes in the composition and activity of microflora can damage soil fertility both in the short and long term.

Considerable attention has been given in recent years to the development of methods for measuring the biomass of the organisms in different soils. More recently, microbial biomass has also been recommended as a biological indicator of soil quality, although there are several ways to quantify the structure (Kennedy and Smith, 1995).

Diaz-Ravina *et al.* (1995) and Hossain *et al.* (1995) described that the microbial biomass of soils has been used to reveal effects of forest management. The microbial biomass C-to-N ratio is often used to describe the structure and the state of the microbial community. Some studies hypothesized that a high microbial C-to-N ratio reflects a higher proportion of fungi in the microbial biomass, whereas, a low value suggests that bacteria predominate the microbial populations (Pitchel and Hayes, 1990; Joergensen, 1995). Assuming this hypothesis is true, the microbial C-to-N ratio in humus horizons of forest soils along the deposition gradient could imply that the microbial biomass at sites with high deposition loads (i.e., with higher pH) was dominated by bacteria, while fungi were predominant in the low-load coniferous forest sites (i.e., with lower pH). Several studies documented that after lime application bacteria became dominant over fungi (Wolters *et al.*, 1995). Diaz-Ravina *et al.* (1995) reported the significant effects of the season, the type of soil, the interaction between the type of soil and season, but soil type explained most of the variance on microbial biomass and other soil nutrient flush of forest soils. They have also found a substantial contribution of microbial biomass to plant -available nutrients of forest soils. Further, they noted close relationship of microbial biomass with pH, organic C, total N and moisture contents.

Lee *et al.* (1996) reported higher levels of microbial biomass carbon associated with trafficked compared with non-trafficked areas.

Maithani *et al.* (1996) on their study on the seasonal dynamics of microbial C, N and P during re-growth of a disturbed subtropical humid forest in northeastern India revealed the occurrence of higher microbial biomass during the winter and the lowest during the rainy season at surface soil layers. There was an increasing trend of microbial

biomass from lower in younger stand to the highest in the older stands of the forest re-growths.

Lavahun *et al.* (1996) reported significant variation in the distribution of the microbial biomass C at different depths of a grassland and two arable soils. The biomass C declined from maximum at the upper layer to a minimum at the lower depth in all the study sites. They observed decrease in microbial biomass C associated with decline in organic C content revealing higher organic C mineralized at upper layers leading to higher microbial activity than the lower soils layers.

Fritze *et al.* (1996) reported that microbial biomass C contributed between 1.19 and 1.8 % to the total organic C content in undisturbed humus layers from boreal coniferous forest ecosystems. The decrease of this ratio at the heavily and moderately influenced forest sites may also indicate a disturbance in the C turnover in these soils.

Inputs of plants residues have an obvious influence on seasonal changes of soil microbial C (Chander *et al.*, 1997). The values of microbial biomass C of summer and autumn samples would, therefore, be expected to be greater than those of winter samples. However, they observed an opposite relationship so other mechanisms must be responsible for the decline of microbial biomass C during summer.

Grisi *et al.* (1998) studied the effect of temperature on organic matter and microbial biomass dynamics in temperate and tropical soils and revealed a slow declining trend of microbial biomass in tropical soils at 15 and 35° C than in temperate soils, although at 15^0 C the differences were minimal. They also observed more rapid mobilization of organic matter in the temperate than in the tropical soils at 35° C. It was

concluded that the organic matter in the tropical soils are more degraded, or humified than in the temperate soils.

Wardle (1998) stressed the temporal variability of the soil microbial biomass is an important component of its turnover; and thus contribute to patterns of soil nutrient release and mineralization. The temporal variability of microbial biomass C was most closely related to soil N content in forest, pH and latitude in arable ecosystems and pH, latitude and soil C contents in grasslands. He also stated that both macroclimate and soil quality factors (e.g. soil pH, organic matter content) play important roles in determining microbial biomass and its turnover.

Taylor *et al.* (1999) investigated the microbial biomass in a sequence of northern hardwood forest stands ranging in age since clear cutting from 3 to more than 120 years. Significant variation was observed among sampling periods and was greater in early and late successional stands than mid successional stand. They found that microbial biomass was not very responsive to the environmental factors, however, moisture content was found to be the most important factor often contributing to variation in microbial biomass. It was concluded that the lower microbial biomass in the mid successional stand suggesting that microbial dynamics in forest soils are not controlled by factors directly related to forest harvesting.

Arunachalam and Arunachalam (1999) reported that the microbial biomass carbon values recorded in the 7 yr-old pine-dominated stand are 24-34 % lower than that of the 22 yr-old pine forest. This indicates faster rate of microbial biomass recovery early during revegetation in a disturbed or degraded site. Similar report has also been made in a

regrowing broadleaved forest community at higher altitudes of Meghalaya (Pandey *et al.*, 1996).

Zeller *et al.* (2000) reported the highest C_{mic} values (with no carbon substrate addition) were found in top soil. They found no significant correlation between the organic carbon content and the biomass amount was not significant.

Piao *et al.* (2000a and b) suggested that higher air temperature stimulates turnover rates of soil microbial biomass C.

Hill *et al.* (2000) observed that microbial biomass measurements give little information about qualitative community-level changes and are limited in their ability to describe a particular microbial ecosystem.

The microbial biomass is part of the active soil organic matter pool, and had been proposed as an indicator of the state and change of total soil organic matter (Tian *et al.*, 2001).

Piao *et al.* (2001) stated that it might be possible that the increase in microbial biomass C with decreasing elevation can be ascribed to increase in inputs of roots and weeds into the soil.

Imberger and Chiu (2001) stated that forest surface tend to have both higher level of microbial biomass and ratios of fungal biomass to bacterial biomass than grassland soils. They also concluded that both the fungal and bacterial biomass decreases from the surface horizon to the deeper horizons in the forest and grassland soils. However, they found that the fungal to bacterial ratio showed no sequential change throughout the forest or grassland soils profiles.

Bolter *et al.* (2002) stated that number and biomass of microorganisms (bacteria, yeast, fungi, cyanobacteria, soil algae, protozoa) as well as descriptions and monitoring of microbial communities are basic requirements for understanding microbiological processes, thus they are within the remit of soil ecological research.

Klose *et al.* (2004) reported that microbial biomass C was significantly lower in the humus layers and mineral topsoil (0-10m) of forest soil. The responds of the microbial biomass and soil respiration data to different atmospheric deposition loads were mainly controlled by the content of organic C and cation exchange capacity, while those of enzyme activities were governed by the pH and concentrations of mobile heavy metals.

Enzyme activity

Soil microbial activity is an indicator reflecting soil properties, microbial populations and enzyme activity. It not only reflects soil fertility, but also reflects soil environmental conditions. Studies of enzyme activities in soil are important as they indicate the potential of the soil to support biochemical processes which are essential for the maintenance of soil fertility (Dkhar and Mishra, 1983). Detailed survey of the available literature on the studies of various aspects of soil enzyme activities reveals an extensive research work done in this field during the last three decades of the 20th century. Microbial activity can be analyzed on the basis of some parameters that reflect the behavior of soil microorganisms, such as enzyme activity.

Ramirez-Martinez and McLaren (1966b) and Paulson and Kurtz (1969) reported that soil enzyme activity is independent of the microbial population.

Skujins (1976) reported that the enzymatic activity of a soil depends on both the abiotic factors such as intracellular enzymes within dead and the living microbial cell. He recorded that once the enzyme is stabilized in the soil, it develops resistance to humidity, temperature and other environmental changes.

Soil enzymes are biologically significant as they catalyze various reactions in soil and participate in nutrient cycling (Skujins, 1976; Ladd, 1978; Dick, 1994).

Casida (1977) reported that the determination of enzyme activities in conjugation with soil respiration and composition of soil microflora provides the most reliable index of microbial activity in soil. Enzymes are markedly dependent on pH, ionic strength, inhibitors (or pollutants), moisture regimes, temperature and other environmental factors (Tabatabai and Dick, 1979; Frankenberger and Johanson, 1982; Dick and Tabatabai, 1983), but they may become stabilized in the soil by forming humus-enzyme or clay-enzyme complexes (Makboul and Ottow, 1979).

Tate (1977) mentioned that soil enzymes play an important role in soil mineralization processes and have been related to other soil biological properties. Several worker have studied dehydrogenase, urease and phosphatase activities in different systems under different climate, land use and soil conditions (Beri *et al.*, 1978; Dkhar and Mishra, 1983; Baruah and Mishra, 1984; Rao and Ghai 1985; Tiwari *et al.*, 1989).

Skujins (1978) reported that the soil enzyme activity estimates are often used as indices of microbial activity and soil fertility.

Dash *et al.* (1981) reported positive correlations between enzymes activities and organic C and N.

Frankenberger and Bingham (1982) reported the inhibitory effect of increased soil salinity to the enzyme activities. They also observed decrease in enzyme activity with increasing electrical conductivity or salinity, however, the degree of inhibition varied among the enzymes assayed and the nature and amounts of salts added. The activity of dehydrogenase was severely inhibited by salinity, whereas, the hydrolases showed lesser degree of inhibition.

Frankenberger and Dick (1983) studied an extensive study on the relationship between enzyme activities and microbial growth and activity indices in different soils have revealed high correlation of the enzyme activities with total biomass in soil.

Dormaar *et al.* (1984) studied the impacts of seasons and site management on the enzyme activities of soils in Alberta, Canada. They found highest enzymatic activity of winter months and lower during rainy season. Their results indicated the significant effects of grazing on enzyme activities of soils in two sites.

Rao and Ghai (1985) mentioned that in reclaimed alkali soils, there was marked improvement in physico-chemical properties and enzyme activities of soil under trees and grass cover for 12 years.

Tabatabai and Fu (1992) found that the overall enzyme activity of soil is derived from the activity of accumulated enzymes and from that of proliferating microorganisms. Factors influencing soil microbial activity exert control over soil enzyme production and control on nutrient availability and soil fertility (Sinsabaugh *et al.*, 1993). Soil enzymes activities may correlate well with nutrient availability (Asmar *et al.*, 1994). Bacteria and fungi synthesize and secrete enzymes such as phosphatases, proteases, ureases and pectinases extracellularly. Those microbially secreted enzymes constitute an important

part of the soil matrix as extracellular enzymes, also called abiontic enzymes (Sinsabaugh, 1994). Evaluation of soil organisms is most closely related to soil enzyme activity. It is generally accepted that enzymes in soils originate from animals, plants and microbial sources (Ladd, 1978; McKay, 1991; Schinner *et al.*, 1995).

Miller and Dick (1995); Deng and Tabatabai (1966a and b, 1997) and Klose *et al.* (1999) reported that the activity of soil microorganisms is strongly linked to the activity of enzymes and soil management (including crop rotations, fertilization, tillage and crop residue placement) strongly influence the activity of soil enzymes.

Nevertheless, soil enzyme activity is believed to be sensitive indicator of the effect of environmental factors on microbial functions (Dick, 1994). Thus, because of their role in nutrient cycling, enzymes like acid phosphatase and alkaline phosphatase are suggested to be good indicators of potentially beneficial or harmful effects on the ecosystem.

Kandeler *et al.* (1996); Klose *et al.* (1999); Acosta-Martinez and Tabatabai, (2000) stated that enzymatic activities are determined by the factors that affect size and metabolic activity of microorganisms in soil, moisture, temperature, pH, available nutrients, toxic elements and litter quality. Due to the significant changes in the soil physical and chemical properties of forest stands in emission areas, changes in the size of the microbiota and in enzyme activities in the forest floor and mineral soils could be expected.

The data of enzyme assays have been connected to many characteristics of soil, including microbial growth and activity as the mineralization of organic P esters

(Vuorinen and Saharinen 1996) Microbial activity can be analyzed on the basis of some parameters that reflect the behavior of soil microorganisms, such as enzyme activity.

Tiwari (1996a) reported that the dehydrogenase, urease and acid phosphatase activities showed a decreasing trend with increase in soil depth in a hilly sandy loam profile of northeastern India. His study demonstrated persistent activities of these enzymes to a depth of 2cm in sandy loam soil profile.

Tiwari (1996b) investigated the relationship between enzyme activities, microbial populations and soil respiration in some Indian soils namely, grasslands, garden, orchard, fallow and arable soils of northeastern hill regions. Multiple regression and simple correlation analysis of the studied parameters revealed widest range (40 fold) in urease enzyme activity for various soils whereas the narrowest range (1-5 fold) was recorded for the phosphatase activity. The dehydrogenase activity falls in the range of variation between the two enzymes, urease and phosphatase. The results showed that fungal biomass accounted largely for the variability in dehydrogenase, urease and phosphatase activities.

Kumari and Charya (1997) found significant positive correlationship between soil enzyme activities and microbial population number in four polluted sites of Warangal, Andhra Pradesh, India. They found increased microbial colonies showing increased accumulation of soil enzymes. Positive correlation was observed between enzyme activities and soil nutrients such as nitrates, potassium and organic matter.

Tiwari and Sharma (1998) showed that the organic matter content of the soil is an important factor that regulates enzyme and microbial activities in the Eastern Himalayan high land soils.

Arunachalam *et al.* (1999) stated that the enzyme activities, soil respiration bacterial and fungal populations and microbial biomass were greatly influenced by the levels of nutrients.

Soil enzymes activities are 'sensors' of soil degradation since they integrate information about microbial status, and also, from soil physico-chemical conditions (Wick *et al.*, 1998; Aon and Colaneri, 2001; Baum *et al.*, 2003). They are used as sensors in studies on the influence of soil treatments on soil fertility (Chen *et al.*, 2003).

Boerner *et al.* (2000) revealed that enzymes activities and specific enzyme activities (activity rates on an organic C basis) were higher in the high deposition-load forest sites compared to the moderate and low-load sites. Specific enzyme activity values give an estimate of how suitable the organic matter is to degradation by enymatic reactions, and thus are believed to be measures of organic matter quality

Enzyme activities can vary depending on the sampling date in zones with a seasonal climate (Watanabe and Hayano, 1995; Baum *et al.*, 2003). In Mediterranean ecosystems, the highest activities occur in spring together with the most active growth of plants and microbial activity (Garcia *et al.*, 1997, 2002). In contrast, when the winter is warm and the climate dry, the enzyme activity is usually higher in autumn-winter period, intermediate in spring and lowest in summer (Fioretto *et al.*, 2000, 2001), showing that the climatic conditions determine the phenology of soil enzyme activities.

Ogaya and Peneulas (2004) showed a significant activity of the five enzymes analyzed in both spring and in autumn, but in agreement with those previous studies, the highest activity of all enzymes occurred in spring when, in addition to optimal

temperatures and water availability there was also a higher quantity of litter in the holm oak forest.

Dehydrogenase activity

Dehydrogenase being a respiratory enzyme provides a measure of catabolic activity of soil and it correlated with the activity of microorganisms (Stevenson, 1959; Peterson, 1967; Skujins, 1973, 1976).

Level of dehydrogenase activity in soil is considered to provide some guide to the microbiological activity of soil. This aspect has been investigated intensively since the introduction by Lenhard (1956) of a method for estimation of the activity. He introduced the concept of determining the metabolic activity of microorganisms in soil and other habitats by measuring dehydrogenase activity using 2, 3, 5-triphenyl tetrazolium chloride (TTC) reduction. The method is based on the assumption that in the absence of oxygen, TTC acts quantitatively as the terminal H- acceptor for dehydrogenase system with the formation of red triphenyl-tetrazolium formazan (TPF): $TTC + 2H^+ + 2e^- = TPF + HCL$.

Stevenson (1959) found a close correlation between the oxygen uptake and dehydrogenase activity in the soil samples. According to him there is a close relationship between dehydrogenase activity and bacterial number.

Skujins (1967) discussed the effect of different forms of soil storage for the determination of enzyme activity and concluded that no rules can be established and that conditions differ for each soil and enzyme to be analyzed.

Dehydrogenase activities appear to be greater in the less acid samples and are strongly favored by moisture. Gilot and Dommergues (1967) found that dehydrogenase

activities /g soil organic C in some sub-alpine forest soils were greater in horizons of highest pH values.

Skujins and McLaren (1968) compared the dehydrogenase activity of geologically preserved soils with freshly collected desert and cultivated soils and detected measurable amount of dehydrogenase activity in fresh soils. The activity did not reflect the overall metabolism.

Harris (1968) found high dehydrogenase activity in light sand, as well as in recently wetted soil.

Ross (1970) reported that dehydrogenase activity depends more upon the metabolic state of the microbial population of the soil than the activity of specific free enzymes acting on particular substrates.

Dehydrogenase is an extracellular enzyme in the soil and considered to play an important role in the initial stages of the oxidation of soil organic matter by transferring hydrogen or electron from substrates to acceptors (Ross, 1971). Because of its importance in the organic matter transformation processes and its potential to indicate the available microbiological activity in the soil, dehydrogenase has been the subject of chosen biochemical tool in various fields of agricultural and soil science investigations. He studied some factors influencing the estimation of dehydrogenase activity of pasture soils in New Zealand. He observed that dehydrogenase activities of samples of the same soil collected at different times differ. He suggested that enzyme dehydrogenase is considered to play an important role in the initial stages of the oxidation of soil organic matter by transferring hydrogen or electron from substrate to acceptors.

Ross and McNeilly (1972) while studying the effects of storage on oxygen uptake and dehydrogenase activities of beach forest litter and soil, observed that dehydrogenase activities of some samples maintained at 4^0 C increased but those of all samples held at -20^0 C decreased. According to them fresh samples gave better dehydrogenase activity and if necessary storage at 4^0 C may be preferable for retaining dehydrogenase activity did not correlate with microbial numbers, and an assay of dehydrogenase activity was used to predict the proteolytic, nitrifying and respiratory activities in soils.

Skujins (1973) reported that dehydrogenase activity did not correlate with microbial numbers and phosphatase, which showed scattered activity values throughout the profiles. The activity of dehydrogenase may act as an indicator of the microbiological redox system in soils and can be considered a good measure of microbial oxidative activity (Casida, 1977; Tabatabai, 1982).

Vishwanath *et al.* (1975) studied the behavior of bacteria surviving in chloroform and toluene treated soils with regard to their dehydrogenase activity. Schinner and Gurschler (1976) reported that higher values for enzyme activity, soil organic matter and moisture were found in the top layers of soil. These parameters decreased with increasing depth.

Smith and Pugh (1979) demonstrated that the dehydrogenase assay could provide a valid indicator of soil microbial activity.

Dehydrogenase activity were influenced directly or indirectly by the factors like; pH, moisture and temperature of the soil (Tesarova and Gloser, 1976; Kowalenko *et al.*, 1978; Tiwari *et al.*, 1987). Determination of dehydrogenase activity in soils is based on the use of soluble tetrazolium salts as artificial electron acceptors, which are reduced to

red colored formazans, extracted and then determined colorimetrically (Trevors, 1984; Von Mersi and Schinner, 1991).

Dkhar and Mishra (1983) observed that the dehydrogenase activity of soil was positively correlated with moisture content and the bacterial population.

Tiwari *et al.* (1987a and b) studied the influence of moisture regime on dehydrogenase activity and reported that moisture plays a significant role in soils. They further reported maximum dehydrogenase activity in the summer season and also suggested that soil temperature, moisture and organic carbon were the most important factors regulating the dehydrogenase activity.

Grobler *et al.* (1987) used the dehydrogenase assay in estimation of heterotrophic bacterial activities in the soil. Further, they described the dehydrogenase assessment as more reproducible activity estimate than the glucose mineralization. The synthesis of extracellular enzymes is affected by all the factors that affect microorganisms in the soil (moisture, temperature, available nutrients). Once the enzymes are released, their activity primarily regulated by chemical and physical factors such as litter chemistry, substrate availability and temperature (Sinsabaugh *et al.*, 1981; Sinsabaugh and Linkins, 1987; Sinsabaugh, 1994).

As microorganisms are important source of soil enzymes, the activities of such soil enzyme should normally correlate with microbial activity and therefore, measurements of the activities of these enzymes have been used as an index of biological activity of soil (Tiwari *et al.*, 1988; Chander and Brookes, 1991).

Tezcan *et al.* (1988) stated that during the solarization, increasing soil temperatures, particularly at the top level and the resulting hydro thermal effect causes a reduction in the dehydrogenase activity.

Pitchel and Hayes (1990) reported that the activities of dehydrogenase, phosphatase were significantly inhibited after treatments of soil with alkaline power plant fly ash.

Jha *et al.* (1992) observed positive correlation between dehydrogenase activity and fungal population in forest stands at two altitudes.

Akbaba (1994) and Ozcelik (1996) claimed that enzymes are inactive above 50° C. The highest and the lowest enzyme activities were obtained at 5-10 cm and 0-1 cm, respectively. Lower enzyme activity at the top soil layer is probably due to the lower humidity in soil humidity.

Garcia *et al.* (1994) found that the rainy season enhanced the enzyme activity (DHA) of soils in the south-east arid region of Spain. Other authors also attributed the increase in microbial activity in forest (Gorres *et al.*, 1998) and in grassland soils (Banerjee *et al.*, 2000) due to higher soil moisture content.

Alef (1995) reported that acidic soil below pH 5 tend to show very low dehydrogenase activity and also pointed out that they should be interpreted with care. The data of enzyme assays have been connected to many characteristics of soil, including microbial growth and activity as the mineralization of organic P esters (Vuorinen and Saharinen, 1996).

Tiwari and Sharma (1998) recorded increased activities of dehydrogenase and urease soil enzymes with increased altitude up to 1100 msl in two mountain ranges of

Arunachal Pradesh, northeastern India. According to their values the soil organic matter content was important factor that regulates the enzyme activities in highland soils.

In an effort to assess soil quality using microbiological and biochemical procedures, Filip (1998) revealed that dehydrogenase activity measurement in soil samples affected by natural and anthropogenic activities may respond as one of the suitable indicators of soil quality.

Camina *et al.* (1998) while studying the dehydrogenase activity in acid soils rich in organic matter showed that Iodonitrotetrazolium formazan (INTF) was adsorbed by the soils with an intensity that closely relates with soil carbon content. They also suggested that a mixture of 1:1 Dimethyl-formamide-ethanol (DMF-ethanol) was more effective than methanol in extracting INTF, thereby improving estimates of dehydrogenase activity

Parham *et al.* (2002) investigated the enzyme activity in the soil and observed that dehydrogenase activity in the soil was significantly higher at the surface and decreased with increasing soil depth, with the exception of manure-treated soils. Dehydrogenase activity showed higher activity in the 20-30 cm manure-treated soil than in the 10-20 cm soil.

Quilchano and Maranon (2002) have reported that DHA measured in a forest soil in autumn samples was almost double that measured in summer samples at the same location. Significant differences were detected between the DHA values in the two seasons. They also reported that the increase in soil water content in autumn would favor the increase in microbiological activity (and hence in DHA), especially considering the low water potential values measured in summer.

Consuelo and Teodoro (2002) found that the Mediterranean forest soil properties (pH, K, Ca, Mg, and soil moisture) showed significant correlations with dehydrogenase activity. They found that the dehydrogenase activity of forest soil in autumn was almost double than in summer. During the dry season, dehydrogenase activity of forest control soils was higher than in the thinned and shrub-cleared forest. Dehydrogenase is significantly correlated with pH, K, and soil moisture in the Mediterranean forest soils.

Baum *et al.* (2003) found that a decrease of enzyme activity in Mediterranean ecosystems under drier conditions might be critical because of the decrease in nutrient supply and consequently, this might have a direct effect on WUE (Water Use Efficiency). Moreover, the re-wetting effect can reduce the bacterial population impairing soil enzyme activity.

Hawrot *et al.* (2005) stated that activity of the enzymes can give information on the kind and duration of the effects of pollutants on the metabolic activity of soil. They reported that regardless of the pollution rate, higher dehydrogenase activity was recorded in sandy soil.

Urease activity

Urease is a hydrolase enzyme responsible for hydrolytic conversion of the substrate, urea into carbon dioxide and ammonia. Due to its applied importance in the N economy of soil, the urease activity has been extensively investigated. It is well established, that soil microorganisms can produce urease, and there are reports that urease activity in soils may increase on addition of organic substances that promote microbial

activity (Gibson, 1930; Conrad, 1942a and b; Chin and Kroontje, 1963; Moe, 1967 and Balasubramanian *et al.*, 1972).

Kuprevich (1951) considered urease activity as an indicator of total biological activity and fertility of the soil. Stojanovic (1959) found marked seasonal variation in urease activity in Mississippi soils. Stojanovic (1959) and Vasilenko (1962) reported that urease enzyme activity is not a stable indicator of the biological activity of the soil. They opined that the difference in the amount of urea hydrolyzed can be attributed to change in the conditions of the soil due to season. Seasonal variations in the enzymatic activities of soil reported in a number of studies are biologically important because they change the quantity and quality of substrates upon which they act and are also responsible for altering the rate of various soil processes influenced by seasonal changes (Freytag, 1965; Galstyan, 1965; Ramirez-Martinez and McLaren, 1966).

Acccording to Skujins (1967), the enzymatic activity of stored soil samples depends on the enzyme considered. McGarity and Myers (1967) obtained significant fluctuations in urease over time of storage even during period as short as 4 days, with no satisfactory explanation.

The presence of urea in large amounts in the soil allows its use as nitrogen fertilizer worldwide. A few studies regarding relationships between urease activity and other soil properties have indicated that urease activity tends to increase in organic matter content and that sandy or calcareous soil tend to have a lower activity than heavy textured or non-calcareous soils (McGarity and Myers, 1967; Skujins, 1967; Skujins and McLaren, 1968, 1969; Myers and McGarity, 1968).

McGarity and Myers (1967) found that urease activity in Australian surface soils was highly related with pH, but they could not detect a significant relationship between pH and urease activity in a subsequent study of five samples (Myers and McGarity, 1968).

Gould *et al.* (1973) observed a significant relationship between urease activity and organic carbon in profile samples of an Alberta soil, but Pancholy and Rice (1973) found no such relationship in nine Oklahama soil. They concluded that the level of urease activity in these soils was determined by the type of vegetation.

Dalal (1975) has shown that the urease activity in Trinidad and Iowa soils was significantly correlated to organic carbon, total nitrogen and cation exchange capacity of soil.

Skujins (1976) reported that urease is primarily produced by microorganisms and its activity generally correlates with organic matter content.

Zantua and Bremner (1976, 1977) showed that soil urease increased after the addition of organic substances promoting microbial growth but subsequently decreased and eventually stabilized at the initial level.

Pettit *et al.* (1977) reviewed that soil urease showed more stability against the various degradative forces in soils in comparison to other soil enzymes. They further suggested that more stable fractions of soils urease apparently occur in active organic matter extracts.

Speir (1977) observed a strong positive relationship between urease activity and soil pH. He also found that enzyme activities were correlated significantly with several soil chemical properties related to the amount of organic matter. Studies of Tabatabai

(1977) on urease activity showed that it is concentrated in surface soils and decreases with depth. Urease activity was proportional to organic carbon distribution in soil profile and was significantly correlated with organic carbon.

Zantua *et al.* (1977) studied surface samples of 21 diverse Iowa soils representing a varied pH, texture and organic matter content to determine the relationships between soil urease activity and other soil properties. They reported that the activity was correlated significantly with organic carbon, total N and cation-exchange capacity; however, it was not significantly correlated with pH, silt or $CaCO_3$ equivalent.

Urease activity correlates positively with microbial biomass, organic carbon, soil temperature, moisture, total nitrogen and phosphorus contents of soil (Beri *et al.,* 1978; Nannipieri *et al.,* 1978; Dash *et al.,* 1981; Dkhar and Mishra, 1983; Baruah and Mishra, 1984; Sahrawat, 1984; Tiwari *et al.,* 1987b).

Burns (1978, 1982) reported that urease activity would be influenced by the type and density of vegetational cover, climate and soil type.

Verstraetan (1978) found significant indirect effects of pH on soil urease activities. He suggested that soil pH greatly influence the relationships between urease activity and organic matter content.

Stott and Hagedon (1980) examined the interrelationship between selected soil characteristics and urease activities under two forest vegetations one native grassland and three clover /grass pastures in Benton country, Oregon. Highly positive correlation was observed between urease activity and soil organic matter. Seasonal fluctuation in the urease activity was recorded where fluctuations in the activity levels were related to moisture and temperature conditions of the soils.

According to Speir and Ross (1981) soil air drying generally increases urease activity in soil. Moller (1981) noticed that urease activity was found to correlate significantly with C/N ratio and soil pH values. They regarded the activity of urease as an indicator of the biological activity of the tested soils. Dash *et al.* (1981) could establish a positive correlation between urease activity, organic C and total N. Further they reported that high temperature also affects the activity in tropical soils.

Nor (1982) studied the urease activity in several Malaysian soils and indicated that the soils have varying capacity to hydrolyze urea. Soil pH and urease activity correlated well, but neither organic carbon content nor cation-exchange capacity had any significant relationship.

O'toole *et al.* (1982) reported the significant correlations between urease activity and organic C and total N contents. They further, suggested that organic matter content, pH and temperature may be accounted for the variation in urease activity.

Sahrawat (1983) investigated the relationships between soil urease activity and other properties of some tropical wetland soils. Results of his study indicated that urease activity was correlated significantly with total N and organic carbon content. Sahrawat (1984) studied the effects of temperature and moisture on urease activity in semi-arid tropical soils, he noted that the activity increased with increase in temperature from 10^0C to a maximum at 60^0 C and further, increase in temperature, urease activity decreased which was nearly totally inhibited at 100^0 C. Urease activity increased with increase in moisture content up to field capacity and remained constant with further increase in moisture.

O'toole and Morgan (1984) investigated the thermal stabilities of urease enzymes in some Irish soils. Their results showed that soil urease enzymes were quite resistant to thermal degradation although activities did not decline during incubation. O'Toole *et al.* (1985) suggested that urease activity was affected by temperature, soil factors, such as moisture content, pH, organic matter and number of microorganisms.

Rao and Ghai (1985) estimated the urease activity of alkaline and reclaimed soils. They found that urease correlated positively with organic C and N and negatively correlated with the pH of soil. They further, demonstrated that organic C may be accounted for most of the variations in enzyme activity.

Tiwari *et al.* (1987b) noted higher urease activity during April in the surface soil which was attributable to higher organic carbon, bacterial population, temperature and favourable moisture content.

Baath (1989) showed that metal inhibition partly explained the decrease in enzyme activities in soil with high metal contents. A decrease in enzyme concentrations was, however, still evident.

Palma and Conti (1990) studied the effects of various treatments of sample and seasonal variation upon urease activities on surface samples of Argentine Agricultural soils. They noted highest urease activity during summer and lowest during winter. The variation in the urease activity in different soil types under different vegetation revealed the activity of urease enzyme is related to the type of vegetation.

Joshi *et al.* (1991) reported that soil with higher microbial population harbored higher urease activity.

Rao *et al.* (1995) observed higher urease activity during rainy season than in winter in sub-tropical pine forest soils collected from different age group stands.

Tiwari and Mishra (1995) studied seasonal variation in urease activity in hilly soils under grassland and forests of northeastern India. Their results showed higher activity of urease under forest and grassland during rainy summer season and lower during winter season.

Kandeler *et al.* (1996) demonstrated that microbial biomass and enzyme activities decreased with increasing heavy pollution using salts of heavy metals but the amount of decrease differed among the enzymes. Urease and alkaline phosphatase activities increased with high application dosage rates and this changing was found statistically significant.

Klose and Tabatabai (1997, 1998) showed that chloroform fumigation of soils resulted in an increase in urease activity of ten different Iowa surface soils when assayed in the presence of toluene.

Klose and Tabatabai (1999) also studied the relationship between urease and microbial biomass C and revealed highly significant relationship between urease activity and microbial biomass C.

Soil enzyme activity and soil microbe were closely related to soil nutrient. Zhang *et al.* (2002) reported that soil sucrase, urease and neutral phosphatase decreased as years of deforestation and soil profile depth increased.

Nourbakhsh and Monreal (2004) reported that there were no significant correlations between urease activity and soil textural properties, pH and bacterial and fungal populations.

Sardans and Peneulas (2005) reported that urease activity was always higher in the upper 0-15 cm layer than in the lower 15-30 cm layer. They also found that reduction of 10 % and 21 % of soil moisture produced by runoff exclusion decreased urease activity by 10-67 % and 42-60 % respectively. They also reported that the enzyme activities strongly decreased with soil depth and were greater in spring than in autumn.

Phosphatase activity

Phosphatase activity is essential for conversion of organic substrates containing phosphorus into inorganic form through hydrolysis in the soil. Phosphatase being an important enzyme in soil is an oxidoreductatase which plays a key role in P-cycle of the environment. The term phosphatase has been used to describe a broad group of enzymes that hydrolyze organic phosphorus (P) compounds, pyrophosphates, metaphosphates, and inorganic polyphosphates which occur in soils. It is generally accepted that plants utilize only inorganic P and since a large proportion of soil P is organically bound, the mineralization of this organic fraction can be an important factor in plant nutrition. Both acid and alkaline phosphodiesterases and alkaline phosphotriesterase exist in soils.

Phosphatase activity measurements provide an index of potentially available phosphate in soil. Rogers *et al.* (1942) described that acid phosphatase, which is present in plant roots is predominant in acid soils and is responsible for the hydrolysis of organic phosphorus in soils.

Soil phosphatase activity and the general mineralization of organic P have been reviewed extensively (Cosgrove, 1967, 1977; Ramirez-Martinez, 1968; Halstead and McKercher, 1975; Hayman, 1975; Dalal, 1977; Speir and Ross, 1978).

Cosgrove (1967) described that soil phosphatases hydrolyse phosphate esters to inorganic P which is then available to higher plants and soil microorganisms.

Tabatabai and Bremner (1969) reported that the pH 6.5 - 6.9 buffers should be used for the determination of soil phosphatase because for most soils the optimum pH of acid phosphatase falls within this range.

The development of an easy and simple method of assaying phosphatase activity in soil systems by using p-nitrophenyl phosphate (Tabatabai and Bremner,1969) as the substrate of phosphorus hydrolysis in laboratory conditions have brought the research in this field to an emerging field of soil enzymology. Phosphatase activity can be influenced by numerous factors and soil properties play a key role among them.

Skujins (1976) opined that phosphatase activity can be used to estimate the general microbial activity in soil.

In soil ecosystems, phosphatase and arylsulphatase are believed to play pivotal roles in phosphorus and sulphur cycle, respectively (Speir and Ross, 1978).

Harrison and Pearce (1979) investigated the type and parent material in woodland soils. The results of their study demonstrated that intensity of phosphatase activity and soil properties differed with soil depth, soil type, season and vegetation type.

Chhonkar and Tarafdar (1984) found a significant positive correlation between phosphatase activities and the organic carbon, phosphorus and bacterial population but

they did not find any correlation with soil pH. Their results revealed that none of the phosphatases was found to be correlated with clay content.

Camara *et al.* (1985) recorded the phosphatases activity between 435 and 464 μg p-nitrophenol $h^{-1}g^{-1}$ of soil. The results showed greater enzyme substrate affinity in soil when the size of soil particles decreased.

Trasar-Cepeda and Gil- Sotres (1987) studied phosphatase activity of acid soils with high organic matter content in forest soils. They found higher activity of acid phosphatase between pH 5 and 6, which appeared to depend on organic activity of soil suggesting that enzyme originating from litter was progressively inhibited as it penetrated the soil.

Soil phosphatase activity is also affected by soil moisture and soil depth. Harrison (1983, 1987), Herbien and Neal (1990) have emphasized the soil moisture. It affects phosphomonoesterases by promoting the development of microbial communities. There is a balance between soil moisture and soil biocenose.

Garcia *et al.* (1993) indicated that phosphatase activity might be an indicator of organic matter in the composting process. Soil microorganisms and plant can utilize soil organic P by means of phosphatases (Hino, 1989; Pant *et al.*, 1994). In areas of Central Spain soil enzyme activity decreased proportionally to the decrease of plant cover (Garcia *et al.*, 2002).

Hoffman and Elias-Azar (1995) reported that the intensity of phosphatase activity in soils has been found to soil physical and chemical properties, such as soil pH, contents of nitrogen, organic matter and plant available phosphorus.

Sousa and Nahas (1998) studied the enzymatic activity of soil samples stored at temperatures of 5 to -12^{0}C and at room temperature for 0-32 weeks. They reported that alkaline phosphatase and dehydrogenase activity decreased compared to control samples stored at low temperatures, acid phosphatase activity showed no significant change.

Lyons *et al.* (1998) observed soil phosphatase, is mostly of plant and microbial origin and consists of alkaline and acid phosphatases. The lowest enzyme activity was recorded in dry soils and this increased with increased in moisture content in soil at maximum moisture holding capacity.

Baligar *et al.* (1998b) found very close associations between soil phosphatase activity and moisture content, organic C in acid soils of the Appalachian Mountains.

Kang and Freeman (1999) revealed that phosphatase enzyme mediates the release of inorganic phosphorus from organically bound phosphorus which returned to soil as litter and other organic debris. The intensity of phosphatase is important as it affects the rate of phosphorus cycling. Hydrogen ion concentration is a dominant controlling factor for the phosphatase activities in soil.

Kathleen *et al.* (2001) reported that P addition consistently reduced phosphatase activity and P uptake capacity in Hawaiian rain forests. Investment in acquisition of a nutrient was greatest when that nutrient was limiting to growth, and plants appeared to allocate excess N to construction of extra-cellular phosphatase to acquire P.

Dilly and Nannipieri (2001) found that the presence of P usually decrease phosphates activity.

Turner *et al.* (2002) and Baum *et al.* (2003) reported that phosphatase activities were higher in the first 15cm layer of soil profile than the values reported by other authors, but this was not the case for the 15-30cm layer.

Cookson (2002) reported the effects of soil organic carbon; salinity and organic content were significant in accounting for over 80 % of the variance in phosphatase activity. Soil salinity ratio of 1:5 (soil: water) extracts appeared to almost entirely inhibit phosphatase activity.

Chen (2003) reported that the activity of the enzymes analyzed decreased with soil depth, confirming a result that has been widely observed. The asymmetrical inputs of litter through soil profile due to the aboveground litter incorporation on the soil surface together with a better aeration account for the higher soil enzyme activity in the 0-15cm than in the lower 15-30cm. He observed decreased in soil enzyme activities through soil profile in forest and agriculture soils when there are organic inputs in the soil surface.

Klose *et al.* (2001; 2003a) reported that the activities of acid phosphatase in the humus layer and mineral topsoil decreased with decreasing deposition loads. In the mineral topsoil (0-10 cm), no consistent trend was found for the specific enzymes activities.

Sarapatka (2003) found that soil phosphatases play a major role in the mineralization processes of organic phosphorus substrates. The activity of soil phosphatase can be influenced by numerous factors and soil properties and farming systems play a key role among them. Positive correlations were found between enzymatic activity and organic carbon, and with nitrogen; and between acid phosphatase activity and total phosphorus. He also observed negative correlation with the quality of humus

(humic: fulvic acids ratio) and available phosphorus, and between acid phosphatase activity and clay content and pH.

Sardans *et al.* (2004) observed that acid and alkaline phosphatase activities were twice as high in spring than in autumn. In the spring sampling no effects of drought on acid phosphatase and on alkaline phosphatase were observed in the upper 0-15 cm layer or in the lower 15-30 cm layer. In autumn, significantly higher acid and alkaline phosphatase activities were observed in the 0-15cm soil layer than in the 15-30 cm soil layer in all treatments. No interactions between soil layers treatments were observed in the ANOVA analysis.

Physico-chemical characteristics of soil

Physico-chemical characteristics such as temperature, moisture, pH and nutrient contents of the soil regulate the population and activity of macro and microorganisms. Chemical and biological properties of the soil in turn are regulated by the soil organic matter, which is one of the major pools of carbon and nutrients, and also regulates to a large extent the physical properties of the soil. Interaction between plants, microbes and the physico-chemical processes in soil together with the soil organic matter dynamics influence nutrient and water availability to the plants. Conversion of natural to agricultural systems may cause drastic changes in the environment for soil biota. The buffer provided by dense vegetation against fluctuations in microclimate is lost with land clearing, resulting in large extremes in soil temperature and moisture conditions. A change from natural vegetation to annual crops often decreases organic matter input. The

physico-chemical characters of the soil may regulate the microbial population and their activities (Waksman, 1927 and Warcup, 1950; Mishra, 1966; Tiwari *et al.,* 1987).

Waksman (1927) considered soil pH, organic matter, moisture, temperature, aeration and nature of the crop grown to be responsible for the distribution and abundance of microbe in soil.

Saksena (1955) suggested that higher moisture content of soil was favourable for the growth of fungi as long as there was no water logging.

In tropical forest soils, where temperature is not limiting, Wong and Nortcliff (1955) found that soil moisture fluctuations between wet and dry seasons largely regulate nitrogen mineralization rates. Orput and Curtis (1957) reported that soil moisture was an important factor in controlling the distribution of fungi.

Yadav and Badolka (1973) found that the soils of deodar forests of Uttar Pradesh (India) were deficient in phosphorus.

Bieleski (1973) studied the phosphorus transport and its availability to the plants in soils. He stated that phosphate is relatively immobile and this may be the reason for the little variations noted along depths.

Halstead and Mckercher (1975) stated that as much as 5-10 % of the organic phosphorus is associated with living microbial tissue.

Zinke *et al.* (1978) reported that the high temperature releases cations in the surface soils thereby increasing the potassium concentration of the soil.

Singh (1980) studied the nutrient cycling in a subtropical humid forest of Meghalaya and reported that most of the nutrients were accumulated at 10-40cm soil

depth. He further, observed that below this depth, nutrient concentration declines drastically.

Lynch and Panting (1980a) found that sieving wet soil and grinding moist soil were found to be affected by the C contents of soils.

Many soil microorganisms are known to be intolerant of low soil moisture contents (Reid, 1980; Harris, 1981; Paul and Clark, 1989) and changes in soil moisture status can result in rapid shift in the magnitude of the soil microbial biomass (Bottner, 1985; Schnurer *et al.*, 1986).

Due to the importance of nitrogen in forest productivity ecosystem and nutrient cycling, research often includes measurement of soil N transformation rates as indices of potential availability and ecosystem losses of nitrogen. Keeney (1980) stated that although forest soil nitrogen (N) pools can be quite large, nitrogen availability often limits forest growth and productivity.

Ewel *et al.* (1981) have reported a 30 % reduction in soil C and 23 % reduction in N at upper soils after burning in Costa Rican wet forests.

A variety of extracting solutions are used to assess plant-available K in different parts of the world (Knudsen *et al.,* 1982). Ion exchange resins have been used to measure the potassium released in soil (Pratt 1951; Arnold, 1958; Havlin and Westfall, 1985; Heming and Rowell, 1985).

Relationships between annual precipitation and the content of soil organic C of top soils have been established for many regions (Spain *et al.*, 1983).

Dkhar (1983) reported maximum concentration of available phosphorus in the soil during the month of July in jhum, terrace and valley lands and suggested that this maybe

due to the greater microbial activity and release of soluble phosphate because of suitable temperature.

Orchard and Cook (1983); Kieft *et al.* (1987); Skoop *et al.* (1990) showed a positive relationships between soil moisture content or water potential and microbial activities or microbial biomass.

Snyder and Harter (1984) found soil carbon increased from 3 to 10 year following disturbance.

Schnurer *et al.* (1985) and Dick (1994) reported that the physical and chemical properties of soils can directly affect the structure, spatial distribution and activity of microbial populations and enzymes in soils, which are potential early indicators of soils health and quality.

McLean and Watson (1985) stated that the highest and biggest practical difficulty in assessing available potassium reserves of soil arises from the soil's ability to release K from non-exchangeable sources.

McGill *et al.* (1986) suggested that temperature affects only the activity, and not the size of the soil microbial biomass.

Soil microorganisms and plants can utilize soil organic P by means of phosphatases (Tarafdar and Junk 1987; Hino, 1989; Pant *et al.*, 1994b).

Soil K is often subdivided into soluble, rapidly exchangeable, slowly exchangeable and structural forms (Sparks, 1987). This arbitrary subdivision account for differences in bioavailability.

Swank and Crossley (1988); Yanai (1992); Likens and Bormann (1995); Zhang and Mitchell (1995) investigated undisturbed mature hardwood ecosystems in humid,

eastern North America cycle nutrients, particularly P. They reported that P conservation may vary considerably in forest ecosystems.

The increase in organic C with precipitations reflects the increase in plant productivity, and hence the increase of plant C to soil (Burke *et al.*, 1989).

Albaladejo and Diaz (1990); Parr and Papendick (1997) revealed that chemical and physical soil parameters have been used to measure soil quality in long-term experiments. Cassman and Munns (1990) found a significant relationship between soil moisture and temperature and on net N mineralization.

Rastin *et al.* (1990) investigated a number of biological and biochemical factors in different horizons from the upper and lower slopes of a spruce forest. They reported that NH_4^- and NO_3^- N concentrations in the soil solution showed significant correlation with most of the biological and biochemical soil factors investigated.

Insam *et al.* (1991) also suggested that N availability has little effect on microbial biomass C, although their results showed some relationship between microbial biomass C and total N of soil.

Vitousek and Howarth (1991) observed that in most ecosystems, N availability is controlled by several aspects of the physical and chemical environment, including availability of other nutrients (partly a function of parent material), temperature, and moisture.

Tiwari *et al.* (1992) reported seasonal variation in P contents of soils. The P concentrations reached maximum peak during spring which might be related to rapid release of the nutrients from liter. Further, they reported temporal and depth wise variation in organic C, total N and P from pineapple orchard stands of northeastern India.

Simard *et al.* (1992) reported that uptake of K from soil solution, is buffered by readily exchangeable forms, which in turn, is slowly replenished by soil K reserves (slowly exchangeable and structural forms). Dynamic reactions exist between solution, exchangeable, fixed and mineral phases of soil K.

Doran (1994) showed that characterization of active N pools in soils can provide valuable information for evaluating soil health.

Kennedy and Smith (1995) stated that the cycling of elements in soils and thus, ecosystems functioning, is governed largely by microbial activity, the effects of cropping systems and fertilization on the soil biota are of ecological importance.

Juo *et al.* (1995) reported that a linear decrease in soil pH up to 4 was reported after forest clearing which resulted inhibition of plant growth in Nigeria after 13 years of regeneration period.

Nayak and Srivastava (1995) reported that higher status of organic carbon content in the surface horizon which could be ascribed to slow microbial activity under low temperature and acidic environments of the shifting cultivated lands of this region.

Singh *et al.* (1995) found presence of higher N content at upper soil layers which decreased with increase in soil depth in the natural forest soils followed by bamboo forest and jhum fallow of northeastern India.

Chantigny *et al.* (1996) found a correlation between the water-soluble organic C and microbial biomass C under various annual and perennial species, suggesting differences in C deposition from the plant species.

Arunachalam *et al.* (1996) reported a decreasing trend of soil moisture content, bulk density and water holding capacity with increase in degree of forest disturbance in pine forests of Meghalaya in northeastern India.

Deforestation of natural forests and their subsequent tillage practices in Central Zgarous mountain of Iran resulted in almost 50 % decrease in organic matter content and total nitrogen, a 10-15 % decrease in soluble ions when compared to the undisturbed forest soils (Hajabbasi *et al.*, 1997).

Chardon *et al.* (1997) reported that 70-90 % of total P from leachates of soil columns and lysimeters were in organic form and dissolved organic P (DOP), as a percentage of total P, increased with increasing soil depth, suggesting that DOP is an important form of P in soil solution and leachate.

Carpenter *et al.* (1998) estimated that P accumulation rates in the USA and several European countries range from 8 to 40 kg P ha^{-1} $year^{-1}$ with an average rate of 22 kg P ha^{-1} $year^{-1}$.

Gebauer *et al.* (1998) have shown that microbial activity is inhibited in acidic soils, whereas in soils with a higher pH they can potentially compete with the fine roots for N.

Saikh *et al.* (1998) reported significant reduction in organic C, total N, and C: N ratios as a result of deforestation in Simlipal National Park in India.

Arunachalam *et al.* (1998) reported maximum P input during winter and spring seasons in the 7, 13 and 16 years old forest re-growths of north eastern India. The level of P was more or less same in all the four seasons under the study.

Saikh *et al.* (1998) reported significant reduction in organic C, but no significant changes in total P levels as a result of deforestation and cultivation in Simlipal National park in India. Topsoil P content is usually greater than in the subsoil due to the sorption of added P and greater biological activity of organic material.

P dynamics in riparian forests were shown to have significant spatial and temporal variability (Lyons *et al.*, 1998) and were linked to hydrologic P transport.

Ajwa *et al.* (1999) found that inorganic N was negatively correlated with urease enzymes activity, due to low soil inorganic N caused by plant uptake and microbial mobilization.

Acid soils are common in temperate humid zones and are frequently dedicated to forestry exploitations (Fernandez and Carballas, 2000).

Soil P content varies with the parent material, extent of pedogenesis, soil texture and management factors, such as rate and type of P applied and soil cultivation (Sharpley, 2000).

Treseder and Vitousek (2001) reported that nitrogen availability markedly increased investment in extracellular phosphatase.

Valued for stock of soil organic carbon at regional, national, continental, or global level is essential information for discussing changes in carbon contents or fluxes in various scales. However, problem arising from soil sampling, soil variability, and depth create wide difficulties in the estimation of C reserves (Swift, 2001).

Couteaux *et al.* (2001) observed that the moisture content response was close to the optimal value at the boreal and Atlantic sites, whereas it was reduced by summer

drought at the Mediterranean sites. In addition they reported that the seasonality of moisture content was less pronounced in the northern sites.

Jha *et al.* (2001) observed maximum moisture content in August and the least moisture in May. They found that during January, February, July and August moisture content decreased with increasing soil depth while in remaining months moisture content increased with increasing soil depth.

Lehmann (2001) stated that soil nutrient availability was not only related to the amount of nutrient applied but was also influenced by the tree species. Nutrient return by litter fall and litter quality played an important role in soil P dynamics.

After addition of carbon on highly weathered rainforest soils and younger soil from the same area, Cleveland *et al.* (2002) found that the increase in microbial activity was more limited by phosphorus availability in the weathered soil.

Ilstedt *et al.* (2003) observed an intricate difference between nitrogen and phosphorus, where phosphorus seemed to limit the initial growth, while nitrogen limited the final plateau. This was argued to be due to phosphorus fixation making phosphorus more difficult to extract than nitrogen, but in total more phosphorus was available than nitrogen in relation to the needs of the microbes.

Giesler *et al.* (2004) hypothesized that the presence of P sorption sites in the humus layer affects microbial growth dynamics because more C has to be allocated to P-fixing capacity.

A decrease in P content of the soil and biomass together with a decrease in the soil P-available forms in response to a more pronounced drought has been observed in Mediterranean forests (Sardans and Peneulas, 2004).

RESULTS

Quantitative Estimation of Fungal and Bacterial Populations of the Soil

Fungal population

The fungal population exhibited almost a similar trend of monthly variations in the soils of the two different forest stands during the study periods of 2002 and 2003. The soil of the undisturbed forest harbored higher fungal population as compared to that of disturbed forest. Maximum fungal population was observed in the month of September during the first and second year of study periods in both the study sites and the minimum was recorded in the months of March and June in the undisturbed and disturbed forests soils respectively in the first year whereas, in the second year, the minimum was recorded in the months of May and January in the undisturbed and disturbed forest soils respectively. In general, the population was found to be higher in summer rainy seasons and lower population was observed in winter months. The fungal population decreased with increase in depth (Fig. 2).

Bacterial population

The bacterial population also exhibited a similar trend of monthly variations throughout the study periods as that of fungal population. The soil of the undisturbed forest harbored higher bacterial population than that of the disturbed forest. The maximum bacterial population was recorded in the month of September and minimum was observed in the month of July in both the study sites during year 2002. In the year 2003, the maximum bacterial population was recorded in the month of January in both

the study sites and the minimum was observed in the months of February and June in the undisturbed and disturbed forest soils respectively. The bacterial populations decreased with increase in depth (Fig.3).

Distribution of Fungal and Bacterial Species and Their Percentage Relative Abundance

Table 2 depicts list of fungal species isolated from the soils of both the study sites at three different depths i.e. 0-10cm, 10-20cm and 20-30cm. Altogether 95 fungal species were isolated of which 12 belonged to Zygomycotina, 80 belonged to Deuteromycotina and 3 Mycelia Sterilia. Qualitatively, there was not much difference in the fungal species isolated from the soil at both the study sites. Zygomycotina was represented by 6 genera and 12 species in which *Mortierella parvispora*, *M. rammaniana* and *Pythium intermedium* were found in all depths of both the study sites. Deuteromycotina was represented by 23 genera and 80 species out of which *Acremonium butyri, A. murorum, Aspergillus candidus, A. fumigatus, A. niger, Cladosporium cladosporioides, Humicola fuscoatra, Paecilomyces carneus, P. lilacinus, Penicillium brevicompactum, P. chrysogenum, P. expansum, P. frequentans, P. janthinellum, P. jensenii, P. lanosum, P. nigricans, P. oxalicum, P. purpurogenum, P. simplicissimum, P. restrictum, P. rubrum, Penicillium* sp, *P. waksmanii, Phoma eupyrena, Trichoderma viride, Verticillium alboatrum, V. chlamydosporium* and white sterile mycelia were found in all the three depths of both the study sites. Majority of the fungal species isolated were common to both the study sites. In the undisturbed forest soil, the dominant fungal species found at 0-10cm depth were *Penicillium brevicompactum, P. rubrum, P. jensenii, Scopulariopsis brumptii, Verticillium alboatrum ;* at 10-20cm depth, *Aspergillus alutaceus, A. fumigatus,*

Eupenicillium javanicum, P. brevicompactum, P. chrysogenum, P. expansum, Trichoderma viride and at 20-30cm depth, *A. fumigatus, Cladosporium cladosporioides, P. brevicompactum, P. jensenii, P. rubrum, T. viride.* In the disturbed forest soil, the dominant fungal species were *Acremonium kiliense, Absidia glauca, Arthrobotrys conoides, P. brevicompactum, P. rubrum, T. viride, Scopulariopsis brumptii, Verticillium alboatrum* at 0-10cm depth; *Acremonium butyri, Acremonium kiliense, C. cladosporioides, Humicola grisea, Mortierella vinacea, P. chrysogenum, P. janthinellum, P. rubrum, P. waksmani, Phoma eupyrena, Verticillium alboatrum* at 10-20cm depth and *C. cladosporioides, P. brevicompactum, P. chrysogenum, P. janthinellum, P. jensenni, P. purpurogenum, P. rubrum, T. viride, Pythium intermedium and Paecilomyces lilacinus* at 20-30cm depth. (Table 3-14).

However, a few species were restricted to each study site. *Acremonium fusidioides, A. sclerotiorum, Aspergillus carneus, A. clavatus, A. versicolor, Aspergillus* sp., *Eupenicillium lapidosum, Heteroconium chaetospira, Nectria ventricosa, Penicillium daleae, P. rugulosum, P. stoloniferum, P. variable, Pythium irregulare, Trichoderma harzianum, T. pseudokoningii, T. piluiliferum* and yellow sterile mycelium could be isolated only from the soil of undisturbed forest whereas, *Aspergillus terreus, Botryotrichium piluiliferum, Eupenicillium sheari, Mucor racemosus, Penicillium granulatum, Plectosphaerella cucumerina, Trichoderma polysporum and Verticillium catenulatum* were restricted to that of the disturbed forest. *Trichoderma* sp., *Penicillium* sp. was isolated frequently from both the study sites. *Fusarium* sp. was isolated occasionally from both the study sites. *Fusarium* sp. was mostly isolated from deeper layer of the soil (Tables 3 to14).

Shannon index of general diversity of fungal species in the first year (2002) ranged from 1.32 to 2.51, 0.38 to 2.34 and 0.50 to 2.25 at 0-10cm, 10-20cm and 20-30cm respectively in the disturbed forest soil, whereas, in the undisturbed forest, it ranged between 1.61 to 2.36, 1.19 to 2.29 and 0.64 to 2.14 at 0-10cm, 10-20cm and 20-30cm respectively. In the second year (2003), the Shannon diversity index ranged from 1.38 to 2.60, 1.21 to 2.40 and 1.09 to 2.43 at 0-10cm, 10-20cm and 20-30cm respectively in the disturbed forest soil, whereas, in the undisturbed forest soil it ranged between 1.36 to 2.53, 0.87 to 2.11 and 1.21 to 1.98 at 0-10cm, 10-20cm and 20-30cm respectively. Shannon's diversity index showed less variation in the disturbed forest as compared to that in undisturbed forest soil (Fig. 4).

A total of 7 bacterial species could be isolated from the soils of both the study sites at different depths (Table 15). These were *Arthrobacter* sp., *Bacillus* sp., *B. cereus, B. subtilis, Micrococcus* sp., *Pseudomonas* sp. and *Rhizobium* sp. Majority of the bacterial species isolated were common to both the forest soils. *Bacillus cereus* was isolated only from the soil of disturbed forest. The dominant bacterial species were *Arthrobacter* sp., *Bacillus* sp., *B. subtilis and Micrococcus* sp. The bacterial species viz., *Arthrobacter* sp,*Bacillus cereus*, *Pseudomonas* sp. and *Rhizobium* sp. were found to occur less frequently in the both the study sites (Tables 16 to 27).

Shannon index of general diversity of bacterial species in the first year (2002) ranged from 0.73 to 1.57, 0.77 to 1.36 and 0.33 to 1.55 at 0-10cm, 10-20cm and 20-30cm respectively in the disturbed forest soil and between 0.25 to 1.37, 0.37 to 1.58 and 0.40 to 1.59 at 0-10cm, 10-20cm and 20-30cm respectively in the undisturbed forest soil. In the second year (2003), the Shannon diversity index ranged from 0.75 to 1.35, 0.59 to 1.26

and 0.78 to 1.49 at 0-10cm, 10-20cm and 20-30cm respectively in the disturbed forest soil, whereas, in the undisturbed forest soil, it ranged from 0.35 to 1.53, 0.09 to 1.49 and 0.69 to 1.56 at 0-10cm, 10-20cm and 20-30cm respectively. Shannon's diversity index in bacteria showed less variation in disturbed forest soil than that in undisturbed forest soil (Fig.5).

The one way analysis of variation (ANOVA) result showed significant variation ($P \leq 0.05$) of fungal population at the three different depths between the two study sites. The one way analysis of variation (ANOVA) result of bacterial population varied significantly ($P \leq 0.05$) at the three different depths between the two study sites (Table 28).

The fungal population varied significantly ($P \leq 0.05$) among all the three depths of each study site. While the bacterial population also varied significantly ($P \leq 0.05$) among the three depths of both the study sites and insignificance variation was observed at DF (10-20cm x 20-30cm) and UDF (10-20cm x 20-30cm) (Table 29).

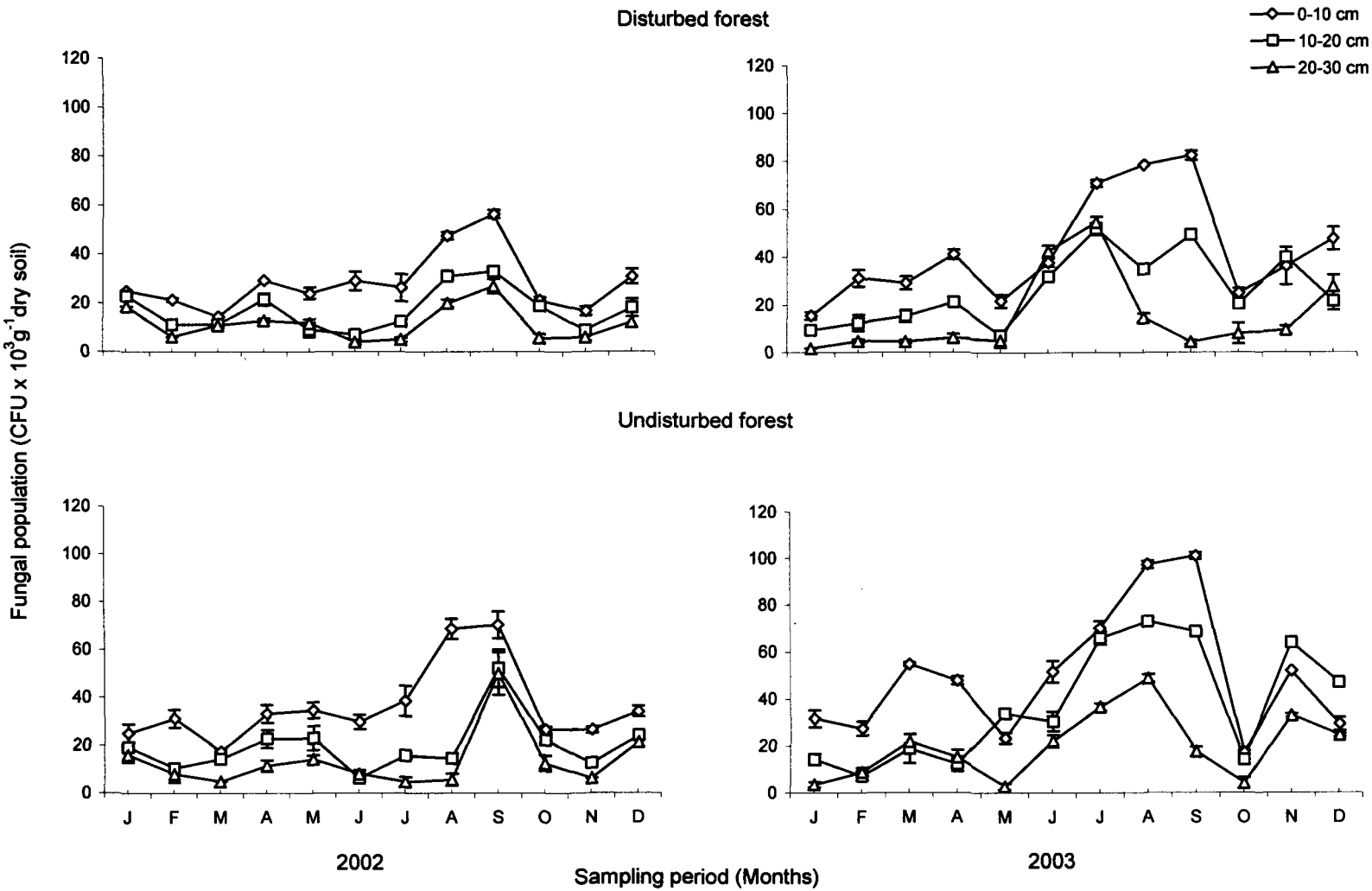

Fig. 2 Fungal population in disturbed and undisturbed forest soils at three different depths 0-10cm,10-20cm and 20-30cm

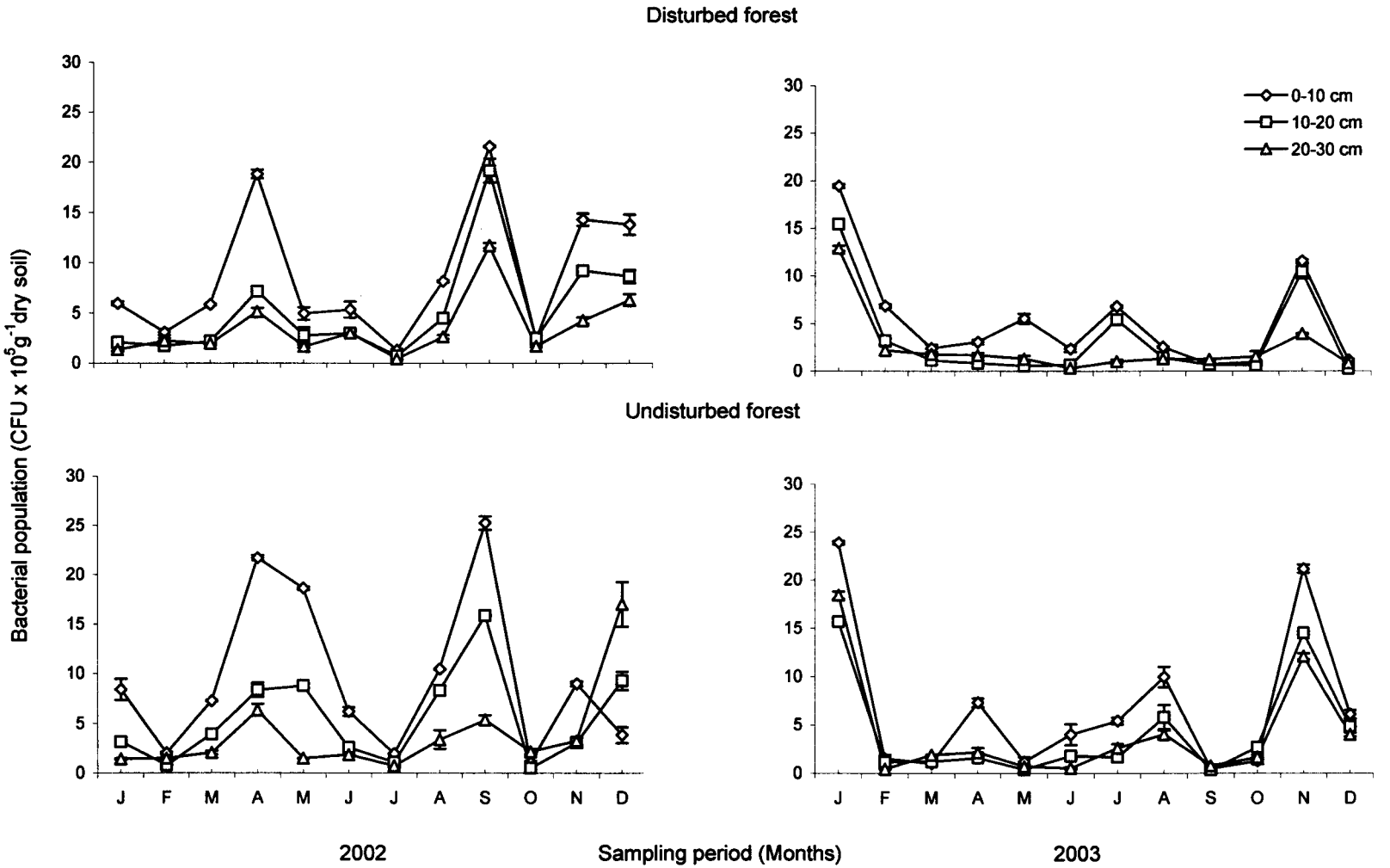

Fig. 3 Bacterial population in disturbed and undisturbed forest soils at three different depths 0-10cm,10-20cm and 20-30cm

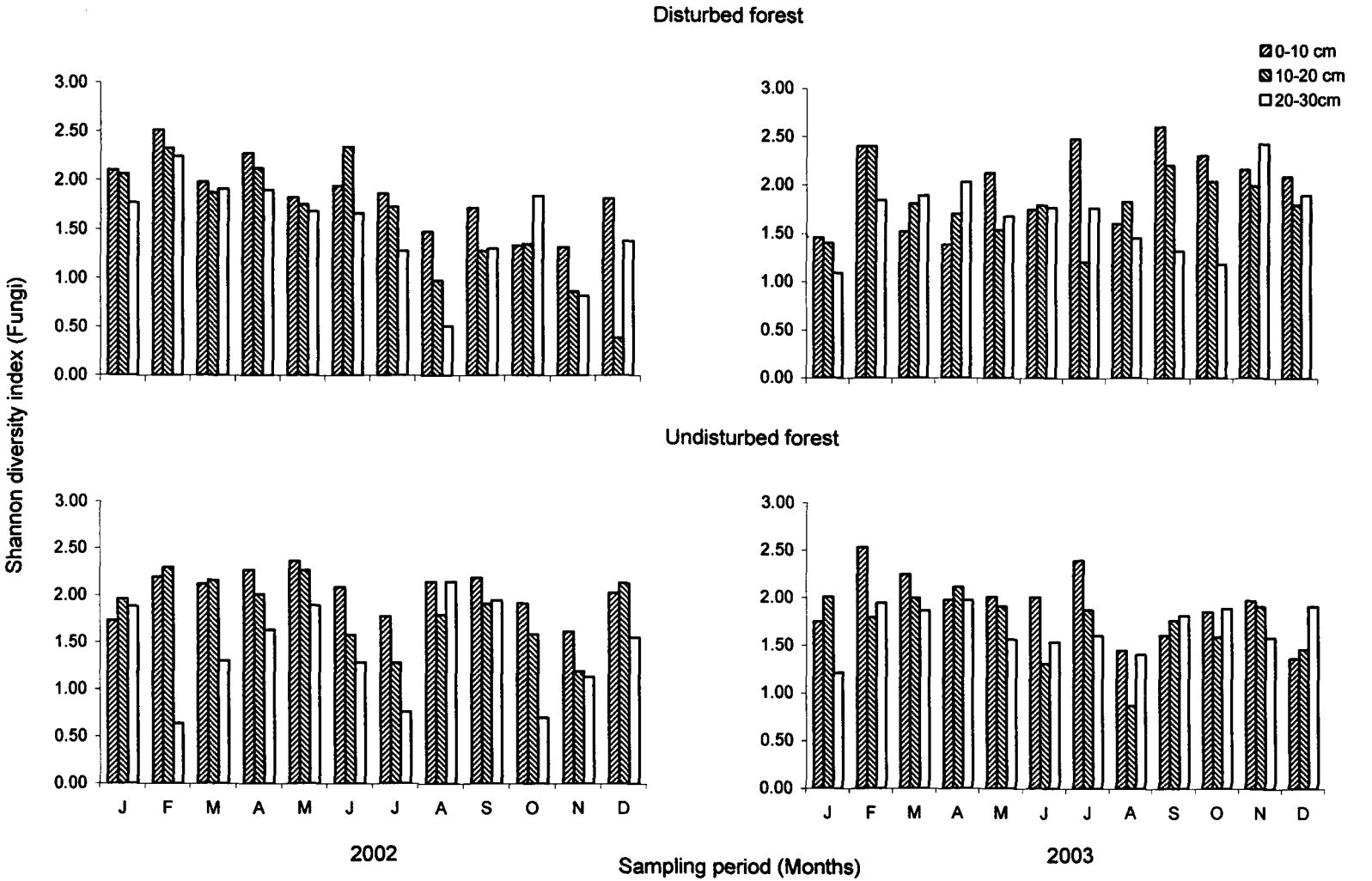

Fig. 4 Shannon diversity index (fungi) in disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20-30cm

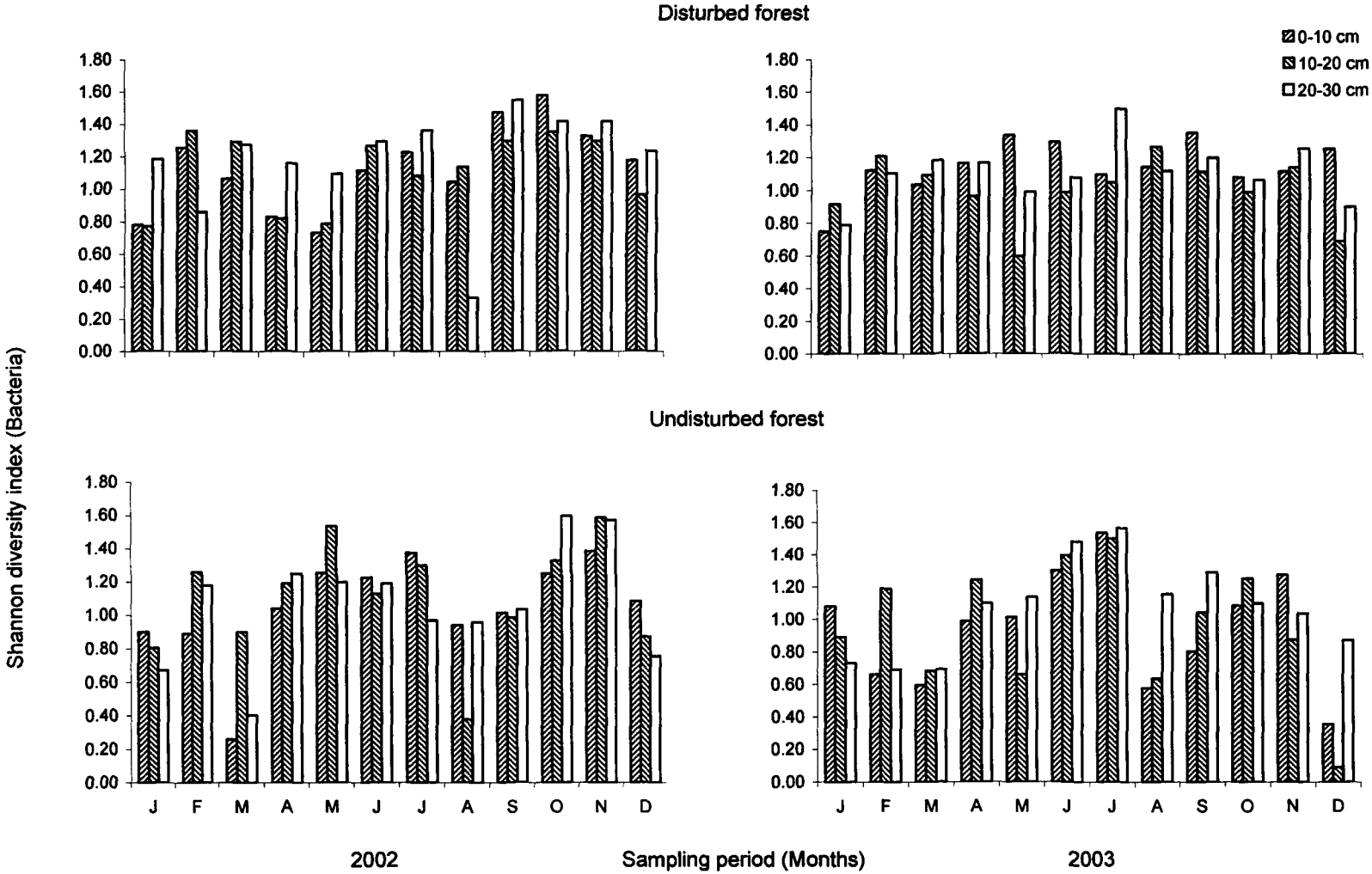

Fig. 5 Shannon diversity index (bacteria) in disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20-30cm

Table 2 List of fungal species isolated from disturbed and undisturbed forest soils

Sl. No	Fungal Species	Disturbed Forest	Undisturbed Forest
1	*Absidia cylindrospora*	+	+
2	*A. glauca*	+	+
3	*Acremonium butyri*	+	+
4	*A. cerealis*	+	+
5	*A. fuscidioides*	-	+
6	*A. kiliense*	+	+
7	*A. murorum*	+	+
8	*A. sclerotiorum*	-	+
9	*A. strictum*	+	+
10	*Arthrobotrys conoides*	+	+
11	*Aspergillus* sp.	-	+
12	*A. alutaceus*	+	+
13	*A. candidus*	+	+
14	*A. carneus*	-	+
15	*A. clavatus*	-	+
16	*A. flavus*	+	+
17	*A. fumigatus*	+	+
18	*A. niger*	+	+
19	*A. terreus*	+	-
20	*A. versicolor*	-	+
21	*Botryotrichum piluliferum*	+	-
22	Brown sterile mycelia	+	+
23	*Chaetomium crispatum*	+	+
24	*Chaetosphaeria vermicularioides*	+	+
25	*Cladosporium cladosporioides*	+	+
26	*Eupenicillium brefeldianum*	+	+
27	*E. javanicum*	+	+
28	*E. lapidosum*	-	+
29	*E. sheari*	+	-
30	*Fusarium redolens*	+	+
31	*Gliocladium roseum*	+	+
32	*Helminthosporium velutinum*	+	+
33	*Heteroconium chaetospira*	-	+
34	*Humicola fuscoatra*	+	+
35	*H. grisea*	+	+
36	*Mammaria echinobotryoides*	+	+
37	*Mortierella hyalina*	+	+
38	*M. parvispora*	+	+
39	*M. polycephala*	+	+
40	*M. ramaniana*	+	+
41	*M. vinaceae*	+	+
42	*M. humilis*	+	+
43	*Mucor racemosus*	+	-
44	*Nectria ventricosa*	-	+
45	*Paecilomyces carneus*	+	+
46	*P. lilacinus*	+	+
47	*P. marquandii*	+	+
48	*P. variotii*	+	+
49	*Penicillium* sp.	+	+
50	*P. brevicompactum*	+	+

51	*P. canescens*	+	+
52	*P. chrysogenum*	+	+
53	*P. daleae*	-	+
54	*P. expansum*	+	+
55	*P. frequentans*	+	+
56	*P. funiculosum*	+	+
57	*P. granulatum*	+	-
58	*P. janthinellum*	+	+
59	*P. jensenii*	+	+
60	*P. lanosum*	+	+
61	*P. lividum*	+	+
62	*P. nigricans*	+	+
63	*P. oxalicum*	+	+
64	*P. purpurogenum*	+	+
65	*P. rugulosum*	-	+
66	*P. restrictum*	+	+
67	*P. rubrum*	+	+
68	*P. sacculum*	+	+
69	*P. simplicissimum*	+	+
70	*P. stoloniferum*	-	+
71	*P. thomnii*	+	+
72	*P. variable*	-	+
73	*P. waksmanii*	+	+
74	*Phoma eupyrena*	+	+
75	*P. medicaginis*	+	+
76	*Plectosphaerella cucumerina*	+	-
77	*Pythium intermedium*	+	+
78	*P. irregulare*	-	+
79	*Rhizopus oryzae*	+	+
80	*Scopulariopsis brevicaulis*	+	+
81	*S. brumpti*	+	+
82	*S. fusca*	+	+
83	*Torula herbarum*	+	+
84	*Trichoderma harzianum*	-	+
85	*T. koningii*	+	+
86	*T. polysporum*	+	-
87	*T. pseudokoningii*	-	+
88	*T. piluliferum*	-	+
89	*T. viride*	+	+
90	*Verticillium alboatrum*	+	+
91	*V. catenulatum*	+	-
92	*V. chlamydosporium*	+	+
93	*V. dahliae*	+	+
94	White sterile mycelia	+	+
95	Yellow sterile	-	+

+ = present
- = absent

Table 3 Monthly variation of fungal population (per gram dry soil x 10^3) in the disturbed forest soil at depth 0-10cm (2002)
Values in parentheses are percentage relative abundance

Sl. No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Absidia glauca*	-	-	-	-	-	-	-	-	-	7.50 (35.71)	-	-
2	*Acremonium butyri*	-	-	-	-	1.09 (4.57)	-	0.51 (1.98)	3.06 (6.45)	-	-	-	-
3	*A. kiliense*	-	-	-	-	-	-	-	-	14.36 (34.68)	-	-	-
4	*A. murorum*	-	-	-	-	-	-	0.51 (1.98)	-	-	3.00 (14.28)	-	-
5	*Arthrobotrys conoides*	-	-	-	-	-	-	-	-	-	-	7.50 (37.51)	-
6	*Aspergillus alutaceus*	1.45 (5.88)	-	-	-	-	-	-	-	-	-	-	-
7	*A. fumigatus*	-	-	2.38 (16.60)	-	-	-	-	-	-	-	-	-
8	*A. niger*	-	-	-	-	-	2.71 (7.90)	3.14 (12.04)	-	-	-	-	-
9	*Botryotrichum piluliferum*	-	-	3.82 (26.6)	-	-	-	-	-	-	-	-	2.03 (6.65)
10	*Helminthosporium velutinum*	-	-	-	-	-	1.08 (3.14)	-	-	-	-	-	-
11	*Humicola fuscoatra*	-	0.45 (2.41)	1.91 (13.3)	-	-	-	-	-	0.54 (1.32)	-	-	-
12	*H. grisea*	-	-	-	-	-	-	-	-	-	4.50 (21.42)	-	-
13	*Mammaria echinobotryoides*	2.42 (9.76)	-	-	-	-	-	-	1.01 (2.13)	-	-	-	-
14	*Mortierella vinacea*	-	1.38 (7.32)	-	-	-	-	-	-	-	-	6.00 (30.01)	-
15	*Paecilomyces carneus*	-	-	-	-	-	6.54 (19.04)	1.03 (3.97)	-	-	-	-	-
16	*P. variotii*	-	-	-	4.37 (14.08)	-	-	-	-	-	-	-	-
17	*P. lilacinus*	-	-	-	-	-	-	1.03 (3.97)	-	6.06 (14.68)	-	-	-

18	*Penecillium brevicompactum*	4.37 (17.64)	0.91 (4.83)	-	-	6.23 (26.14)	3.27 (9.52)	-	-	-	-	-	11.76 (38.33)
19	*P. canescens*	-	-	-	-	-	-	-	-	-	-	-	2.03 (6.65)
20	*P. chrysogenum*	-	3.69 (19.47)	-	4.86 (15.63)	-	-	-	-	-	-	-	-
21	*P. expansum*	-	-	-	-	-	-	-	-	-	-	-	3.57 (11.65)
22	*P. frequentans*	-	1.84 (9.73)	-	0.96 (3.28)	-	-	1.57 (6.02)	-	-	-	-	4.60 (15.00)
23	*P. funiculosum*	2.91 (11.76)	-	-	-	-	-	-	-	-	-	-	-
24	*P. janthinellum*	-	-	-	-	-	7.08 (20.61)	-	-	-	-	-	-
25	*P. jensenii*	-	-	-	-	3.89 (16.33)	-	-	-	3.86 (9.32)	-	-	2.56 (8.35)
26	*P. lanosum*	-	-	-	2.42 (7.79)	-	-	-	-	-	-	-	-
27	*P. nigricans*	1.94 (7.82)	-	-	-	-	-	-	-	-	-	-	-
28	*P. oxalicum*	3.88 (15.64)	-	-	-	-	-	-	-	-	-	-	-
29	*P. purpurogenum*	1.45 (5.88)	0.45 (2.41)	0.94 (6.60)	3.94 (12.67)	-	-	-	-	-	-	-	-
30	*P. restrictum*	-	1.38 (7.32)	-	-	-	-	-	4.07 (8.60)	-	-	-	-
31	*P. rubrum*	-	0.45 (2.41)	-	-	-	-	7.87 (30.12)	1.01 (39.78)	-	-	-	-
32	*P. sacculum*	-	-	-	-	3.62 (15.23)	-	-	-	-	-	-	-
33	*P. simplicissimum*	-	-	-	1.45 (4.69)	4.20 (17.64)	-	-	-	-	-	-	-
34	*P. thomnii*	-	-	-	3.88 (12.67)	-	-	-	-	-	-	-	-
35	*P. waksmanii*	4.37 (17.34)	-	-	-	-	6.54 (19.04)	-	-	-	-	-	1.01 (3.30)
36	*Phoma eupyrena*	-	1.38 (7.32)	0.47 (3.30)	-	-	-	5.76 (22.04)	1.01 (2.13)	-	-	-	-
37	*P. medicaginis*	-	-	-	-	-	-	-	-	-	-	3.00 (15.00)	-

38	*Pythium intermedium*	-	0.92 (4.83)	1.43 (10.00)	3.94 (12.67)	-	-	5.76 (22.04)	-	7.18 (17.32)	6.00 (28.87)	-	-
39	*Scopulariopsis brumpti*	-	1.84 (9.73)	-	0.96 (3.28)	1.55 (6.53)	-	-	-	-	-	-	-
40	*Trichoderma polysporum*	-	1.38 (7.32)	-	-	-	-	-	-	-	-	-	-
41	*T. koningii*	-	0.45 (2.41)	0.94 (6.60)	-	-	-	-	-	-	-	-	-
42	*T. viride*	-	-	2.38 (16.60)	1.94 (6.24)	-	2.17 (6.33)	3.66 (14.03)	16.87 (35.98)	3.86 (9.32)	-	-	-
43	*Verticillium dahliae*	1.45 (5.88)	-	-	-	-	-	0.51 (1.98)	-	-	-	-	0.306 (10.00)
44	*V. alboatrum*	-	0.45 (2.41)		2.42 (7.98)	3.11 (13.07)	4.90 (14.28)	-	-	-	-	-	-
45	*V. catenulatum*	-	-	-	-	-	-	-	1.01 (2.13)	-	-	-	-
46	*V. chlamydosporium*	-	-	-	-	-	-	0.51 (1.98)	1.53 (3.22)	5.52 (13.32)	-	3.49 (17..51)	-
47	*White sterile mycelia*	-	1.84 (9.73)	-	-	-	-	-	-	-	-	-	-

Table 4 Monthly variation of fungal population (per gram dry soil x10^3) in the disturbed forest soil at depth 10-20cm (2002)
Values in parentheses are percentage relative abundance

Sl. No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Acremonium butyri*	-	-	-	-	1.47 (16.67)	0.52 (7.62)	-	0.48 (1.57)	-	0.49 (26.30)	4.39 (50.00)	-
2	*A. kiliense*	-	-	-	-	-	-	-	-	10.63 (51.47)	-	-	-
3	*A. murorum*	3.69 (16.63)	-	-	0.47 (1.94)	-	-	0.97 (7.92)	-	-	-	-	-
4	*Arthrobotrys conoides*	-	0.98 (9.54)	-	-	-	-	-	-	-	-	1.45 (16.67)	-
5	*Aspergillus fumigatus*	-	-	0.92 (8.61)	-	1.47 (16.67)	0.52 (7.62)	-	-	-	-	-	-
6	*A. flavus*	-	1.40 (13.64)	-	-	-	-	-	-	-	-	-	-
7	*A. niger*	-	-	-	-	-	-	0.48 (3.96)	-	-	-	-	-
8	*Botryotrichum piluliferum*	-	-	1.40 (13.05)	4.29 (17.64)	-	-	-	-	-	-	-	0.47 (2.67)
9	*Cladosporium cladosporioides*	-	1.40 (13.64)	-	-	-	-	-	-	-	-	-	-
10	*Eupenecillium sheari*	-	-	-	-	-	-	-	-	-	-	-	2.42 (13.54)
11	*Humicola grisea*	-	-	-	0.47 (1.94)	-	-	-	-	-	6.37 (34.20)	-	-
12	*Mammaria echinobotryoides*	0.91 (4.13)	-	-	-	-	0.52 (7.62)	-	-	-	-	-	-
13	*Mortierella polycephala*	-	-	-	-	-	-	-	-	-	-	-	2.90 (16.22)
14	*M. vinacea*	-	-	-	-	-	0.52 (7.62)	-	-	-	-	2.91 (33.33)	-
15	*Paecilomyces carneus*	-	0.98 (9.54)	1.40 (13.05)	-	-	-	-	-	-	-	-	-
16	*P. lilacinus*	-	-	-	-	-	-	5.41 (43.93)	-	2.52 (12.20)	-	-	-
17	*Penecillium chrysogenum*	1.38 (6.25)	-	-	-	-	-	-	-	-	-	-	-
18	*P. frequentans*	-	0.98 ((9.54)	-	4.29 (17.64)	-	0.52 (7.62)	0.48 (3.96)	-	-	-	-	-
19	*P. funiculosum*	-	-	-	1.43 (5.88)	-	-	-	-	-	-	-	1.45 (8.11)

20	*P. granulatums*	-	-	-	-	-	-	-	-	-	2.94 (15.79)	-	-
21	*P. janthinellum*	-	-	-	2.86 (11.76)	-	-	-	-	-	-	-	-
22	*P. jensenii*	-	-	-	-	1.95 (22.16)	-	-	-	-	-	-	5.32 (29.76)
23	*P. lividum*	-	-	0.92 (8.61)	-	-	-	-	-	-	-	-	-
24	*P. nigricans*	4.16 (18.75)	-	-	-	-	0.52 (7.62)	0.97 (7.92)	-	-	-	-	-
25	*P. oxalicum*	2.77 (12.5)	-	-	-	-	0.52 (7.62)	-	-	-	-	-	-
26	*P. purpurogenum*	-	0.46 (4.50)	1.86 (17.36)	1.43 (5.88)	-	1.11 (16.16)	-	2.44 (7.90)	-	-	-	-
27	*P. restrictum*	-	-	-	-	-	1.11 (16.16)	-	-	-	-	-	-
28	*P. rubrum*	-	-	2.80 (26.10)	2.37 (9.76)	1.95 (22.16)	-	1.47 (12.00)	15.69 (50.79)	-	-	-	-
29	*P. sacculum*	-	0.46 (4.50)	-	-	-	-	-	-	-	-	-	-
30	*P. thomnii*	-	-	-	3.88 (12.67)	-	-	-	-	-	-	-	-
31	*P. waksmanii*	8.79 (39.58)	-	-	-	-	-	-	-	-	-	-	-
32	*Phoma eupyrena*	-	-	-	-	-	-	-	12.00 (39.68)	-	-	-	-
33	*Pythium intermedium*	-	0.98 (9.54)	-	3.81 (15.64)	-	-	0.97 (7.92)	-	1.00 (4.80)	4.41 (23.69)	-	-
34	*Scopulariopsis fusca*	-	0.46 (4.50)	-	-	-	-	-	-	-	-	-	-
35	*Torula herbarum*	-	-	-	-	-	0.52 (7.62)	-	-	-	-	-	-
36	*Trichoderma koningii*	-	-	1.40 (13.05)	-	-	-	-	-	-	-	-	-
37	*T. viride*	-	1.40 (13.64)	-	2.86 (11.76)	0.97 (11.00)	0.52 (7.62)	1.47 (12.00)	-	5.06 (24.48)	-	-	-
38	*Verticillium alboatrum*	-	-	-	-	0.97 (11.00)	-	-	-	-	-	-	5.32 (29.76)
39	*V. chlamydosporium*	-	-	-	-	-	-	-	-	1.51 (7.35)	-	-	-
40	White sterile mycelia	-	0.98 (9.54)	-	-	-	-	-	-	-	-	-	-

Table 5 Monthly variation of fungal population (per gram dry soil x10^3) in the disturbed forest soil at depth 20-30cm (2002) Values in parentheses are percentage relative abundance

Sl. No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Acremonium butyri*	-	-	-	1.95 (14.77)	-	-	-	-	-	0.50 (9.02)	-	-
2	*A. murorum*	0.25 (12.76)	-	-	-	-	-	-	-	2.04 (18.14)	-	-	-
3	*A. strictum*	-	-	-	-	-	-	-	-	-	-	1.41 (25.00)	-
4	*Aspergillus alutaceus*	-	-	-	1.95 (14.77)	-	-	-	-	-	-	-	-
5	*A. fumigatus*	-	-	-	-	2.20 (19.00)	-	-	-	-	-	-	-
6	*Chaetosphaeria vermicularioides*	-	-	-	-	-	0.52 (12.40)	-	-	-	0.50 (9.02)	-	-
7	*Gliocladium roseum*	-	-	-	0.97 (7.33)	-	-	-	-	-	-	-	-
8	*Humicola fuscoatra*	-	0.50 (6.60)	-	-	-	-	-	-	-	-	-	-
9	*Mortierella parvispora*	-	1.01 (13.20)	-	-	-	0.52 (12.40)	-	-	-	-	-	-
10	*M. humilis*	-	-	-	1.95 (14.77)	-	-	-	-	-	-	-	-
11	*Paecilomyces carneus*	-	-	1.49 (12.5)	-	-	-	-	-	-	-	-	-
12	*P. lilacinus*	-	-	-	-	-	-	1.49 (30.03)	-	-	-	-	-
13	*Penecillium brevicompactum*	-	1.01 (13.20)	1.49 (12.5)	1.95 (14.77)	1.09 (9.42)	-	-	-	-	-	-	2.89 (24.01)
14	*P. canescens*	-	-	-	-	-	0.53 (12.40)	-	-	-	-	-	-
15	*P. chrysogenum*	-	-	1.49 (12.5)	2.94 (22.22)	-	-	-	-	-	-	-	2.89 (24.01)
16	*P. expansum*	-	-	-	-	-	-	-	-	-	-	2.35 (41.75)	-
17	*P. frequentans*	-	1.01 (13.20)	-	-	-	-	-	-	-	-	-	-

18	*P. funiculosum*	0.411 (20.46)	-	-	-	-	-	-	-	-	-	-	3.37 (27.97)
19	*P. granulatums*	-	-	-	-	-	-	-	-	-	1.00 (18.30)	-	-
20	*P. jensenii*	0.360 (17.92)	-	-	-	1.65 (14.28)	0.52 (12.40)	-	-	-	-	-	-
21	*P. lanosum*	-	-	-	-	-	-	-	-	-	-	0.93 (16.50)	-
22	*P. lividum*	-	-	1.99 (16.62)	-	-	-	-	-	-	-	-	-
23	*P. nigricans*	-	-	-	-	-	0.52 (12.40)	0.98 (19.81)	4.07 (20.51)	-	-	-	-
24	*P. oxalicum*	0.256 (12.76)	-	-	-	-	-	-	-	-	-	-	-
25	*P. purpurogenum*	-	-	1.49 (12.5)	-	-	1.60 (37.59)	-	-	-	0.50 (9.01)	-	-
26	*P. restrictum*	-	1.01 (13.20)	-	-	-	-	-	-	-	-	-	-
27	*P. rubrum*	-	-	2.54 (21.25)	-	3.86 (33.28)	-	-	15.79 (79.46)	-	-	-	-
28	*P. simplicissimum*	-	-	1.04 (8.75)	-	-	-	-	-	-	-	-	-
29	*P. waksmanii*	3.09 (15.38)	-	-	-	-	-	-	-	-	-	-	-
30	*Phoma eupyrena*	-	0.50 (6.60)	-	-	-	-	-	-	-	-	0.47 (8.25)	-
31	*Pythium intermedium*	-	0.50 (6.60)	-	-	-	-	1.99 (39.93)	-	3.07 (27.28)	1.00 (18.30)	-	-
32	*Scopulariopsis brevicaulis*	-	-	-	-	-	-	-	-	-	050 (9.02)	-	-
33	*Trichoderma viride*	-	1.01 (13.20)	-	1.47 (11.11)	1.09 (9.42)	-	0.49 (9.90)	-	4.61 (40.92)	1.50 (27.32)	-	-
34	*Verticillium dahliae*	0.41 (20.46)	-	-	-	-	-	-	-	-	-	-	-
35	*V. alboatrum*	-	0.50 (6.60)	-	-	1.65 (14.28)	-	-	-	-	-	0.46 (8.25)	2.89 (24.01)
36	*V. chlamydosporium*	-	-	-	-	-	-	-	-	1.53 (13.64)	-	-	-
37	White sterile mycelia	-	0.50 (6.60)	-	-	-	-	-	-	-	-	-	-

Table 6 Monthly variation of fungal population (per gram dry soil x10^3) in the disturbed forest soil at depth 0-10cm (2003)
Values in parenthesis are percentage relative abundance

Sl No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Absidia cylindrospora*	-	-	-	-	-	0.59 (1.57)	-	-	-	-	-	-
2	*Acremonium butyri*	-	1.37 (4.41)	-	-	1.02 (4.83)	-	-	-	-	-	-	-
3	*A. cerealis*	-	-	5.21 (17.91)	-	-	-	-	-	-	-	-	-
4	*Acremonium murorum*	-	-	-	-	-	-	-	-	-	-	0.99 (2.75)	-
5	*Aspergillus alutaceus*	-	-	-	-	-	-	-	-	5.41 (6.57)	-	0.99 (2.75)	-
6	*A. candidus*	-	-	-	-	-	-	2.52 (3.55)	-	5.41 (6.57)	-	-	-
7	*A. flavus*	-	-	-	-	-	-	2.00 (2.82)	-	-	3.39 (13.63)	-	-
8	*A. fumigatus*	-	-	4.34 (14.91)	-	-	-	-	-	-	2.83 (11.38)	-	-
9	*A. niger*	-	-	-	-	-	-	1.51 (2.13)	-	-	-	-	-
10	*A. terreus*	-	-	-	-	-	-	0.49 (0.70)	-	-	-	-	-
11	*Botryotrichum piluliferum*	-	-	0.43 (1.48)	-	-	-	-	-	10.81 (13.14)	-	-	-
12	*Brown sterile mycelia*	-	-	-	-	1.02 (4.83)	-	-	-	-	-	-	-
13	*Chaetomium crispatum*	-	0.45 (1.46)	-	-	-	-	-	-	-	-	-	-
14	*Cladosporium cladosporioides*	-	-	-	-	-	1.19 (3.14)	-	41.38 (52.77)	0.53 (0.65)	-	-	3.67 (7.85)
15	*Eupenicillium brefeldianum*	-	0.44 (1.46)	-	-	-	-	-	8.17 (10.42)	-	-	-	0.45 (0.97)
16	*E. javanicum*	-	-	-	-	-	0.59 (1.57)	-	-	-	-	-	0.45 (0.97)
17	*E. sheari*	-	-	-	-	-	-	-	-	-	-	-	4.12 (8.82)
18	*Fusarium redolens*	-	-	-	-	-	-	-	-	-	-	3.00 (8.33)	-

19	*Gliocladium roseum*	-	-	-	-	0.51 (2.41)	-	-	0.54 (0.69)	-	-	-	-
20	*Mammaria echinobotryoides*	-	-	-	9.30 (22.67)	-	-	-	-	-	-	-	-
21	*Mortierella hyalina*	-	-	-	-	-	-	-	-	4.33 (5.27)	-	-	3.20 (6.85)
22	*M. parvispora*	-	1.37 (4.41)	-	-	-	-	-	-	-	-	1.50 (4.17)	-
23	*M. rammaniana*	-	-	-	-	-	-	-	-	1.07 (1.36)	-	1.50 (4.17)	-
24	*M. vinacea*	-	0.91 (2.91)	-	-	-	-	0.49 (0.70)	-	-	-	-	-
25	*M. humilis*	-	-	-	-	-	-	-	-	-	-	4.50 (12.50)	-
26	*Paecilomyces carneus*	-	-	-	-	-	2.39 (6.33)	-	-	-	2.25 (9.07)	-	-
27	*P. marquandii*	-	-	-	-	0.51 (2.41)	-	-	-	-	-	-	-
28	*P. variotii*	-	-	-	9.72 (23.69)	-	-	-	-	-	-	-	-
29	*Penicillium brevicompactum*	-	2.29 (7.37)	-	-	2.58 (12.22)	12.59 (33.33)	-	-	1.07 (1.30)	-	0.99 (2.75)	12.82 (27.44)
30	*P. canescens*	-	-	-	-	-	-	5.53 (7.80)	-	-	-	-	-
31	*P. chrysogenum*	4.27 (26.70)	-	-	-	-	7.79 (20.62)	11.04 (5.60)	-	-	-	-	-
32	*P. daleae*	-	-	-	-	-	-	-	0.54 (0.69)	-	-	-	-
33	*P. expansum*	-	-	-	-	-	-	0.99 (1.40)	-	-	-	-	-
34	*P. frequentans*	4.12 (13.23)	-	-	-	-	-	-	-	-	-	-	-
35	*P. funiculosum*	-	-	-	-	-	-	-	2.71 (3.46)	-	-	-	-

36	*P. janthinelum*	-	-	-	-	-	4.78 (12.69)	8.03 (11.34)	-	-	-	-	-
37	*P. jensenii*	-	2.29 (7.37)	-	-	0.29 (29.26)	-	3.51 (4. 95)	3.81 (4.85)	2.71 (3.29)	-	-	9.62 (20.59)
38	*P. lanosum*	-	-	-	-	0.51 (2.41)	-	-	9.26 (11.81)	10.81 (13.14)	-	-	-
39	*P. lividum*	-	-	-	-	-	-	-	-	4.35 (5.27)	2.25 (9.07)	-	-
40	*P. nigricans*	-	4.12 (13.23)	-	-	-	-	1.51 (2.13)	-	8.12 (9.86)	-	-	-
41	*P. restrictum*	-	0.45 (1.46)	-	0.42 (1.02)	-	-	-	-	-	3.39 (15.88)	-	-
42	*P. rubrum*	-	-	2.60 (8.96)	-	0.51 (2.41)	8.39 (22.24)	7.03 (9.93)	-	5.41 (6.57)	3.39 (13.63)	9.50 (26.38)	0.45 (0.97)
43	*P. rugulosum*	-	-	-	-	-	0.59 (1.57)	-	-	-	-	-	0.91 (1.94)
44	*P. sacculum*	-	-	-	-	-	-	-	1.08 (1.38)	2.71 (3.29)	-	0.99 (2.75	4.12 (8.82)
45	*P. simplicissimum*	-	-	-	-	2.06 (9.73)	-	-	2.17 (2.77)	-	-	-	-
46	*P. stoloniferum*	-	-	-	-	-	-	-	1.08 (1.38)	-	0.56 (2.25)	-	-
47	*P. thomnii*	-	-	-	1.27 (3.09)	-	-	-	-	-	-	-	-
48	*Penicillium* sp.	1.55 (10.00)	-	-	-	-	-	-	-	-	-	-	3.20 (6.85)
49	*Phoma eupyrena*	-	-	4.34 (14.91)	-	-	-	-	-	-	-	-	-
50	*P. medicaginis*	1.04 (6.70)	3.20 (10.28)	-	-	-	-	-	-	-		-	-
51	*Plectosphaerella cucumerina*	-	-	-	-	-	-	-	-	-	0.56 (2.25)	-	-
52	*Pythium intermedium*	4.13 (26.70)	0.45 (1.46)	-	-	-	-	2.52 (3.55)	7.64 (9.71)	-	1.12 (4.49)	-	-
53	*Rhizopus oryzae*	-	-	-		-	-	-	-	0.54 (0.65)	-	-	-

54	*Scopulariopsis brevicaulis*	-	-	-	-	-	-	9.04 (12.76)	-	-	-	-	-
55	*S. brumptii*	-	-	-	16.08 (39.19)	-	-	-	-	-	-	5.00 (13.87)	-
56	*Torula herbarum*	-	-	-	-	-	-	-	-	-	-	5.00 (13.87)	-
57	*Trichoderma viride*	-	2.29 (7.37)	-	-	-	-	8.03 (11.34)	-	5.41 (6.57)	2.83 (11.38)	-	3.67 (7.85)
58	*T. harzianum*	-	-	-	-	-	-	6.02 (8.51)	-	-	-	-	-
59	*T. polysporum*	-	-	-	-	3.09 (14.63)	-	-	-	-	-	-	-
60	*T. koningii*	-	5.49 (17.64)	-	-	-	-	-	-	-	-	-	-
61	*Verticillium chlamydosporium*	-	-	-	-	-	-	-	-	-	1.12 (4.49)	-	
62	*V. alboatrum*	5.69 (36.70)	0.45 (1.46)	-	0.84 (2.04)	0.51 (2.41)	-	-	-	6.49 (7.89)	-	-	-
63	White sterile mycelia	-	-	-	-	2.06 (9.73)	-	-	-	7.56 (9.91)	0.56 (2.25)	0.99 (2.75)	-

Table 7 Monthly variation of fungal population (per gram dry soil x10^3) in the disturbed forest soil at depth 10-20 cm (2003)
Values in parenthesis are percentage relative abundance

Sl. No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Acremonium murorum*	-	-	-	1.69 (7.98)	-	-	-	-	-	-	-	-
2	*Aspergillus candidus*	-	-	-	-	-	-	1.40 (2.70)	-	-	-	-	-
3	*A. fumigatus*	-	-	-	-	2.05 (15.97)	-	-	-	-	1.01 (5.50)	10.40 (26.24)	-
4	*A. niger*	-	-	-	-	-	-	0.19 (3.59)	-	-	-	0.49 (1.24)	-
5	*A. terreus*	-	-	-	-	-	-	0.92 (1.78)	-	-	-	-	-
6	*Botryotrichum piluliferum*	-	-	-	0.84 (3.96)	-	-	-	-	1.05 (6.00)	-	-	-
7	*Brown sterile mycelia*	-	-	-	0.42 (1.98)	1.02 (7.92)	-	-	-	-	-	-	-
8	*Cladosporium cladosporioides*	-	-	-	-	-	-	-	9.72 (27.94)	0.48 (3.00)	-	-	7.03 (34.04)
9	*Eupenicillium brefeldianum*	-	-	-	-	-	-	-	-	-	0.50 (2.75)	-	-
10	*Humicola fuscoatra*	-	-	0.89 (7.18)	-	-	-	-	-	-	0.50 (2.75)	-	-
11	*H. grisea*	-	-	-	0.84 (3.96)	-	-	-	-	-	-	-	-
12	*Mortierella hyalina*	-	-	-	-	-	-	-	-	-	-	-	0.11 (10.66)
13	*M. parvispora*	-	-	-	-	-	-	-	-	-	-	0.98 (2.47)	-
14	*M. rammaniana*	-	-	-	-	-	-	-	-	-	-	3.97 (10.01)	-
15	*M. vinacea*	-	0.44 (3.54)	-	-	-	-	-	-	-	-	-	-
16	*M. humilis*	-	-	-	-	-	-	-	-	-	0.50 (2.75)	-	-
17	*Mucor racemosus*	-	-	-	-	-	-	0.46 (0.89)	-	-	-	-	-

18	*Paecilomyces carneus*	-	-	1.65 (10.79)	-	-	2.69 (7.37)	-	-	-	-	-	-
19	*Penicillium. brevicompactum*	-	0.44 (3.54)	-	-	-	3.23 (8.82)	-	2.04 (5.87)	1.59 (9.09)	1.01 (5.50)	3.97 (10.01)	0.87 (4.21)
20	*P. chrysogenum*	1.56 (16.67)	0.44 (3.54)	-	-	-	6.99 (19.10)	32.68 (63.05)	-	-	-	-	-
21	*P. expansum*	-	-	-	-	-	-	-	6.65 (19.10)	-	3.55 (19.42)	-	-
22	*P. frequentans*	-	-	-	2.10 (9.96)	-	-	-	-	-	-	9.91 (25.0)	-
23	*P. funiculosum*	-	-	-	0.84 (3.96)	-	-	-	-	-	-	-	-
24	*P. granulatums*	2.08 (22.17)	-	-	-	-	-	-	-	-	-	-	-
25	*P. janthinellum*	-	-	-	2.10 (9.96)	-	11.84 (32.33)	9.34 (18.03)	-	-	-	1.98 (4.99)	-
26	*P. jensenii*	-	-	-	-	3.08 (24.0)	-	-	0.51 (1.46)	-	-	1.49 (1.48)	4.390 (21.25)
27	*P. lanosum*	-	-		-	-	-	4.20 (8.11)	1.54 (4.41)	0.48 (3.00)	-	-	-
28	*P. lividum*	-	-	1.65 (10.79)	-	-	-	-	-	1.05 (6.00)	-	-	-
29	*P. nigricans*	-	-	-	-	-	-	-	-	1.05 (6.00)	-	-	-
30	*P. purpurogenum*	-	0.44 (3.54)	4.55 (29.76)	-	-	-	-	-	-	-	-	-
31	*P. restrictum*	-	0.44 (3.54)	-	-	-	-	-	-	-	-	-	-
32	*P. rubrum*	1.05 (11.17)	0.44 (3.54)	0.82 (5.35)	-	3.59 (27.97)	3.76 (10.28)	-	0.51 (1.46)	3.17 (18.18)	5.59 (30.58)	-	-
33	*P. rugulosum*	-	-	-	-	-	-	-	8.19 (23.51)	-	-	-	-
34	*P. sacculum*	-	-	-	-	-	-	-	-	2.11 (12.09)	0.50 (2.75)	3.46 (8.74)	1.32 (6.38)
35	*P. simplicissimum*	-	-	2.07 (13.54)	-	-	-	-	4.10 (11.78)	-	-	0.98 (2.47)	-
36	*Penicillium* sp.	0.52 (5.50)	-	-	-	-	-	-	-	-	-	-	0.44 (2.11)
37	*Pythium intermedium*	-	1.77 (14.26)	-	2.10 (9.96)	-	-	-	1.54 (4.41)	-	-	-	-

38	*Scopulariopsis brevicaulis*	-	0.89 (7.18)	-	-	-	-	-	-	-	-	-	-
39	*S. brumptii*	-	1.77 (14.26)	-	-	-	-	-	-	-	-	-	-
40	*Torula herbarum*	-	-	-	-	-	-	-	-	-	-	1.98 (4.99)	-
41	*Trichoderma koningii*	-	0.44 (3.54)	3.72 (24.33)	-	-	5.38 (14.69)	-	-	-	2.55 (13.92)	-	-
42	*T. polysporum*	-	2.67 (21.43)	-	-	-	2.69 (7.37)	-	-	-	-	-	-
43	*T. viride*	-	0.44 (3.54)	0.41 (2.68)	-	3.08 (24.0)	-	0.92 (1.78)	-	0.48 (3.00)	1.52 (8.33)	-	1.75 (8.49)
44	*Verticillium alboatrum*	4.17 (44.50)	0.89 (7.18)	-	-	-	-	-	-	2.65 (15.18)	-	-	-
45	White sterile mycelia	-	-	-	10.14 (47.99)	-	-	-	-	.3.17 (18.18)	1.01 (5.50)	-	2.64 (12.76)

Table 8 Monthly variation of fungal population (per gram dry soil x10^3) in the disturbed forest soil at depth 20-30cm (2003) Values in parenthesis are percentage relative abundance

Sl. No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Acremonium butyri*	-	-	-	0.93 (15.24)	-	-	-	-	-	-	-	-
2	*A. cylindrospora*	-	-	-	-	-	1.61 (3.79)	-	-	-	-	-	-
3	*Aspergillus alutaceus*	-	-	-	0.47 (7.62)	-	-	6.27 (11.49)	-	0.53 (12.36)	-	0.49 (5.21)	
4	*A. candidus*	-	-	-	-	-	-	3.38 (6.19)	-	-	-	-	-
5	*A. flavus*	-	-	-	-	-	0.53 (12.53)	-	-	-	-	-	-
6	*A. fumigatus*	-	-	-	-	-	9.68 (22.79)	-	-	-	-	0.49 (5.21)	-
7	*A. niger*	-	-	-	-	-	-	1.93 (3.53)	-	-	-	-	-
8	*A. terreus*	-	-	-	-	-	-	0.48 (0.88)	-	-	-	-	-
9	*Botryotrichum piluliferum*	-	-	-	0.93 (15.24)	-	-	-	-	-	-	-	-
10	*Brown sterile mycelia*	-	-	-	-	-	-	0.48 (0.88)	-	-	-	-	-
11	*Chaetomium crispatum*	-	-	-	-	0.56 (11.00)	-	-	-	-	-	-	-
12	*Cladosporium cladosporidioides*	-	-	-	-	-	-	-	5.21 (35.69)	-	-	-	0.87 (3.30)
13	*Gliocladium roseum*	-	-	-	0.47 (7.62)	-	-	-	-	-	-	-	-
14	*Humicola fuscoatra*	-	-	-	-	-	-	-	-	-	-	0.49 (5.21)	-
15	*Mortierella rammaniana*	-	-	-	-	-	-	-	-	-	-	0.98 (10.43)	2.19 (8.35)
16	*M. humilis*	-	-	-	0.47 (7.61)	-	-	-	-	-	-	0.49 (5.21)	-
17	*Paecilomyces carneuss*	-	0.47 (8.99)	-	-	-	-	-	-	-	-	-	-
18	*P. lilacinus*	-	0.95 (17..98)	-	-	-	-	-	-	-	4.02 (50.09)	-	-
19	*Penicillium brevicompactum*	-	-	0.41 (8.99)	0.93 (15.24)	-	1.06 (2.51)	-	-	1.59 (37.45)	-	-	3.94 (15.00)

20	*P. chyrsogenum*	0.55 (33.30)	-	0.41 (8.99)	0.93 (15.24)		0.53 (1.25)	18.36 (33.63)	-	-	-	-	-
21	*P. expansum*	-	-	-	-	-	2.15 (5.05)		0.52 (3.53)	-	-	-	-
22	*P. frequentans*	-	0.47 (8.99)	-	-	-	-	-	-	-	-	-	-
23	*P. funiculosum*	-	-	-	-	-	-	-	-	-	-	-	3.94 (15.00)
24	*P. granulatums*	0.55 (33.30)	-	-	-	-	-	-	-	-	-	-	-
25	*P. janthinellum*	-	-	-	-	-	17.78 (40.52)	18.36 (33.63)	-	-	-	-	5.68 (21.65)
26	*P. jensenii*	-	-	-	-	1.69 (33.33)	-	-	-	-	-	1.97 (21.01)	-
27	*P. lanosum*	-	-	-	-	-	4.83 (11.39)	1.45 (2.65)	-	-	-	-	4.82 (18.35)
28	*P. lividum*	-	-	0.41 (8.99)	-	-	-	-	-	-	-	-	-
29	*P. purpurogenum*	-	0.95 (17..98)	0.41 (8.99)	-	-	-	-	-	-	-	-	-
30	*P. rubrum*	-	-	0.41 (8.99)	-	-	1.06 (2.51)	0.96 (1.75)	-	-	0.49 (6.19)	-	0.43 (1.65)
31	*P. sacculum*	-	-	-	-	-	-	-	1.03 (7.07)	1.054 (24.72)	-	0.98 (10.43)	-
32	*P. simplicissimum*	-	-	0.41 (8.99)	-	-	-	-	2.61 (17.89)	-	-	-	-
33	*P. waksmanni*	-	-	-	-	-	-	-	-	-	-	0.49 (5.21)	-
34	*Penicillium* sp.	-	-	-	-	-	-	-	-	-	-	0.49 (5.21)	-
35	*Pythium intermedium*	-	0.47 (8.99)	-	-	-	-	0.48 (0.87)	5.21 (35.69)	-	-	0.49 (5.21)	-
36	*Scopulariopsis brevicaulis*	-	1.43 (27.25)	-	-	-	-	-	-	-	-	-	
37	*Torula herbarum*	-	-	-	-	-	-	-	-	-	-	0.98 (10.43)	-
38	*Trichoderma. harzianum*	-	-	-	-	-	-	-	-	-	2.00 (24.95)	-	-
39	*T. polysporum*	-	-	-	-	0.55 (11.00)	3.22 (7.59)	-	-	-	-	-	-
40	*T. viride*	-	0.47 (8.99)	0.41 (8.99)	0.93 (15.24)	1.11 (22.00)	-	1.45 (2.65)	-	-	1.51 (18.76)	0.49 (5.21)	-
41	*Verticillium alboatrum*	0.55 (33.30)	-	0.41 (8.99)	-	0.55 (11.00)	0.53 (1.25)	-	-	-	-	-	-
42	White sterile mycelia	-	-	-	-	0.55 (11.00)	-	1.93 (3.53)	0.52 (3.54)	1.05 (24.72)	-	0.49 (5.21)	4.37 (16.65)

Table 9 Monthly variation of fungal population (per gram dry soil x 10^3) in the undisturbed forest soil at depth 0-10cm (2002)
Values in parentheses are percentage relative abundance

Sl. No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Acremonium butyri*	-	-	-	-	-	-	0.45 (1.28)	-	-	-	-	-
2	*A. fusidioides*	-	-	-	-	-	-	-	-	-	-	-	2.64 (9.50)
3	*A. kiliense*	-	-	-	-	-	-	-	-	4.41 (6.30)	-	-	-
4	*A. murorum*	2.18 (8.74)	-	-	-	-	-	2.74 (7.24)	-	9.80 (13.99)	0.83 (3.14)	-	-
5	*A. strictum*	-	-	-	-	-	-	-	-	-	0.83 (3.14)	-	-
6	*Aspergillus candidus*	-	-	-	4.45 (11.53)	-	-	-	-	-	-	-	-
7	*A. clavatus*	-	-	-	1.48 (3.84)	-	-	-	-	-	-	-	-
8	*A. fumigatus*	-	-	-	2.96 (7.69)	-	-	-	-	-	-	-	-
9	*A. niger*	-	-	-	-	-	3.73 (13.00)	-	-	-	-	-	-
10	*Cladosporium cladosporioides*	-	-	-	2.46 (6.38)	-	-	-	-	-	-	-	-
11	*Eupeniciullium brefeldianum*	-	-	-	-	-	-	-	-	-	0.41 (1.60)	-	-
12	*Helminthosporium velutinum*	-	-	-	-	-	0.94 (3.3)	-	-	-	-	-	-
13	*Heteroconium chaetospira*	-	-	-	-	-	-	-	-	-	0.41 (1.60)	-	-
14	*Humicola grisea*	-	-	-	-	-	-	0.45 (1.28)	-	-	-	-	-
15	*Mammaria echinobotryoides*	-	-	-	-	-	3.34 (11.65)	-	-	-	2.09 (7.90)	-	-
16	*Mortierella parvispora*	-	0.41 (3.81)	-	3.94 (10.23)	-	-	-	-	-	-	-	-
17	*M. polycephala*	-	-	-	-	-	-	0.45 (1.28)	-	-	-	-	-

18	*M. humilis*	-	-	-	2.96 (7.69)	-	-	-	-	-	-	-	-
19	*Paecilomyces carneus*	-	1.24 (11.55)	-	-	-	-	1.37 (3.62)	-	-	0.66 (3.17)	-	-
20	*P. lilacinus*	-	-	-	-	-	-	-	-	13.74 (19.60)	-	-	-
21	*Penecillium brevicompactum*	8.31 (33.32)	1.99 (18.41)	2.47 (15.80)	7.90 (20.50)	6.01 (18.02)	9.55 (33.35)	10.52 (27.89)	-	1.47 (2.10)	-	-	-
22	*P. canescens*	-	-	-	-	-	-	-	-	-	-	-	3.96 (14.28)
23	*P. chrysogenum*	0.43 ((1.74)	-	-	-	-	-	-	11.7 (19.86)	0.97 (1.38)	-	9.23 (35.69)	4.40 (15.85)
24	*P. daleae*	-	-	-	-	-	0.47 (1.65)	-	-	-	-	-	-
25	*P. expansum*	-	-	-	-	-	-	-	8.67 (14.69)	-	-	-	4.40 (15.85)
26	*P. frequentans*	-	1.62 (15.01)	0.81 ((5.24)	-	2.73 (8.28)	3.34 (11.65)	-	3.46 (5.87)	2.94 (4.20)	-	-	-
27	*P. funiculosum*	-	-	-	-	-	0.47 (1.65)	-	-	-	-	-	-
28	*P. jensenii*	6.57 (26.32)	-	1.64 (10.55)	-	3.28 (9.85)	1.90 (6.65)	-	5.63 (9.55)	-	-	-	4.40 (15.85)
29	*P. lanosum*	-	1.62 (15.01)	-	-	3.28 (9.94)	-	-	-	-	-	2.77 (10.71)	2.64 (9.50)
30	*P. nigricans*	-	0.82 (8.08)	3.28 (21.11)	-	-	2.38 (8.30)	10.98 (29.89)	-	2.44 (3.48)	-	4.61 (17.84)	-
31	*P. oxalicum*	2.63 (10.53)	-	-	-	-	-	-	-	-	-	-	-
32	*P. purpurogenum*	0.43 (1.74)	-	-	-	-	-	-	-	0.97 (1.39)	-	-	-
33	*P. restrictum*	-	-	1.23 (7.90)	-	-	-	-	7.81 (13.24)	16.20 (23.10)	0.83 (3.14)	-	-
34	*P. rubrum*	-	-	1.64 (10.55)	6.82 (18.07)	3.83 (11.47)	3.34 (11.65)	-	10.84 (18.38)	3.91 (5.58)	12.19 (46.0)	5.07 (19.61)	-
35	*P. sacculum*	-	-	-	-	-	-	-	4.76 (8.07)	-	-	-	-
36	*P. simplicissimum*	-	-	-	-	2.18 (6.55)	-	-	-	-	-	-	1.32 (4.76)
37	*P. stoloniferum*	-	-	-	-	2.18 (6.55)	-	-	-	-	-	-	-

38	*P. thomnii*	-	-	-	-	2.18 (6.55)	-	-	-	-	-	-	-
39	*P. waksmanii*	1.747 (7.00)	-	-	-	3.83 (1.47)	-	-	-	-	1.26 (4.80)	-	-
40	*Penicillium.* sp.	-	0.82 (8.08)	-	-	-	0.94 (3.30)	-	-	-	-	-	-
41	*Phoma eupyrena*	-	-	-	2.46 (6.38)	-	-	-	-	-	-	-	-
42	*P. medicaginis*	-	-	-	-	-	-	-	-	-	0.83 (3.14)	-	-
43	*Pythium intermedium*	-	-	-	-	-	-	6.40 (16.88)	2.16 (3.662)	0.97 (1.39)	0.83 (3.14)	-	-
44	*P. irregulare*	-	0.07 (8.08)	-	-	-	-	-	-	-	-	-	-
45	*Scopulariopsis brevicaulis*	-	-	-	-	-	-	-	-	-	-	3.23 (12.48)	-
46	*S. fusca*	-	-	1.23 (7.90)	-	-	-	-	-	-	-	-	-
47	*Trichoderma harzianum*	-	-	-	-	-	-	-	2.16 (3.67)	-	-	-	-
48	*T. viride*	2.63 (10.53)	-	1.64 (10.55)	-	-	-	2.74 (7.24)	-	3.43 (4.89)	-	-	-
49	*Verticillium dahliae*	-	-	-	-	0.54 (1.63)	-	-	0.85 (1.46)	4.90 (6.99)	-	-	3.96 (14.28)
50	*V. alboatrum*	-	0.82 (8.08)	1.23 (7.90)	-	1.08 (3.25)	-	-	-	-	1.26 (4.84)	-	-
51	*V. chlamydosporium*	-	-	-	-	2.18 (6.55)	-	0.90 (2.42)	-	-	3.78 (14.39)	-	-
52	White sterile mycelia	-	0.41 (3.81)	2.05 (13.17)	2.96 (7.69)	-	1.43 (5.00)	0.45 (1.28)	0.85 (1.46)	3.91 (5.58)	-	0.91 (3.53)	-

Table 10 Monthly variation of fungal population (per gram dry soil x10^3) in the undisturbed forest soil at depth 10-20cm (2002) Values in parentheses are percentage relative abundance

Sl. No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Absidia cylindrospora*	-	-	-	1.47 (5.68)	-	-	-	-	-	-	-	-
2	*Acremonium butyri*	-	-	0.80 (5.68)	-	-	-	0.85 (5.83)	-	-	-	-	-
3	*A. fusidioides*	-	-	-	-	-	-	-	-	-	-	-	2.51 (10.00)
4	*A. murorum*	-	-	-	-	-	-	2.15 (13.83)	-	2.25 (4.33)	-	2.50 (11.76)	-
5	*Aspergillus alutaceus*	-	-	-	4.42 (17.0)	-	-	-	-	-	-	-	2.93 (11.65)
6	*A. carneus*	-	3.22 (10.8)	-	-	-	-	-	-	-	-	-	-
7	*A. candidus*	-	-	-	-	2.58 (10.64)	-	-	-	-	-	-	-
8	*A. clavatus*	-	-	-	-	-	0.86 (14.25)	-	-	-	-	-	-
9	*A. flavus*	-	-	-	-	1.56 (6.4)	-	-	-	-	-	-	-
10	*Cladosporium cladosporioides*	-	-	-	-	-	-	-	-	-	0.81 (3.66)	-	-
11	*Eupenecillium javanicum*	-	-	-	-	-	-	-	-	-	-	-	1.26 (5.00)
12	*Gliocladium roseum*	-	3.63 (12.0)	0.40 (2.84)	-	-	-	-	-	-	-	-	-
13	*Humicola fuscoatra*	-	-	1.62 (11.46)	-	1.02 (4.23)	-	-	-	-	-	-	-
14	*Mammaria echinobotryoides*	1.22 (5.77)	-	-	-	-	0.43 (7.17)	-	-	-	-	-	-
15	*Moetierella rammaniana*	-	-	2.03 (14.31)	-	-	-	-	-	-	-	-	-
16	*M. vinacea*	-	2.82 (9.30)	-	-	-	-	-	-	-	-	-	-
17	*Paecilomyces carneus*	-	-	-	-	-	-	-	-	-	-	4.58 (21.50)	-

18	*P. variotii*	-	-	-	-	-	-	-	2.41 (16.66)	-	-	-	-
19	*P. lilacinus*	-	3.22 (10.8)	-	-	-	-	-	-	12.22 (23.49)	-	-	-
20	*Penicillium brevicompactum*	3.25 (15.34)	-	1.22 (8.62)	5.39 (20.7)	-	2.63 (42.91)	9.53 (61.06)	-	-	-	-	-
21	*P. canescens*	-	-	-	-	-	-	-	-	-	-	-	4.19 (16.65)
22	*P. chrysogenum*	3.25 (15.34)	3.22 (10.4)	2.94 (11.30)	2.94 (11.30)	-	0.43 (7.08)	0.45 (2.75)	-	-	-	-	3.77 (15.00)
23	*P. expansum*	-	3.22 (10.8)	-	-	-	-	-	1.60 (11.08)	-	-	-	2.51 (10.00)
24	*P. frequentans*	-	-	-	-	-	0.43 (7.17)	0.42 (2.75)	0.79 (5.50)	-	-	-	-
25	*P. funiculosum*	1.62 (7.67)	-	-	-	-	-	-	-	-	-	-	-
26	*P. janthinellum*	-	-	-	-	-	-	-	-	-	-	-	3.77 (15.00)
27	*P. jensenii*	5.71 (26.88)	-	1.22 (8.62)	-	4.68 (19.23)	-	-	2.81 (19.42)	-	-	-	2.93 (11.65)
28	*P. lividum*	-	-	-	-	-	-	-	-	-	-	-	1.26 (5.00)
29	*P. nigricans*	-	2.82 (9.20)	3.25 (22.93)	-	-	0.86 (14.25)	-	-	2.71 (5.22)	-	-	-
30	*P. oxalicum*	1.22 (5.77)	-	-	-	-	-	-	-	-	-	-	-
31	*P. purpurogenum*	-	-	-	-	-	-	-	-	3.16 (6.08)	-	-	-
32	*P. restrictum*	-	-	-	-	-	-	-	-	6.79 (13.05)	-	-	-
33	*P. rubrum*	-	-	2.03 (14.31)	3.43 (13.23)	-	0.86 (14.25)	-	4.83 (33.33)	4.07 (7.83)	10.65 (48.24)	-	-
34	*P. rugulosum*	-	-	-	-	3.12 (12.82)	-	-	-	-	-	-	-
35	*P. sacculum*	-		-	-	-	-	-	0.39 (2.75)	-	-	-	-
36	*P. simplicissimum*	-	-	-	-	2.07 (8.50)	-	-	-	-	-	-	-
37	*P. waksmanii*	0.80 (3.80)	-	-	-	-	-	-	-	-	-	-	-

38	*Phoma eupyrena*	-	-	0.80 (5.68)	3.92 (15.11)	-	-	-	-	-	-	-	-
39	*Pythium intermedium*	-	-	-	2.44 (9.43)	-	-	1.73 (11.03)	0.79 (14.16)	1.35 (2.61)	2.46 (11.11)	-	-
40	*P. irregulare*	-	-	-	-	-	-	-	-	-	1.64 (7.38)	-	-
41	*Scopulariopsis brevicaulis*	-	-	1.62 (11.46)	-	-	-	-	-	-	-	-	-
42	*Trichiderma harzianum*	-	-	-	-	-	-	-	0.39 (2.75)	-	-	-	-
43	*T. koningii*	-	-	-	-	3.63 (14.93)	-	-	-	-	-	2.91 (13.70)	-
44	*T. piluiliferum*	-	-	-	-	-	-	-	-	3.61 (6.95)	-	-	-
45	*T. pseudokoningii*	-	-	-	-	-	-	-	1.20 (8.33)	-	-	-	-
46	*T. viride*	3.67 (17.31)	-	-	-	1.56 (6.4)	-	-	-	15.84 (30.44)	-	-	-
47	*Verticillium dahliae*	3.67 (10.53)	2.01 (6.70)	-	-	2.07 (8.50)	-	0.42 (2.75)	-	-	2.86 (12.94)	-	-
48	*V. alboatrum*	0.40 (1.90)	2.82 (9.20)	1.96 (7.38)	1.96 (7.38)	1.56 (6.40)	-	-	-	-	2.86 (12.94)	11.27 (52.94)	-
49	*V. chlamydosporium*	-	-	-	-	-	-	-	-	-	0.40 (1.83)	-	-
50	White sterile mycelia	-	3.22 (10.8)	0.80 (5.68)	-	0.51 (2.11)	-	-	-	-	0.40 (1.83)	-	-

Table 11 Monthly variation of fungal population (per gram dry soil x10^3) in the undisturbed forest soil at depth 20-30cm (2002) Values in parentheses are percentage relative abundance

Sl. No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Acremonium butyri*	-	-	-	2.57 (22.65)	-	-	-	-	12.79 (25.69)	-	-	-
2	*A. kiliense*	-	-	-	-	-	-	-	-	11.03 (21.15)	-	-	-
3	*A. murorum*	2.79 (14.87)	-	-	-	-	-	0.83 (18.03)	-	5.29 (10.63)	-	-	-
4	*A. sclerotiorum*	-	-	-	-	-	-	-	-	-	0.41 (3.30)	-	-
5	*Aspergillus fumigatus*	-	-	-	-	-	2.95 (43.73)	-	-	-	-	-	-
6	*Aspergillus* sp.	-	-	-	2.06 (18.15)	-	-	-	-	-	-	-	-
7	*Chaetosphaeria vermicularioides*	-	-	-	-	-	-	-	-	-	-	0.42 (4.125)	-
8	*Humicola grisea*	-	-	-	3.10 (27.29)	-	-	-	-	-	-	-	-
9	*Mammaria echinobotryoides*	1.19 (6.38)	-	-	-	-	-	-	-	-	1.24 (10.00)	-	-
10	*Paecilomyces carneus*	-	-	-	-	3.13 (22.22)	-	-	-	0.43 (0.88)	-	-	-
11	*P. marquandii*	-	-	-	-	1.03 (7.33)	-	-	-	-	-	-	-
12	*P. variotii*	-	-	-	-	-	-	-	0.79 (14.16)	-	-	-	-
13	*P. lilacinus*	-	-	-	-	-	-	-	-	10.14 (20.37)	-	-	-
14	*Penecillium brevicompactum*	-	3.99 (66.60)	2.01 (41.50)	-	-	-	-	-	-	-	-	-
15	*P. canescens*	-	-	-	-	-	0.83 (12.38)	-	-	-	-	-	4.19 (16.65)
16	*P. chrysogenum*	-	-	0.80 (16.5)	-	-	-	-	0.39 (7.08)	12.79 (25.69)	-	1.67 (16.6)	1.62 (28.54)
17	*P. daleae*	-	-	-	-	-	-	-	-	-	-	1.67 (16.6)	1.62 (28.54)

18	*P. expansum*	-	-	-	-	1.56 (11.11)	-	-	0.79 (14.16)	-	-	-	-
19	*P. frequentans*	-	-	-	-	-	-	-	-	-	-	-	0.80 (14.16)
20	*P. jensenii*	-	-	0.80 (16.5)	-	-	-	-	0.39 (7.08	0.87 (1.76)	-	2.94 (29.12)	0.80 (14.16)
21	*P. lividum*	-	-	-	-	-	-	-	-	-	-	2.94 (29.12)	0.80 (14.16)
22	*P. nigricans*	0.35 (19.15)	-	-	0.51 (4.50)	-	1.26 (18.76)	-	-	-	-	-	0.80 (14.16)
23	*P. oxalicum*	0.35 (19.15)	-	-	-	-	-	-	-	-	-	-	0.80 (14.16)
24	*P. restrictum*	-	-	-	-	-	-	-	0.79 (14.16)	0.87 (1.76)	-	-	-
25	*P. rubrum*	-	-	-	-	2.60 (18.44)	-	-	0.39 (7.08)	0.87 (1.76)	9.95 (80.0)	5.05 (50.00)	-
26	*P. stoloniferum*	-	-	-	-	1.56 (11.11)	-	-	-	-		5.05 (50)	-
27	*P. thomnii*	-	-	-	0.51 (4.50)	-	-	-	-	-	-	-	-
28	*P. waksmanii*	0.35 (19.15)	-	-	-	-	-	-	-	-	-	-	-
29	*Phoma eupyrena*	-	-	1.21 (25.00)	-	-	-	-	-	-	-	-	-
30	*Pythium intermedium*	-	-	-	1.02 (9.00)	-	-	-	0.79 (14.16)	-	-	-	-
31	*P. irregulare*	-	-	-	-	-	-	-	-	0.87 (1.76)	-	-	-
32	*Rhizopus oryzae*	-	-	-	-	-	2.53 (24.95)	-	-	-	-	-	-
33	*Trichoderma viride*	2.39 (12.77)	-	-	-	2.08 (14.77)	-	3.35 (72.67)	-	2.64 (5.42)	-	-	-
34	*Verticillium dahliae*	-	-	-	-	-	-	-	0.79 (14.16)	-	-	-	-
35	*V. alboatrum*	1.59 (8.49)	2.00 (33.40)	-	1.55 (13.64)	2.08 (14.77)	-	-	-	-	0.82 (6.60)	-	-
36	*V. chlamydosporium*	-	-	-	-	-	-	-	-	3.52 (7.07)	-	-	-
37	White sterile mycelia	-	-	-	-	-	-	0.41 (9.02)	0.39 (7.08)	1.32 (2.66)	-	-	-

Table 12 Monthly variation of fungal population (per gram dry soil x10^3) in the undisturbed forest soil at depth 0-10cm (2003) Values in parentheses are percentage relative abundance

Sl No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Absidia cylindrospora*	-	-	-	-	-	0.52 (1.01)	-	-	-	-	-	-
2	*A. glauca*	-	-	-	-	-	-	-	-	0.94 (0.93)	0.50 (2.91)	-	-
3	*Acremonium murorum*	-	-	0.51 (0.89)	-	-	-	-	-	-	-	-	-
4	*A. cerealis*	-	-	-	-	-	-	-	-	-	-	-	0.45 (1.55)
5	*Arthrobotrys conoides*	-	0.40 (1.49)	-	-	-	-	-	-	-	-	-	-
6	*Aspergillus alutaceus*	-	-	-	-	-	0.52 (1.01)	1.45 (2.07)	-	-	-	2.67 (5.13)	-
7	*A. candidus*	-	-	-	12.97 (27.16)	-	-	1.45 (2.07)	-	-	-	2.23 (4.28)	-
8	*A. clavatus*	-	-	-	2.07 (4.34)	-	-	-	-	-	-	-	-
10	*A. fumigatus*	-	-	-	0.51 (1.08)	-	-	0.96 (1.37)	-	-	-	15.56 (29.92)	-
11	*A. niger*	-	-	-	-	-	-	4.83 (6.89)	-	-	-	-	-
12	*Brown sterile mycelia*	-	-	-	-	-	-	4.35 (6.21)	-	-	-	-	-
13	*Cladosporium cladosporioides*	-	-	-	0.51 (1.08)	-	-	-	3.30 (3.39)	-	-	-	2.29 (7.83)
14	*Eupenicillium sheari*	-	-	0.51 (0.89)	-	-	5.26 (10.19)	-	-	-	3.03 (17.65)	-	-
15	*E. lapidosum*	-	0.40 (1.49)	-	-	-	-	-	-	-	-	-	-
16	*E .brefeldianum*	-	-	-	-	-	-	-	-	3.31 (3.28)	-	-	-
17	*Gliocladium roseum*	-	-	-	-	-	-	-	-	0.47 (0.46)	-	-	-
18	*Helminthosporium velutinum*	-	-	-	0.51 (1.08)	-	-	-	-	-	-	-	-

19	*Humicola fuscoatra*	-	-	-	-	-	-	1.93 (2.75)	-	-	-	1.77 (3.41)	-
20	*H. grisea*	8.91 (28.21)	-	-	-	-	-	-	-	-	-	-	-
21	*Mortierella parvispora*	-	-	-	5.19 (10.86)	-	-	-	-	-	-	-	-
22	*M. rammaniana*	-	0.82 (2.99)	-	-	-	-	-	-	-	-	-	-
23	*M. vinacea*	4.05 (12.81)	-	3.09 (5.45)	-	-	-	-	-	-	-	-	-
24	*Mucor hiemalis*	-	-	0.51 (0.89)	11.42 (23.89)	-	-	-	-	0.47 (0.46)	1.00 (5.83)	-	-
25	*Nectria ventricosa*	-	-	-	-	-	-	4.35 (6.21)	-	-	-	-	-
26	*Paecilomyces carneus*	-	-	1.04 (1.83)	-	-	-	-	-	-	-	-	-
27	*P. lilacinus*	-	1.63 (5.97)	-	-	0.50 (2.15)	4.22 (8.17)	-	-	-	-	-	-
28	*P. brevicompactum*	4.46 (14.12)	3.28 (11.94)	3.09 (5.45)	2.07 (4.34)	-	-	-	70.41 (53.89)	33.18 (32.86)	1.52 (8.83)	3.11 (5.97)	-
29	*P. chrysogenum*	-	2.86 (10.43)	-	-	-	-	19.32 (27.58)	-	-	-	-	-
30	*P. expansum*	4.86 (15.38)	-	-	-	1.52 (6.52)	2.64 (5.11)	-	-	-	-	-	-
31	*P. frequentans*	-	2.86 (10.43)	8.25 (14.54)	-	0.50 (2.15)	-	-	-	-	-	2.67 (5.13)	-
32	*P. janthinellum*	-	-	-	-	-	1.04 (2.02)	10.62 (15.16)	-	-	-	-	-
33	*P. jensenii*	-	0.40 (1.49)	7.74 (13.64)	-	9.65 (41.29)	-	-	1.09 (1.12)	27.48 (27.23)	-	4.44 (8.54)	5.04 (17.21)
34	*P. lanosum*	-	2.45 (8.96)	-	-	-	6.32 (12.24)	5.32 (7.59)	-	1.42 (1.41)	5.56 (32.39)	-	-
35	*P. nigricans*	-	-	5.68 (10.01)	-	-	-	1.93 (2.75)	-	26.66 (26.28)	-	14.67 (28.21)	-
36	*P. oxalicum*	-	-	-	-	-	-	1.45 (2.07)	-	-	-	-	-
37	*P. purpurogenum*	-	0.40 (1.42)	-	-	-	-	1.45 (2.07)	-	-	-	-	-
38	*P. restrictum*	-	-	6.19 (10.91)	-	-	-	-	-	-	-	-	-
39	*P. rubrum*	-	0.40 (1.49)	-	8.30 (17.38)	1.01 (4.31)	14.22 (27.55)	-	7.15 (7.34)	-	0.50 (2.91)	-	-

40	*P. rugulosum*	-	-	-	-	-	7.38 (14.24)	-	11.51 (11.86)	-	-	-	-
41	*P. sacculum*	-	-	-	-	-	-	-	2.75 (2.82)	-	-	-	-
42	*P. simplicissimum*	-	-	-	-	0.50 (2.15)	3.68 (7.13)	-	14.85 (15.25)	-	-	-	-
43	*P. stoloniferum*	-	-	-	-	1.01 (4.31)	-	-	-	-	-	-	0.45 (1.55)
44	*P. thomnii*	-	-	-	0.51 (1.08)	-	-	-	-	-	-	-	-
45	*P. waksmanii*	-	-	-	-	1.01 (4.31)	-	-	-	-	-	-	-
46	*Penicillium* sp.	-	0.40 (1.49)	-	0.51 (1.08)	-	-	-	-	-	-	-	-
47	*Phoma eupyrena*	-	-	0.51 (0.89)	2.07 (4.34)	-	-	-	-	-	-	-	-
48	*Pythium irregulare*	-	0.82 (2.99)	-	-	-	-	-	-	-	-	-	-
49	*P. intermedium*	-	0.40 (1.49)	-	-	-	-	0.96 (1.37)	3.84 (3.95)	-	-	-	-
50	*Scopulariopsis brevicaulis*	-	-	-	-	0.50 (2.15)	-	-	-	-	-	-	-
51	*S. brumptii*	-	-	-	-	-	-	-	-	-	0.50 (2.91)	-	9.62 (32.82)
52	*S. fusca*	-	-	8.25 (14.54)	-	-	-	-	-	-	-	-	-
53	*Trichoderma harzianum*	-	-	-	-	1.52 (6.52)	-	-	-	-	-	-	-
54	*T. koningii*	-	0.82 (2.99)	-	-	-	-	-	-	3.31 (3.28)	-	-	-
55	*T. viride*	4.05 (12.80)	4.91 (17.91)	10.34 (18.19)	1.03 (2.15)	-	-	2.42 (3.46)	-	0.47 (0.46)	-	-	-
56	*Torula herbarum*	-	-	-	-	-	-	-	-	0.47 (0.46)	-	2.67 (5.13)	-
57	*Verticillium alboatrum*	5.26 (16.65)	0.82 (2.98)	1.02 (1.79)	-	-	-	-	-	0.93 (0.93)	1.00 (5.83)	-	11.44 (39.05)
58	*V. chlamyosporium*	-	-	-	-	1.52 (6.52)	-	0.96 (1.37)	-	-	-	-	-
59	*V. dahliae*	-	-	-	-	0.50 (2.15)	-	-	-	-	-	-	-
60	White sterile mycelia	-	2.86 (10.34)	-	-	3.55 (15.19)	5.79 (11.22)	6.28 (8.96)	-	1.42 (1.41)	3.53 (20.56)	2.23 (4.28)	-

Table 13 Monthly variation of fungal population (per gram dry soil x10^3) in the undisturbed forest soil at depth 10-20cm (2003) Values in parentheses are percentage relative abundance

Sl No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Absidia cylindrospora*	-	-	-	0.50 (4.13)	-	0.44 (1.46)	-	-	-	-	-	-
2	*A. glauca*	-	-	-	-	-	-	-	-	8.51 (12.41)	-	-	-
3	*Acremonium kiliense*	-	-	-	-	0.45 (1.34)	-	-	-	-	-	-	-
4	*A. murorum*	-	-	2.01 (10.79)	-	-	0.89 (2.91)	-	-	-	-	-	-
5	*A. strictum*	-	-		0.50 (4.13)	-	-	-	-	-	-	-	-
6	*Aspergillus alutaceus*	-	-	-	4.09 (33.38)	-	-	-	-	-	-	-	-
7	*A. candidus*	-	-	-	-	8.23 (24.32)	-	2.61 (3.85)	-	-	-	-	-
8	*A. carneus*	-	0.39 (5.50)	-	-	-	-	-	-	-	-	-	-
9	*A. flavus*	-	-	-	-	12.35 (36.48)	-	-	-	-	-	044 (0.85)	-
10	*A. fumigatus*	-	-	-	-	-	-	-	-	-	-	23.11 (34.58)	-
11	*A. niger*	-	-	-	-	-	-	0.82 (1.27)	-	-	-	-	-
12	Brown sterile	-	-	-	1.01 (8.33)	-	-	-	-	-	-	-	-
13	*Chaetomium crispatum*	-	-	-	-	-	-	-	-	-	-	-	0.88 (1.87)
14	*Cladosporium cladosporidioides*	1.98 (13.89)	-	-	-	-	-	-	8.94 (12.24)	-	-	0.42 (0.62)	1.75 (3.74)
15	*Eupenicillium javanicum*	-	-	-	-	-	-	-	49.67 (68.02)	-	-	-	-
16	*E. sheari*	-	-	-	-	-	-	-	-	-	0.46 (3.30)	-	-
17	*E. brefeldianum*	-	-	-	-	-	-	-	-	1.34 (1.96)	-	-	-

18	*Fusarium redolens*	-	-	-	-	-	-	-	-	-	0.46 (3.30)	-	-
19	*Gliocladium roseum*	-	0.79 (11.17)	1.00 (5.35)	-	-	-	-	13.90 (19.04)	-	-	-	-
20	*Helminthosporium velutinum*	-	-	-	0.50 (4.13)	-	-	-	-	-	-	-	-
21	*Humicola fuscoatra*	-	-	1.52 (8.11)	-	-	-	-	-	-	-	0.82 (1.25)	-
22	*Mammaria echinobotryoides*	-	-	-	-	-	-	0.43 (0.63)	-	-	-	-	-
23	*Mortierella parvispora*	-	-	-	-	-	-	-	-	0.90 (1.31)	-	-	-
24	*M. rammaniana*	-	-	4.55 (24.33)	-	-	-	-	-	0.44 (0.65)	-	0.83 (1.25)	2.20 (4.68)
25	*M. vinacea*	-	1.19 (16.67)	-	-	-	0.89 (2.91)	-	-	-	-	-	-
26	*Mucor hiemalis*	-	-	-	-	-	-	1.74 (2.56)	-	0.90 (1.31)	0.46 (3.30)	-	-
27	*Nectria ventricosa*	-	-	-	-	-	0.44 (1.46)	-	-	-	-	-	-
28	*Paecilomyces carneus*	-	-	-	0.50 (4.13)	-	-	-	-	-	-	-	-
29	*Paecilomyces lilacinus*	-	0.39 (5.50)	-		-	-	-	-	-	-	-	-
30	*Penicillium brevicompactum*	-	-	1.52 (8.11)	1.01 (8.33)	-	-	0.86 (1.27)	-	15.67 (22.87)	-	4.63 (6.92)	1.32 (2.80)
31	*P. chrysogenum*	-	0.80 (11.17)	-	1.01 (8.25)	-	-	9.15 (13.46)	-	23.29 (33.98)	-	8.41 (12.58)	-
32	*P. daleae*	-	-	-	-	-	-	-	-	-	0.46 (3.30)	-	-
33	*P. expansum*	-	2.79 (38.88)	-	-	-	-	-	-	-	-	-	-
34	*P. frequentans*	-	-	-	-	2.29 (6.77)	3.25 (10.28)	-	-	-	0.46 (3.30)	-	-
35	*P. funiculosum*	1.58 (11.11)	-	-	-	-	-	-	-	-	-	0.83 (1.25)	-
36	*P. janthinellum*	-	-	-	-	-	-	18.73 (27.56)	-	-	-	-	-
37	*P. jensenii*	-	-	4.55 (24.33)	-	1.37 (4.05)	-	-	-	-	-	13.87 (20.75)	5.71 (12.15)

38	*P. lanosum*	1.98 (13.92)	-	-	-	-	-	-	-	-	0.46 (3.30)	-	3.96 (8.41)
39	*P. nigricans*	-	-	1.52 (8.11)	-	-	-	18.30 (26.92)	-	-	-	5.89 (8.81)	-
40	*P. oxalicum*	2.37 (16.67)	-	-	-	-	-	-	-	-	-	-	-
41	*P. purpurogenum*	-	-	-	-	-	-	6.54 (9.62)	-	-	-	-	-
42	*P. rubrum*	1.58 (11.11)	-	0.50 (2.68)	1.53 (12.5)	2.74 (8.11)	17.45 (57.34)	-	-	-	-	5.46 (8.17)	-
43	*P. rugulosum*	-	-	-	-	0.91 (2.68)	-	-	-	-	-	-	-
44	*P. simplicissimum*	-	-	-	-	1.83 (5.39)	6.27 (20.59)	-	-	-	3.27 (23.30)	-	-
45	*P. variable*	-	-	-	-	-	0.89 (2.91)	-	-	-	-	-	-
46	*Penicillium.* sp.	1.98 (13.89)	-	-	-	-	-	-	-	-	-	-	-
47	*Phoma eupyrena*	-	-	1.52 (8.11)	-	-	-	-	-	-	-	-	-
48	*Phoma medicaginis*	7.11 (16.67)	-	-	-	-	-	-	-	-	-	-	-
49	*Pythium intermedium*	-	-	-	0.50 (4.13)	-	-	0.86 (1.27)	0.49 (0.67)	-	-	-	-
50	*Scopulariopsis brevicaulis*	-	-	-	-	-	-	-	-	-	0.46 (3.30)	-	-
51	*Trichoderma koningii*	-	-	-	-	-	-	-	-	-	0.46 (3.30)	-	-
52	*T. viride*	-	-	-	-	0.45 (1.34)	-	7.84 (11.54)	-	4.93 (7.19)	-	-	27.27 (57.94)
53	*Torula herbarum*	-	-	-	-	-	-	-	-	0.44 (0.65)	-	-	-
54	*Verticillium alboatrum*	0.39 (2.77)	0.39 (5.50)	-	0.50 (8.33)	-	-	-	-	11.19 (16.33)	-	-	3.52 (7.48)
55	*V. dahliae*	-	-	-	-	1.37 (4.05)	-	-	-	-	-	-	-
56	White sterile mycelia	-	0.39 (5.50)	-	-	2.74 (8.11)	-	-	-	0.90 (1.31)	7.03 (50.00)	-	0.44 (0.93)
57	Yellow sterile mycelia	-	-	-	-	-	-	-	-	-	-	2.11 (3.15)	-

Table 14 Monthly variation of fungal population (per gram dry soil x10^3) in the undisturbed forest soil at depth 20-30cm (2003) Values in parentheses are percentage relative abundance

Sl No.	Fungi	J	F	M	A	M	J	J	A	S	O	N	D
1	*Absidia cylindrospora*	-	0.39 (4.52)	-	-	-	-	-	-	-	-	-	-
2	*A. glauca*	-		-	-	-	-	-	-	1.31 (7.50)	-	-	-
3	*Acremonium butyri*	-	-	-	2.61 (17.27)	-	-	-	-	-	-	-	-
4	*A. murorum*	-	-	0.50 (2.30)	-	-	-	-	-	-	-	-	-
5	*Aspergillus alutaceus*	-	-	-	1.56 (10.34)	-	-	0.87 (2.39)	0.48 (0.99)	-	-	-	-
6	*A. candidus*	-	-	-	-	-	-	6.17 (16.88)	-	-	-	-	-
7	*A. flavus*	-	-	-	-	-	-	-	-	-	-	2.56 (7.89)	-
8	*A. fumigatus*	-	-	-	-	-	-	-	-	-	-	15.81 (48.68)	-
9	*A. versicolor*	-	-	-	-	-	-	-	-	-	-	0.43 (1.30)	-
10	*Botryotrichum piluliferum*	-	-	0.50 (2.30)	-	-	-	-	-	-	-	-	-
11	*Cladosporium cladosporidioides*	-	-	-	-	-	-	-	26.35 (54.00)	-	-	-	0.87 (3.54)
12	*Eupenicillium sheari*	-	-	-	-	-	-	-	-	1.31 (7.50)	-	-	0.43 (1.77)
13	*E. brefeldianum*	-	-	-	-	-	-	-	-	-	0.45 (9.91)	-	-
14	*Fusarium redolens*	-	-	-	-	-	-	-	-	-	-	-	0.43 (1.77)
15	*Humicola fuscoatra*	-	1.59 (18.22)	-	1.05 (6.92)	-	-	-	-	-	-	0.42 (1.30)	-
16	*H. grisea*	-	-	-	1.05 (6.93)	-	0.43 (1.94)	-	-	-	-	-	-
17	*Mammaria echinobotryoides*	-	-	-	-	-	-	1.32 (3.61)	-	-	-	-	-

18	*Mortierella hyalina*	-	-	-	-	-	-	-	-	-	-	-	0.43 (1.77)
19	*M. parvispora*	-	-	-	-	-	-	-	-	2.62 (15.0)	-	-	-
20	*M. rammaniana*	-	-	2.03 (9.28)	-	-	-	-	-	5.23 (30.00)	-	0.85 (2.61)	2.19 (8.94)
21	*M. humilis*	-	0.39 (4.52)	-	-	-	-	-	-	0.43 (2.48)	-	-	-
22	*Paecilomyces carneus*	-	-	1.52 (6.98)	-	0.44 (16.50)	1.73 (7.82)	-	-	-	-	-	-
23	*P. marquandii*	-	-	-	-	0.87 (33.00)	-	-	-	-	-	-	-
24	*Penicillium brevicompactum*	-	0.80 (9.18)	-	-	-	3.02 (13.71)	0.44 (1.19)	1.46 (3.00)	-	0.90 (19.82)	6.83 (21.04)	6.13 (25.01)
25	*P. canescens*	-	-	-	-	-	-	-	-	-	0.90 (19.82)	-	-
26	*P. chrysogenum*	0.39 (11.11)	-	-	-	-	-	-	-	-	-	-	-
27	*P. expansum*	-	-	-	-	0.44 (16.50)	0.86 (3.88)	-	-	-	-	-	-
28	*P. frequentans*	-	2.80 (22.87)	-	-	-	-	-	-	-	-	-	1.31 (5.36)
29	*P. janthinellum*	-	-	-	-	-	1.73 (7.82)	17.61 (48.17)	-	-	-	-	-
30	*P. jensenii*	-	-	4.06 (18.63)	-	-	-	-	6.84 (14.00)	-	-	2.56 (7.89)	9.19 (37.49)
31	*P. lanosum*	-	-	-	-	-	-	-	-	-	-	-	0.43 (1.77)
32	*P. nigricans*	-	-	5.59 (25.61)	3.12 (20.68)	-	-	-	-	-	-	0.43 (1.30)	-
33	*P. oxalicum*	1.55 (44.44)	-	-	-	-	-	0.44 (1.19)	-	-	-	-	-
34	*P. purporugenum*	-	-	-	-	-	-	1.32 (3.61)	-	-	-	-	-
35	*P. restrictum*	-	-	5.59 (6.98)	-	-	-	-	-	-	-	-	-
36	*P. rubrum*	1.178 (33.33)	-	-	-	-	11.70 (52.94)	-	1.95 (4.00)	-	0.45 (9.91)	2.14 (6.59)	-
37	*P. rugulosum*	-	-	-	-	-	-	-	8.30 (17.00)	-	-	-	-

38	*P. simplicissimum*	-	-	0.50 (2.30)	-	-	0.86 (3.88)	-	0.48 (0.99)	-	-	-	-
39	*P. stoloniferum*	-	-	-	-	0.44 (16.50)	-	-	-	-	-	-	1.31 (5.36)
40	*P. thomnii*	-	-	-	1.56 (10.34)	-	-	-	-	-	-	-	-
41	*Penicillium* sp.	-	1.59 (18.22)	-	-	-	-	-	-	-	-	-	-
42	*Phoma eupyrena*	-	-	1.52 (6.98)	-	-	-	-	-	-	-	-	-
43	*Pythium intermedium*	0.39 (11.11)	-	-	3.12 (20.68)	-	1.73 (7.82)	0.44 (1.19)	2.93 (6.00)	-	-	-	0.43 (1.77)
44	*Scopulariopsis brevicaulis*	-	-	-	-	-	-	-	-	-	0.45 (9.91)	-	-
45	*S. brumptii*	-	-	-	-	-	-	-	-	-	0.45 (9.91)	-	-
46	*Trichoderma koningii*	-	-	-	-	-	-	-	-	-	0.90 (19.82)	-	-
47	*T. viride*	-	-	-	-	0.44 (16.50)	-	2.21 (6.04)	-	0.43 (2.48)	-	-	0.87 (3.54)
48	*Verticillium alboatrum*	-	1.20 (13.69)	-	1.03 (6.83)	-	-	-	-	3.94 (22.50)	-	-	0.433 (1.77)
49	*V .chlamydosporium*	-	-	-	-	-	-	5.72 (15.65)	-	-	-	-	-
50	White sterile mycelia	-	0.80 (9.18)	-	-	-	-	-	-	0.22 (12.53)	-	0.43 (1.30)	-

Table 15 Bacterial species isolated from disturbed and undisturbed forest soils

Bacterial Species	Disturbed Forest	Undisturbed Forest
Arthrobacter sp.	+	+
Bacillus cereus	+	-
Bacillus subtilis	+	+
Bacillus sp.	+	+
Micrococcus sp.	+	+
Pseudomonas sp.	+	+
Rhizobium sp.	+	+

+ = present

- = absent

Table 16 Monthly variation of bacterial population (per gram dry soil x10^5) in the disturbed forest soil at depth 0-10cm (2002) Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	-	1.29 (42.11)	-	-	-	0.38 (7.22)	-	0.10 (1.25)	2.07 (9.63)	0.13 (5.21)	0.63 (4.38)	0.23 (1.67)
Bacillus cereus	-	-	0.05 (0.82)	-	-	-	-	-	-	-	-	-
B. subtilis	1.77 (30.29)	0.37 (12.02)	2.81 (48.15)	10.46 (55.63)	12.72 (62.50)	3.27 (61.85)	0.55 (42.87)	2.63 (32.29)	4.98 (23.11)	0.65 (27.09)	7.53 (52.72)	-
Bacillus sp.	3.82 (65.15)	0.02 (36.84)	0.10 (17.69)	7.08 (37.65)	7.27 (35.71)	1.09 (20.62)	0.37 (28.58)	4.68 (57.37)	8.29 (38.51)	0. 88 (36.46)	2.00 (14.01)	7.67 (55.66)
Micrococcus sp.	-	-	1.94 (33.33)	0.85 (4.53)	0.36 (1.78)	0.33 (6.18)	0.29 (22.45)	0.41 (5.01)	4.15 (19.26)	0.30 (12.50)	3.00 (21.01)	3.32 (24.12)
Pseudomonas sp.	0.27 (4.56)	0.07 (2.26)	-	-	-	0.22 (4.12)	0.08 (6.12)	0.10 (1.25)	2.07 (9.63)	0.20 (8.34)	0.75 (5.25)	1.41 (10.20)
Rhizobium sp.	-	0.21 (6.77)	-	-	-	-	-	0.23 (2.82)	-	0.25 (10.41)	0.38 (2.63)	1.15 (8.35)

Table 17 Monthly variation of bacterial population (per gram dry soil x 10^5) in the disturbed forest soil at depth 10-20cm (2002) Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.07 (3.37)	0.77 (45.21)	-	0.14 (2.00)	-	-	-	0.22 (4.94)	0.73 (3.95)	0.12 (4.95)	0.61 (6.61)	0.09 (1.12)
Bacillus cereus	-	0.16 (9.59)	-	-	-	-	-	-	-	-	-	-
B. subtilis	0.69 (33.72)	0.05 (2.74)	0.70 (32.26)	5.01 (70.23)	0.64 (4.13)	1.38 (46.43)	0.29 (41.36)	0.12 (64.83)	6.69 (35.97)	0.98 (39.60)	2.43 (26.45)	-
Bacillus sp.	12.96 (62.94)	0.54 (31.53)	0.58 (26.88)	1.67 (23.41)	9.33 (60.32)	6.63 (22.32)	0.22 (31.02)	0.71 (15.94)	7.76 (41.69)	0.86 (34.65)	4.87 (52.91)	6.05 (70.03)
Micrococcus sp.	-	-	070 (32.26)	0.26 (3.68)	3.07 (19.84)	0.53 (17.85)	0.19 (27.57)	0.37 (8.24)	1.39 (7.49)	0.27 (10.89)	0.44 (4.76)	1.55 (17.93)
Pseudomonas sp.	-	0.07 (4.11)	0.19 (8.60)	0.05 (0.67)	2.43 (15.71)	0.39 (13.39)	-	0.12 (3.79)	2.03 (10.89)	0.25 (9.89)	0.48 (5.29)	0.85 (9.80)
Rhizobium sp.	-	0.12 (6.85)	-	-	-	-	-	0.09 (2.19)	-	-	0.36 (3.97)	0.34 (3.92)

Table 18 Monthly variation of bacterial population (per gram dry soil x 10^5) in the disturbed forest soil at depth 20-30cm (2002)
Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.16 (11.54)	0.23 (10.34)	-	0.25 (4.93)	0.28 (2.08)	0.23 (0.90)	0.13 (29.45)	0.28 (10.36)	1.44 (12.28)	0.08 (4.48)	0.11 (2.77)	0.22 (3.44)
Bacillus subtilis	0.08 (51.93)	1.64 (73.56)	0.75 (38.46)	1.72 (34.49)	7.74 (58.33)	1.31 (44.14)	0.13 (29.45)	-	2.56 (21.93)	0.81 (47.78)	1.76 (41.44)	-
Bacillus sp.	0.33 (24.98)	0.23 (10.34)	0.67 (34.62)	2.26 (45.32)	3.76 (28.33)	0.80 (27.03)	0.09 (23.52)	2.42 (89.63)	3.85 (32.89)	0.38 (22.39)	1.18 (27.63)	3.49 (55.55)
Micrococcus sp.	-	0.03 (1.14)	0.25 (12.81)	0.76 (15.27)	0.99 (7.50)	0.54 (18.01)	0.07 (17.64)	-	1.92 (16.45)	0.30 (17.91)	0.78 (18.23)	1.20 (19.16)
Pseudomonas sp.	0.16 (11.54)	0.03 (1.14)	0.27 (14.10)	-	0.28 (2.08)	029 (9.91)	-	-	1.92 (16.45)	0.13 (7.46)	0.31 (7.18)	0.84 (13.41)
Rhizobium sp.	-	0.08 (3.45)	-	-	0.22 (1.67)	-	-	-	-	-	0.12 (2.76)	0.529 (8.43)

Table 19 Monthly variation of bacterial population (per gram dry soil x 10^5) in the disturbed forest soil at depth 0-10cm (2003)
Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.21 (1.06)	0.09 (1.34)	-	0.27 (8.97)	-	-	0.13 (1.85)	0.82 (32.26)	0.11 (14.77)	-	0.07 (0.65)	-
Bacillus cereus	-	-	0.09 (3,60)	-	-	-	-	-	-	-	-	-
B. subtilis	6.18 (31.83)	3.25 (47.49)	1.39 (57.66)	1.59 (51.73)	1.73 (31.16)	0.99 (42.31)	4.27 (62.73)	0.82 (32.26)	0.22 (29.63)	0.34 (36.36)	7.51 (64.79)	0.49 (41.99)
Bacillus sp.	12.80 (65.92)	2.29 (33.44)	0.67 (27.93)	0.85 (27.58)	-	0.45 (19.23)	0.58 (8.49)	0.89 (35.48)	0.19 (25.93)	3.66 (39.39)	1.50 (12.96)	0.37 (31.99)
Micrococcus sp.	-	0.07 (1.00)	0.26 (10.81)	0.36 (11.72)	0.98 (17.76)	0.59 (25.64)	1.25 (18.45)	0.03 (1.06)	-	0.23 (24.24)	1.13 (9.72)	0.18 (15.99)
Pseudomonas sp.	0.28 (1.46)	-	-	-	0.93 (16.74)	0.29 (12.82)	0.58 (8.49)	-	-	-	1.13 (9.72)	0.12 (9.99)
Rhizobium sp.	-	0.02 (16.73)	-	-	1.91 (34.42)	-	-	-	0.22 (29.63)	-	0.25 (2.16)	-

Table 20 Monthly variation of bacterial population (per gram dry soil x 10^5) in the disturbed forest soil at depth 10-20cm (2003) Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.16 (1.01)	2.76 (21.91)	-	-	-	-	0.21 (3.88)	0.08 (5.37)	-	-	0.17 (1.65)	-
Bacillus subtilis	7.53 (48.74)	0.44 (3.53)	0.58 (50.92)	0.44 (53.85)	0.39 (71.43)	0.37 (56.02)	0.07 (68.96)	0.87 (60.70)	0.37 (53.83)	0.33 (54.13)	6.93 (66.19)	0.11 (45.41)
Bacillus sp.	5.94 (38.45)	0.08 (0.71)	0.01 (27.28)	0.27 (33.33)	-	0.16 (24.0)	0.77 (14.22)	0.18 (12.49)	0.13 (19.22)	0.18 (29.17)	0.74 (7.09)	0.14 (54.49)
Micrococcus sp.	-	5.51 (43.82)	0.23 (20.02)	0.11 (12.82)	0.15 (28.57)	0.13 (20.0)	0.35 (6.47)	0.15 (10.71)	0.01 (23.07)	0.10 (16.67)	0.82 (7.80)	-
Pseudomonas sp.	0.63 (4.05)	-	0.02 (1.80)	-	-	-	0.28 (5.17)	0.05 (3.57)	-	-	0.57 (5.43)	-
Rhizobium sp.	0.08 (0.51)	3.78 (30.03)	-	-	-	-	0.07 (1.29)	0.10 (7.14)	0.03 (3.81)	-	1.24 (11.82)	-

Table 21 Monthly variation of bacterial population (per gram dry soil x10^5) in the disturbed forest soil at depth 20-30cm (2003) Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.06 (0.44))	0.21 (9.78)	-	0.09 (5.63)	0.17 (13.05)	-	0.14 (14.28)	0.16 (10.53)	-	-	-	-
Bacillus sp.	4.50 (34.93)	1.67 (7.60)	0.82 (46.51)	0.52 (31.18)	0.59 (45.66)	0.13 (41.67)	0.36 (35.71)	0.08 (5.26)	0.61 (48.93)	0.70 (45.91)	1.97 (25.16)	0.46 (57.13)
Bacillus subtilis	7.97 (61.82)	1.22 (55.43)	0.70 (27.91)	0.08 (47.88)	-	0.11 (33.33)	0.27 (26.21)	0.57 (38.59)	0.35 (27.65)	0.43 (27.87)	1.97 (50.31)	0.28 (34.27)
Micrococcus sp.	-	-	0.37 (20.93)	0.26 (15.49)	-	0.08 (25)	0.14 (14.29)	-	0.19 (14.89)	0.40 (26.23)	0.54 (13.84)	0.07 (8.57)
Pseudomonas sp.	0.36 (2.82)	-	0.08 (4.64)	-	0.53 (41.33)	-	0.09 (9.52)	-	-	-	0.35 (8.81)	-
Rhizobium sp.	-	0.59 (27.17)	-	-	-	-	-	0.68 (45.61)	0.11 (8.51)	-	0.07 (1.89)	-

Table 22 Monthly variation of bacterial population (per gram dry soil x 10^5) in the undisturbed forest soil at depth 0-10cm (2002)
Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.09 (1.04)	0.27 (13.54)	-	0.74 (3.42)	0.27 (1.47)	0.23 (3.87)	0.14 (7.06)	0.28 (2.70)	0.49 (1.95)	0.11 (4.82)	0.44 (4.90)	-
Bacillus subtilis	4.59 (54.68)	0.17 (8.34)	6.74 (92.90)	12.31 (56.85)	9.59 (51.62)	3.11 (50.38)	0.34 (17.65)	5.03 (48.23)	15.17 (60.12)	0.79 (36.54)	1.98 (22.06)	-
Bacillus sp.	3.29 (39.06)	1.45 (72.92)	0.51 (7.09)	6.18 (28.54)	5.21 (28.02)	1.87 (30.23)	0.94 (48.24)	4.86 (46.57)	7.36 (29.18)	0.97 (44.24)	4.62 (51.47)	1.89 (49.39)
Micrococcus sp.	-	0.02 (1.03)	-	2.42 (11.19)	1.78 (9.59)	0.59 (9.69)	0.34 (17.65)	0.43 (4.16)	1.35 (5.35)	0.21 (9.61)	0.88 (9.80)	1.39 (36.58)
Pseudomonas sp.	044 (5.21)	-	-	-	1.37 (7.34)	0.36 (5.81)	0.18 (9.41)	-	0.86 (3.40)	0.11 (4.82)	0.62 (6.86)	0.35 (9.15)
Rhizobium sp.	-	0.08 (4.16)	-	-	0.36 (1.92)	-	-	-	-	-	0.44 (4.90)	0.19 (4.88)

Table 23 Monthly variation of bacterial population (per gram dry soil x10^5) in the undisturbed forest soil at depth 10-20cm (2002)
Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	-	0.10 (12.85)	-	0.34 (4.12)	0.91 (10.39)	-	-	-	0.54 (3.42)		0.10 (3.42)	-
Bacillus subtilis	8.37 (26.79)	-	1.12 (28.78)	4.18 (50.0)	1.69 (19.29)	1.32 (51.28)	0.46 (41.99)	7.55 (91.51)	6.73 (42.43)	0.20 (40.12)	0.65 (21.22)	-
Bacillus sp.	2.08 (66.67)	0.36 (46.15)	2.37 (60.74)	2.46 (29.41)	2.99 (34.13)	0.77 (29.92)	0.28 (25.99)	0.38 (4.61)	7.93 (50.00)	0.12 (24.09)	1.15 (37.66)	6.88 (74.18)
Micrococcus sp.	-	-	0.41 (10.48)	0.86 (10.29)	1.43 (16.32)	0.35 (13.68)	0.15 (13.99)	0.14 (1.69)	0.32 (2.00)	0.08 (16.02)	0.46 (15.06)	1.31 (14.08)
Pseudomonas sp.	0.12 (3.92)	0.12 (15.39)	-	0.27 (3.24)	0.78 (8.90)	0.13 (5.13)	0.19 (17.99)	-	0.34 (2.14)	0.102 (20.12)	0.27 (8.90)	1.09 (11.73)
Rhizobium sp.	0.08 (1.30)	0.20 (25.62)	-	-	0.26 (2.96)	-	-	0.18 (2.18)	-	-	0.42 (13.70)	0.33 (3.52)

Table 24 Monthly variation of bacterial population (per gram dry soil x10^5) in the undisturbed forest soil at depth 20-30cm (2002) Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.56 (39.45)	0.59 (40.54)	-	0.52 (8.16)	-	-	-	0.03 (1.19)	0.04 (0.81)	0.23 (10.58)	0.19 (5.73)	-
Bacillus subtilis	0.86 (60.58)	-	0.28 (13.86)	3.23 (51.02)	0.03 (51.78)	0.93 (50.57)	-	2.26 (67.06)	3.31 (61.73)	0.83 (38.46)	1.03 (31.85)	-
Bacillus sp.	-	0.59 (40.54)	1.76 (86.16)	1.55 (24.49)	0.34 (23.21)	0.49 (26.44)	0.32 (42.84)	0.58 (17.36)	1.32 (24.69)	0.52 (24.03)	1.07 (33.12)	12.97 (76.39)
Micrococcus sp.	-	-	-	0.91 (14.29)	0.21 (14.30)	0.25 (13.79)	0.34 (45.70)	0.42 (12.57)	0.35 (6.58)	0.19 (8.65)	0.25 (7.64)	2.53 (14.91)
Pseudomonas sp.	-	0.12 (8.11)	-	0.13 (2.04)	0.16 (10.71)	0.17 (9.19)	0.08 (11.43)	0.06 (1.79)	0.33 (6.17)	0.23 (10.59)	0.37 (11.47)	1.06 (6.21)
Rhizobium sp.	-	0.16 (10.82)	-	-	-	-	-	-	-	0.17 (7.69)	0.33 (10.19)	0.42 (2.48)

Table 25 Monthly variation of bacterial population (per gram dry soil x10^5) in the undisturbed forest soil at depth 0-10 cm (2003) Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.57 (2.68)	0.23 (15.07)	-	0.26 (3.55)	0.15 (13.05)	0.39 (9.93)	0.48 (8.92)	0.39 (3.89)	0.05 (10.04)	-	-	-
Bacillus sp.	3.44 (16.27)	0.10 (6.85)	0.72 (71.79)	4.59 (62.99)	0.74 (63.06)	1.97 (49.67)	2.17 (40.17)	0.58 (5.82)	-	0.53 (41.18)	13.33 (55.81)	5.49 (90.22)
Bacillus subtilis	14.72 (69.57)	1.17 (78.09)	0.28 (28.23)	1.64 (22.42)	0.05 (4.35)	0.95 (23.84)	1.59 (29.46)	8.55 (86.15)	0.33 (69.96)	0.40 (31.38)	3.33 (13.95)	0.53 (8.65)
Micrococcus sp.	-	-	-	0.80 (11.03)		0.53 (13.25)	0.85 (15.62)	0.30 (3.05)	0.09 (19.99)	0.35 (27.47)	3.58 (14.98)	0.07 (1.13)
Pseudomonas sp.	2.43 (11.48)	-	-	-	0.22 (19.36)	0.13 (3.31)	0.36 (6.69)	-	-	-	0.03 (9.30)	-
Rhizobium sp.	-	-	-	-	-	-	0.29 (5.36)	0.11 (1.11)	-	-	1.07 (5.95)	-

Table 26 Monthly variation of bacterial population (per gram dry soil x10^5) in the undisturbed forest soil at depth 10-20cm (2003) Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.06 (0.34)	0.21 (19.33)	-	0.31 (19.35)	-	0.20 (11.25)	0.17 (10.40)	0.52 (9.01)	0.16 (43.78)	-	-	-
Bacillus sp.	11.68 (66.93)	-	0.68 (57.43)	0.74 (46.78)	0.23 (62.53)	0.67 (37.49)	0.33 (19.48)	4.72 (81.54)	-	0.94 (34.78)	10.51 (72.57)	0.09 (1.81)
Bacillus subtilis	2.53 (14.49)	0.62 (53.44)	0.51 (42.54)	0.38 (24.19)	0.14 (37.52)	0.56 (31.23)	0.01 (38.96)	0.49 (8.58)	0.07 (18.76)	1.05 (39.13)	2.48 (21.63)	4.74 (98.19)
Micrococcus sp.	-	0.12 (10.35)	-	0.15 (9.67)	-	0.29 (16.25)	0.28 (16.88)	-	1.34 (37.52)	0.33 (17.39)	0.63 (4.35)	-
Pseudomonas sp.	1.42 (8.15)	-	-	-	-	0.07 (3.75)	0.24 (14.28)	-	-	0.23 (8.69)	0.21 (1.45)	-
Rhizobium sp.	1.76 (10.08)	0.19 (17.23)	-	-	-	-	-	0.05 (0.86)	-	-	-	-

Table 27 Monthly variation of bacterial population (per gram dry soil x10^5) in the undisturbed forest soil at depth 20-30 cm (2003) Values in parentheses are percentage relative abundance

Bacteria	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Arthrobacter sp.	0.09 (0.53)	0.20 (47.62)	-	0.34 (16.04)	0.04 (6.49)	0.07 (12.5)	0.15 (5.93)	0.76 (18.90)	0.19 (25.71)	-	-	-
Bacillus sp.	8.32 (45.19)	-	0.88 (47.29)	1.27 (60.49)	0.07 (54.89)	0.13 (25)	0.44 (16.95)	2.44 (60.97)	-	0.57 (34.72)	5.57 (46.19)	2.38 (59.56)
Bacillus subtilis	9.96 (54.07)	0.22 (52.38)	0.99 (52.69)	0.39 (18.52)	0.15 (22.59)	0.19 (37.5)	0.99 (38.14)	0.27 (6.71)	0.26 (34.28)	0.50 (30.54)	2.22 (18.41)	1.31 (32.79)
Micrococcus sp.	-	-	-	0.16 (7.41)	-	0.09 (16.67)	6.61 (25.42)	0.34 (8.54)	0.24 (31.45)	0.57 (34.72)	4.27 (35.40)	0.31 (7.65)
Pseudomonas sp.	0.04 (0.21)	-	-	-	0.11 (16.13)	0.04 (8.33)	0.29 (11.02)	-	0.07 (8.57)	-	-	-
Rhizobium sp.	-	-	-	-	-	-	0.13 (5.09)	0.19 (4.88)	-	-	-	-

Table 28 One-way analysis of variance (ANOVA) of the microbial populations between the disturbed forest (DF) and undisturbed forest (UDF) soils at three different depths 0-10cm, 10-20cm and 20-30cm ($P \leq 0.05$)

Soil properties	Source of variation	F-ratio	P- level
Fungal population	DF(0-10cm) x UDF(0-10cm)	4.79	3.02×10^{-2}
	DF(0-10cm) x UDF(10-20cm)	4.59	3.37×10^{-2}
	DF(0-10cm) x UDF(20-30cm)	47.61	1×10^{-3}
	DF(10-20cm) x UDF(0-10cm)	50.64	1×10^{-3}
	DF(10-20cm) x UDF(10-20cm)	5.76	1.76×10^{-2}
	DF(10-20cm) x UDF(20-30cm)	4.97	2.73×10^{-2}
	DF(20-30cm) x UDF(0-10cm)	94.87	$1X10^{-3}$
	DF(20-30cm) x UDF(10-20cm)	26.71	1×10^{-6}
	DF(20-30cm) x UDF(20-30cm)	-	-
Bacterial population	DF(0-10cm) x UDF (0-10cm)	-	-
	DF(0-10cm) x UDF(10-20cm)	5.41	2.14×10^{-2}
	DF(0-10cm) x UDF(20-30cm)	10.86	1.24×10^{-3}
	DF(10-20cm) x UDF(0-10cm)	13.09	4.10×10^{-4}
	DF(10-20cm) x UDF(10-20cm)	-	-
	DF(10-20cm) x UDF(20-30cm)	-	-
	DF(20-30cm) x UDF(0-10 cm)	28.19	1×10^{-3}
	DF(20-30cm) x UDF(10-20cm)	7.53	6.84×10^{-3}
	DF(20-30cm) x UDF(20-30cm)	-	-

Note: Insignificant values are marked with '–' sign

Table 29 One way analysis of variance (ANOVA) of the microbial populations among the three different depths 0-10cm, 10-20cm and 20-30cm in the disturbed forest (DF) and undisturbed forest (UDF) soils (P≤0.05)

Soil properties	Source of variation	F-ratio	P- level
Fungal population	DF(0-10cm x 10-20cm)	27.12	$1x10^{-6}$
	DF(0-10cm x 20-30cm)	66.62	$1x10^{-3}$
	DF(10-20cm x 20-30cm)	13.99	$2.66x10^{-4}$
	DF(0-10cm x 10-20cm x 20-30cm)	39.02	$1x10^{-3}$
	UDF(0-10cm x 10-20cm)	16.61	$7.6x10^{-5}$
	UDF(0-10cm x 20-30cm)	74.09	$1x10^{-3}$
	UDF(10-20 cm x 20-30cm)	16.27	$8.9x10^{-5}$
	UDF(0-10cm x 10-20cm x 20-30cm)	34.07	$1x10^{-3}$
Bacterial population	DF(0-10cm x 10-20cm)	8.36	$4.44x10^{-3}$
	DF(0-10cm x 20-30cm)	24.27	$2x10^{-6}$
	DF(10-20 cm x 20-30cm)	-	-
	DF(0-10cm x 10-20cm x 20-30cm)	12.56	$7x10^{-6}$
	UDF(0-10cm x 10-20cm)	9.84	$2.07x10^{-3}$
	UDF(0-10cm x 20-30cm)	15.72	$1.16x10^{-4}$
	UDF(10-20cm x 20-30cm)	-	-
	UDF(0-10cm x 10-20cm x 20-30cm)	10.26	$5.5x10^{-5}$

Note: Insignificant values are marked with '–' sign

Microbial biomass carbon

Similar trend of monthly variations in microbial biomass carbon (C_{mic}) was observed in both the study sites. Comparing both the study sites, it was observed that the soil of the undisturbed forest had higher C_{mic} than that of disturbed forest soil. In the first year of study period, the maximum C_{mic} at 0-10cm was observed in the month of January and the minimum C_{mic} was observed in the month of June in both the forest soils. At 10-20cm depth, maximum C_{mic} in the disturbed forest was observed in the month of November and the minimum was observed in the month of August, whereas, in the undisturbed forest soil at 10-20cm depth, the maximum C_{mic} was observed in the month of January and the minimum C_{mic} was recorded in the month of November. At 20-30cm depth, the maximum C_{mic} was observed in the month of January and the minimum was observed in the month of August in the disturbed forest soil, whereas, in the undisturbed forest soil, the maximum C_{mic} was observed in the month of April and the minimum was observed in the month of November. In the second year at 0-10cm, in the disturbed forest soil, the maximum C_{mic} was observed in the month of November and the minimum was observed in the month of March whereas, in the undisturbed forest soil the maximum C_{mic} was observed in the month of May and the minimum was recorded in the month of August. At 10-20cm the maximum C_{mic} was observed in the month of July and the minimum was observed in the month of September in the disturbed forest soil whereas, the maximum C_{mic} was recorded in the month of January and the minimum was recorded in the month of August in the undisturbed forest and in the third depth i.e., at 20-30cm in the disturbed forest soil, the maximum C_{mic} was observed in the month of April and the minimum was observed in the month of October, whereas, in the undisturbed forest soil,

the maximum C_{mic} was recorded in the month of May and the minimum was recorded in the month of August. The C_{mic} decreased with increase in depth (Fig 6). In general, the maximum C_{mic} was observed in autumn to winter season and minimum was observed during summer season. Table 30 depicts the range (depth wise) of C_{mic} in the disturbed and undisturbed forest soil.

The overall C_{mic} values ranged between 47.77 and 977.50 μg C g^{-1} dry soil in the disturbed forest soil, whereas, in case of undisturbed forest soil, it ranged between 38.64 and 983.27 μg Cg^{-1} dry soil. In the second year i.e. 2003, C_{mic} ranged between 44.24 and 742.55 μg Cg^{-1} dry soil in the disturbed forest site, whereas, in case of undisturbed soil, it was between 47.65 and 1102.01 μg Cg^{-1} dry soil (Table 30).

The one way analysis of variance (ANOVA) of C_{mic} varied significantly ($P \leq 0.05$) between the two study sites at three different depths and insignificant variation was observed at DF (0-10cm) x UDF (0-10cm), DF (10-20cm) x UDF (10-20cm) and DF (20-30cm) x UDF (20-30cm) (Table 31). Similarly, significant variation ($P \leq 0.05$) among the three different depths in both the forest soils were also observed (Table 32).

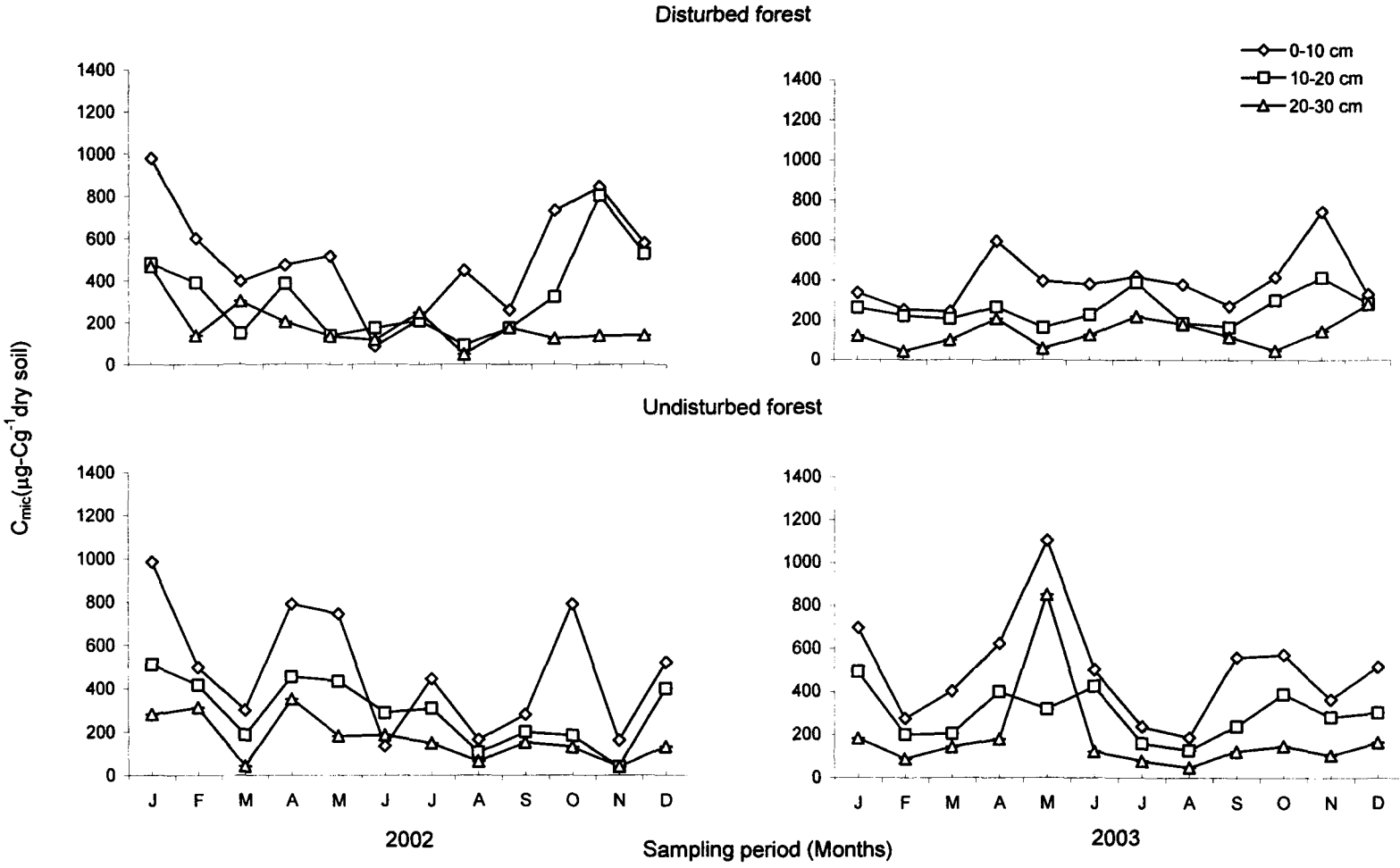

Fig. 6 Microbial biomass carbon (C_{mic}) in disturbed and undisturbed forest soils at three different depths 0-10cm,10-20cm and 20-30cm

Table 30 Values (range) of microbial biomass carbon (C_{mic}) in the disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20- 30cm during the study periods 2002-2003. Values in parentheses indicate the mean and standard error

Soil properties	Year	Depth (cm)	Disturbed Forest	Undisturbed Forest
Microbial biomass C (C_{mic})	2002	0-10	85.19-977.50 (510.06±7.18)	134.43-983.27 (483.47±20.70)
	2003	0-10	244.01- 742.55 (396.58±19.28)	186.20-1102.67 (502.69±22.75)
	2002	10-20	92.00-804.43 (320.22±22.07)	39.13-510.20 (292.96±29.62)
	2003	10-20	163.55-413.90 (257.06±17.67)	126.64- 494.01 (294.93±24.31)
	2002	20-30	47.77-465.80 (185.8±14.71)	38.64- 350.31 (167.85±12.52)
	2003	20-30	44.24- 280.13 (137.16±13.04)	47.65- 849.41 (185.40±11.07)

Table 31 One way analysis of variance (ANOVA) of the microbial biomass carbon (C_{mic}) between the disturbed forest (DF) and undisturbed forest (UDF) soils at three different depths 0-10cm, 10-20cm and 20-30cm ($P \leq 0.05$)

Soil properties	Source of variation	F-ratio	P-level
Microbial biomass C (C_{mic})	DF(0-10cm) x UDF(0-10cm)	-	-
	DF(0-10cm) x UDF(10-20cm)	28.17	$1x10^{-3}$
	DF(0-10cm) x UDF(20-30cm)	76.59	$1x10^{-3}$
	DF(10-20cm) x UDF(0-10cm)	31.68	$1x10^{-3}$
	DF(10-20cm) x UDF(10-20cm)	-	-
	DF(10-20 cm) x UDF(20-30cm)	17.26	$5.6x10^{-5}$
	DF(20-30cm) x UDF(0-10cm)	100.45	$1x10^{-3}$
	DF(20-30cm) x UDF(10-20cm)	43.40	$1x10^{-3}$
	DF(20-30cm) x UDF(20-30cm)	-	-

Note: Insignificant values are marked with '–' sign

Table 32 One way analysis of variance (ANOVA) of the microbial biomass carbon (C_{mic}) among the three different depths 0-10cm, 10-20cm and 20-30cm in the disturbed forest (DF) and undisturbed forest (UDF) soils ($P \leq 0.05$)

Soil properties	Source of variation	F-ratio	P-level
Microbial biomass C (C_{mic})	DF(0-10cm x 10-20cm)	27.57	$1x10^{-6}$
	DF(0-10cm x 20-30cm)	112.20	$1x10^{-3}$
	DF(10-20cm x 20-30cm)	33.25	$1x10^{-3}$
	DF(0-10cm x10-20cm x 20-30cm)	57.63	$1x10^{-3}$
	UDF(0-10cm x 10-20cm)	32.05	$1x10^{-3}$
	UDF(0-10cm x 20-30cm)	75.01	$1x10^{-3}$
	UDF(10-20cm x 20-30cm)	21.34	$9x10^{-6}$
	UDF(0-10 cm x 10-20cm x20-30cm)	47.73	$1x10^{-3}$

Note: Insignificant values are marked with '–' sign

Soil Enzyme Activities

Dehydrogenase activity

A monthly fluctuation in dehydrogenase activity was observed in both the forest sites during the two year study periods. Comparing both the study sites, it was observed that the activity was almost same in both the forest soils. However, the soil of the undisturbed forest showed slightly higher activity than that of disturbed forest soils.

In the first year of study period i.e. 2002, the maximum activity was observed at 0-10cm depth in the month of August and the minimum activity was observed in the month of February in both the forest soils. At 10-20cm, maximum activity was observed in the month of May in both the forest stands and the minimum activity was observed in the months of April and February in the disturbed and undisturbed forest soils respectively. At 20-30cm depth, the maximum activity was observed in the month of May in both the forest stands and the minimum activity was observed in the months of July and March in the disturbed and undisturbed forest soils respectively. In the second year of study period i.e. 2003, at depth 0-10cm, maximum dehydrogenase activity was observed in the months of October and May in the disturbed and undisturbed forest soils respectively. The minimum activity at the same depth was observed in the month of November in both the forest stands. At depth 10-20cm, the maximum activity was observed in the months of September and August in the disturbed and undisturbed forest soils respectively. The minimum activity at the same depth was observed in the months of February and November in the disturbed and the undisturbed forest soil respectively. At 20-30cm depth, the maximum activity was observed in the months of September and

October in the disturbed and the undisturbed forest soils respectively. The minimum activity was observed in the month of February in the disturbed site and in the month of November in the undisturbed forest soils. The enzyme activity decreased with increase in depth (Fig.7).

Table 33 depicts the range (depth wise) of dehydrogenase activity in both the forest stands during the two year of study periods. The overall dehydrogenase activity in the disturbed forest soil, ranged from 0.08 to 1.56 mgTPFg^{-1}dry soil $24h^{-1}$ during 2002 and in the second year i.e. 2003, the range was between 0.09 to 1.54 mgTPFg^{-1}dry soil $24h^{-1}$. In the undisturbed forest soil during 2002, range varied from 0.04 to 1.61mgTPFg^{-1}dry soil $24h^{-1}$ and in 2003, it ranged from 0.13 to 1.85 mgTPFg^{-1}dry soil $24h^{-1}$.

The one way analysis of variance (ANOVA) of dehydrogenase activity varied significantly ($P \leq 0.05$) between the two sites at the three different depths and showed insignificant variation at DF (0-10cm) x UDF (0-10cm) and DF (20-30cm) x UDF (20-30cm) (Table 34). A significant variation ($P \leq 0.05$) among the three different depths was observed among the three different depths in both the study sites (Table 35).

Urease activity

The urease activity showed less variation in the first year, throughout the study periods in both the study sites. Comparing both the study sites, it was observed that the urease activity was slightly higher in the disturbed forest soil than the undisturbed forest soil. The activity was low in the months of January to May and thereafter it increased till November in both the study sites. In the first year, in the disturbed forest soils, the months of June, July and October showed maximum urease activity at 0-10cm, 10-20cm

and 20-30cm respectively. The minimum urease activity was observed in the month of January at the three depths 0-10cm, 10-20cm and 20-30cm, whereas, in the undisturbed forest soils, the maximum urease activity was observed in the month of June at 0-10cm and 10-20cm depths. At 20-30cm depth, the maximum activity was observed in the month of August. The minimum activity was observed in the month of April at depth 0-10cm and in the month of December at depths 10-20cm and at 20-30cm. During the second year of study period i.e. 2003, in the disturbed forest soils, the maximum activity was observed in the month of June at all the three different depths. The minimum activity was observed in the month of April at 0-10cm and in the month of March at depths 10-20cm and 20-30cm. In the undisturbed forest soil, the maximum activity was observed in the month of June at 0-10cm and in the month of October at depths 10-20cm and 20-30cm. The minimum activity was observed in the month of April at depths 0-10cm and 10-20cm and in the month of December at 20-30cm (Fig. 8).

Table 33 depicts the range (depth wise) of urease activity in the disturbed and undisturbed forest soils at all the three different depths. The overall urease activity in the disturbed forest soil, during the first year, ranged from 0.14 to 1.63 mg NH_4^+-N g^{-1} dry soil $3h^{-1}$ and in the second year, it ranged between 0.05 to 1.65 mg NH_4^+-N g^{-1} dry soil $3h^{-1}$, whereas, the enzyme activity in the undisturbed forest soil ranged between 0.24 to 1.60 mg NH_4^+-N g^{-1} dry soil $3h^{-1}$ during 2002 and in the second year i.e. 2003, it ranged from 0.14 to 1.64 mg NH_4^+-N g^{-1} dry soil $3h^{-1}$. However the activity was higher at the surface layer and decreased with increase in depth.

The one way analysis of variance showed significant variation ($P<0.05$) of urease activity between the two study sites at DF (0-10cm) x UDF (20-30cm) and DF (20-30cm)

x UDF (0-10cm) (Table 34) and among the depths at UDF (0-10cm x 10-20cm) and UDF (0-10cm x 20-30cm) (Table 35).

Phosphatase activity

Similar trend of monthly variations was observed in both the study sites. Slightly higher activity was found in the soil of disturbed forest as compared to undisturbed forest soil. During the first year of study period, it was observed that the months of July and November showed maximum and minimum activity at all the three different depths in both the disturbed and undisturbed forest soils respectively. In the second year of study period, in the disturbed forest soil, the maximum and the minimum activity was observed in the months of June and December respectively at all the three different depths. In the undisturbed forest soil at 0-10cm, the maximum activity was observed in the month of June and in the month of July at 10-20cm and 20-30cm. The minimum activity was observed in the month of December at all the three different depths (Fig. 9). The phosphatase activity was found to be higher during summer than that of winter months. Phosphatase activity decreased with increase in depth.

Table 33 depicts the range (depth wise) of phosphatase activity in both the forest soils at the three different depths. The overall range of phosphatase activity during the first year, in the disturbed forest soils was between 72.91 and 1552.63 μg PNP released g^{-1} dry soil h^{-1}, whereas, in the second year i.e. 2003, it ranged between 72.61 and 1779.29 μg PNP released g^{-1} dry soil h^{-1}. In the undisturbed forest soil, it ranged from 28.03 to 1710.86 μg PNP released g^{-1} dry soil h^{-1} during the first year and in the second year, it ranged between 78.81 and 1405.58 μg PNP released g^{-1} dry soil h^{-1}.

The one way analysis of variance (ANOVA) of phosphatase activity showed significant variation ($P \leq 0.05$) between the two study sites at DF (0-10cm) x UDF (10-20cm), DF (0-10cm) x UDF (20-30cm), DF (10-20cm) x UDF (0-10cm) and DF (20-30cm) x UDF (0-10cm) (Table 34). Significant variation ($P \leq 0.05$) was also observed among the different depths of the two study sites and insignificant variation was observed at DF (10-20cm x 20-30cm) and UDF (10-20cm x 20-30cm) (Table 35).

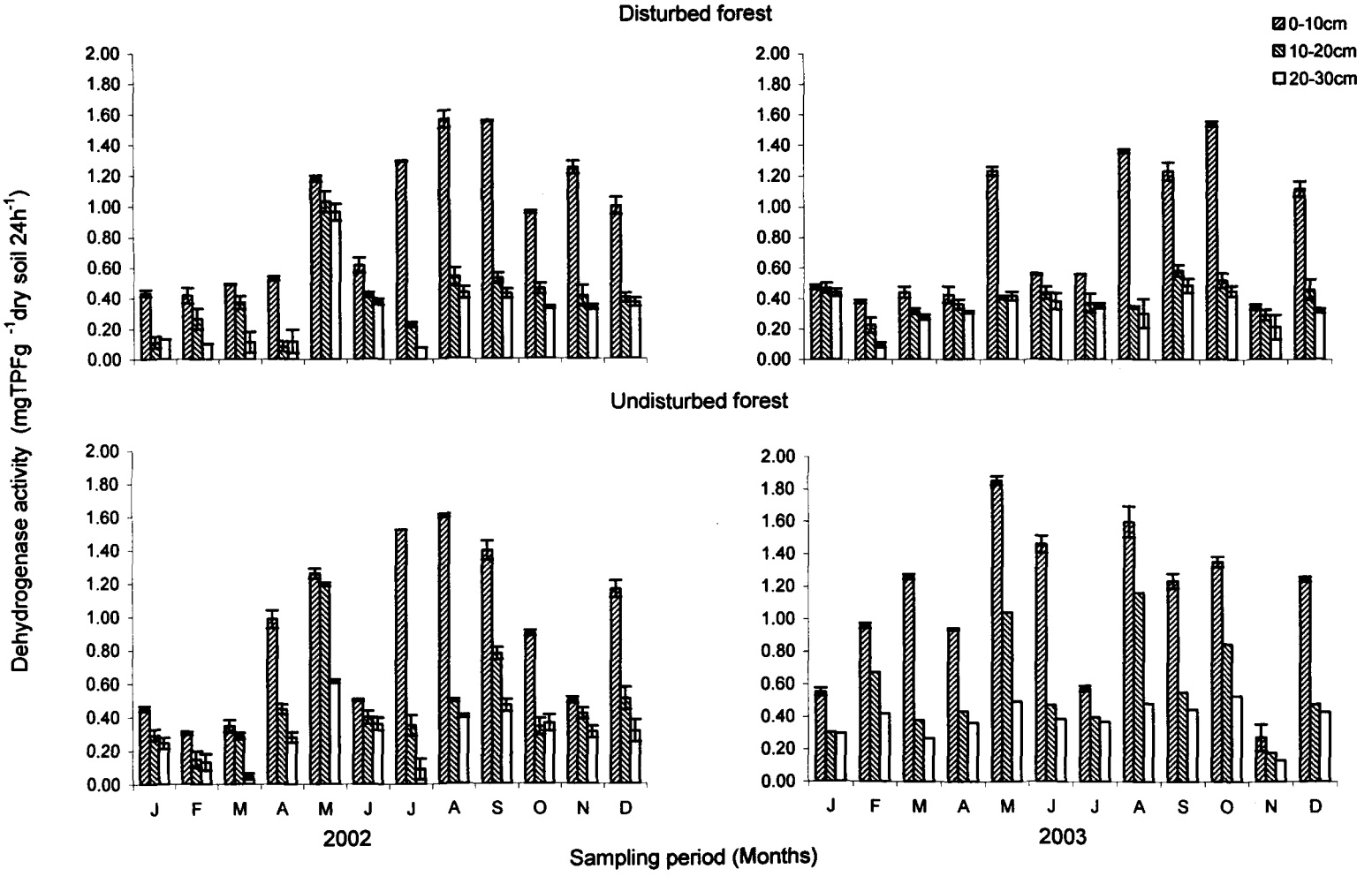

Fig. 7 Dehydrogenase activity in disturbed and undisturbed forest soils at three different depths 0-10cm,10-20cm and 20-30cm

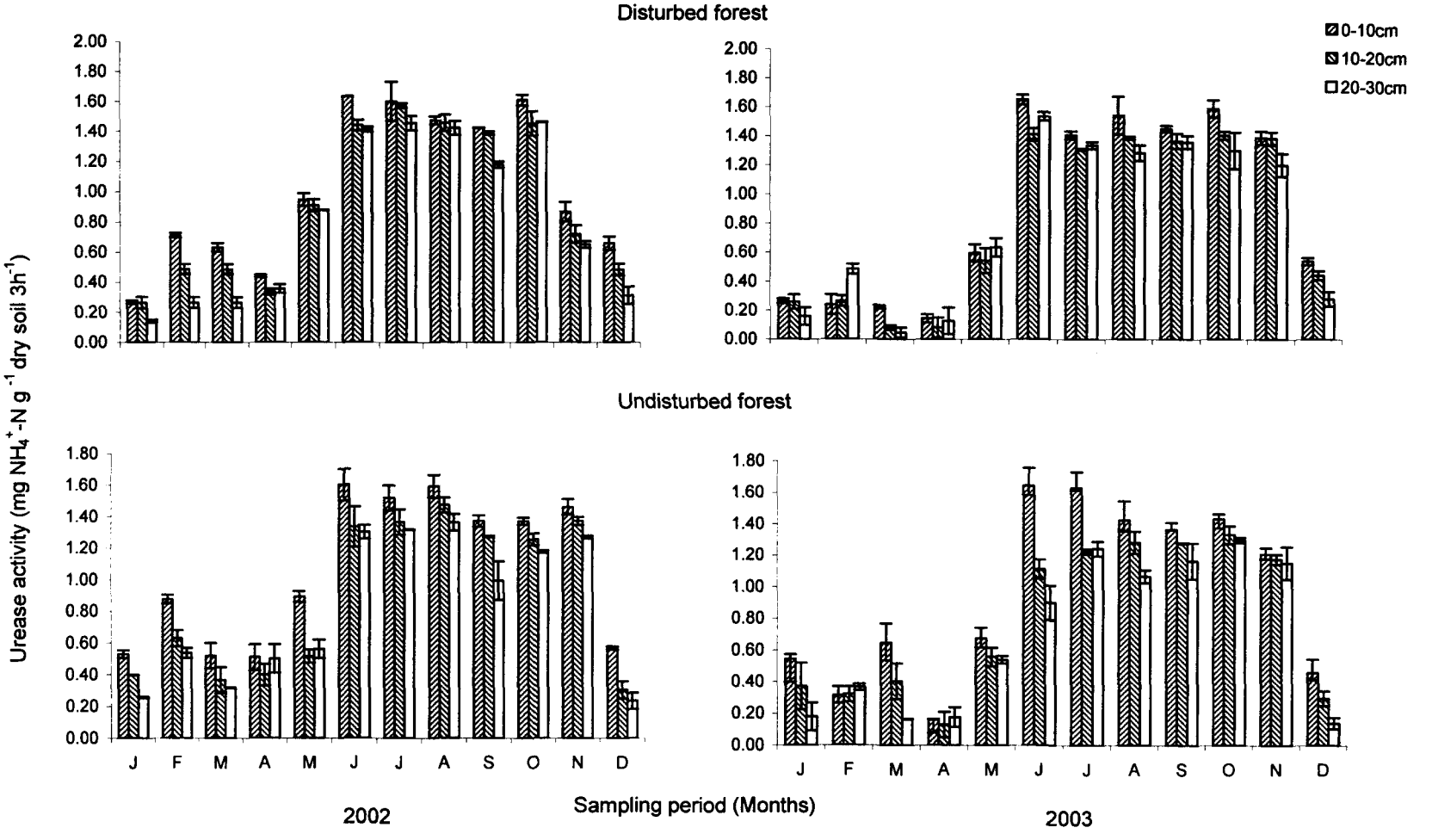

Fig. 8 Urease activity in disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20-30cm

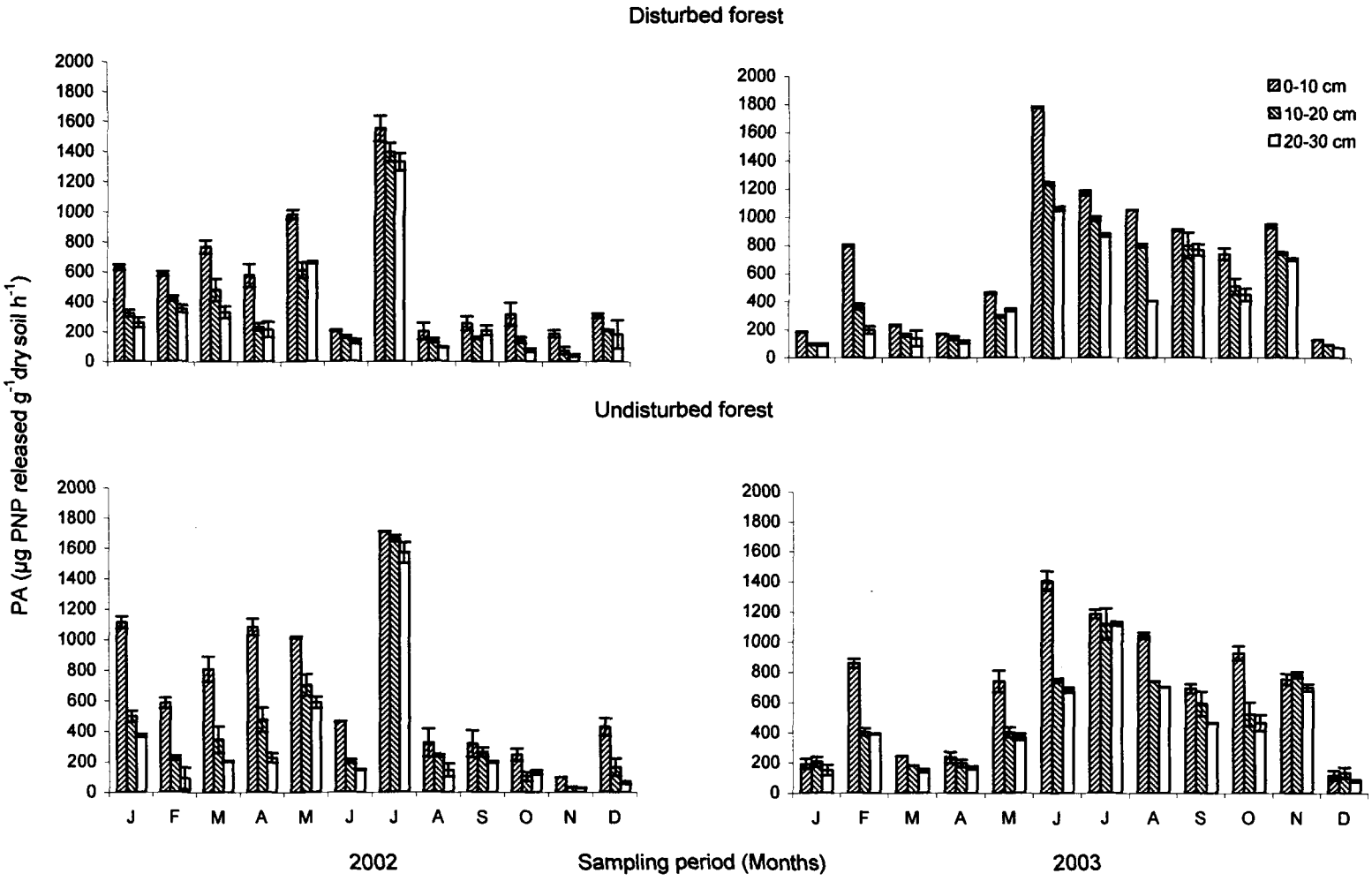

Fig. 9 Phosphatase activity in disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20-30cm

Table 33 Values (range) of biochemical characteristics in the disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20- 30cm during the study periods 2002-2003. Values in parentheses indicate the mean and standard error

Soil properties	Year	Depth (cm)	Disturbed Forest	Undisturbed Forest
Dehydrogenase	2002	0-10	0.42-1.56 (0.94±0.02)	0.31-1.61 (0.99±0.01)
	2003	0-10	0.35- 1.54 (0.81±0.01)	0.27-1.85 (1.10±0.02)
	2002	10-20	0.23- 1.03 (0.40±0.02)	0.15-1.19 (0.57±.0.01)
	2003	10-20	0. 23- 0.58 (0.39± 0.02)	0.30-1.16 (0.57±0.02)
	2002	20-30	0.08-0.96 (0.32±0.01)	0.04-0.62 (0.30±0.895)
	2003	20-30	0.09- 0.48 (0.34±0.02)	0.13-0.52 (0.38±0.02)
Urease	2002	0-10	0.44-1.63 (1.02±0.02)	0.51-1.60 (1.07±0.03)
	2003	0-10	0.23-1.65 (0.92±0.03)	0.16-1.64 (0.96± 0.04)
	2002	10-20	0.27- 1.57 (0.92±0.02)	0.31-1.47 (0.89±0.02)
	2003	10-20	0.08-1.41 (0.83±0.02)	0.29-1.33 (0.79±0.04)
	2002	20-30	0.14-1.46 (0.82±0.02)	0.24-1.36 (0.82±0.02)
	2003	20-30	0.05-1.53 (0.81±0.04)	0.14-1.29 (0.69±0.03)
Phosphatase	2002	0-10	187.44-1552.63 (547.16±23.96)	99.57-1710.86 (682.20±24.02)
	2003	0-10	128.76-1779.29 (712.74 ± 5.33)	116.85-1405.58 (699.67±20.97)
	2002	10-20	72.91-1394.75 (362.79 ± 16.17)	33.73-1665.31 (409.94±21.54)
	2003	10-20	89.62-1240.38 (518.57±12.42)	137.28-1120.14 (503.48±21.71)
	2002	20-30	77.48-1329.76 (324.45± 14.37)	28.03-1568.69 (315.17±14.69)
	2003	20-30	72.61-1063.65 (435.00±14.16)	78.81-1122.18 (453.87±9.86)

Table 34 One way analysis of variance (ANOVA) of the biochemical characteristics between the disturbed forest (DF) and undisturbed forest (UDF) soils at three different depths 0-10cm, 10-20cm and 20-30cm (P≤0.05)

Soil properties	Source of variation	F-ratio	P-level
Dehydrogenase	DF(0-10cm) x UDF(0-10cm)	-	-
	DF(0-10cm) x UDF(10-20cm)	33.29	$1x10^{-3}$
	DF(0-10cm) x UDF(20-30cm)	98.73	$1x10^{-3}$
	DF(10-20cm) x UDF(0-10cm)	105.50	$1x10^{-3}$
	DF(10-20cm) x UDF(10-20cm)	9.24	$2.8x10^{-3}$
	DF(10-20 cm) x UDF(20-30cm)	4.67	$3.2x10^{-2}$
	DF(20-30cm) x UDF(0-10cm)	132.67	$1x10^{-3}$
	DF(20-30cm) x UDF(10-20cm)	24.04	$3x10^{-6}$
	DF(20-30cm) x UDF(20-30cm)	-	-
Urease	DF(0-10cm) x UDF(0-10cm)	-	-
	DF(0-10cm) x UDF(10-20cm)	-	-
	DF(0-10cm) x UDF(20-30cm)	6.29	$1.3x10^{-2}$
	DF(10-20cm) x UDF(0-10cm)	-	-
	DF(10-20cm) x UDF(10-20cm)	-	-
	DF(10-20 cm) x UDF(20-30cm)	-	-
	DF(20-30cm) x UDF(0-10cm)	5.33	$2.2x10^{-2}$
	DF(20-30cm) x UDF(10-20cm)	-	-
	DF(20-30cm) x UDF(20-30cm)	-	-
Phosphatase	DF(0-10cm) x UDF(0-10cm)	-	-
	DF(0-10cm) x UDF(10-20cm)	6.92	$9.4x10^{-3}$
	DF(0-10cm) x UDF(20-30cm)	11.48	$9.1x10^{-4}$
	DF(10-20cm) x UDF(0-10cm)	10.33	$1.6x10^{-3}$
	DF(10-20cm) x UDF(10-20cm)	-	-
	DF(10-20cm) x UDF(20-30cm)	-	-
	DF(20-30cm) x UDF(0-10cm)	17.73	$4.5x10^{-5}$
	DF(20-30cm) x UDF(10-20cm)	-	-
	DF(20-30cm) x UDF(20-30cm)	-	-

Note: Insignificant values are marked with '–' sign

Table 35 One way analysis of variance (ANOVA) of the biochemical characteristics among the three different depths 0-10cm, 10-20cm and 20-30cm in the disturbed forest (DF) and undisturbed forest (UDF) soils (P≤0.05)

Soil properties	Source of variation	F-ratio	P-level
Dehydrogenase	DF(0-10cm x 10-20cm)	72.84	$1x10^{-3}$
	DF(0-10cm x 20-30cm)	97.44	$1x10^{-3}$
	DF(10-20cm x 20-30cm)	5.84	$1.6x10^{-2}$
	DF(0-10cm x10-20 cm x 20-30cm)	74.35	$1x10^{-3}$
	UDF(0-10cm x 10-20cm)	57.17	$1x10^{-3}$
	UDF(0-10cm x 20-30cm)	134.76	$1x10^{-3}$
	UDF(10-20cm x 20-30cm)	23.42	$3x10^{-6}$
	UDF(0-10cm x 10-20cm x20-30cm)	80.85	$1x10^{-3}$
Urease	DF(0-10cm x 10-20cm)	-	-
	DF(0-10cm x 20-30cm)	-	-
	DF(10-20cm x 20-30cm)	-	-
	DF(0-10cm x10-20cm x 20-30cm)	-	-
	UDF(0-10cm x 10-20cm)	4.65	$3.2x10^{-1}$
	UDF(0-10cm x 20-30cm)	10.38	$1.6x10^{-3}$
	UDF(10-20cm x 20-30cm)	-	-
	UDF(0-10cm x 10-20cm x20-30cm)	5.41	$5x10^{-3}$
Phosphatase	DF(0-10cm x 10-20cm)	6.18	$1.4x10^{-1}$
	DF(0-10cm x 20-30cm)	11.40	$9.4x10^{-4}$
	DF(10-20cm x 20-30cm)	-	-
	DF(0-10cm x10-20cm x 20-30cm)	6.43	$1.9x10^{-3}$
	UDF(0-10cm x 10-20cm)	11.75	$7.9x10^{-4}$
	UDF(0-10cm x 20-30cm)	17.86	$4.2x10^{-5}$
	UDF(10-20cm x 20-30cm)	-	-
	UDF(0-10cm x 10-20cm x20-30cm)	10.57	$4.2x10^{-5}$

Note: Insignificant values are marked with '–' sign

Soil Physico-Chemical Characteristics

Soil temperature

A similar trend of monthly variations was observed in the soils of the disturbed and undisturbed forests. Highest soil temperature was recorded at surface soil as compared to subsurface layers. Undisturbed forest soil had slightly higher temperature than that of disturbed forest soil. In the undisturbed forest soil, the months of June, July and August recorded highest soil temperature. In the first year, at 0-10cm and 10-20cm, the highest temperature was recorded in the month of July and minimum was recorded in the month of January in both the study sites. At 20-30cm, the maximum soil temperature was recorded in the month of September and the minimum was recorded in the month of January in the disturbed forest. In the undisturbed forest soil, the maximum temperature was recorded in the month of July and minimum was recorded in the month of January at 20-30cm. In the second year of study period, i.e. 2003, in the disturbed forest soil, the maximum soil temperature at 0-10cm was observed in the month of July and minimum was observed in the month of January, whereas at 10-20cm soil depth, maximum soil temperature was recorded in the month of June and minimum was observed in the month of January. At 20-30cm soil depth, the maximum and minimum soil temperatures were recorded in the months of September and January respectively. Whereas in the undisturbed forest soil, at 0-10cm soil depth, the maximum and minimum temperatures were recorded in the month of July and December respectively. At 10-20cm soil depth, maximum and minimum temperatures were recorded in the month of August and

December respectively. At 20-30cm, the maximum and minimum soil temperatures were recorded in the months of July and March respectively (Fig. 10).

Table 36 depicts the range (depth wise) of soil temperatures in the two forests at the three different depths. In the first year, in the disturbed forest, the overall soil temperature ranged from 10.10^0 C to 18.50^0 C, whereas, in the second year, it ranged between 10.00^0 C and 18.10^0 C. In the first year, in the undisturbed forest, the soil temperature ranged between10.10^0 C to 20.20^0 C, whereas, in the second year, it ranged between 10.00^0 C to19.80^0 C.

Soil moisture content

Less variation was observed in the soil moisture content in both the study sites. However, higher moisture content was found in the disturbed forest soils than in the undisturbed forest soils. In the disturbed forest soil, the maximum and minimum moisture content at 0-10cm was recorded in the months of September and February respectively. At 10-20cm depth, the maximum and minimum moisture content was in the months of June and January, whereas at 20-30cm, the months of May and November recorded maximum and minimum moisture content respectively. In the second year of study period, it was observed that the maximum and minimum moisture content was in the months of June and April respectively at 0-10cm. At 10-20cm, the months of June and March recorded maximum and minimum moisture content, whereas at depth 20-30cm, the months of May and March recorded maximum and minimum moisture content respectively (Fig.11).

In the undisturbed forest during the first year of study period, at 0-10cm the maximum and minimum moisture content was recorded in the months of May and March and in the months of May and February at 10-20cm whereas at 20-30cm, the maximum and minimum moisture content was found in the months of May and January respectively. In the second year of study period, the maximum moisture content was observed in the month of August and the minimum was found in the month of January. The maximum moisture content at 10-20cm was found in the month of April and the minimum in the month of January. At 20-30cm the maximum and minimum moisture content was found in the months of August and January respectively (Fig.11).

Table 36 depicts the range (depth wise) of moisture content in the disturbed and undisturbed forest soils at the three different depths. In the disturbed forest soils, during the first year, the overall moisture content ranged between 27.93 to 39.70 % and between 19.10 to 43.63 % in the second year. In the undisturbed forest soil, it ranged from 16.47 to 39.20 % during the first year and 14.27 % to 39.40 % in the second year.

pH

The pH of the soil showed very less variations in all the months throughout the study periods in both the forest soils. The pH was acidic in both the study sites. Comparing both the study sites it was observed that the soil of the undisturbed forest was more acidic than that of the disturbed forest (Fig. 12).

Table 36 depicts the range (depth wise) of pH in the disturbed and undisturbed forest soils at the three different depths. In the first year, the overall range of soil pH in the disturbed forest ranged from 5.58 to 6.53 and from 5.34 to 6.86 in the second year. In

the undisturbed forest the pH ranged from 5.19 to 6.52 in the first year and from 5.09 to 6.27 in the second year.

Soil organic carbon

The soil organic carbon was found to be higher in the disturbed forest than that in undisturbed forest. The surface soil contained highest organic carbon followed by sub-surface layers. The maximum organic carbon content in the disturbed forest was found in the month of August and the minimum was found in the month of September at all the three different depths i.e. 0-10cm, 10-20cm and 20-30cm. In the second year of study period at 0-10cm, it was observed that the month of January contained maximum organic carbon and July contained minimum organic carbon. At 10-20cm, the months of September and July showed maximum and minimum organic carbon content respectively. At 20-30cm the maximum and minimum organic carbon content was found in the months of January and July respectively (Fig. 13).

In the undisturbed forest soil, the maximum organic carbon content was found in the month of June and minimum in the month of February at 0-10cm. At 10-20cm and 20-30cm depths, the maximum organic carbon was found in the month of May and the minimum was found in the month of February. In the second year of study period, at 0-10cm depth, the maximum and minimum organic carbon was found in the months of August and February respectively. At 10-20cm, the maximum and minimum organic carbon content was found in the months of August and February respectively. At 20-30cm soil layer, the maximum and minimum organic carbon was found in the months of June and February respectively (Fig. 13).

Table 36 depicts the range (depth wise) of soil organic carbon in both the forest stands at the three different depths. In the first year, in the disturbed forest, the organic carbon content ranged between 1.92 to 6.13 %. During the second year, it ranged between 1.66 to 7.08 %. The organic carbon in the undisturbed forest soil ranged from 0.44 to 5.68 % in the first year and 1.40 to 6.63 % in the second year. The organic carbon decreased with increase in depth.

Total nitrogen

A monthly fluctuation was observed during the two year of study periods in both the forest soils. It was observed that the soil of disturbed forest contained more nitrogen as compared to that of undisturbed forest. At 0-10cm and 10-20cm depths in the disturbed forest soil, the maximum nitrogen was found in the month of May and the minimum in the month of August, whereas, at the third depth i.e., 20-30cm, the maximum and minimum nitrogen content was recorded in the months of April and August respectively. In the second year, the month of May showed maximum and November recorded minimum nitrogen content at 0-10cm. At 10-20cm, the maximum nitrogen content was found in the month of January and the minimum was found in the month of July, whereas, at 20-30cm, the maximum and minimum was found in the months of May and July respectively (Fig. 14).

In the first year of study period, in the undisturbed forest soil, the months of May and August also showed maximum and minimum nitrogen content at 0-10cm and 10-20cm respectively, whereas, at 20-30cm, the maximum and minimum nitrogen content was found in the months of July and August respectively. In the second year, maximum nitrogen content was found in the months of May and the minimum was recorded in the

month of December at 0-10cm. The months of April and February showed maximum and minimum nitrogen content at 10-20cm, whereas at 20-30cm, the maximum nitrogen content was found in the month of May and the minimum was recorded in the month of February (Fig.14).

Table 36 depicts the range (depth wise) of nitrogen in the two forest stands at the three different depths. The overall nitrogen content ranged from 0.34 % to 2.52 % during the first year in the disturbed forest and in the second year, the nitrogen content ranged from 0.29 % to 2.30 %. In the undisturbed forest soil, it ranged from 0.05 % to 1.93 % during the first year. During second year, the nitrogen content ranged from 0.26 % to 1.73 %. It was observed that in most cases, the nitrogen content decreased with increased in depth.

Available phosphorus

In the first year of study period, monthly fluctuations were observed throughout in both the study sites. In the disturbed forest soil, at 0-10cm, phosphorus was maximum in the month of February and the minimum in the month December. At 10-20cm depth, the maximum and minimum phosphorus was recorded in the months of February and May respectively, whereas, at 20-30cm depth, the maximum and minimum phosphorus was in the months of November and June respectively. In the undisturbed forest soil, in the same year at 0-10cm, the maximum phosphorus was recorded in the month of February and the minimum in May. At 10-20cm, the phosphorus was maximum in the month of January and minimum in the month of May, whereas, at the 20-30cm, the maximum and minimum phosphorus was found in the months of October and May respectively (Fig.15).

In the second year of study period, it was observed that phosphorus content increased in the months of January and February and thereafter decreased. In the disturbed forest soil it was observed that the month of February at 0-10cm contained maximum phosphorus and the month of May showed minimum. At 10-20cm and 20-30cm depths, the months of January and May showed maximum and minimum phosphorus content respectively (Fig. 15).

In the undisturbed forest soil, same trend was observed where the months of January and February showed maximum phosphorus at all the three different depths and the minimum phosphorus content was found in the month of May at all the three different depths (Fig. 15).

Table 36 showed the range (depth wise) of available phosphorus in both the forest stands at the three different depths. The overall range of phosphorus in the disturbed forest soil was between 2.33 and 51.39 $\mu g g^{-1}$dry soil in the first year. In the second year the range was between 1.99 and 57.28 $\mu g g^{-1}$dry soil. In the undisturbed forest soil the range was from 3.99 to 59.94 $\mu g g^{-1}$dry soil. In the second year, it was between 2.99 to 63.60 $\mu g g^{-1}$dry soil.

Comparing both the study sites it was observed that the undisturbed forest soils contained more phosphorus as compared to that of the disturbed forest soils. The amount of phosphorus decreased with increased in depth.

Exchangeable potassium

In the first year of study period, it was observed that the variation was very less in both the study sites at almost all the three different depths. However, in the first year in

the disturbed forest, at 0-10cm, the maximum was found in the months of May and July and the minimum in the month of September. At 10-20cm, the maximum potassium content was observed in the month of May and the minimum in the month of April, whereas at the third depth i.e. 20-30cm, the maximum and minimum potassium content was recorded in the months of May and November respectively (Fig. 16).

In the undisturbed forest, during the first year of study period at 0-10cm the maximum and minimum potassium content was found in the months of May and November respectively. At 10-20cm and 20-30cm, the maximum and minimum was found in the months of September and November respectively (Fig. 16).

In the second year of study period, in the disturbed forest, it was observed that the potassium content increased from the months of February to July and thereafter a fall was observed. At 0-10cm, the maximum potassium content was recorded in the month of May and the minimum was found in the month of December. At 10-20cm, the maximum and minimum potassium content was found in the months of July and December respectively. At 20-30cm, the maximum and minimum was recorded in the months of May and December respectively. In the undisturbed forest, similar trend of monthly variation was observed, where at 0-10cm, the maximum and minimum was recorded in the months of May and December respectively. At 10-20cm, the maximum and minimum was observed in the months of March and December respectively. At 20-30cm, the months of April and December showed maximum and minimum potassium content respectively (Fig. 16). At depth 20-30cm, minimum potassium was found in the soil of both the study sites whereas maximum potassium was recorded between March to July at different depths in both the study sites.

maximum potassium was recorded between March to July at different depths in both the study sites.

Table 36 showed the range of the potassium at the three different depths in both the study sites. The potassium decreased with increase in depth. In the disturbed forest soil, the overall range of potassium was 1.00 % to 4.63 % in the first year. In the second year, the range was from 1.59 % to 11.25 %. In the undisturbed forest the range varied from 0.63 % to 3.38 % in the first year. In the second year, the range varied from 1.25 % to 10.04 %.

Comparing both the study sites, it was observed that the potassium content was found to be higher in the soil of the disturbed forest as compared to that of the undisturbed forest soil.

The correlation coefficient of the various biological, biochemical and physico-chemical characteristics of the two forest soils was analyzed. It was observed that in the disturbed forest soil, the fungal population showed a positive significant correlation with C_{mic} ($r= 0.22$, $P\leq 0.001$), dehydrogenase activity ($r=0.45$, $P\leq 0.001$), urease activity ($r=0.39$, $P\leq 0.001$), phosphatase activity ($r=0.34$, $P\leq 0.001$), soil temperature ($r=0.38$, $P\leq 0.001$), moisture content ($r=0.14$, $P\leq 0.05$), organic carbon ($r= 0.25$, $P\leq 0.001$) and potassium ($r=0.19$, $P\leq 0.001$). It showed a negative correlation with pH ($r= -0.17$, $P\leq 0.01$), whereas, the bacterial population was positively significant with C_{mic} ($r=0.29$, $P\leq 0.001$), dehydrogenase activity ($r=0.20$, $P\leq 0.001$), moisture content ($r=0.14$, $P\leq 0.02$), total nitrogen ($r=0.23$, $P\leq 0.001$), available phosphorus ($r=0.43$, $P\leq 0.001$) and negatively correlated with phosphatase activity ($r=-0.22$, $P\leq 0.001$), and potassium ($r=-0.22$, $P\leq 0.001$) (Table 37). In the undisturbed forest soil, the fungal population was positively

moisture content (r=0.38, P≤ 0.001), organic carbon (r= 0.44, P≤ 0.001), total nitrogen (r= 0.23, P≤ 0.001, potassium (r= 0.28, P≤ 0.001) and negatively correlated with pH (r= -0.17, P≤ 0.01). Whereas the bacterial population was found to be positively correlated with C_{mic} (r=0.15, P≤ 0.05), total nitrogen (r=0.29, P≤ 0.001), available phosphorus (r=0.29, P≤ 0.001) (Table 38).

C_{mic} was positive significantly correlated with dehydrogenase activity (r=0.23, P≤ 0.001), organic carbon (r=0.15, P≤ 0.05), total nitrogen (r=0.29, P≤ 0.001), available phosphorus (r=0.24, P≤ 0.001) and negatively correlated with soil temperature (r=-0.17, P≤ 0.05) in the disturbed forest soil (Table 37), whereas, in the undisturbed forest soil, C_{mic} showed a positive correlation with dehydrogenase activity (r=0.42, P≤ 0.001), phosphatase activity (r=0.19, P≤ 0.001), moisture content (r=0.25, P≤ 0.001), organic carbon (r=0.44, P≤ 0.001), total nitrogen (r=0.59, P≤ 0.001), available phosphorus (r=0.59, P≤ 0.05), and negatively correlated with urease activity (r=-0.19, P≤ 0.001) (Table 38).

Dehydrogenase activity showed a significant positive correlation with urease activity (r=0.36, P≤ 0.001), phosphatase activity (r=0.14, P≤ 0.05), soil temperature (r=0.36, P≤ 0.001), moisture content (r=0.37, P≤ 0.001), organic carbon (r=0.54, P≤ 0.001) and total nitrogen (r=0.42, P≤ 0.001) in the disturbed forest soil (Table 37). In the undisturbed forest soil, dehydrogenase activity showed a positive significant correlation with urease activity (r=0.24, P≤ 0.001), phosphatase activity (r=0.64, P≤ 0.001), soil temperature (r=0.28, P≤ 0.001), moisture content (r= 0.58, P≤ 0.001), organic carbon (r= 0.64, P≤ 0.001), total nitrogen (r=0.47, P≤ 0.001) and potassium (r=0.28, P≤ 0.001) (Table 38).

In the disturbed forest soil, urease activity showed a positive significant correlation with phosphatase activity (r=0.49, P≤ 0.001), soil temperature (r=0.64, P≤ 0.001), moisture content (r=0.58, P≤ 0.001), organic carbon (r=0.24, P≤ 0.001) (Table 37), whereas, in the undisturbed forest soil it was observed that the urease activity showed a positive significant correlation with phosphatase activity (r=0.36, P≤ 0.001), soil temperature (r=0.65, P≤ 0.001), and negatively correlated with C_{mic} (r=-0.19, P≤ 0.01) and pH (r=-0.17, P≤ 0.01) (Table 38).

In the disturbed forest, phosphatase activity showed significantly positive correlation with soil temperature (r=0.42, P≤ 0.001), moisture content (r=0.32, P≤ 0.001), organic carbon (r=0.15, P≤ 0.05) and potassium (r=0.28, P≤ 0.001) and negatively correlated with bacterial population (r=-0.22, P≤ 0.001) (Table 37). In the undisturbed forest, phosphatase activity showed a significant positive correlation with soil temperature (r=0.50, P≤ 0.001), moisture content (r=0.24, P≤ 0.001), organic carbon (r=0.26, P≤ 0.001), total nitrogen (r=0.45, P≤ 0.001) and potassium (r=0.23, P≤ 0.001) (Table 38).

The moisture content in the disturbed forest showed a significant positive correlation with pH (r=0.19, P≤ 0.001), organic carbon (r=0.28, P≤ 0.001), total nitrogen (r=0.19, P≤ 0.01) and negatively correlated with soil temperature (r=-0.42, P≤ 0.001) (Table 37), whereas in the undisturbed forest soil moisture content showed positive correlation with organic carbon (r=0.59, P≤ 0.001), total nitrogen (r=0.46, P≤ 0.001), potassium (r=0.36, P≤ 0.001) and negatively correlated with soil temperature (r=-0.25, P≤ 0.001) and available phosphorus (r=-0.39, P≤ 0.001) (Table 38).

The pH showed a negative correlation with potassium (r=-0.26, P≤ 0.001) in the disturbed forest soil (Table 37), whereas, in the undisturbed forest it showed negative correlation with soil temperature (r=-0.26, P≤ 0.001), available phosphorus (r=-0.17, P≤ 0.01) and potassium (r=-0.40, P≤ 0.001) (Table 38).

The organic carbon showed a positive significant correlation with total nitrogen (r=0.32, P≤ 0.001) in the disturbed forest (Table 37) and with total nitrogen (r=0.54, P≤ 0.001) and potassium (r=-0.29, P≤ 0.001) in the undisturbed forest soil (Table 38).

Total nitrogen showed a significant positive correlation with soil temperature at (r=0.14, P≤ 0.05) in the disturbed forest soil (Table 37) and with soil temperature (r=0.11, P≤ 0.01), potassium (r=0.14, P≤ 0.05) in the undisturbed forest soil (Table 38).

Available phosphorus showed a negative correlation with potassium (r=-0.17, P≤ 0.01) and soil temperature (r=-0.25, P≤ 0.001) in the disturbed forest soil (Table 37) and negatively correlated with soil temperature (r=-0.17, P≤ 0.01), moisture content (r=-0.39, P≤ 0.001), pH (r=-0.17, P≤ 0.01) in the undisturbed forest soil (Table 38).

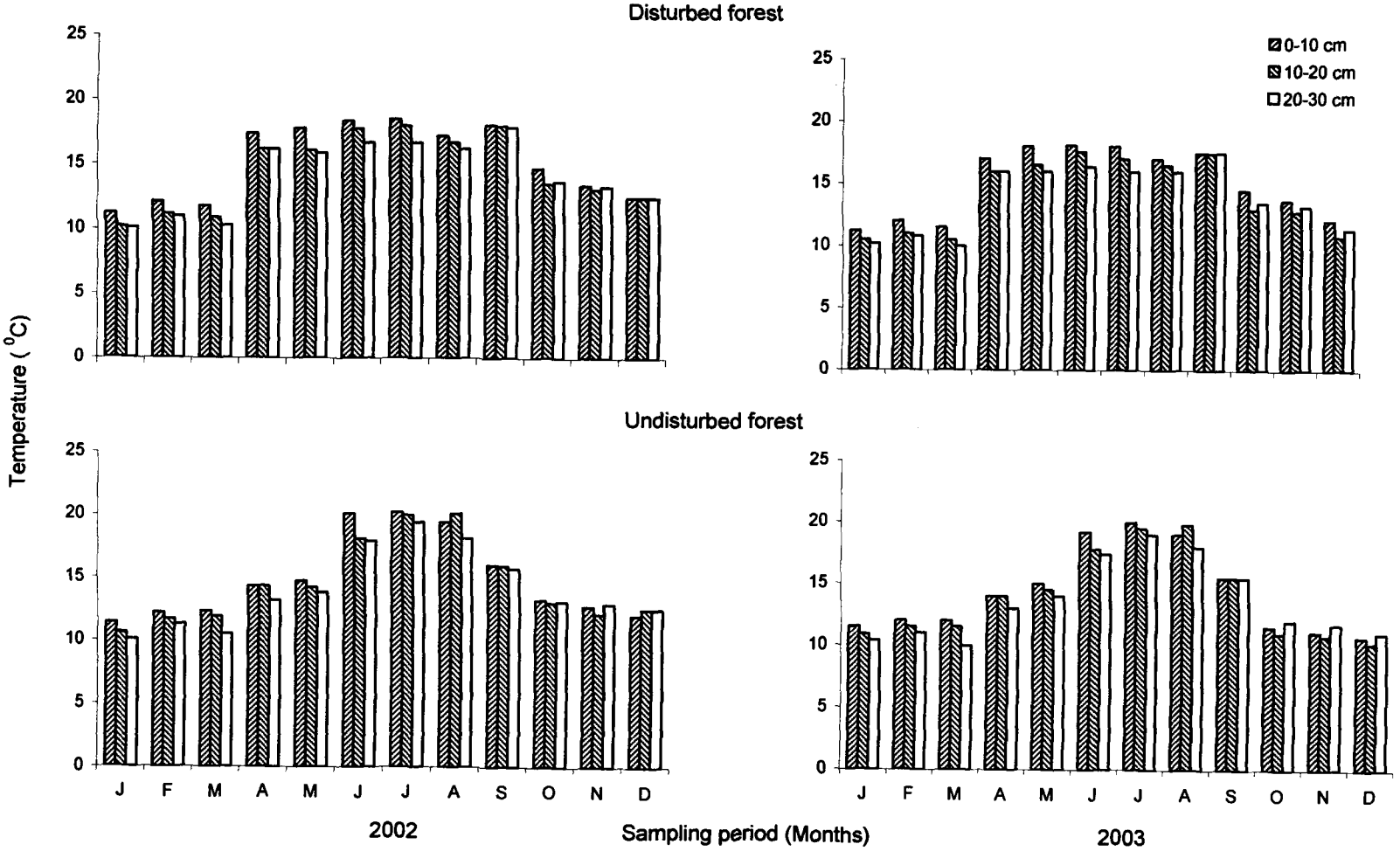

Fig. 10 Temperature in disturbed and undisturbed forest soils at three different depths 0-10cm,10-20cm and 20-30cm

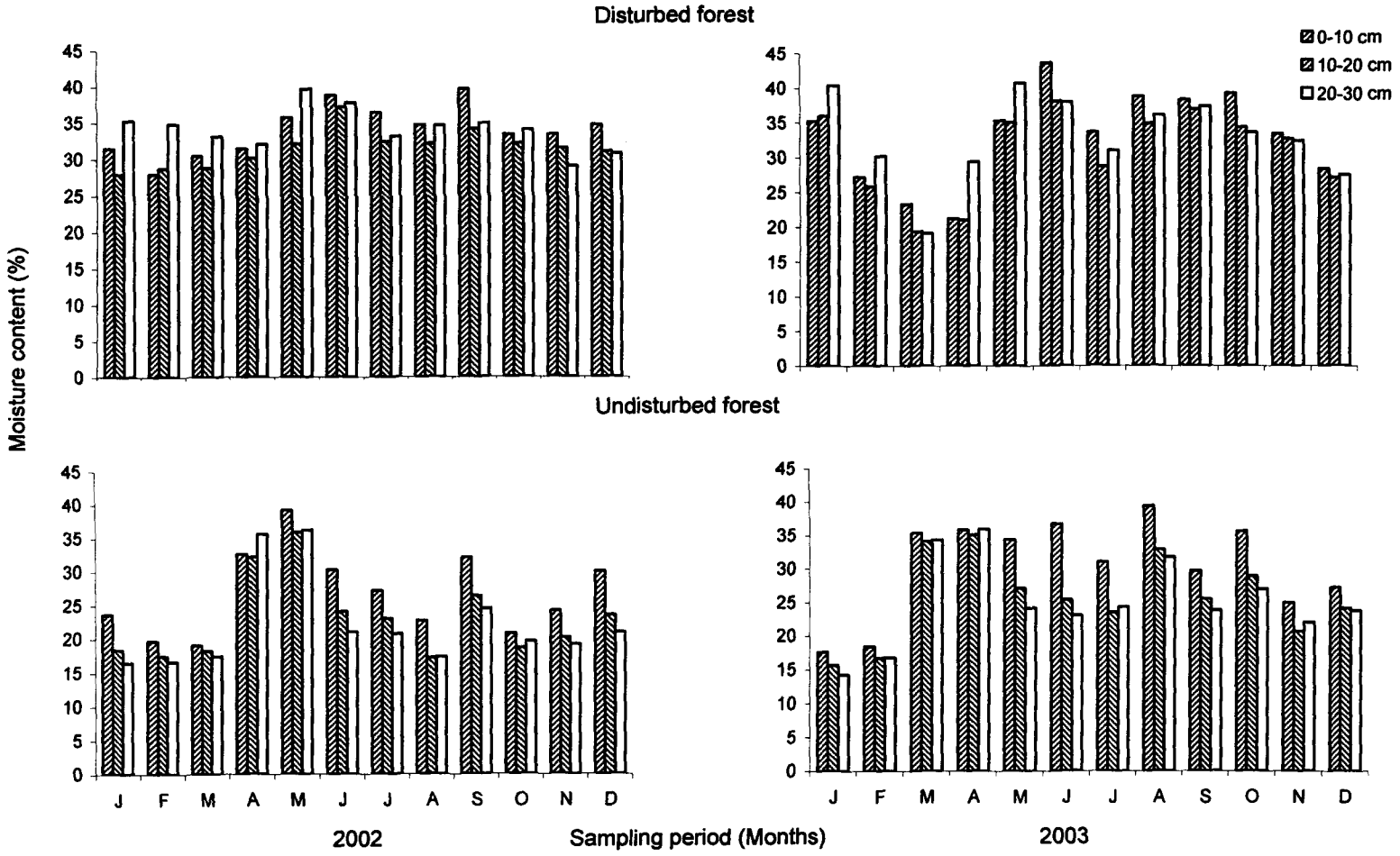

Fig. 11 Moisture content in disturbed and undisturbed forest soils at three different depths 0-10cm,10-20cm and 20-30cm

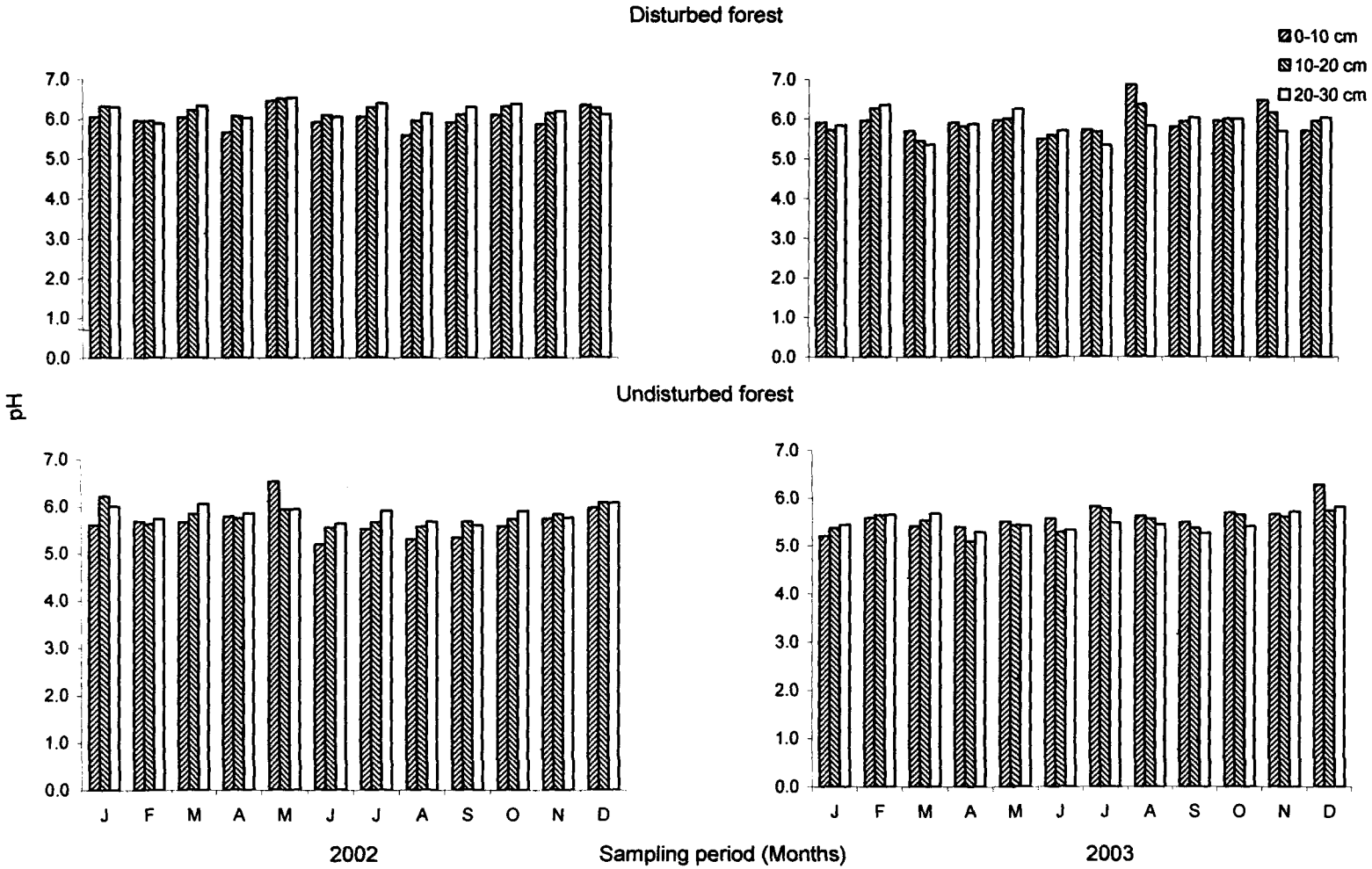

Fig. 12 pH in disturbed an d undisturbed forest soil at three different depths 0-10cm, 10-20cm and 20-30cm

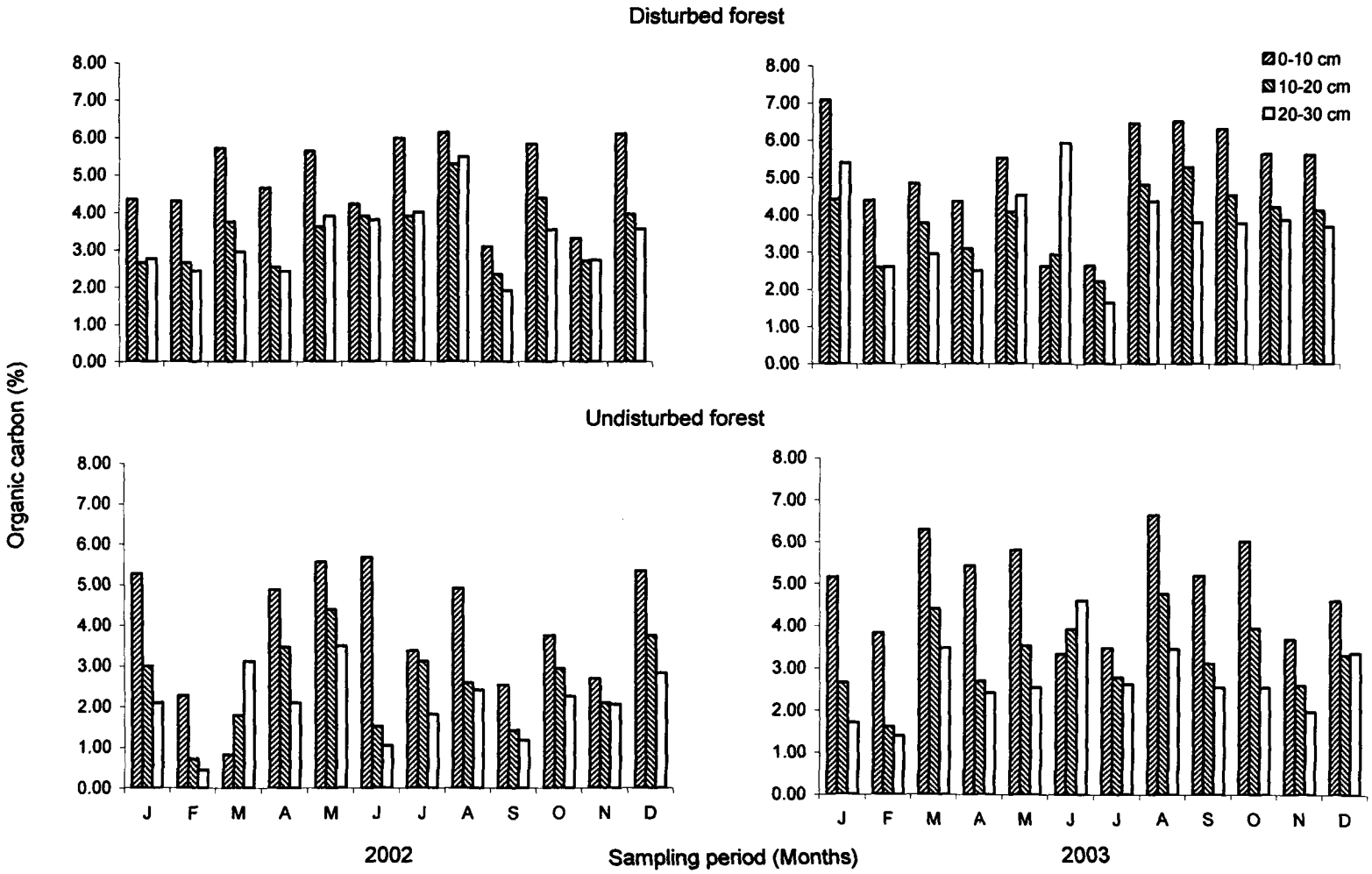

Fig 13 Organic carbon in disturbed and undisturbed forest soils at three different depths 0-10cm,10-20cm and 20-30cm

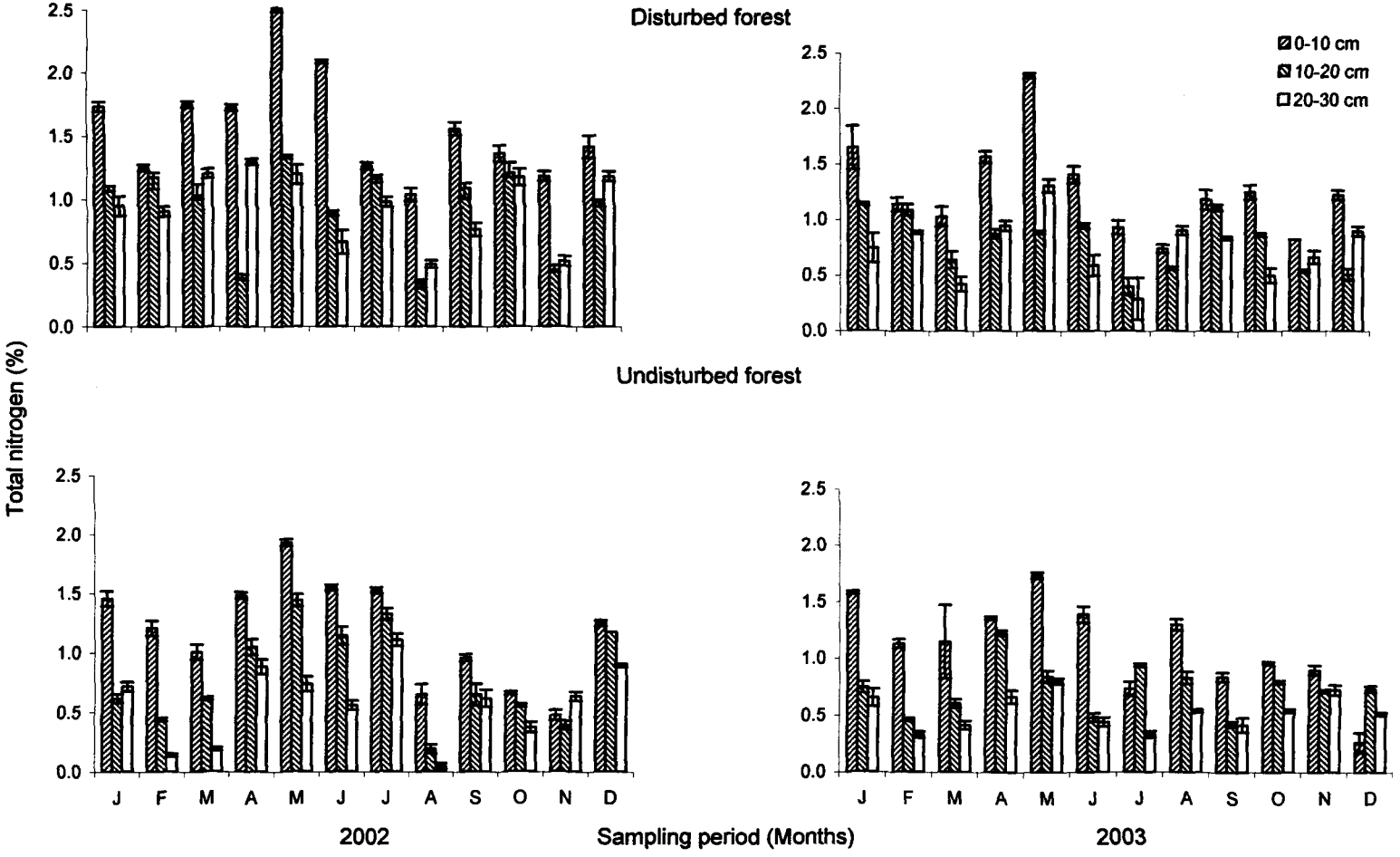

Fig. 14 Total nitrogen in disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20-30cm

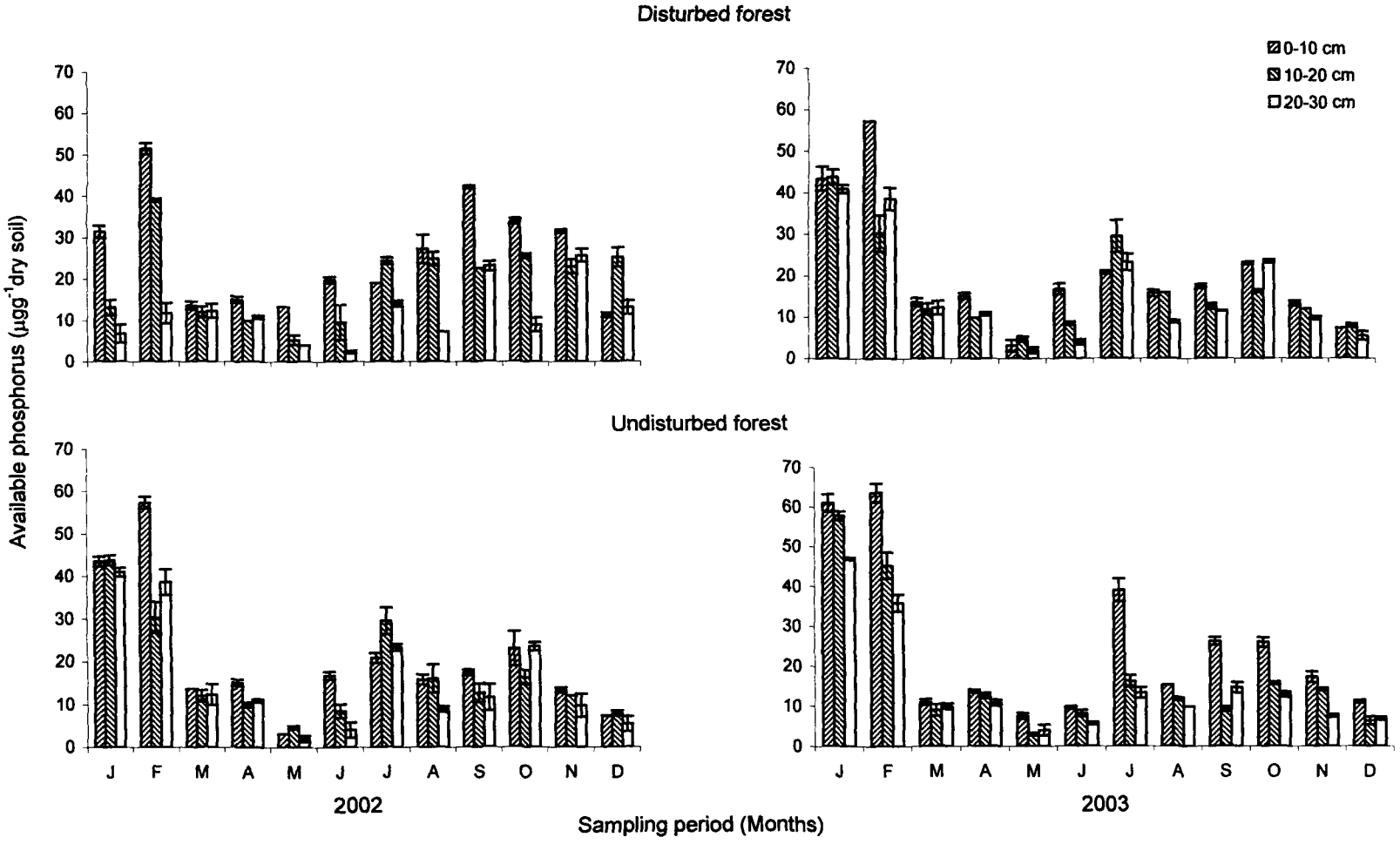

Fig. 15 Available phosphorus in disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20-30cm

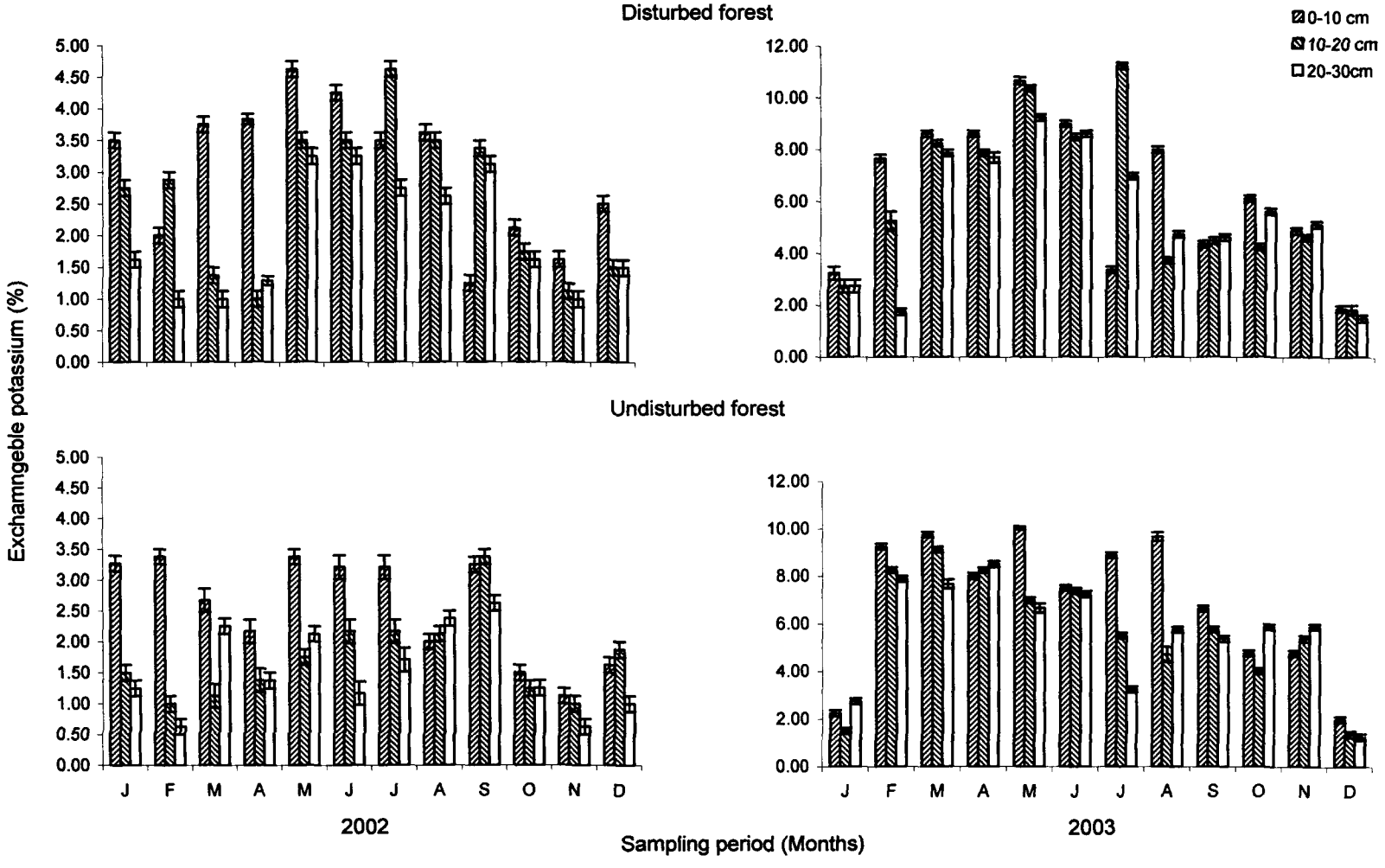

Fig. 16 Exchangeable potassium in disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20-30cm

Table 36 Values (range) of physico-chemical characteristics of disturbed and undisturbed forest soils at three different depths 0-10cm, 10-20cm and 20-30cm during the study periods of 2002 and 2003. Values in parentheses indicate the mean and standard error

Study sites	Year	Depth (cm)	Soil Temperature (C)	Moisture Content (%)	pH	Organic Carbon (%)	Total Nitrogen (%)	Available Phosphorus ($\mu g g^{-1}$ dry soil)	Exchangeable Potassium (%)
Disturbed forest	2002	0-10	11.20-18.50 (15.26±0.84)	27.97-39.63 (34.01±0.42)	5.58-6.46 (5.99±0.04)	3.09-6.13 (4.94±0.09)	1.04-2.52 (1.45±0.03)	11.29-51.39 (30.64±0.52)	1.25-4.63 (3.21±0.07)
	2003	0-10	11.20-18.10 (15.05±0.82)	21.20-43.63 (33.13±0.42)	5.49-6.86 (5.96±0.10)	2.62-7.08 (5.16±0.05)	0.74-2.30 (1.21±0.03)	3.10-57.28 (20.58±0.49)	1.89-10.67 (5.67±0.08)
	2002	10-20	10.20-18.00 (14.52±0.85)	27.93-37.20 (31.51±0.35)	5.94-6.51 (6.18±0.02)	2.35-3.96 (3.48±0.13)	0.34-1.34 (0.89±0.02)	5.33-39.07 (22.67±0.73)	1.00-4.63 (2.57±0.07)
	2003	10-20	10.50-17.60 (14.14±0.86)	19.40-38.07 (30.84±0.34)	5.46-6.37 (5.91±0.02)	2.22-5.29 (3.84±0.08)	0.40-1.14 (0.86±0.03)	4.66-43.79 (16.95±0.67)	1.83-11.25 (5.75±0. 09)
	2002	20-30	10.10-17.90 (14.21±0.79)	29.03-39.70 (34.11±0.31)	5.89-6.53 (6.22 ±0.02)	1.92-5.48 (3.29±0.07)	0.49-1.30 (1.10±0.03)	2.33-25.75 (12.37±0.63)	1.00-3.25 (2.0±0.07)
	2003	20-30	10.00-17.50 (13.92±0.79)	19.10-40.73 (32.98±0.25)	5.34-6.35 (5.86±0.03)	1.66-5.93 (3.75±0.09)	0.29-1.31 (0.75±0.03)	1.99-40.96 (15.94±0.54)	1.59-9.25 (5.93±0.08)
Undisturbed forest	2002	0-10	11.40-20.20 (14.88±0.95)	19.03-39.20 (26.78±0.39)	5.19-6.52 (5.65±0.08)	0.82-5.68 (3.92±0.09)	0.48-1.93 (1.05±0.02)	10.21-59.94 (34.23±0.57)	1.13-3.38 (2.15±0.08)
	2003	0-10	10.70-19.20 (14.29±0.99)	17.67-39.40 (30.52±0.59)	5.20-6.27 (5.60±0.05)	3.33-6.63 (4.95±0.10)	0.26-1.73 (1.06±0.03)	7.54-63.60 (25.12±0.65)	2.00-10.04 (5.57±0.07)
	2002	10-20	10.60-20.10 (14.53±0.95)	17.20-35.90 (22.93±0.27)	5.55-6.21 (5.78±0.04)	0.72-3.75 (2.56±0.09)	0.19-1.18 (0.72±0.02)	4.88-37.29 (22.29±0.94)	1.00-3.38 (1.62±0.08)
	2003	10-20	10.20-19.80 (13.92±1.02)	15.70-35.07 (25.81±0.30)	5.09-5.77 (5.51±0.02)	1.62-4.75 (3.27±0.07)	0.43-1.23 (0.73±0.02)	2.99-57.78 (17.48±0.58)	1.38-9.13 (4.85±0. 08)
	2002	20-30	10.10-19.40 (14.04±0.89)	16.47-36.23 (22.14±0.29)	5.59-6.07 (5.84±0.05)	0.44-3.48 (2.06±0.09)	0.05-1.11 (0.58±0.03)	3.99-25.64 (10.91±0.92)	0.63-2.63 (1.91-0.08)
	2003	20-30	10.00-17.40 (13.58±0.91)	14.27-35.90 (25.08±0.23)	5.27-5.82 (5.49±0.02)	1.40-3.45 (2.71±0.09)	0.33-0.80 (0.69±0.02)	3.99-46.95 (14.89±0.46)	1.25-8.50 (5.39±0.08)

Table 37 Correlation coefficient (r) values among microbial populations (Fungi and Bacteria), enzymes activities and physico-chemical characteristics in the disturbed forest soil

Soil properties	Cmic	DHA	URA	PA	ST	MC	pH	OC	TN	AP	K
FP	0.22^{c}	0.45^{c}	0.39^{c}	0.34^{c}	0.38^{c}	0.14^{a}	-0.17^{a}	0.25^{c}	NS	NS	0.19^{c}
BP	0.29^{c}	0.20^{c}	NS	-0.22^{c}	NS	0.14^{a}	NS	NS	0.23^{c}	0.43^{c}	-0.22^{c}
C_{mic}		0.23^{c}	NS	NS	-0.17^{a}	NS	NS	0.15^{a}	0.29^{c}	0.24^{c}	NS
DHA			0.36^{c}	0.14^{a}	0.36^{c}	0.37^{c}	NS	0.54^{c}	0.42^{c}	NS	NS
URA				0.49^{c}	0.64^{c}	0.58^{c}	NS	0.24^{c}	NS	NS	NS
PA					0.42^{c}	0.32^{c}	NS	0.15^{a}	NS	NS	0.28^{c}
ST						-0.42^{c}	NS	NS	0.14^{a}	-0.25^{c}	0.33^{c}
MC							0.19^{c}	0.28^{c}	0.19^{b}	NS	NS
pH								NS	NS	NS	-0.26^{c}
OC									0.32^{c}	NS	NS
TN										NS	NS
AP											-0.17^{b}

(**Note**: FP=fungal population, BP=bacterial population, C_{mic}=microbial biomass carbon, DHA=dehydrogenase activity, URA=urease activity, PA=Phosphatase activity ST=soil temperature, MC=moisture content, OC=organic carbon, TN=total nitrogen, AP=available phosphorus, K=exchangeable potassium)
Values marked with a, b and c is significant at $P \leq 0.05$, $P \leq 0.01$ and $P \leq 0.001$ respectively;
Insignificant values are marked with 'NS'

Table 38 Correlation coefficient (r) values among microbial populations (Fungi and Bacteria), enzymes activities and physico- chemical characteristics in the undisturbed forest soil

Soil properties	Cmic	DHA	URA	PA	ST	MC	pH	OC	TN	AP	K
FP	NS	0.51^{c}	0.38^{c}	0.31^{c}	0.38^{C}	0.38^{c}	-0.17^{b}	0.44^{c}	0.23^{c}	NS	0.28^{c}
BP	0.15^{a}	NS	NS	NS	NS	NS	NS	NS	0.29^{c}	0.29^{c}	NS
C_{mic}		0.42^{c}	-0.19^{c}	0.19^{c}	NS	0.25^{c}	NS	0.44^{c}	0.59^{c}	0.14^{a}	NS
DHA			0.24^{c}	0.26^{c}	0.28^{c}	0.58^{c}	NS	0.64^{c}	0.47^{c}	NS	0.28^{c}
URA				036^{c}	0.65^{c}	NS	-0.17^{b}	NS	NS	NS	NS
PA					0.50^{c}	0.24^{c}	NS	0.26^{c}	0.45^{c}	NS	0.23^{c}
ST						-0.25^{c}	-0.26^{c}	NS	0.11^{b}	-0.17^{b}	0.13^{a}
MC							NS	0.59^{c}	046^{c}	-0.39^{c}	0.36^{c}
pH								NS	NS	-0.17^{b}	-0.40^{c}
OC									0.54^{c}	NS	0.29^{c}
TN										NS	0.14^{a}
AP											NS

(**Note**: FP=fungal population, BP=bacterial population, C_{mic}=microbial biomass carbon, DHA=dehydrogenase activity, URA=urease activity, PA=Phosphatase activity ST=soil temperature, MC=moisture content, OC=organic carbon, TN=total nitrogen, AP=available phosphorus, K=exchangeable potassium)
Values marked with a, b and c is significant at $P \leq 0.05$, $P \leq 0.01$ and $P \leq 0.001$ respectiviely;
Insignificant values are marked with 'NS'

DISCUSSION

Microbial Population

Fungal and bacterial populations

The higher fungal and bacterial populations observed in the undisturbed forest soil than that in the disturbed forest could be due to better availability of nutrients and environmental conditions which favored their growth. The higher bacterial population in the undisturbed forest soil could be due to the favorable soil moisture, relative high temperature and the availability of organic matter and mineral nutrients. The abundance and activity of soil microorganisms are influenced by soil type, nutrient status, pH, moisture as well as plant factors (e.g. species, age). Jha *et al.* (1992) also reported the similar findings. The disturbance of soil and vegetation has an adverse effect on the distribution of microbial population number. The development of fungal communities may be influenced by soil moisture content and associated effects on tree growth and litter and exudates inputs (Anderson *et al.*, 2003).

Maximum fungal population was observed in the month of September in both the study sites during the study periods of 2002-2003. Tiwari *et al.* (1991) also reported highest number of fungi in spring-summer season while least number of fungi was recorded during winter season. The soil moisture content was related to the fungal population and was responsible for the higher population number in the rainy season. The lower number of fungal population was due to the lower soil moisture in the dry winter season. The seasonal variation in the fungal spectrum of the soil might be due to seasonal

variations in soil moisture, temperature, pH and organic matter of the soil (Baruah, 1983; Chauhan *et al.* (1985) and Agarwal and Chauhan (1988). Liu *et al.* (2000) stated soil moisture, soil temperature and substrate availability as the most important factors that influence soil microbial growth and population density. Soil microbial diversity (as measured by substrate utilization) and activity were generally reported to decrease with disturbance.

It was observed that the surface layer had higher fungal and bacterial populations as compared to that at lower layer in both the forest stands. The higher fungal and bacterial populations at 0-10 cm might be due to the higher organic matter content, nutrient status and better aeration in the surface layer (Balasubramanium *et al.,* 1972) and moisture regime (Selvaraj and Rangaswamy, 1978; Clarholm and Rosswall, 1980). The decrease in the population was linked to the decrease in oxygen level affecting the species composition and number of soil microbes at the subsurface layer. (Tiwari *et al.,* 1986). Low population may be linked to run-off losses of fungal propagules along with plant materials from the hill slope due to heavy rainfall in the study areas which was also reported by (Kshattriya *et al.,* 1992; Maithani, 1996). Tiwari *et al.* (1986) and Tiwari *et al.* (1987a, b) suggested that the presence of soil moisture content was the major factor controlling the distribution of fungal community. Tiwari and Sharma (1998) also reported that the fungal and the bacterial populations in highland soils increased with increase in altitude up to 1100 m, but thereafter, the populations declined sharply. They also observed positive correlation between fungal and bacterial populations with organic matter content of the soil. Behera and Mukerji (1985) studied the seasonal variation and

distribution of microfungi in forest soils of Delhi and noted that surface soils harbored the highest population and species number which gradually declined with increase in depth.

Almost similar trend of temporal and depth wise variations in microbial populations and their activities show that most of these parameters are governed by same or similar set of environmental factors. Decrease in the fungal population with increasing soil depth is related to the organic carbon content of the soil (Tiwari *et al.,* 1991). Among several factors affecting microbial population and activity, moisture and nutrient regime and soil depth are important factors. Zabinski *et al.* (1997) compared the functional diversity of soil microbial communities in heavily impacted sub-alpine campsites and adjacent undisturbed areas using the Biolog method of carbon utilization profiles. They reported that microbial communities differentiate in response to disturbance in the top 6cm of soil, while below 6cm there were no recognizable differences between disturbed and undisturbed soil communities. Analysis of the factors that differentiate the upper microbial communities between disturbed and undisturbed sites revealed that the percent of total carbon sources utilized was significantly less in the disturbed (54 %) than in undisturbed areas (95 %). They also compared among the total culturable actinomycetes, bacteria, and fungi and revealed no difference in overall number of colony forming units (CFU) on disturbed and undisturbed sites, but a marked decrease in actinomycetes on disturbed sites.

Additional factors which are not directly linked to the specific environmental conditions found within the soil profiles may also contribute to the differentiation of microbial communities. These factors may include: the selective transport of microbes through the soil profile by water or bioturbation (Abu-Ashour *et al.,* 1994; Balkwill *et al.,*

1998; Kieft *et al.*, 1998). Communities are responsive to pH, organic matter and nutrient concentrations (Egerton-Wharton and Allen, 2000). Zvyaginstev (1994) studied management in the case of the forest and suggested that soil properties which change with soil depth control the composition of microbial populations.

Although soils in tropical forests often are low in fertility, they also tend to be heterogeneous (Sollins *et al.*, 1994). It is quite clear that the pattern of distribution of fungi is affected by many environmental factors. Nutritional conditions (Rehder and Schafer, 1978) and microclimate factors must have a decisive influence on the distribution of individual species. Fierer *et al.* (2003) reported that soil water content may be partially responsible for differentiation of microbial communities with soil depth. The variability in soil moisture is much higher at the soil surface than at greater depths. Previous studies by Schimel *et al.* (1999) and Wilkinson *et al.* (2002) have shown that the variability in soil moisture can influence the composition of soil bacterial and fungal communities. In the present study, most of the fungal species were not confined to a particular horizon. Only a few species were restricted to a specific depth. Distribution of most of the fungal species to all depths might also be due to activities of soil fauna (earthworms) because their feeding and borrowing activity homogenize the species composition along all the depths.

Microbial biomass carbon

Microbial biomass carbon was lower in the disturbed than in the undisturbed forest soils. The reason for high microbial biomass C in the undisturbed forest soil could be due to the presence of litters, twigs, herbs and the tree canopy which render a moist

environment in the soil and favor high microbial activity and hence high microbial biomass carbon. Srivastava (1992) and Mao *et al.* (1992) reported maximum microbial biomass C from mixed forest. The activities of microorganisms in the soil are highly influenced by many factors amongst which soil moisture, temperature and density are the most important (Anderson, 1975; Krockel and Stolp, 1986; Akira *et al.,* 2000). Zeller *et al.* (2000) reported the highest C_{mic} values (with no carbon substrate addition) were found in top soil. They observed that the correlation between the organic carbon content and the biomass amount was not significant.

Not much variation was observed in microbial biomass C throughout the study periods in both the study sites. However, the winter season (October -January) showed high microbial biomass C when compared to other seasons during the first year. Maithani *et al.* (1996) also reported high microbial biomass carbon during the winter. The factors influencing microbial biomass C are soil moisture and temperature (Campbell and Biederbeck 1976), physical disturbance of the soil (Doran, 1987) and interaction with soil fauna (Beare *et al.,* 1992). During the second year, monthly variations were observed in microbial biomass C in both the study sites. Lavahun *et al.,* (1996); Bardgett and Cook (1998), Sneh *et al.* (2000) and Parham *et al.* (2002) also reported similar findings. The variations in soil microbial biomass C might be due to soil type and seasonal fluctuations, which was controlled by climatic factors and geomorphic conditions (Piao *et al.,* 2000 and Diaz-Ravina *et al.* 1995). They concluded that variations in soil microbial biomass C resulted in increase in soluble organic C due to higher turnover rate of the former at warm air temperature. Thus there was a marked seasonal change in soil microbial biomass C. Inputs of plants residues have an obvious influence on seasonal changes of soil microbial

C (Chander *et al.*, 1997). The values of microbial biomass C of summer and autumn samples would, therefore, be expected to be greater than those of winter samples. However, they observed an opposite relationship so other mechanisms must be responsible for the decline of microbial biomass C during summer. Diaz-Ravina *et al.* (1995) reported significant effects of the season, the type of soil, the interaction between the type of soil and season, but soil type explained most of the variance on microbial biomass and other soil nutrient flush of forest soils. They have also found a substantial contribution of microbial biomass to plant -available nutrients of forest soils. Further, they noted close relationship of microbial biomass with pH, organic C, total N and moisture contents.

The microbial biomass C was found to be higher in the surface soil layer and decreased with increase in depth. Maithani *et al.* (1996) also observed that the surface soil layer (0-10cm) had significantly higher microbial biomass C than the sub-surface layer. Large pool of organic matter at the soil surface supports a uniquely large and active soil microbial community. Lavahun *et al.* (1996) also reported significant variation in the distribution of the microbial biomass C at different depths of grassland and arable soils. They found that biomass C declined from maximum at the upper layer to a minimum at the lower depth in their study sites. They observed decrease in microbial biomass C associated with decline in organic C content revealing higher organic C mineralized at upper layers leading to higher microbial activity than the lower soils layers.

Microbial biomass carbon was found to show significant variation with moisture content, total nitrogen and organic carbon in both the forest soils. Wardle (1998) stressed the temporal variability of the soil microbial biomass is an important component of its

turnover; and thus contribute to patterns of soil nutrient release and mineralization. The temporal variability of microbial biomass C was most closely related to soil N content in forest, pH, latitude and soil C contents in most ecosystems. He also stated that both macroclimate and soil quality factors (e.g. soil pH, organic matter content) play important roles in determining microbial biomass and its turnover. Insam (1990) reported that studies on intimate relationship between climatic conditions and the microbial soil C pool and respiratory C flux showed the best possible prediction of microbial biomass C and its ratio to total organic C by using climatic variables such as precipitation: evaporation ratio in soil with equilibrium C content. Further he found that longer dry period reduced microbial biomass and increased with increase in moisture content.

Soil Enzyme Activities

Dehydrogenase activity

Dehydrogenase activity was found to be higher in the soil of undisturbed forest site in comparison to the disturbed forest site. This could be due to higher organic matter contents. Similar findings were reported by Camina *et al.* (1998) and Tiwari *et al.* (2002). The high dehydrogenase activity in the forest soil was related to high nitrogen, organic carbon content and increased microbial populations. Kumari and Charya (1997) found significant positive correlationship between soil enzyme activities and microbial population number in four polluted sites of Warangal, Andhra Pradesh, India. They found increased microbial colonies showing increased accumulation of soil enzymes. Positive correlation was observed between enzyme activities and soil nutrients such as nitrates, potassium and organic matter. Enzyme activities are determined by the factors that affect

size and metabolic activity of microorganisms in soil. Due to the significant changes in the soil physical and chemical properties of forest stands, changes in the size of the micro biota and in enzyme activities in the forest floor and mineral soils could be expected. The dehydrogenase activity is influenced directly or indirectly by the factors such as pH, moisture content and temperature of the soil (Tesarova and Gloser, 1976; Kowalenko *et al.*, 1978; Tiwari *et al.*, 1987, Von Merci and Schinner 1999). Dehydrogenase activities appear to be greater in the less acid samples and are strongly favored by moisture. Gilot and Dommergues (1967) found that dehydrogenase activities per gram soil organic C in some sub-alpine forest soils were greater in horizons of highest pH values.

High dehydrogenase activity at the 0-10cm soil layer was observed in both the forest stands. This might be due to the accumulation of litter, shrubs and other materials on the surface soil layers which increase the substrate for both the forest stands by the saprophytic microbes. Increase in microbial community results in increase in dehydrogenase activity. Similar observations was also reported by Khan, 1970; Das, 1980; Dkhar and Mishra, 1983; Baruah and Mishra, 1984 and Tiwari *et al.*, 1987b. The decrease in enzyme activities suggested that microbial activity was inhibited by unfavorable environmental conditions prevailing in the deeper region of the soil (Duxbury and Tate, 1981). Low microbial population in the deeper soils may be responsible for lesser microbial activities in deeper layer.

Urease activity

Not much variation was observed in urease activities throughout the study periods in both the study sites. However, urease activity was observed to be slightly higher in the disturbed as compared to undisturbed forest soils. Higher urease activity might be due to

the availability of nutrients and other environmental factors like temperature, moisture content and type of vegetation. This was also reported by Pancholy and Rice (1973) who showed that urease activity is related to type of vegetation and the quality of incorporated organic materials in the soil. The same result was also reported by Palma and Conti (1990). The enzyme activity showed positive correlation with organic carbon in the disturbed forest soil. With increase in organic carbon content enzyme activities increased. Speir (1977), Beri *et al.* (1978) and Ojeniyi (1980) reported that soil urease activity was largely controlled by the organic carbon status of the various soils. The positive correlation between urease activity and organic carbon suggests that the organic carbon content may be accounted for most of the variations in urease activity (Dkhar and Mishra, (1983); Dalal, (1975) and Tabatabai (1977).

Urease activity decreased with increase in depth. This finding was in accordance with the report of Tiwari *et al.* (1987a); Tiwari (1988, 1996); Dkhar (1983). Singh (2002) also showed the decrease in activity down the soil profile. Higher urease activity during the study period in the surface soil may be due higher organic content, bacterial population and favorable moisture (Dalal, 1975; Speir, 1977; Tabatabai, 1977; Beri *et al.* 1978; Dkhar and Mishra, 1983; Gonzalez and Fuente, 1984; O'Toole *et al.* 1985; Tiwari *et al.* 1987b). Sahrawat (1984) noted that urease activity increased with the increasing temperature. Higher urease activity also indicated that it is largely governed by the soil temperature, moisture, organic carbon. On the other hand, Skujins (1976), Beri *et al.* (1978); Dkhar and Mishra (1983); Sahrawat (1983) and Rao and Ghai (1985) found that the activity was principally associated with the organic carbon content of the soil.

Variations in urease activity were shown to be caused mainly by changes in organic matter content of soils (Bremner and Mulvaney, 1978).

Phosphatase activity

The phosphatase activity was found to be almost same in both the study sites. Higher phosphatase activity in the months of June and July could be attributed to higher temperature, organic matter and availability of nutrients on the forest floor and also the activity of the plant growth in which enzymes were probably being generated by roots and microorganisms, for these are major sources of phosphatases (Nannipieri *et al.*, 1979; Appiah and Thomas, 1982; Chhonkar and Tarafdar, 1984). The increase in phosphatase activity during this period appeared to be related to the leaching of phosphatase from leaf liter and decomposition processes (Brown, 1974). Soil temperature, moisture, organic carbon, total nitrogen and potassium were found to be important factors affecting the phosphatase activity. Singh (2002) also reported similar observations in the degraded and undegraded forests soils of Arunachal Pradesh. The phosphatase activity may be influenced by seasonal and climatic changes in the forest sites. Tiwari (1988) in his investigation reported that phosphatase activity was found to be regulated by the organic carbon content and soil temperature. No significant correlation was observed between phosphatase activity and available phosphorus. The plants and microbes regulate mineralization in response to nutrients supply. When nutrients supply is low, enzymes are induced and nutrients are mineralized but when nutrients supply is high, enzymes are suppressed and mineralization ceases. Relationship between nutrient content and enzyme activity is regulated by negative feedback mechanisms. Dilly and Nannipieri (2001) found that the presence of P usually decrease phosphates activity.

The phosphatase activity was observed to be higher in the surface soil layer as compared to the layer below it. This could be due to the accumulation of organic matter, pH which enhances the microbial population which in turn increases enzyme activity at the surface soil layer. Similar findings were reported by Trasar-Cepeda and Gil-Sotres (1987); Tiwari (1988; 1996a and b); Tiwari *et al.* (1989a and b); Singh (2000) who showed the decrease in enzyme activity with increase in depth.

Soil Physico-Chemical Characteristics

Temperature

Increased soil temperature in the undisturbed forest site during the three months i.e. June, July and August could be due to effect of the solar radiation and the heating up of the surrounding soil surface. Studies have shown that the nearer the angle of incidence of the sun's rays approaches the perpendicular, the greater will be the absorption. Variation in temperature was recorded in both the forest soils; however, it was observed that the surface soil layers showed more temperature than the subsurface layers. Higher soil temperature of pineapple stands during rainy season at the surface layer was also reported by Tiwari (1988). Variations in soil temperature were also affected by the altitude of the study sites and also by the climate of the particular place. Low soil temperature in deeper soils may be due to the insulation effect of the upper soil and litter layer. Baruah (1983) studied the temporal and depth-wise variations in soil temperature, moisture and pH of rice field soil and noted higher soil temperature from surface (0-10cm) soils which decreased with increasing depth. A thick layer of ground vegetation, limit the amount of heat the soil can absorb during the summer. Other soil characteristics,

such as soil moisture, may also be involved. Soil temperatures can provide a long term measure of how the climate is changing because soil temperatures, especially at deeper levels, tend to reflect long term changes. Similar trend of temporal and depth wise variations in soil temperature and soil pH was noted by Baruah (1983) and Dkhar (1983) in rice and maize fields soil respectively.

Moisture content

The moisture content was found to be significantly higher in the disturbed forest than in the undisturbed forest soil. This can be attributed to a combination of the higher infiltration rate allowing more water into the profile in the disturbed soil, as well as the water extraction by the plants. The lower moisture content in the undisturbed forest soil was the result of quick runoff from the slopes and low water retention capacity of the soil. Temperature and uneven topography of the undisturbed forest also play important role in the runoff of water from the site. Dynamic of soil moisture is related to soil physical properties, topography, weather, and soil cover. Das (1980) studied the physico-chemical characteristics of forest soil of Meghalaya (India) and noted higher moisture content during the months of May-December and the values of moisture content dropped in the preceding months. Tiwari (1988) also reported the same from pineapple orchard soil where he suggested that the increase or decrease in soil moisture was related to the amount of rainfall. Much variations in soil moisture content were observed depth wise, however, in most cases it was found to be higher at surface soil than at subsurface layers Singh (2002) also reported maximum moisture content from the undegraded site at the surface soil layer.

Organic carbon

The soil organic carbon was found to be higher in the disturbed forest than that in undisturbed forest. The surface soil contained highest organic carbon followed by sub-surface layers in both the forest soils. Similar findings were also reported by Singh (1980). The higher concentration of nutrients in surface (0-10 cm) soils might be due to higher organic matter content in the surface layer (Blackmore, 1966; Gupta and Rorison, 1975; Dkhar 1983). Surface layer is continuously enriched by the nutrients released from decomposing litters. In addition to regulating the oxygen content of the soil, moisture partly regulates the availability and movement of nutrients to the microbes.

The decline in resource availability through the soil profile is predominantly a function of decreasing carbon concentrations i.e., a reduction in carbon quality with depth (Richter and Markewitz, 1995; Ajwa *et al.*, 1998; Trumbore, 2000). The temporal variations in nutrients are attributable to the variations in 'biological activity' occurring in the soil. The nutrient levels of deeper soils mainly depend on the mineralization process at the surface layer because the nutrients released in surface layer percolate down the profile along with the water. The higher organic carbon content on the surface soil layer may be due to the accumulation of organic matter on the forest floor after decomposition of litters. This was also reported by Brown *et al.* (1994) and Singh *et al.* (1995).

Nitrogen, phosphorus and potassium

Physico-chemical properties of both forest soils were found to be quite similar in all the soil depths. Monthly fluctuation in nitrogen was observed during the two year of study periods in both the forest soils. The soil of disturbed forest contained more nitrogen

as compared to that of undisturbed forest. It was observed that in most cases, the nitrogen content decreased with increase in depth. A marked variation was observed in both the study sites. Singh (2002) also reported the same. High nitrogen content in the disturbed forest soil can be explained by the preference of trees which assimilate ammonium which is the dominant mineral nitrogen compound in acidic soils. This was reported by Buchmann *et al.* (1995) and Gebauer and Schulze (1997). In addition, microbial activity is inhibited in acidic soils, whereas in soils with a higher pH they can potentially compete with nitrogen.

Comparing both the study sites it was observed that the undisturbed forest soils contained more phosphorus as compared to that of the disturbed forest soils. The available phosphorus content did not show much variation with increase in depths. Similar result was reported by Tiwari (1988) suggesting that the monthly variation may be related to the rapid release of this nutrient from the litter at the same period. The concentration of available phosphorus remained more or less constant in the deeper soils. Phosphate is relatively immobile and this may be the reason for the little variations noted along three depths. The low phosphorus content during summer months corresponds to the increased uptake by the growing plant species. Tiessen *et al.,* (1984) reported that the role of phosphorus in the functioning of forest ecosystems is, however, severely hampered by difficulties in identifying and quantifying available P in forest soils as phosphorus occurs in soil both in organic and inorganic forms.

It was observed that the potassium content was found to be higher in the soil of the disturbed forest as compared to that of the undisturbed forest soil. The concentration of the exchangeable potassium was found to be maximum in surface (0-10cm) soil and it

decreased with increase in soil depth. Similar result was reported by Tiwari (1988). The content of available potassium and immediately available potassium is always higher in uppermost soil layer than that in lower soil layers and it is closely related to the organic matter content of the soil. Potassium content of the soil also depends on topography of the study sites as well as on quality of a soil. Their content are varied tremendously not only among the soil types and but also in a soil type. Exchangeable potassium concentration was lower during winter months. This was also recorded by Singh (1980) from the *Shorea robusta* forest soils. Higher concentration of exchangeable potassium may be due to the rapid release of the nutrient from the decomposing organic matter (Tukey *et al.,* 1958). On lower location, the potassium content in the soil is higher but on higher location, its content is lower. This could be the reason for the low content of exchangeable potassium in the undisturbed forest soil which was sloppy in topography and located slightly higher than the disturbed forest soil. Almost all high land soils suffered from potassium losses due to leaching and soil erosion. Haagsma and Miller (1963) described that the rate of release of non-exchangeable potassium to exchangeable form has been found to be a temperature dependent phenomenon. Moisture content of the soil was also found to be directly related to the availability of potassium (Kuchenbuch *et al.,* 1986).

SUMMARY

The present investigations were carried out in two sub tropical forest stands viz., (i) disturbed and (ii) undisturbed forests of Law Lyngdoh situated at Nongkrem, 20 km away from Shillong in the East Khasi Hills District of Meghalaya. The geographical position of the study site is between latitude 25°34' N and longitude 91°46' E and at an altitude of 1786 msl.

The soil samples were collected at three different depths (0-10cm, 10-20cm and 20-30cm) from five places at each study site for a period of two years i.e. 2002 and 2003 at monthly intervals. The soil samples of each site were mixed to make the composite samples separately and the following studies were made:

For isolation of fungi and bacteria, serial dilution plate method (Johnson and Curl, 1972) was followed using rose bengal agar medium (Martin, 1950) and nutrient agar medium respectively. The inoculated Petri plates for fungi and bacteria were incubated separately at 25 $\pm 1^0$ C for 5 days and at 30 $\pm 1^0$ C for 24 hours respectively. Three replicates were maintained for each sample. The fungal and bacterial populations were estimated on per gram dry soil basis. Soil microbial biomass carbon (C_{mic}) was determined by the technique of Anderson and Ingram (1993). Dehydrogenase, urease and phosphatase activities of the soils were estimated by the methods of Casida (1977), McGarity and Myer's (1967), Tabatabai and Bremner (1969) respectively. pH was measured in a 1:5 soil water suspension using electric digital pH meter. Moisture content of the soil was measured by drying 10 g of soil in an oven at 105^0 C for 24 h and reweighing the dried samples till a constant weight was obtained. The soil used in the

analyses of organic carbon, total nitrogen, available phosphorus and exchangeable potassium was air dried and ground to pass through a 0.2 mm sieve. Soil organic carbon was determined by colorimetric method (Anderson and Ingram, 1993). Total N and available P were determined by Kjeldahl's method and molybdenum blue method respectively. Exchangeable K was extracted in ammonium- acetate buffer (pH 7) and the same was read in flame- photometer (Allen, 1974).

The results showed that the fungal and bacterial populations exhibited almost a similar trend of monthly variations in the soils of the two different forest stands during the study periods of 2002 and 2003. The soil of the undisturbed forest harbored higher fungal and bacterial populations as compared to that of disturbed forest. Both fungal and bacterial populations decreased with increase in depth. Altogether 95 fungal species were isolated of which 12 belonged to Zygomycotina, 80 belonged to Deuteromycotina and 3 Mycelia Sterilia. *Acremonium butyri, A. murorum, Aspergillus candidus, A. fumigatus, A. niger, Cladosporium cladosporioides, Humicola fuscoatra, Paecilomyces carneus, P. lilacinus, Penicillium brevicompactum, P. chrysogenum, P. expansum, P. frequentans, P. janthinellum, P. jensenii, P. lanosum, P. nigricans, P. oxalicum, P. purpurogenum, P. simplicissimum, P. restrictum, P. rubrum, Penicillium* sp, *P. waksmanii, Phoma eupyrena, Trichoderma viride, Verticillium alboatrum, V.* chlamydosporium and white sterile mycelia were found in all the three depths of both the study sites. A few species were restricted to each study site. *Acremonium fuscidioides, A. sclerotiorum, Aspergillus carneus, A. clavatus, A. versicolor, Aspergillus* sp., *Eupenicillium lapidosum, Heteroconium chaetospira, Nectria ventricosa, Penicillium daleae, P. rugulosum, P. stoloniferum, P. variable, Pythium irregulare, Trichoderma harzianum, T.*

pseudokoningii, T. piluliferum and yellow sterile mycelium could be isolated only from the soil of undisturbed forest whereas, *Aspergillus terreus, Botryotrichum piluliferum, Eupenicillium sheari, Mucor racemosus, Penicillium granulatum, Plectosphaerella cucumerina, Trichoderma polysporum* and *Verticillium catenulatum* were restricted to that of the disturbed forest.

A total of 7 bacterial species could be isolated from the soils of both the study sites at different depths. These were *Arthrobacter* sp., *Bacillus* sp., *B. cereus, B. subtilis, Micrococcus* sp., *Pseudomonas* sp. and *Rhizobium* sp. Majority of the bacterial species isolated were common to both the forest soils. *Bacillus cereus* was isolated only from the soil of disturbed forest. The dominant bacterial species were *Arthrobacter* sp., *Bacillus* sp., *B. subtilis* and *Micrococcus* sp. The bacterial species viz., *Arthrobacter* sp., *Bacillus cereus*, *Pseudomonas* sp., and *Rhizobium* sp., were found to occur less frequently in the both the study sites.

Shannon's diversity index for fungi and bacteria showed less variation in disturbed forest soil as compared to undisturbed forest soil.

Monthly variations in microbial biomass carbon showed similar trend of (C_{mic}) in both the study sites. Comparing both the study sites, it was observed that the soils of the undisturbed forest had higher C_{mic} than that of disturbed forest soil. In the first year of study period, the maximum C_{mic} was observed in the months of January in both the forest soil and the minimum was observed in the months of August and November in the disturbed and undisturbed forest soil respectively. Whereas in the second year, the maximum C_{mic} was observed in the months of November and May and the minimum was

observed in the months of October and August in the disturbed and undisturbed forest soils respectively.

The dehydrogenase activity showed slightly higher activity than that of disturbed forest soils. There was a monthly fluctuation in dehydrogenase activity in both the forest sites during the two year of study periods. Comparing both the study sites, it was observed that the activity was almost same in both the forest soils. In the first year, the months of August (0-10cm) showed higher activity in both the forest soils and the lower activity was observed in the months of July (20-30cm) and March (20-30cm) in the disturbed and undisturbed forest soils respectively. During the second year, it was observed that the months of October and May (0-10cm) showed highest dehydrogenase activity in both the forest soils and the minimum activity was observed in the months of February and November (20-30m) in the disturbed and undisturbed forest soils respectively.

In the first year, the urease activity showed less variation throughout the study period in both the study sites. It was observed that the urease activity was slightly higher in the disturbed forest soil than the undisturbed forest soil. The activity decreased in the months of January to May and thereafter it increased till November in both the study sites. The activity was higher at the surface layer and decreased with increase in depth.

In case of phosphatase activity, similar trend of monthly variations were observed in both the study sites. The soil of disturbed forest showed slightly higher activity than undisturbed forest soil. In the first year of study periods, it was observed that the month of July has higher phosphatase activity at all the three different depths in both the forest soils. In the second year, it was observed that the activity decreased in the months of

January to May in both the forest soil and then increased in the months of June and thereafter it showed a decreasing trend. The phosphatase activity was found to be higher during summer than that of winter months. Phosphatase activity decreased with increase in depth.

Among various physico-chemical characteristics, temperature, moisture content, pH, organic carbon, total nitrogen, available phosphorus and exchangeable potassium of soils of both the study sites were studied. Highest soil temperature was recorded at surface soil as compared to subsurface soils. Undisturbed forest soils had slightly higher temperature than that of disturbed forest soils. There were not much variations of moisture content throughout the months in the soils of both the forest stands during the study periods. However, higher moisture content was found in the disturbed forest soil than that in the undisturbed forest.

The pH was acidic in both the study sites. Comparing both the study sites it was observed that the soil of the undisturbed forest was more acidic than that of the disturbed forest.

Soil organic carbon clearly showed that at depth 0-10cm, soil held higher organic carbon content as compared to other two depths as in both the forest soils throughout the study periods. Monthly variations were also observed throughout the study periods. The soil organic carbon was found to be higher in the disturbed forest than that in undisturbed forest. The surface soil contained highest organic carbon followed by sub-surface layers.

Nitrogen showed a monthly fluctuation during the two year of study periods in both the forest soils. It was observed that the soil of disturbed forest contained more

nitrogen as compared to that of undisturbed forest. The nitrogen content was found to be highest at the 0-10cm and decreased with increased in depth.

In the first year, the months of February contained high amount of available phosphorus in both the study sites. In the second year of study period the months of January and February contained high amount of available phosphorus at all the three different depths. Comparing both the study sites it was observed that the undisturbed forest soils contained more phosphorus as compared to that of the disturbed forest soils. The amount of phosphorus decreased with increased in depth.

There was less variation of exchangeable potassium in both the study sites. Comparing both the study sites, it was observed that the potassium content was found to be higher in the soil of the disturbed forest as compared to that of the undisturbed forest soil.

The correlation coefficient of the various biological, biochemical and physico-chemical characteristics of the two forest soils was analyzed. It was observed that in the disturbed forest soil, the fungal population showed a positive significant correlation with C_{mic} ($r= 0.22$, $P\leq 0.001$), dehydrogenase activity ($r=0.45$, $P\leq 0.001$), urease activity ($r=0.39$, $P\leq 0.001$), phosphatase activity ($r=0.34$, $P\leq 0.001$), soil temperature ($r=0.38$, $P\leq 0.001$), moisture content ($r=0.14$, $P\leq 0.05$), organic carbon ($r= 0.25$, $P\leq 0.001$) and potassium ($r=0.19$, $P\leq 0.001$). It showed a negative correlation with pH ($r= -0.17$, $P\leq 0.01$), whereas, the bacterial population was positively significant with C_{mic} ($r=0.29$, $P\leq 0.001$), dehydrogenase activity ($r=0.20$, $P\leq 0.001$), moisture content ($r=0.14$, $P\leq 0.02$), total nitrogen ($r=0.23$, $P\leq 0.001$), available phosphorus ($r=0.43$, $P\leq 0.001$) and negatively correlated with phosphatase activity ($r=-0.22$, $P\leq 0.001$), and potassium ($r=-0.22$, $P\leq$

0.001) (Table 37). In the undisturbed forest soil, the fungal population was positively correlated with dehydrogenase activity (r=0.51, P≤ 0.001), urease activity (r= 0.38, P≤ 0.001), phosphatase activity (r= 0.31, P≤ 0.001), soil temperature (r=0.38, P≤ 0.001), moisture content (r=0.38, P≤ 0.001), organic carbon (r= 0.44, P≤ 0.001), total nitrogen (r= 0.23, P≤ 0.001), potassium (r= 0.28, P≤ 0.001) and negatively correlated with pH (r= -0.17, P≤ 0.01). Whereas the bacterial population was found to be positively correlated with C_{mic} (r=0.15, P≤ 0.05), total nitrogen (r=0.29, P≤ 0.001), available phosphorus (r=0.29, P≤ 0.001) (Table 38).

C_{mic} was positive significantly correlated with dehydrogenase activity (r=0.23, P≤ 0.001), organic carbon (r=0.15, P≤ 0.05), total nitrogen (r=0.29, P≤ 0.001), available phosphorus (r=0.24, P≤ 0.001) and negatively correlated with soil temperature (r=-0.17, P≤ 0.05) in the disturbed forest soil (Table 37), whereas, in the undisturbed forest soil, C_{mic} showed a positive correlation with dehydrogenase activity (r=0.42, P≤ 0.001), phosphatase activity (r=0.19, P≤ 0.001), moisture content (r=0.25, P≤ 0.001), organic carbon (r=0.44, P≤ 0.001), total nitrogen (r=0.59, P≤ 0.001), available phosphorus (r=0.59, P≤ 0.05), and negatively correlated with urease activity (r=-0.19, P≤ 0.001) (Table 38).

Dehydrogenase activity showed a significant positive correlation with urease activity (r=0.36, P≤ 0.001), phosphatase activity (r=0.14, P≤ 0.05), soil temperature (r=0.36, P≤ 0.001), moisture content (r=0.37, P≤ 0.001), organic carbon (r=0.54, P≤ 0.001) and total nitrogen (r=0.42, P≤ 0.001) in the disturbed forest soil (Table 37). In the undisturbed forest soil, dehydrogenase activity showed a positive significant correlation with urease activity (r=0.24, P≤ 0.001), phosphatase activity (r=0.64,

P≤ 0.001), soil temperature (r=0.28, P≤ 0.001), moisture content (r= 0.58, P≤ 0.001), organic carbon (r= 0.64, P≤ 0.001), total nitrogen (r=0.47, P≤ 0.001) and potassium (r=0.28, P≤ 0.001) (Table 38).

In the disturbed forest soil, urease activity showed a positive significant correlation with phosphatase activity (r=0.49, P≤ 0.001), soil temperature (r=0.64, P≤ 0.001), moisture content (r=0.58, P≤ 0.001), organic carbon (r=0.24, P≤ 0.001) (Table 37), whereas, in the undisturbed forest soil it was observed that the urease activity showed a positive significant correlation with phosphatase activity (r=0.36, P≤ 0.001), soil temperature (r=0.65, P≤ 0.001), and negatively correlated with C_{mic} (r=-0.19, P≤ 0.01) and pH (r=-0.17, P≤ 0.01) (Table 38).

In the disturbed forest, phosphatase activity showed significantly positive correlation with soil temperature (r=0.42, P≤ 0.001), moisture content (r=0.32, P≤ 0.001), organic carbon (r=0.15, P≤ 0.05) and potassium (r=0.28, P≤ 0.001) and negatively correlated with bacterial population (r=-0.22, P≤ 0.001) (Table 37). In the undisturbed forest, phosphatase activity showed a significant positive correlation with soil temperature (r=0.50, P≤ 0.001), moisture content (r=0.24, P≤ 0.001), organic carbon (r=0.26, P≤ 0.001), total nitrogen (r=0.45, P≤ 0.001) and potassium (r=0.23, P≤ 0.001) (Table 38).

The moisture content in the disturbed forest showed a significant positive correlation with pH (r=0.19, P≤ 0.001), organic carbon (r=0.28, P≤ 0.001), total nitrogen (r=0.19, P≤ 0.01) and negatively correlated with soil temperature (r=-0.42, P≤ 0.001) (Table 37), whereas in the undisturbed forest soil moisture content showed positive correlation with organic carbon (r=0.59, P≤ 0.001), total nitrogen (r=0.46, P≤ 0.001),

potassium (r=0.36, P≤ 0.001) and negatively correlated with soil temperature (r=-0.25, P≤ 0.001) and available phosphorus (r=-0.39, P≤ 0.001) (Table 38).

The pH showed a negative correlation with potassium (r=-0.26, P≤ 0.001) in the disturbed forest soil (Table 37), whereas, in the undisturbed forest it showed negative correlation with soil temperature (r=-0.26, P≤ 0.001), available phosphorus (r=-0.17, P≤ 0.01) and potassium (r=-0.40, P≤ 0.001) (Table 38).

The organic carbon showed a positive significant correlation with total nitrogen (r=0.32, P≤ 0.001) in the disturbed forest (Table 37) and with total nitrogen (r=0.54, P≤ 0.001) and potassium (r=-0.29, P≤ 0.001) in the undisturbed forest soil (Table 38).

Total nitrogen showed a significant positive correlation with soil temperature at (r=0.14, P≤ 0.05) in the disturbed forest soil (Table 37) and with soil temperature (r=0.11, P≤ 0.01), potassium (r=0.14, P≤ 0.05) in the undisturbed forest soil (Table 38).

Available phosphorus showed a negative correlation with potassium (r=-0.17, P≤ 0.01) and with soil temperature (r=-0.25, P≤ 0.001) in the disturbed forest soil (Table 37) and negatively correlated with soil temperature (r=-0.17, P≤ 0.01), moisture content (r=-0.39, P≤ 0.001), pH (r=-0.17, P≤ 0.01) in the undisturbed forest soil (Table 38).

CONCLUSION

Hence we can conclude that microbial populations and activities are governed by a set of environmental factors and nutrients status present in the soil. Among various physico-chemical characteristics of soil: temperature, moisture content, organic carbon and potassium were found to play an important role in influencing the fungal population whereas, pH did not show significant effect on the microbial populations and their activities in both the forest stands. The biochemical properties of the soil like dehydrogenase, urease and phosphatase activities contribute to the populations of fungi and bacteria in the two forest stands. Other important factors which influence soil microbial activities and nutrients dynamics are climate and topography. It appears that within the same climatic conditions, the seasonality of microbial activity differs depending on the type of vegetation.

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1	SIDBI-Sponsored STUP on Medicinal and Aromatic Plants, Shillong, Meghalaya (Workshop)	CIMAP, Lucknow in collaboration with State Council Of Science, Technology and Environment, Meghalaya.	Shillong	27-30th Sept. 2000	Participant
2	Intellectual Property Rights.	North Eastern Hill University, Shillong.	Shillong	24-25th May 2002	Participant
3	Atomic Energy & Societal Development in India.	Department of Atomic Energy, Mumbai.	Shillong	18th Sept. 2002	Participant
4	72nd Annual Session of NASI and Symposium.	The National Academy of Sciences, India.	Shillong	25-27th Oct. 2002	Participant
5	National Roving Seminar on Patenting in Biotechnology.	Department of Biotechnology, Ministry of Science & Technology, New Delhi.	Shillong	26th Oct. 2002	Participant
6	National Seminar on impact of Increasing Human Population on Natural Resources(IPN)	ISCON, Varanasi and Department of Botany, Banaras Hindu University,Varanasi.	Varanasi	16-18th Oct. 2003	Participated and presented Paper

8. List of research publications:

1. Dkhar, M. and Dkhar, M.S. 2006. Microbial diversity and biomass carbon of disturbed and undisturbed forest soils. *Soil Biol. Ecol.* (in press).
2. Dkhar, M. and Dkhar, M.S. 2006. Microbial populations and dehydrogenase activity in disturbed and undisturbed deciduous forest soils (communicated).
3. Dkhar, M. and Dkhar, M.S. 2006. Urease and phosphatase activities in disturbed and undisturbed deciduous forest soils (communicated).

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