STUDIES ON THE EFFECT OF "BUN" CULTIVATION ON SOIL MICROBES AND THEIR ACTIVITIES

ABSTRACT

BY

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ABSTRACT

The practice of shifting cultivation is well known amongst the hill tribes of India especially in the Northeast region. Although shifting cultivation is understood by many workers from various fields of studies but in the context of the Khasi of Meghalaya, it exists in various forms. Besides the generally known slash and burn practice of jhum cultivation, there is a special type of shifting cultivation practiced particularly in the Khasi and Jaintia Hills of Meghalaya known as "bun" strips cultivation for potato and other crops. It is practice in three different ways according to the land distribution: i) Syiem system, where the king, owner of the land distributes land, ii) Village head system, where the leader of the clan earmarks land for the village and iii) Individual owner, who have the land in the name of single or joint families. The first and second systems of cultivation practice are orthodox and never go for permanent cultivation but the third category is adaptable. Nearly 38.4 percent of the land utilization area is under the Neolithic agricultural practice of 'BUN' cultivation.

The research investigation focus on the enumeration and isolation of soil microorganisms and the effect of 'BUN' cultivation on the soil microbial community (fungal and bacterial population), soil microbial biomass C (C_{mic}), physico-chemical properties (moisture content, pH, organic carbon, N, P and K) and the soil enzyme activities.

The investigation was carried out at Mawlali village of Nongkrem area (Altitude of 1785 m above the sea level; $25^0 35^l$ N latitude and $91^0 47^l$ E

longitude), Shillong, Meghalaya on cabbage crop cultivation for two consecutive cropping seasons in the month of May 2001 to December 2002. The whole valley of the Nongkrem area is covered with agricultural land, where bun cultivation is the main agricultural practice that is extensively practiced by the local people.

The average annual rainfall in the year 2001 and 2002 is 1200 cm with average temperature of 18-20°C. The climate of the study area has been very much controlled by the south-west monsoons and north-east winter winds which are neither too warm in summer nor too cold in winter. The year are divided into four seasons spring, summer, autumn and winter season respectively. The cabbage crop cultivation is conducted on the basis of local cultivation practices called 'BUN" cultivation where a bun strips are made and burn. The adjacent forest site has also been chosen for the comparative study as control where *Myrica esculenta* and *Pinus khasiana* are the dominant species.

The experimental fields were divided into two sites as bun cultivation site (BCU) and forest site (FS) with three replicates each at different depths of the soil. Soil samples were collected from surface (0-10 cm) layer (SL), middle (10-20 cm) layer (ML) and subsurface (20-30 cm) layer (SSL) respectively. The soil shows different types of textural class at each level of the soil. The surface layer (0-10 cm) of both forest and bun cultivation soil are sandy loam whereas the middle layer (10-20 cm) of forest soil is loamy and bun cultivation middle layer soil is sandy loam. Both forest and bun cultivation subsurface layer (20-30 cm) soil has the sandy loam type of soil.

At the beginning of the experiments microbial population (fungi and bacteria) increases both in bun cultivation and forest sites. Fungal population is higher at the surface layer (0-10 cm depth) in both the sites and decreases as the soil depth decreases. The middle layer (10-20 cm depth) of bun cultivation has higher bacterial population during the second year. In general, the surface layer recorded the maximum microbial population. The highest fungal population is recorded at bun cultivation site whereas highest bacterial population was recorded at forest site. The middle and subsurface layers (20-30 cm depth) of bun cultivation maintained a very low fungal population.

The optimum species diversity of fungi was observed at bun cultivation site at middle and subsurface layers whereas surface layer of forest site contributed the highest fungal population. Bacterial species diversity was highest at middle layer at both bun cultivation and forest sites. In general, peak microbial species diversity was noted at middle layers in both the sites. The maximum similarity index of fungal species was observed at a paired layers of middle and subsurface layers (ML x SSL) during the first year at both bun cultivation and forest sites, whereas during the second year maximum fungal species similarity index is observed at a paired layers of surface and middle layers (SL x ML) at bun cultivation site and surface and subsurface layers (SL x SSL) at forest site. The minimum similarity index of fungal species was observed at a paired layers of surface and subsurface (SL x SSL) at bun cultivation site and surface and middle layers (SL x ML) at forest site. The maximum similarity index of bacterial species was noted at

paired layers of surface and subsurface layers (SL x SSL) at both bun cultivation and forest sites. Minimum similarity index of bacterial species was observed at a paired layers of middle and subsurface layers (ML x SSL) at bun cultivation site and surface and subsurface layers (SL X SSL) at forest site.

The maximum distribution of fungal and bacterial species was noted at surface layer at forest site. Minimum fungal species distribution was observed at subsurface layer and minimum bacterial distribution was noted at middle layer of bun cultivation. In general, microbial distributions are higher at the forest site. The significant variation ($P \leq 0.05$) of microbial population and positive correlation ($P \leq 0.05$) among the surface, middle and subsurface soil layers were observed. Insignificant variation of fungal population was observed between the surface, middle and subsurface soil layers. Fungal population shows insignificant correlation ($P \leq 0.05$) between surface, middle and subsurface soils layers at different sites.

Bacterial population has negative correlation ($P \leq 0.05$) with fungal population at subsurface soil layer at bun cultivation site. A total of 45 fungal species has been isolated and identified, out of which *Aspergillus sp.*, and *Pennicillium sp.* are the dominant species. A fungal species like *Penicillium granulatum, P. Jensenii, Rhizopus oryzae* etc. and *Penicillium waksmanii, Phoma eupyrena, Colletotrichum dematium* etc. are some of the species that are isolated only at a single layer at bun cultivation and forest sites. A total of 7 bacterial species were isolated where *Arthrobacter sp.* and *Bacillus sp.* are

the dominant species. *Azotobacter sp.* was isolated at middle soil layer at forest site and *Arthrobacter sp.* at surface soil layer at forest site.

In general bun cultivation site shows higher soil temperature range than the forest site. Soil moisture content was higher during the cropping season and decreases at the post harvest at bun cultivation site. There is no much different in soil moisture content at surface, middle and subsurface layers in both the sites.

The soil pH decreases from pre-sowing to post harvest. Higher pH was noted at surface layer at bun cultivation site and middle layer at forest site. Surface layer contributed higher soil organic carbon and significant variation ($P \leq 0.05$) between surface, middle and subsurface layers was observed at both bun cultivation and forest sites.

Burning increases the soil total nitrogen while decreases during the post harvest. Surface layer of bun cultivation site has higher soil nitrogen content as compared to forest site. There is a significant variation ($P \leq 0.05$) of soil nitrogen content between surface, middle and subsurface layers at bun cultivation site and positive correlation ($P \leq 0.05$) of soil nitrogen content between middle and subsurface layers at forest site was observed.

The peak soil nitrogen content was observed at bun cultivation site during pre-sowing and inconsistent distribution of soil nitrogen in each monthly sampling was noted at surface, middle and subsurface layers at both the sites. The inconsistent distribution of soil phosphorus content was observed at both bun cultivation and forest sites during first year whereas significant variation ($P \leq 0.05$) between surface, middle and subsurface layers

was also observed. Higher increases in soil phosphorus content was noted at the middle and subsurface layers of bun cultivation site during the first year and higher decreases in soil phosphorus was noticed at surface layer at forest site during the second year. The surface layer showed highest soil exchangeable potassium and decreases as the soil depth decreases at bun cultivation site inconsistent distribution of soil exchangeable potassium was observed at forest site.

The soil microbial biomass carbon (C_{mic}) shows slight increases at the initial stage where higher microbial biomass carbon (C_{mic}) was observed during post harvest at both bun cultivation and forest sites. Bun cultivation has higher microbial biomass carbon (C_{mic}) accumulation at soil subsurface layer whereas middle layer showed high accumulation at forest site. Inconsistent distribution of soil microbial biomass carbon (C_{mic}) during monthly soil sampling was noticed at both bun cultivation and forest sites and insignificant variation between same layers of different sites was also observed. The subsurface layer at bun cultivation site showed insignificant correlation of soil microbial biomass carbon (C_{mic}) with surface and middle layers but there is a positive correlation of soil microbial biomass carbon (C_{mic}) with soil microbial population and dehydrogenase enzyme.

There is an inconsistent distribution of soil dehydrogenase enzyme during the monthly soil sampling at bun cultivation site whereas surface soil layer has maximum activities at forest site and decreases as the soil depth decreases. Soil phosphatase enzyme increases rapidly during pre-sowing and decreases as the maturity of the soil at bun cultivation site.

Higher soil phosphatase enzyme activity was recorded during the first year and the peak soil phosphatase enzyme activity was observed at the surface soil layer at both bun cultivation and forest sites. Soil urease enzyme activity increases during the cropping season and the highest soil urease enzyme activity was observed at the surface soil layer in both the sites. Soil urease enzyme showed an inconsistent accumulation during the monthly sampling at forest site.

The significant variation of soil enzymes (dehydrogenase, urease and phosphatase) between surface, middle and subsurface layers at bun cultivation and forest sites was observed. The surface soil enzymes (dehydrogenase, urease and phosphatase) showed a positive correlation with middle and subsurface soil enzymes in both the sites.

Local cropping management (BUN) enhances the soil nutrient availability, microbial biomass carbon (C_{mic}) and increases the soil microbial (fungal and bacterial) population.

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10TH APRIL, 2006

I, M. KHONGSAI, hereby, declare that the subject matter of this thesis entitled "Studies on the effect of "Bun" cultivation on soil microbes and their activities" 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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General Introduction

Shifting cultivation or Jhumming, variously known as rotational bush-fallow agriculture, swidden cultivation, or slash and burn cultivation, is an ancient form of agriculture still commonly practiced in many parts of the humid tropics, which is a system consisting mainly of cutting, drying, burning and clearing of the forest where plantation of the crops has been processed on the cleared area. As the nutrient status falls and the crops become poorer in production, the area is left as fallow to grow secondary vegetation until the forest regenerates. Within India, it is still widely practiced predominantly in the hill tracts of eastern and Northeast India. Although it showed that, far from being primitive and inefficient, jhum is an indigenous system of organic multiple cropping well suited to the heavy rainfall areas of the hill tracts (Ramakrishnan, 1992).

The practice of shifting cultivation is well known amongst the hill tribes of India especially in the Northeast region. Although shifting cultivation is understood by many workers from various fields of studies but in the context of the Khasi of Meghalaya, it exists in various forms. Besides the generally known slash and burn practice of jhum cultivation, there is a special type of shifting cultivation practiced particularly in the Khasi and Jaintia Hills of Meghalaya known as "bun" strips cultivation for potato and other crops. This is different in form from the general open slash and burn method but considered to be more damaging to the soil and water regimes. It is perhaps unique in the whole world. The practice consists of the cutting and collection

of trees, branches, bushes, dried grass, etc. and heaping them up in strips of 3 by 8-10 ft. size. The bun strips are covered by soil and upturned sods to which fire is lighted for burning the collected materials in the heaped up strips. The effect is a smothered burning with lots of smoke produced. It is to particularly note that these strips are laid down along the slope instead of across the slope. The soil is dug and scraped beside the strips themselves in order to heap up the strips. The portion where the dug up soil is done is generally reduced to the sub-soil, which enhances the momentum of the flowing water around them. As this process goes on, these portion of the dug up soil become deeper and bigger and ultimately from small to medium-sized gullies in between the bun strips. No actual study has been undertaken about the loss of the fertile topsoil and other related matters in connection with "bun" cultivation. However, from general observation, it may be deduced that the destruction caused by this practice in terms of soil and water erosion is tremendous, as the heaped up soil is dug up while harvesting potato and other crops, leaving open the raked up soil for easy erosion along the slope.

According to the experimental finding of the Indian Council of Agriculture Research Complex at Shillong, the average soil loss in shifting cultivation is estimated at about 41 tonnes per hectare annually. This works out to about 10.25 million tonnes of soil in the jhum cycle of five years or so. Similarly, the same experiment has shown that along with the soil loss, there is also corresponding loss of soil nutrients at an estimated 175.75 tonnes of organic carbon, 26.5 tonnes of P_2O_5 (phosphate) and 1750 tonnes of K_2O (potash) in the same period (Patiram, 1995).

Bun cultivation is practice in three different ways according to the land distribution. First, the Syiem system where the king, owner of the land distributes land. Secondly, the village head system where the leader of the clan earmarks land for the village and thirdly, the individual owner who have the land in the name of joint families. The first two categories are orthodox and never go for permanent cultivation. However, the third category is adaptable.

Bun cultivation has a great significant on agricultural practices where, the land which is previously burned will have even surface than other moraine areas as the stones and other particles are easily removed from this area, minimum labour is required for bringing large area or forest under cultivation, no fertilizers are required, so even a poor man can effort to cultivate and the crops are usually strikingly free from disease, as the soil is not infected by disease organisms. In agro-ecosystems, fallow is the principal means for restoring soil fertility after cultivation and maintaining agriculture yields (Kleinman *et al.,* 1995).

Nearly 38.4 percent of the land utilisation area is under the Neolithic agricultural practice of 'Bun' cultivation. This kind of traditional agricultural practice has further aggravated the deterioration of soil capability. In the process, not only did the land convert into wasteland, the soil characteristics can no longer support other potential traditional crops like sweet potato, flaminga spices, local millet etc. Species capable of adapting to the agriculture landscape may be limited directly by the disturbance regimes of

grazing, planting, harvesting and indirectly by the abundance of plant and insect foods (Alison and Pierre, 1995).

The shifting cultivation has other effects on the ecosystem, most of which are associated with the use of fire. Besides converting plant materials to ash, fire also stimulates the shedding and germination of seeds. It also causes impairment to water bodies, the biological resources and the atmospheric condition. The burning of vegetation also greatly reduced the availability of nutrients and increases the incidence of floods as well as accelerating siltation (Wirawan, 1993). Fire is a natural disturbance that may result in changes in the amount of composition of soil nutrients (Knapp and Seastedt, 1986; Garcia and Rice, 1994) and increases plant productivity by increasing photosynthetically active radiation and soil temperature, especially when it occurs in the early growing season (Hayes and Seastedt, 1989). Although burning has been reported to have no consistent long-term effect on organic C or total N content in the surface 15 cm of grassland soils, reductions in organic matter content and the size of the microbial biomass in the surface few cm are commonly recorded (Ojima *et al.,* 1994; Blair, 1997).

Agriculture productivity is based on nutrients mineralized from the liable soil organic matter fraction, especially under low external input farming prominent in the humid tropics (Noordwijk *et al.,* 1997). Aside from the nutrients released by mineralization, organic matter may improve the availability or solubility of inorganic nutrients. Agriculture soils receive nutrients in the form of organic matter, manure, chemical fertilizers and through weathering of the parent rocks. Soil is exposed to nutrient loss

through run-off, leaching and crop harvest. Physico-chemical characteristics of agricultural soils are also determined by the type of crop and agricultural practices (Fujisaka, 1990, Lewis *et al.,* 1991).

Management practices like crop rotation, mulching tillage and application of fertilizers and pesticides may have diverse effects on various enzymes and microbial activities of soil (Ladd, 1985; Dick *et al.,* 1987; Tabatabai, 1994a). Burning and N fertilization may affect soil enzyme activities and management practices that minimize the addition of organic matter to soil may reduce enzyme activities and thus affect the ability of soils to supply the nutrients for plant growth. Therefore changes in enzymes and microbial activities could alter the availability of nutrients for plant uptake (Dick, *et al.,* 1988a), and these changes are potentially sensitive indicators of soil quality (Dick, 1994). Soil microbial biomass and numbers of microorganisms were influenced by and exhibited qualitative changes in response to, long-term fertilization. Crop rotation also exerted effects on chemical and microbial properties of the soil (Belay *et al.*, 2002).

Physico-chemical characteristics of agricultural soils are also determined by the type of crop and agricultural practices (Fujisaka, 1990, Lewis *et al.,* 1991). In agriculture ecosystems, the community of soil microorganisms and other organisms is influenced direct or indirect by cultivation, the use of organic and inorganic fertilizers and the application as well by environmental pollutants (Bloem *et al.,* 1994; Matson *et al.,* 1997).

Soil is a fundamental component of agro-ecosystem, it act as biological entities with complex biochemical reaction (Dick, 1994). Analysis of soil is very important for enumeration of microbial species diversity, function and activities of soil microorganisms. The size and activity of the microbial populations depend on quantity and quality of soil organic matter, soil texture, soil pH and other properties of soil (Kaiser *et al.*, 1992).

The physical and chemical properties of soils can directly influence the structure, spatial distribution and activity of microbial population and enzymes in soils, which are potential early indicator of soil health and quality (Schnurer *et al.,* 1985; Dick, 1994). The differentiation between the different pools of enzyme activities in soils is important, because enzyme activities of microbial biomass can be used as indices of nutrient cycling and the health of agricultural ecosystems (Klose and Tabatabai, 1999). Soil microbial populations may act as early indicators of changes in soil quality as they can respond much rapidly than soil C or N (Kennedy and Papendick, 1995). It contributes to the maintenance of soil quality through decomposition, nutrient cycling and availability and soil aggregation.

Soil organic matter fractions that are considered important include microbial biomass, organic C and N. Potentially mineralizable C and N may be good indicators of changes in soil organic C and N induced by crop rotation, fertilization or tillage practices since they represent the more active fraction of soil organic matter (Campbell *et al.,* 1989). Long-term cropping system practices would be expected to affect these fractions of soil organic matter (SOM).

The diversity of plant communities in forest and agro-ecosystem has received a great deal of scientific attention but microbial diversity tends to be still ignored. Dick (1992, 1994) described that soil is a fundamental component of agro-ecosystem; it acts as a biological entity with complex biochemical reaction. Little is known of the importance of functional diversity of soil microbial communities for the sustained functioning of terrestrial ecosystems (Beare *et al.,* 1995; Moore *et al.,* 1995; Pankhurst *et al.,* 1996). Limited information is available in the northeast region regarding the importance of microbial diversity in the functioning of soil (Pankhurst *et al.,* 1996; Giller *et al.,* 1997). However, meager information is available on distribution of soil organic matter, nitrogen, phosphorus, and microbial population and enzyme activities under hilly region in general (Nayak and Srivastava, 1995), while no significance attempt has been made so far to study the microbiology under traditional agricultural soil system of the region in particular.

In agriculture ecosystems, the microbial communities are influenced direct or indirect by cultivation, the use of organic and inorganic fertilizers and the application as well by environmental pollutants (Bloem *et al.,* 1994; Matson *et al.,* 1997). The debris from primary producers accumulates on the soil surface and also within the mineral horizon along the network of roots and rhizomes. Beside this, a secondary addition of organic matter comprises microbial remains, bacterial cells, fungal hyphae and mineral corpses and faeces that have in the main are produced during the decomposition process in the litter layer. The substances that are added in the soil are continuously

being exposed to various microbial activities, which ultimately release the nutrients locked up in the debris through decomposition. The microorganisms in the soil are also essential for their participation in the soil formation, work conservation and sometimes biological control of certain pathogens. By burning, the vegetation cover is completely destroyed. The ash, which is formed after burning the area, is used up by the crops planted in first or second year of burning. But much of this ash is lost by leaching or erosion in such area. Thus such area becomes very poor in nutrients. Finally, the entire forest system is destroyed by Jhumming and it takes a pretty long time to be substantiated by a vegetation of the original kind. Burning also reduced microbial diversity and most of enzyme activities (Xiaoyun *et al.*, 2000).

The adverse climatic conditions added with poor soil quality and small farm holdings are the major limitations for the agriculture and other land use activities. The whole system of agriculture depends in many important ways on microbial activities and there appears to be a tremendous potential for making use of microorganisms in increasing crop production. Microbial encompasses a spectrum of microscopic organisms and plays an important role in nutrient acquisition for plants (N, P and K) and other microelements. Immobilization of nitrogen (N) by soil microbes affects availability to plants after fertilization (Raison *et al.*, 1992) but the magnitude of this effect can be limited by low availability of carbon (C) for microbial growth.

The size and activity of the microbial populations also depend on quantity and quality of soil organic matter, soil texture, soil pH and other

properties of the soil (Kaiser *et al.*, 1992). The primarily C limitation of microbial populations and the importance of easily decomposable organic materials for soil microbial activity (Friedel *et al.,* 1996) are mainly due to higher urease activity.

In agro-ecosystem, human intervention may affect soil biota and therefore, it has a crucial impact on system productivity and its maintenance. Therefore, the depletion of organic matter level in heavily cultivated agricultural situations could considerably affect the future productivity of some soils. Since microorganisms can act as an indicator to show the fertility level and quality of soil, the assessment and precise study on microbial composition, their activity and quality of soil is necessary. By keeping the various aspects as depicted above, the research has been carried out under the following heads:

1. Physico-chemical characteristics of soil (moisture content, pH, organic carbon, N, P and K) in Bun cultivation and Forest soil.
2. Diversity of soil microorganisms (fungi and bacteria) in Bun cultivation and Forest soil.
3. Soil microbial Biomass carbon ($_{Cmic}$) in Bun cultivation and Forest soil.
4. Estimation of soil enzymes (dehydrogenase, urease and phosphatase) in Bun cultivation and Forest soil.

Review of literature

Beets (1990) observed that the main feature of shifting cultivation is the avoidance of the very common tropical problem of soil exhaustion by shifting to a new piece of forest land as soon as the yield of the existing land declines. The technique used by shifting cultivators is to let the fallow accumulates nutrients and mine these nutrients during the cropping period. This system can be optimized by having the correct length of fallow period to accumulate the necessary nutrients for the cropping period.

Ramakrishnan *et al.*, (1992) observed that the fallow periods last anywhere from less than 10 to 30-40 years and the yields decline with decreasing length of the cropping cycle in Northeast India due to decreased soil fertility and increased weed pressure. The increasing population pressure throughout much of the tropics has resulted in shortened fallow periods, resulting in excessive soil erosion, declining fertility, and greater weed problems.

Schauder and Karl, (1993) studied the long term trapping efficiency of a vegetated filter strip under agriculture use and observed the trapping efficiency for a long term but large over-estimate the efficiency for the short term experiments.

Wirawan, (1993) observed the effects on the soil; shifting cultivation has other effects on the ecosystem, most of which are associated with the use of fire. Besides converting plant materials to ash, fire also stimulates the shedding and germination of seeds. It also causes impairment to water

bodies, the biological resources and the atmospheric condition. The burning of vegetation greatly reduces the availability of nutrients and increases the incidence of floods as well as accelerating siltation.

Tilak *et al.,* (1995) reported that various agricultural management practices such as cropping system, cultivation practices, soil organic amendments and pesticide application alter the microbial dynamic in the agro-ecosystem.

Buchi, (1997) studied the shifting cultivation and its impact on forest ecosystem and observed that the effects of fire on biological resources relate to the losses of the type and the numbers of species of both plants and animals. Repeated burns within the same area can change the forest cover into single-layer vegetation dominated by the fire-resistant *Imperata* grass.

Lee, (1997) studied the effects of shifting cultivation on soils in Sarawak, and observed that, the nutrient contents and soils in a chronosequence of sites of varying period after shifting cultivation. The assumption of the approach is that all study sites had relatively uniform soil and site conditions before the disturbance.

Soil nutrients

Westerman and Kurtz (1973) suggested that increased crop soil N uptake was due to stimulation of microbial activity through various agricultural management which increased availability of soil Nitrogen and thus making more soil N available for plants.

Cassman and Munns, (1980) study the soil physical properties as affected by soil moisture, temperature and depth and discovered a significant relationship between soil moisture and temperature and its consequent effect on net N mineralization.

Norman *et al.,* (1984) observed that the largest pools of nutrients are the vegetation and uppermost layer of soil where many nutrients are lost in the process of land preparation. Thus more shallowly rooted herbaceous crop species are unable to recover nutrients leached a meter or more into the soil through cultivation.

Rice *et al.,* (1986) concluded that after 5 to 10 years of cropping, soil organic matter concentrations apparently approached new steady-state levels. This concentration was greater in the non-tillage system of cropping due to relative accretion of organic matter on the soil surface.

Insam *et al.,* (1991) found that soil nutrient contents (N, P, K) were significantly correlated with microbial biomass C in the crop yield of three ultisols.

Tiessen *et al.,* (1992) observed that soil mineral N contents (NO_3 and NH_4) were highest under natural vegetation and lowest after cultivation that recovered with increasing fallow period. The root biomass from native vegetation decomposes upon cultivation and contributes to the carbon and nitrogen pools. The relatively slow rates of additions of organic matter and nutrient uptake by the re-establishing native vegetation are more effective in building up and maintaining soil organic matter levels than the periodic

additions of crop residues under cultivation. Nutrient limitations on the cultivated sites prevent organic matter maintenance or increase unless carbon inputs are accompanied by substantial fertilization.

Wienhold and James, (1992) studied the effect of prescribed fire on nitrogen and phosphorus in Arizona chapparal soil plant systems and observed that ammonium production increased and nitrate production decreased fire, cumulative N mineralization did not differ between the pre-burn and post-burn soils. Fire resulted in the loss of N and a redistribution of N and P to system components that are highly susceptible to erosion.

Halenda and Christine, (1993) observed that clearing and burning of logged over forest for shifting cultivation resulted in a complete removal of all living vegetation. The effects of clearing and burning on the soil within a month after burning, were increase in C, K, Ca and Mg; decreases in N; and no change in P. Increases were the results of additions from ash and unburned organic matter leaching into the soil. Decreases in N were attributed to volatilization and the leaching of N from the soil profile. The establishment of a cover of shrubs and secondary species occurred one and a half years after abandonment, was.

Garcia and Rice, (1994) found that microbial activity was a regular dynamics in tall-grass prairie; net mineralization increased inorganic at the beginning of the growing season, and the N returned to the soil upon plant senescence at the end of the growing season was conserved by microbial immobilization. There is an increased in inorganic N to enhanced N

mineralization due to higher soil temperature after the removal of detritus during the first year of burning and decreased after repeated burning. Most of the readily decomposable organic C added to soil is rapidly consumed by microorganisms (Van Veen and Kulkman, 1990; Ladd *et al.*, 1994). Analysis of soil biological processes regulating soil nutrient conservation is essential for defining sustainable production systems (Gupta and Malik, 1996).

Alegre and Cassel, (1996) studied the dynamic of soil physical properties under alternative systems to slash and burn and observed that half of the N remained in the system but losses of other nutrients were high, especially P and K. Most of the N loss occurs by volatilization during burning. Losses of P and K occur through wind erosion of the ash and by leaching during the first few heavy rains following the burning.

Soon and Arshad, (1996) found that the C/N and C/P ratios of soil organic matter decrease slightly with cropping. Exchangeable K was decreased by cropping systems containing a legume crop to a greater extent than those without a legume crop and most of the decrease occurred in the 0-15 cm depth. Continuous cultivation caused significant losses of C, N and P.

Boyer and Groffman, (1996) reported that the total Soil organic Carbon (SOC) was higher in forest than in crop systems, but the difference was restricted to the surface layers and concentrations of water-soluble and bioavailable SOC were higher in agricultural soils than in forest soils. Agricultural soils generally have lower amounts of total and microbial C than

forest soils and may support equal or greater rates of microbial activity than forest soils due to increased production of water-soluble C.

Ajwa *et al.,* (1999) observed that the pH values of the soils and total N and C were similar across sampling dates and were not significantly ($P<0.05$) affected by burning. The C-N ratio of the biomass in the unburned treatment was similar during the growing season but in burned treatment, this ratio varied widely and increased. Correlation coefficients between inorganic N and soil enzymes showed that inorganic N was negatively correlated (r=-0.4935, $P<0.001$) with urease enzymes activity, possibly due to low soil inorganic N caused by plant uptake and microbial mobilization. The significant effect of burning on urease activity may suggest that this enzyme can also be used as an indicator of microbial activity.

Kathleen *et al.,* (2001) found 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. This response suggests that plants in the P-limited site lack necessary P reserves to maximize growth.

Parham *et al.,* (2003) observed the pH values in manure-treated soils almost uniform in the soil profiles from 0-10 cm depth (4.2-5.7) with that of untreated control soil around pH 5. Manure application increased soil pH significantly. N and P levels are higher in manure-treated than untreated

soils. Cultivated soils presented higher amounts of total and available P and organic C than uncultivated soils (Borie and Rubio, 2003).

Soil microorganisms (Fungi and bacteria)

Dkhar and Mishra, (1987) observed that the microbial population was higher in the soils of permanent agriculture as compared to that of slash and burn type of shifting agriculture. They further reported that the soil of the valley land harboured maximum microbial populations followed by land agricultural and minimum in the soil of jhum land agriculture.

Nannipieri *et al.,* (1990) conducted a study on the relationships between enzymes activities, biomass and microbial growth in a red soil and found none of these measurements suitable for monitoring the microbial growth in the soil system.

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. The changes in temperature and water potential determine changes in microbiological activity, which in turn determines changes in the H+ budget and its physico-chemical properties (Conyers *et al.,* 1995).

Franzluebbers *et al.,* (1994) stated that soil fertility is markedly affected by microbial activity. The microbial biomass, which is a small fraction of the soil's total organic matter content, is a source and sink of nutrients and controls soil organic matter mineralization. Disturbing the biological equilibria with changes in the composition and activity of the micro-flora can damage

soil fertility both in the short and long term cultivation (Brookes, 1995). The specific impact of soil management, seasonal conditions affect an amount of soil microbial activity as well as enzymatic activities (Batra *et al.,* 1997).

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 effects on ecosystem process. Beare *et al.,* (1997) also describe the effects of changing land-use and agricultural intensification on the structure and diversity of decomposer communities in the tropics and suggest some priorities for future research. The specific impact of soil management, seasonal conditions affect an amount of soil microbial activity as well as enzymatic activities (Batra *et al.,* 1997).

Frey *et al.,* (1999) stated that microbial composition may be important determinant of soil organic matter decomposition rates and nutrient turnover and availability in agriculture soils. Tiwari *at al.,* (2002) also mentioned that microbial activities in soil, despite their importance in many of the soil processes, are frequently disturbed as shown by altered soil enzyme activities as a result of agricultural exploitation.

Fungal population

Deldauri, (1975) pointed out that the distribution of soil fungi is determined by the combined effect of moisture, temperature and organic carbon. In organic soil layer, the amount of hyphae showed seasonal

periodicity at three sites and this periodicity was correlated to the soil moisture content (Baath and Soderstrom, 1982).

Upadhyay and Rai, (1979) reported that *Trichoderma sp.* was recorded frequently and in high population from forest and some other containing high organic matter and low pH where *Aspergillus* dominated over *Penicillia* in cultivated soils.

Tiwari *et al.,* (1986) study the temporal and depth-wise variation in microbial population in Pineapple plantation soils and found that reversal in the depth-wise distribution of fungal population during winter in all the plantation soil appears to be due to the effect of low temperature during this period. The effect of low temperature was more pronounced in surface soils and probably due to insulation; fungi of deeper soils were least affected resulting into higher populations in deeper layers.

Shukla and Mishra, (1992) study the influence of soil management systems on the microfungal communities of Potato field and observed that maximum fungal propagules were recorded in valley land followed by terrace land and slope land soils. The numbers of fungal propagules were higher in surface soil, which decreased with increasing soil depth.

Jha *et al.,* (1992) found that the fungal and bacterial populations showed marked variations at different altitudes. In the more degraded forest stand at the lower altitude both the fungal and bacterial population showed a significant positive correlation with organic C (r=0.658, 0.735, respectively), whereas in the less degraded forest stand there was a significant correlation

between fungal population and organic C (r=0.835). At higher altitude fungal population was significantly correlated with soil moisture and organic C. Soil fungal and bacterial biomass significantly decreased ($P<0.05$) with the shift of vegetation from forest to grassland (Imberger and Chiu, 2001).

McLean and Huhta, (2000) study the temporal and spatial fluctuations in moisture affect humus micro-fungal community structure in microcosms and found 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. The microbial population changes after fertilization and fertilizers can directly stimulate the growth of microbial populations as a whole by supplying nutrients and may affect the composition of individual microbial communities in the soil (Khonje *et al.,* 1989; Sarathchandra *et al.,* 1989; Khamis *et al.,* 1990).

Kwa *et al.,* (2000) found that after the amendment of pine sawdust, soil pH had changed in both agriculture and forest site but a small decrease in the number of species was not statistically significant. *Trichoderma harzianum,* an important biological control fungus increased considerably after sawdust application whilst some fungi, such as *Penicillium sp., Pseudogymnascus roseus* and partially Mucorales decreased.

Parham *et al.,* (2003) observed that fungal population decreased significantly with increasing soil pH in soil samples taken in winter,

suggesting fungi are more competitive than bacteria in the acidic environment.

Bacterial population

Winogradsky, (1925), a soil bacteriologist systematically observed soil particles and record the types of bacterial colonies and cells found on them.

Feher, (1933) studied seasonal variation in bacterial numbers in several European forest soils. Separating particles into different size groups by centrifugation, he was able to observe that the majority of bacteria in the soil were either large cocci or coccobacilli, some of which resembled *Azotobacter*. It has been stated that from time to time, most of the bacteria that occur in soil are attached to soil particles and do not occur in large numbers in the soil dilution (Waksman, 1952).

Campbell *et al.*, (1971) found that temperatures, which fluctuated daily, reduce microbial organisms in the soil especially the population of bacteria. A reduction in the aggregate microbial population has relatively little effect on N mineralization due to sheer numbers of organisms responsible for mineralization. Soil moisture directly effects microbial activity and in turns effect soil nitrogen mineralization.

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.

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

Ogawa, (1996) observed that the soil biological property is related primarily to the microbial population. Clearing and burning also cause significant changes in soil microflora. Burning causes partial sterilization of soils followed by a flush of microbial population and eventually a decline approaching new equilibrium levels. A large proportion of bacteria are found after clearing. The total microbial population decreases during the dry season and also with mulching and fertilization. The soil microbial population thus appears to be closely tied up with carbonized substances.

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.

Lahav and Steinberger, (2001) showed that sequencing bacterial functional diversity in agroecosystems would be affected by plant growing stages and human activity (agriculture practice).

Biomass C (C_{mic})

Powlson *et al.,* (1987) showed 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. Henrot and Robertson, (1994) stated 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.

Anderson and Domsch, (1989) stated that the ratio of soil biomass C to organic C ratio is a good indicator of changes in microbial performance caused by environmental conditions. The structure and distribution of C in soil affect biological activity. Organic C and total N were highly correlated with dehydrogenase.

Gestel, (1992) found that microbial biomass carbon was highest in winter-sample soils and the lowest in those summer sampled soils.

Lavahun *et al.,* (1996) observed that there is no significant depth gradient in the plough layer (0-30 cm depth) for biomass carbon and soil organic C contents. The microbial biomass C to soil organic C ratio decreased with depth from maximum of between 1.4 and 2.6% to a minimum of between 0.5 to 0.7% at 70-90 cm in the soil. The surface soil layer (0-10 cm) had significantly higher microbial biomass C content than the subsurface layer (10-20 cm) and the microbial biomass C, N, P were highest in during winter and lowest during rainy season at both the 0-10 cm and 10-20 cm sample depths (Kusum *et al.,* 1996).

Hossain *et al.,* (1995) found that microbial biomass was increased 47% in the top 0-25 mm soil by N and P fertilizers two and a half years after a combined application of these nutrients, but there was no effect of the

combined application in the 25-50 mm depth and the amount of N contained in the microbial biomass was unaffected in both depths.

Maithani, (1996) revealed that values of microbial biomass could provide one of the most satisfactory estimates of the restoration of soil microbial populations. However, detailed information on qualitative changes of soil organic matter (SOM) during the seasons is still lacking (Andreux *et al.*, 1990).

Witter, (1996) found that more C had accumulated per unit C input in peat and sewage sludge-amended soils than in any other soils, suggesting that peat and sewage sludge were more resistant to microbial attack. The linear relationship between microbial C and cumulative C losses suggested that the significantly reduced biomass in the sewage sludge-amended soil was at least partially due to the presence of toxic substances (elevated heavy metal) in this soil and was probably not affected by somewhat low pH (5.3) in this soil.

Lovell and Jarvis, (1996) reported that soil microbial biomass responds rapidly in terms of activity to additions of readily available C (e.g. glucose). The size of the soil microbial biomass (SMB) in a lowland grassland soil was unaffected by changes in N input over 1 year but in the longer term (10 years) a smaller and more active biomass developed in response to regular inputs of N. Long-term fertilizer input appeared to have had the effect of increasing the active component of SMB without a corresponding increase in total biomass.

Goyal *et al.,* (1999) found that the amount of soil organic matter and mineralizable C and N increased with the application of inorganic fertilizers and farmyard manure. The urease and alkaline phosphatase activities of soils increased significantly with the combination of inorganic fertilizers and organic amendments.

Piao *et al.,* (2000) observed that soil microbial biomass C has inverse relationship with atmospheric temperature but no obvious relationship soil moisture and a negative correlation with soluble organic carbon. The highest microbial biomass C occurred in winter and the lowest in summer ranged from 231-723 μg g^{-1} dry soil. The marked changes in soil microbial biomass C at above 20^0C were ascribed to fluctuations of soil moisture, which were controlled by climatic factors and geomorphic conditions. It was concluded that the lowest amounts of soil microbial biomass C measured in the summer resulted in increases in soluble organic C due to higher rates of the former at warmer air temperatures. The surface soil layer (0-10 cm) had significantly higher microbial biomass C than the subsurface layer (10-20 cm) and negatively correlated with soil temperature and pH (Maithani *et al.,* 1996). Seasonal changes in soil microbial C at 0-10 cm depth were significantly correlated with total soil organic C (Piao *et al.,* 2001).

Parham *et al.,* (2002) observed that microbial biomass contents were the highest in the 0-10 cm surface soils, with considerable variations due to the sampling time. It decreased with increasing soil depth in most of the soils but manure treated soils shows a higher microbial biomass C in 20-30 cm than 10—20 cm depth soils. The percentage of total organic carbon

presented as C_{mic} decreased with the increasing soil depth, indicating changes in microbial community, C availability and growth condition in the lower soil profiles. The microbial biomass in both the surface and subsurface sandy loam soils was not significantly affected by the seasonal variation but activity increased by as much as 80% at the summer temperatures in the surface soil (Blume *et al.*, 2002).

Soil Enzymes

Frankenberger and Johanson, (1982) observed that the pH stability of soil enzymes was highly dependent on the soils being assayed and attributed the variation to the diversity of vegetation, microorganisms and soil fauna as sources contributing to the enzyme activity, and also on variation in protective sites.

Malcolm, (1983) studied the assessment of phosphatase activity in soils and found that the use of optimum pH in enzyme activity measurements provides a measure of the maximum potential activity of the enzyme under natural conditions.

Harrison, (1983) found that phosphatase activity in woodland soils is related to soil physical and chemical properties such as soil pH, contents of nitrogen, organic matter (0-5 cm depth), moisture and plant-available phosphorus.

Dick, (1984) found that the urease activity is significantly correlated with organic C in soils with upto 92% of the total variation in urease activities accounted for by organic C concentrations. The highest total urease

activities were obtained in soils under 4-year oats-meadow rotations and the lowest under continuous corn. The higher total activities under multi-cropping systems were caused by a higher activity of both the intracellular and extracellular urease fractions.

Reddy *et al.,* (1987) found that dehydrogenase and phosphatase enzyme activity was inhibited in a sludge-amended soil. The decline in enzyme activities in a sludge-amended soil is due to decrease enzyme synthesis associated with inhibited microbial growth than to direct enzyme inhibition by the metals.

Dick *et al.,* (1988a) observed a strong correlation between dehydrogenase activity and microbial biomass carbon (MBC), acid phosphatase with burning of the residue in the top 20 cm soil.

Baligar *et al.,* (1988) found very close association between soil phosphatase activity and moisture content, organic C and ammonium N concentrations in acid soils of the Appalachian mountain.

Tabatabai, (1994c) stressed that enzymes are usually more stable in the vicinity of the optimum pH for activity and that they are irreversibly denatured by extremes acidity or alkalinity.

Tiwari, (1996) studied that microbial activity in soil as a measured in terms of enzymes (viz. Urease, Dehydrogenase, Invertase, Phosphatase, Amidase, Cellulase enzymes assays), soil respiration and nitrification have been found to be correlated with dynamic of soil nutrients. He also observed that the widest range (40 fold) in enzyme activities for urease in various soils

and the narrowest range (1.5 fold) for phosphatase. Dehydrogenase (19 fold), biomass (7 fold), fungal population (18 fold) and bacterial population (24 fold) also varied greatly in various soil types. He also concluded that arable soils (rice) showed higher activities of urease and dehydrogenase enzymes as compared to the other soils. Microbial count was found to be highest in forest soil whereas agriculture soil showed a lowest value of the fungal and bacterial population.

Eivazi and Tabatabai, (1997) observed that microbial biomass is significant correlated with phosphatase and urease activities in the effect of long-term prescribed burning on enzyme activities in a forest ecosystem. The effects of long-term prescribed burning on enzyme activities in a forest soil and found that long-term burning of the forest floor debris always reduced the activities of all enzymes where burning treatments are more apparent than seasonal variability.

Amador *et al.,* (1997) found that phosphatase activity was highest in poorly drained soil and decreased as drainage improved and the activity increased as root mass increased within the soil fraction. Soil alkaline phosphatase followed the similar trends to organic matter and inorganic phosphorus where increase in content with increase moisture content and a decrease with prolonged flooding.

Hysek and Sarapatka, (1997) in their studies in forest soils showed that soil alkaline phosphatase activity correlated positively with organic C. Phosphatase activity was highest in poorly drained soil and decreased as

drainage improved. Phosphatase activity increased with soil pH in moderately well drained soil with maximal activity occurring at a pH 6.5 and 6.9.

Lyons *et al.,* (1998) observed soil phosphatase, the enzyme that transforms organic P to inorganic P 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.

Ajwa *et al.,* (1999) observed that long term burning and N fertilization of the tall-grass prairie soil reduced MBC and MBN relatively to the unburned-unfertilized treatment but increased activities of urease and acid phosphatase. The large input in dehydrogenase activity in all the treatments between June and August was likely due to C input (via root mass) during this period that enhanced microbial activity. Enzymes are more sensitive indicators of burning disturbances in pristine ecosystems than microbial biomass. Long-term burning appears to alter the rate of organic matter turnover and therefore, affect microbial biomass and the production of enzymes. Many soil enzymes are immediately responsive to soil disturbances or restoration.

Xiaoyun *et al.,* (2000) observed that soil dehydrogenase activity was affected by sub-treatment in all season. Burned plots had markedly lower dehydrogenase activity than any of other treatments. Dehydrogenase activity was higher in the winter rainout and lower in the control and summer rainout

plots in spring. Phosphatase activity was generally highest in the winter rainout plots and lowest in the burn and control plots during the summer and spring. Enzymatic activities were not influenced by intense-grazing treatment, but by substrate treatments on most sampling dates.

Martinez and Tabatabai, (2000) observed that acid phosphatase was predominant in the control plots (pH 4.9) and liming increased the activities of the alkaline phosphatase and phosphodiesterase. The significant increase in soil pH by lime applications may stimulate the microbial population and diversity, resulting in an increase in the soil enzyme activities and thus affecting soil nutrients.

Parham *et al.,* (2002) found that inorganic phosphatase and dehydrogenase were significantly highly in the soil treated with cattle manure. Enzyme activity was significantly higher at the surface and decreased with the increasing soil depth but dehydrogenase activity has higher activity in 20-30 cm than 10-20 cm in manure treated soils. Increasing soil temperatures at the top level causes a reduction in the dehydrogenase activity.

Consuelo and Teodoro, (2002) found that the dehydrogenase activity of forest soil in autumn was almost double that 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.

Chapter- I

Description of the Study sites and Climate

1. 1. Location

The present investigation was conducted at agricultural field of Mawlali village and a forest stand (as control) in areas of Nongkrem, East Khasi hills district, which is 25 kms away from Shillong, the capital of Meghalaya. The topography of the two sites was similar and both of them were adjacent to each other. The Nongkrem areas are covered with agricultural land, where bun cultivations are extensively practiced by the villagers. The study area lies between $25^0 35^l$ N and latitude and $91^0 47^l$ longitude and at an altitude of 1785 m MSL.

1. 2. Climate

The climate of the study area is of sub-tropical monsoonic type, largely controlled and influenced by the south-west monsoons and north-east winter winds. On the basis of general meteorological conditions, the year is divisible into four seasons. The spring season starts from the month of March to April, summer (rainy) from May to September, autumn from October to November and winter from December to February. The average annual rainfall in 2001 and 2002 ranges from 2.5 mm to 2200 mm while the maximum and minimum temperatures vary from 22 – 25^0 C and 5 to 20^0 C respectively. Similarly, the average humidity ranged from 45% - 98%.

1. 3. Soil

The soil shows different types of textural class at each level of the soil. The soil of forest and bun cultivation surface layer (0-10 cm) is sandy loam whereas the middle layer (10-20 cm) of forest soil is loamy and bun cultivation middle layer soil is sandy loam. Both forest and bun cultivation subsurface layer (20-30 cm) soil has the sandy loam type of soil (Table 1.1).

Table 1.1. Sites characteristics of Bun cultivation (BCU) and Forest (FS) site at Surface (SL), middle (ML) and subsurface (SSL) layers.

Soil Properties	**BCU**			**FS**		
Soil textures	***SL***	***ML***	***SSL***	***SL***	***ML***	***SSL***
Clay (%)	*10.8*	*14.1*	*8.1*	*8.0*	*18.5*	*13.8*
Silt+Clay (%)	*37.6*	*44.4*	*35.5*	*41.0*	*57.5*	*37.6*
Silt (%)	*26.7*	*30.3*	*27.3*	*32.9*	*39.0*	*23.8*
Total Sand (%)	*62.3*	*55.5*	*64.4*	*58.9*	*42.4*	*62.3*
Soil textural Class	**Sandy loam**	**Sandy loam**	**Sandy loam**	**Sandy loam**	**Loamy**	**Sandy loam**

1.4. Vegetation

The natural forest in this region is restricted to small areas. The forest stand was dominated by tree species like *Pinus kesiya* followed by *Myrica esculentum, Callicarpa arborea, Myrica esculentum, Schima wallichii.* The herbaceous weedy like *Ageratum conyzoides*, *Eupatorium odoratum* and *Osbeckia crinata* were common.

1. 5. Soil sampling

Soil samples were collected randomly by a soil sampler from five different places at each site from a depth of 0 -10 cm, 10 – 20 cm and 20 - 30 cm every month. Soil samples collected in the month of May was considered as pre-sowing while that collected in the month of December was considered as post harvest soil sample. The soil sampler was cleaned and sterilized after each sampling. All five samples collected randomly were mixed thoroughly to obtain a composite sample. The samples were brought to the laboratory on the same day and kept in the refrigerator at 4^0 C until they were used.

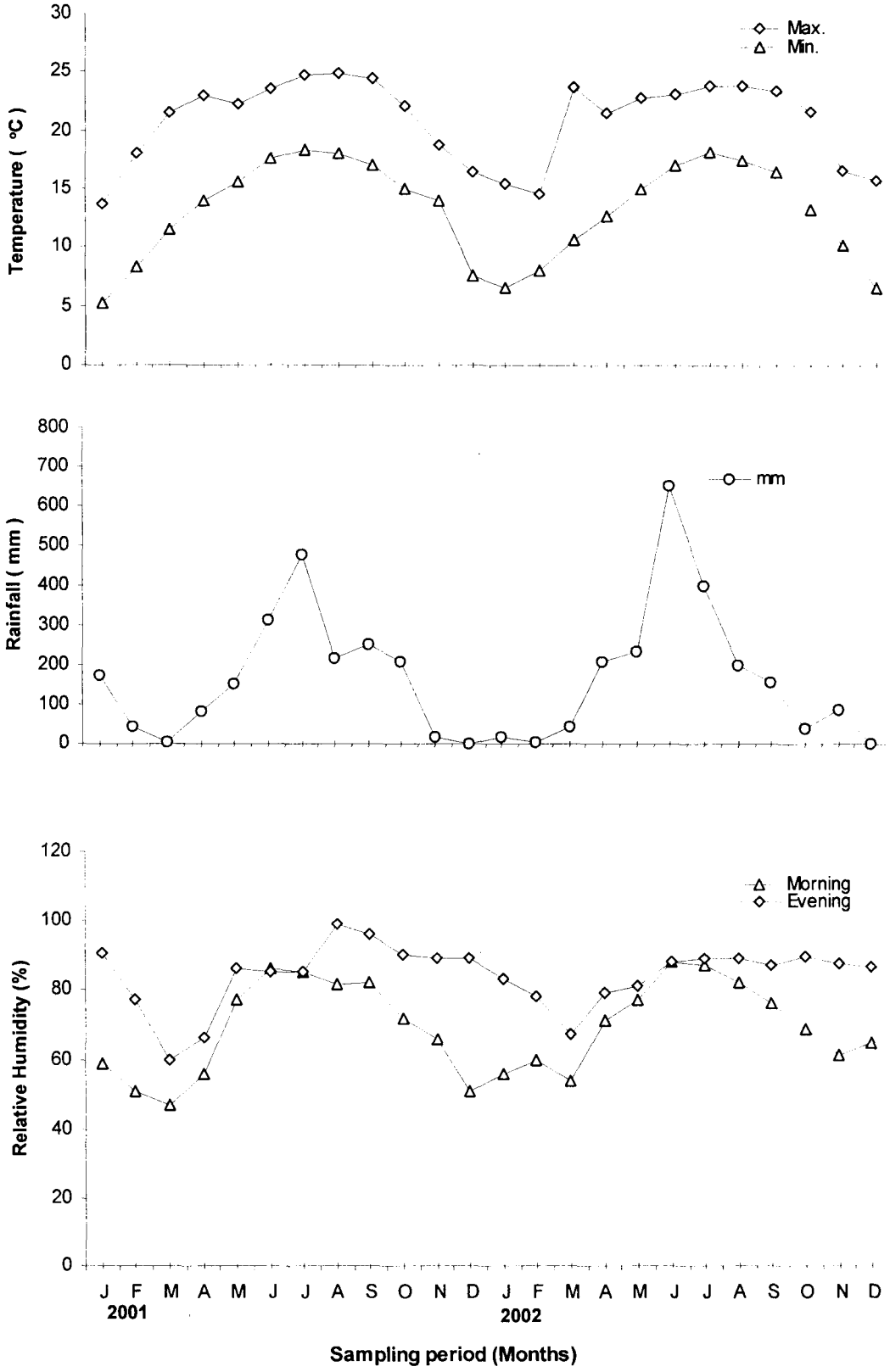

Fig.1.1. Climatogram showing mean monthly variation of temperature, rainfall and relative humidity of the study sites.

Plate 1.1. Photograph showing the preparation of field for bun cultivation.

Plate 1.2. Photograph showing the bun cultivation of Cabbage crop.

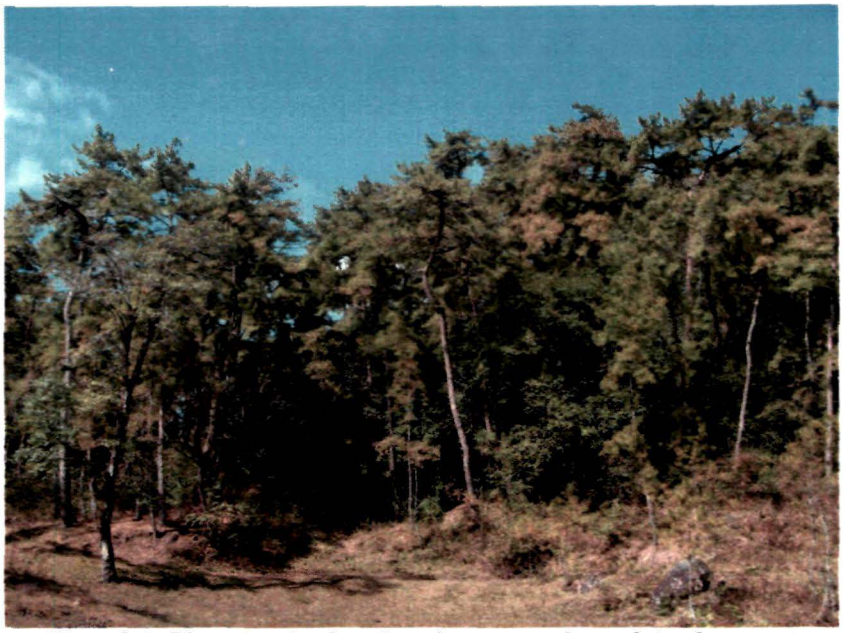

Plate 2.1. Photghaph showing the outer view of the forest at Nongkrem area.

Plate 2.2. Photograph showing the inner view of the forest at Nongkrem area.

Chapter- II

ANALYSIS OF PHYSIC0-CHEMICAL PROPERTIES IN BUN CULTIVATION AND FOREST SOIL

2.1. Introduction

Soil physico-chemical characteristic influenced level of biomass and the activity of microorganisms. Seasonal changes in soil moisture, soil temperature and C input from crop roots, rhizosphere products and crop residues can have a large effect on soil microbial biomass and its activity (Ross, 1971), which in turn effect the ability of soil supply nutrients to plants through soil organic matter turnover (Blonde and Roswall, 1987; McGill *et al.*, 1986).

Burning of plant debris is a natural disturbance that may result in changes in the amount of composition of soil nutrients (Owensby and Wyrill, 1973; Knapp and Seastedt, 1986; Garcia and Rice, 1994) and increases plant productivity by increasing photosynthetically active radiation and soil temperature, especially when it occurs in the early growing season (Hayes and Seastedt, 1989). Agriculture soils receive nutrients in the form of organic matter, manure, chemical fertilizers and through weathering of the parent rocks. Soil is exposed to nutrient loss through run-off, leaching and crop harvest. Physico-chemical characteristics of agricultural soils are also determined by the type of crop and agricultural practices (Fujisaka, 1990, Lewis *et al.,* 1991). Physical properties of soil are considered as important factors, which affect the growth and activity of soil microorganisms (Frankenberger and Dick, 1983). The positive effect of cropping on physical, chemical and biological soil properties are related higher C inputs and

diversity of plant residues returned to soils (Biederbeck *et al.*, 1984; Havlin *et al.*, 1990; Varvel, 1994; Miller and Dick, 1995; Friedel *et al.*, 1996; Robinson *et al.*, 1996).

The physical condition and chemical characteristics of soil play an important role in determining the environment in which biological processes take place and can be defined at different spatial and temporal scales (De Vos *et al.*, 1994). So the physico-chemical properties can determine the suitability of a soil for production of different crop plants (Brady, 1995). The chemical characteristics make a significant contribution in determining the quality and may even determine the maximum quality of a particular soil (Hassink, 1997).

Nutrients in soil exist mostly in organic and inorganic form, both of which are important source of plant and microbial uptake (Lyons *et al.*, 1998). The soil P availability is controlled by environmental conditions such as soil organic matter, moisture content and aeration, which influence microbial activity and eventually transformations of phosphorus (Hedley and Stewart, 1982; Bloom *et al.*, 1985; Read, 1991).

The Conversion of forest ecosystems to agriculture marked a decline in the amounts of total and labile soil organic C. However, analysis of soil organic C changes induced by forest conversion to agriculture has generally been restricted to surface horizons and to pools of total microbial biomass soil organic C. The nutrient transformations associated with traditional shifting cultivation form the basis of agriculture productivity in large areas of semi-arid tropics. Changes in the fertility occur so slowly during the complete

cycle of shifting cultivation that it is difficult to monitor them in field experimentation (Tiessen *et al.*, 1992). The dynamics of biotic components, which are affected by environmental factors such as the characteristics of the soil and temperature, together with the quality of organic matters applied to soil, are of great agronomical importance. Distributions of soil organic carbon and total N contents over the soil profiles indicated movement of soluble manure compounds from the surface to lower soil profiles and a significant proportion of manure-P may also be found in the deeper soil profiles (Parham *et al.,* 2002).

In agriculture soils all forms of P are potentially available to plants. The availability degree will depend on the solubility and structure of chemical forms, on the susceptibility to microbial attack as well as on the soil-root environment. The higher levels of labile P found in forest soils in comparison to agriculture ones together to C/Po ratios are suggesting a faster P cycling in forest ecosystem (Borie and Rubio, 2003).

The cropping systems can influence important soil properties such as soil structure and density, soil pH, the quantity and of nutrient cycles within the soil profile (Tabatabai *et al.,* 1992; Miller and Dick, 1995; Senwo and Tabatabai, 1998). Cropping relies on soil microorganisms to form and decompose soil organic matter as a continuous nutrient supply (Subler and Kirsch, 1998). Bioactive soil organic C is a direct and stable reservoir of carbon and nitrogen that consists of living and dead organic materials subject to rapid biological decomposition (Hountin *et al.,* 1997, Knops and Tilman, 2000; Lu and Pignatello, 2002). Under normal growing conditions,

green plants rapidly reduce nitrate to intermediate compounds that are subsequently converted into amino acid, protein and nucleic acids using the energy obtain from light (Barrett and Burke, 2000; Delgado, 2002). Nitrogen in the ammonium form is strongly held by negatively charged clay and soil organic matter colloids until adsorbed by plants or converted to SOM components or nitrate form by bacteria (Delgado, 2002).

2.2. Methodology

2.2.1. Soil temperature

Soil temperature was measured by using soil thermometer. The soil temperature was taken at monthly interval at surface, middle and subsurface layers.

2. 2.2. Soil moisture content

Moisture content of the soil was determined by oven dry method. 10 g of freshly collected soil sample was kept in a hot air oven at 105^0 C for 24 hours. The soil was weighed again after oven dried. Three replicates were maintained for each soil samples.

The percentage of moisture content was calculated by the formula below.

$$\textit{Moisture Content (\%)} = \frac{\textit{(W1 – W2) g}}{\textit{W1 (g)}} \times 100$$

Where,

W1 = Initial weight of soil before drying

W2 = Weight of soil after drying

2. 2.3. Soil pH

10 g of freshly collected soil was taken in a 100 ml beaker containing 50 ml of distilled water. The soil water mixture was then stirred for 20

minutes on a magnetic stirrer. The solution was kept overnight to allow the soil sample to settle. Three replicates were maintained and the pH was read by using electronic digital pH meter.

2.2.4. Soil organic carbon

Soil organic carbon was estimated by the method of Anderson and Ingram, (1993). Weighed 1 g of ground soil (<0.15 mm) into a 100 ml conical flask (if the soil was dark or was suspected to be high in organic matter used about 0.5 g). 10 ml of 5 % potassium dichromate solution was added to the conical flask and allowed it to completely wet the soil or dissolved the standards solution. 20 ml of concentrated sulphuric acid was then added from a fast burette and swirled the mixture gently, allowed to cool it. To this 50 ml of 0.4 % barium chloride was added. The mixture are swirled thoroughly and then allowed to stand overnight. The Blank was run without soil. The supernatant was then transferred into a colorimetric cuvette and measured the optical density by using Hitachi (220) spectrophotometer at 600 nm.

The percentage of soil organic carbon was calculated by the formula below:

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

Where, W = Weight of soil.

K = Value obtained from the graph.

2.2.5. Total nitrogen

Soil total nitrogen was estimated by using the method of Jackson, (1973). Weighed 1 g of dried finely ground soil (> 2 mm sieved) into a

Kjeldahl digestion flask. To it 6 ml of concentrated sulphuric acid and one piece of kjeltablet was added. This was then digested in a block digestor for about half an hour till the colour turn completely green. The flask was allowed to cool and diluted with 50 ml distilled water. The solution was filtered with Whatman filter paper No. 1. After this, distillation of filtrate was done in a kjeldahl distillation set with 10 ml of the sample solution and 10 ml of 40 % sodium hydroxide. The distillated was then collected in a beaker with 5 ml of boric acid indicator till the pink colour turned greenish. The distillated was then titrated against N/140 hydrochloric acid. The titration was stop when the colour turned pink.

The percentage of total nitrogen was calculated by the following formula:

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

Where, T = Burette reading

Preparation of boric acid indicator

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

2.2.6. Available phosphorus

The available phosphorus was estimated by the 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 shaken in a rotatory shaker for 30 minutes. The mixture was filtered through

Whatman filter paper No. 44. 10 ml of the sample was pipetted out into 50 ml volumetric flask and diluted with distilled water to about two-third of the flask. 2 ml of ammonium molybdate reagent and 2 ml of stannous chloride reagent were added to this solution and the final volume was made up to 50 ml by adding distilled water. Control was maintained without soil sample. The samples are allowed to stand for 30 minutes and thereafter the optical density of the sample was read in a Hitachi (220) spectrophotometer at 700 nm. The calibration curve was prepared from the standard and used in determining mg P in the same aliquot.

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

Preparation of reagents

Olsen's reagent

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

Ammonium molybdate sulphuric reagent

Dissolved 25 g of ammonium molybdate [$(NH_4)_6Mo_7O_{24}.4H_2O$] in about 200 ml of water by slight warming. 280 ml of concentrated sulphuric acid was added (with mixing and cooling) to about 400 ml of water and the solution was mix thoroughly and distilled water was added to make up to 1litre after becoming cool. The mixture was then stored in a dark place.

2.2.7. Exchangeable potassium

The total potassium was estimated by flame photometer method (Allen *et. al*, 1974). 10 gram of air-dried sieved soil was weighted in a 500 ml conical flask. To this 250 ml of ammonium acetate solution was added. The solutions are then shaken in a rotatory shaker for 1 hour and kept overnight. The solutions are shaken again for 5 minutes and filter through Whatman filter paper No. 44. The filtered samples are stored in an extraction bottles for determination of total potassium in a flame photometer. The blank samples are run with standard KCl.

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

Preparation of reagent:

Extractant (Ammonium acetate)

575 ml of glacial acetate was added to 200ml distilled water in a 10 litres container preferably polythene. To this solution 600 ml of 0.880N NH_3 solutions was added slowly by cooling. The total solution was then diluted to 10 litres. The pH was adjusted to 7± 0.005 by adding either few drops of acetic acid or NH_3 solution.

2.3. Results

2.3.1. Soil temperature

Soil temperature increases in the first three months and dropped in both bun (BCU) and forest (FS) sites. Soil temperature in the first year of bun cultivation increases in June and dropped in October at surface layer (SL), middle (ML) and subsurface (SSL) layers respectively, whereas in the

second year the soil temperature increases in June and dropped in September. In the forest site (FS), soil temperature dropped in October in the first year whereas it increases in June and dropped in September during the second year at the three depths (Fig. 2.1).

Soil temperature at the surface layer (SL) ranges from 18.5^{0}C to 12.5^{0}C in July and December during the first year and 18.1^{0}C to 12.1^{0}C in June and December during the second year of bun cultivation (BCU). In forest (FS), soil temperature ranges from 20.2^{0}C to 12.0^{0}C in July and December and 20.0^{0}C to 10.7^{0}C in July and December during the second year (Fig. 2.1).

Soil temperature at the middle layer (ML) ranges from 18.0^{0}C to 12.5^{0}C in July and December during the first year and 17.6^{0}C to 10.8^{0}C in June and December during the second year of bun cultivation. In forest, the soil temperature ranges from 20.1^{0}C to 12.1^{0}C in August and November during the first year and 19.8^{0}C to 10.2^{0}C in August and December during the second year (Fig. 2.1).

Soil temperature at the subsurface (SSL) layer soil ranges from 017.9^{0}C to 12.5^{0}C in September and December during the first year and 17.5^{0}C to 11.4^{0}C in September and December during the second year of bun cultivation. In forest, the soil temperature ranges from 19.4^{0}C to 12.5^{0}C in July and December during the first year and 19.0^{0}C to 11.0^{0}C in July and December during the second year (Fig. 2.1).

The one-way analysis of variance (ANOVA) shows insignificant variation ($P \leq 0.05$) among the different depths of forest and bun cultivation

(Table 2.1) and insignificant variation ($P \leq 0.05$) in between the same depths of the two sites (Table 2.2).

The Correlation coefficient (r) values of soil temperature show positive correlation ($P \leq 0.001$) at surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and forest (FS) sites (Table 2.3).

Correlation coefficient (r) values of soil temperature with various microbial population, biological, biochemical and other physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that soil temperature is positively correlated with pH (0.652 at $P \leq 0.01$), organic carbon (0.715 at $P \leq 0.01$), total nitrogen (0.557 at $P \leq 0.05$), ambient temperature (0.909 at $P \leq 0.01$), rainfall (0.760 at $P \leq 0.01$), fungal population (0.533 at $P \leq 0.05$) and phosphatase enzyme (0.533 at $P \leq 0.05$) respectively (Table 2.4). Soil temperature in the middle layer of bun cultivation (Table 2.5) is positively correlated with pH (0.558 at $P \leq 0.05$), organic carbon (0.716 at $P \leq 0.01$), ambient temperature (0.900 at $P \leq 0.01$), rainfall (0.756 at $P \leq 0.01$) and dehydrogenase enzyme (0.577 at $P \leq 0.05$). In the subsurface layer of bun cultivation (Table 2.6), soil temperature is positively correlated with ambient temperature (0.891 at $P \leq 0.001$) and rainfall (0.0.658 at $P \leq 0.01$).

Correlation coefficient (r) values of soil temperature with various microbial population, biological, biochemical and other physico-chemical properties of soil in forest (FS) site at surface (SL) shows positive correlation with soil moisture content (0.526 at $P \leq 0.05$), potassium (0.584 at $P \leq 0.05$), ambient temperature (0.844 at $P \leq 0.05$), rainfall (0.805 at $P \leq 0.001$), dehydrogenase enzyme (0.647 at $P \leq 0.01$) and negatively correlated with pH

(-0.705 at $P \leq 0.05$) and total nitrogen (-0.525 at $P \leq 0.05$) respectively (Table 2.7). Soil temperature in the middle (ML) layer of forest site (Table 2.8) shows negative correlation with pH (-0.498 at $P \leq 0.05$) and positively correlated with soil moisture content (0.535 at $P \leq 0.05$), organic carbon (0.624 at $P \leq 0.01$), ambient temperature (0.834 at $P \leq 0.001$) and rainfall (0.735 at $P \leq 0.001$). In the subsurface layer of forest site (Table 2.9), soil temperature is negatively correlated with pH (-0.659 at $P \leq 0.01$) and positively correlated with soil moisture content (0.586 at $P \leq 0.05$), potassium (0.644 at $P \leq 0.01$), ambient temperature (0.839 at $P \leq 0.001$) and rainfall (0.775 at $P \leq 0.001$).

2.3.2. Soil moisture content

Soil moisture contents increases during first year of both sites and whereas in the second year it increases and decreases in forest and bun cultivation sites at the three depths. Soil moisture contents in the first year of bun cultivation increases in July and dropped in November at surface layer (SL), middle (ML) and subsurface (SSL) layers respectively, whereas in the second year the soil moisture contents dropped in December. In the forest site (FS), soil moisture contents are very high in September and December during the first year whereas it is very low during the second year at the three (Fig. 2.2).

Soil moisture contents in bun cultivation site ranges from 41.83 to 26.03 % in September at subsurface layer (SSL) and May at surface layer (SL) during the first year. In the second year it ranges from 36.73 to 22.95 %

in May at subsurface layer (SSL) and December at surface (SL) layer (Fig. 2.2).

Soil moisture contents in forest site ranges from 42.73 to 25.06 in September at subsurface layer (SSL) and May at middle layer (ML) whereas in the second year it ranges from 39.43 to 21.30 % in July at surface layer (SL) and December at middle (ML) layer (Fig. 2.2).

The one-way analysis of variance (ANOVA) of soil moisture contents in bun cultivation and forest sites shows insignificant variation ($P<0.05$) at surface, middle and subsurface layers (Table 2.1).

The one-way analysis of variance (ANOVA) of soil moisture contents at surface, middle and subsurface layers in bun cultivation and forest sites (Table 2.1) shows insignificant variation ($P<0.05$).

The Correlation coefficient (r) values of soil moisture contents at surface (SL), middle (ML) and subsurface (SSL) layers of soil in bun cultivation (BCU) and forest (FS) sites (Table 2.3) show positive correlation ($P\leq 0.05$, $P\leq 0.01$) at the three depths.

Correlation coefficient (r) values of soil moisture contents with various microbial population, biological, biochemical and other physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that soil moisture content is negatively correlated with available phosphorus (-0.638 at $P\leq 0.01$) and positively correlated with ambient temperature (0.467 at $P\leq 0.05$), dehydrogenase enzyme (0.278 at $P\leq 0.05$) and urease enzyme (0.666 at $P\leq 0.01$) respectively (Table 2.4). Soil moisture contents in the middle layer of bun cultivation (Table 2.5) is positively correlated with

organic carbon (0.622 at $P \leq 0.01$), ambient temperature (0.538 at $P \leq 0.05$) and phosphatase enzyme (0.533 at $P \leq 0.05$). In the subsurface layer of bun cultivation (Table 2.6), soil moisture content is positively correlated with organic carbon (0.562 at $P \leq 0.01$) and ambient temperature (0.570 at $P \leq 0.01$). Correlation coefficient (r) values of soil moisture content with various microbial population, biological, biochemical and other physico-chemical properties of soil in forest (FS) site at surface (SL) shows positive correlation with available phosphorus (0.486 at $P \leq 0.05$), ambient temperature (0.464 at $P \leq 0.05$) and fungal population (0.533 at $P \leq 0.05$) respectively (Table 2.7). Soil moisture content in the middle (ML) layer of forest site (Table 2.8) shows positive correlation with ambient temperature (0.454 at $P \leq 0.05$) and phosphatase enzyme (0.574 at $P \leq 0.01$). In the subsurface layer of forest site (table 2.9), soil moisture content is positively correlated with ambient temperature (0.613 at $P \leq 0.01$) and phosphatase enzyme (0.597 at $P \leq 0.01$).

2.3.3. Soil pH

The soil pH dropped during the first year but show almost constant during the second year at the three depths in both the sites. Soil pH in bun cultivation site ranges from 7.41 to 5.12 in May at surface layer (SL) and October at middle layer (ML) during the first year. In the second year it ranges from 6.45 to 5.62 in May at surface layer (SL) and August at subsurface (SSL) layer. Soil pH in forest site ranges from 7.64 to 4.67 in May at subsurface layer (SSL) and December at middle layer (ML) whereas, in

the second year it ranges from 6.27 to 5.49 in November at surface layer (SL) and June at subsurface (SSL) layer (Fig. 2.3).

The one-way analysis of variance (ANOVA) of soil pH at bun cultivation and forest sites shows insignificant variation ($P<0.05$) at surface, middle and subsurface layers (Table 2.1).

The one-way analysis of variance (ANOVA) of soil pH at surface, middle and subsurface layers in bun cultivation and forest sites (Table 2.2) shows insignificant variation ($P<0.05$).

The Correlation coefficient (r) values of soil pH at surface (SL), middle (ML) and subsurface (SSL) layers of soil in bun cultivation (BCU) and forest (FS) sites (Table 2.3) is positively correlated ($P\leq 0.05$. $P\leq 0.01$, $P\leq 0.001$).

Correlation coefficient (r) values of soil pH with various microbial population, biological, biochemical and other physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that soil pH is positively correlated with soil temperature (0.652 at $P\leq 0.01$), dehydrogenase enzyme (0.263 at $P\leq 0.05$) and urease enzyme (0.453 at $P\leq 0.05$) respectively (Table 2.4). Soil pH in the middle layer of bun cultivation site (Table 2.5) is positively correlated with soil temperature (0.558 at $P\leq 0.05$) and total nitrogen (0.529 at $P\leq 0.05$). In the subsurface layer of bun cultivation (Table 2.6), soil pH is negatively correlated with available phosphorus (-0.499 at $P\leq 0.05$).

Correlation coefficient (r) values of soil pH with various microbial population, biological, biochemical and other physico-chemical properties of soil in forest (FS) site at surface (SL) shows positive correlation with ambient

temperature (0.458 at $P \leq 0.05$) and negatively correlated with soil temperature (0.705 at $P \leq 0.05$), potassium (-0.476 at $P \leq 0.05$) respectively (Table 2.7). Soil pH in the middle (ML) layer of forest site (Table 2.8) shows negative correlation with soil temperature (-0.498 at $P \leq 0.05$) and positively correlated with dehydrogenase enzyme (0.332 at $P \leq 0.01$). In the subsurface layer of forest site (Table 2.9), soil pH is negatively correlated with soil temperature (-0.659 at $P \leq 0.01$).

2.3.4. Soil organic carbon

Soil organic carbon increases in July and August at the three depths during the first and second year of bun cultivation. In forest site, it increases in July at surface layer and in December at middle and subsurface layer during the first year whereas it increases in August during the second year. Soil organic carbon in bun cultivation ranges from 7.01 to 2.93 % in July at surface layer and December at middle layer. In the second layer it ranges from 5.32 to 1.47 % in August at subsurface layer and December at middle layer. In forest site, soil organic carbon ranges from 6.79 to 2.15 % in July at surface layer and October at subsurface layer during the first year whereas in the second year it ranges from 4.79 to 1.91 % in August and November at subsurface layer (Fig. 2.4).

The one-way analysis of variance (ANOVA) of soil organic carbon at bun cultivation and forest sites shows significant variation ($P<0.05$) at the subsurface layer of bun cultivation and surface layer of forest sites (Table 2.1).

The one-way analysis of variance (ANOVA) of soil organic carbon at surface, middle and subsurface layers in forest site shows significant variation ($P<0.05$) among the three depths. The middle layer of forest site shows significant variation ($P<0.05$) with surface and subsurface layers. Bun cultivation shows insignificant variation at all the three depths (Table 2.2).

The Correlation coefficient (r) values of soil organic carbon at surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and forest (FS) is positively correlated ($P<0.05$) at the three depths except subsurface layer of forest sites shows insignificant correlation ($P<0.05$, $P\leq 0.01$, $P\leq 0.001$) with surface and middle layers of bun cultivation (Table 2.3).

Correlation coefficient (r) values of soil organic carbon with various microbial population, biological, biochemical and other physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that soil organic carbon is positively correlated with ambient temperature (0.579 at $P\leq 0.05$), soil temperature (0.0.715 at $P\leq 0.01$), potassium (0.596 at $P\leq 0.05$), phosphatase enzyme (0.743 at $P\leq 0.001$) and negatively correlated with available phosphorus (-0.517 at $P\leq 0.05$) respectively (Table 2.4). Soil organic carbon in the middle layer of bun cultivation (Table 2.5) is positively correlated with ambient temperature (0.508 at $P\leq 0.05$), potassium (0.499 at $P\leq 0.05$), fungal population (0.626 at $P\leq 0.01$) and phosphatase enzyme (0.680 at $P\leq 0.01$). In the subsurface layer of bun cultivation (Table 2.6), soil organic carbon is positively correlated with ambient temperature (0.572 at $P\leq 0.05$), available phosphorus (0.555 at $P\leq 0.05$), rainfall (0.760 at

$P \leq 0.001$), fungal population (0.602 at $P \leq 0.01$) and urease enzyme (0.651 at $P \leq 0.01$). Correlation coefficient (r) values of soil organic carbon with various microbial population, biological, biochemical and other physico-chemical properties of soil in forest (FS) site at surface (SL) layer (Table 2.7) shows positive correlation with potassium (0.607 at $P \leq 0.05$). Soil organic carbon in the middle (ML) layer of forest site (Table 2.8) shows negative correlation with total nitrogen (-0.537 at $P \leq 0.05$. In the subsurface layer of forest site (Table 2.9), soil organic carbon is positively correlated with available phosphorus (0.582 at $P \leq 0.05$) and urease enzyme (0.610 at $P \leq 0.05$).

2.3.5. Soil total Nitrogen

The soil total nitrogen dropped in June during the first year at surface, middle and subsurface layers in both bun cultivation and forest sites. In the second year, it dropped in November during the second year at the three sites of both bun cultivation and forest sites. Soil total nitrogen increases in October during the year at the three depths in both bun cultivation and forest sites. In the first year, soil total nitrogen is high in May whereas it is high in October and December in both the sites. In bun cultivation site, the soil total nitrogen ranges from 0.094 to 0.018% in October at subsurface layer and May at middle layer during the first year and it ranges from 0.104 to 0.007% in October at surface layer and May at middle layer during the second year. In forest site, soil total nitrogen ranges from 0.098 to 0.014% in October at surface layer and September at subsurface layer during the first year and it

ranges from 0.126 to 0.008% in December at middle layer and November at subsurface layer during the second year (Fig. 2.5).

The one-way analysis of variance (ANOVA) of soil total nitrogen in bun cultivation and forest sites shows insignificant variance ($P<0.05$) at surface, middle and subsurface layers (Table 2.1).

The one-way analysis of variance (ANOVA) of soil total nitrogen at surface, middle and subsurface layers in bun cultivation and forest sites shows significant variation ($P<0.05$) only among the three layers of bun cultivation site whereas insignificant variation ($P<0.05$) among the other layers of bun cultivation and forest site (Table 2.2).

The Correlation coefficient (r) values of soil total nitrogen at surface (SL), middle (ML) and subsurface (SSL) layers of soil in bun cultivation (BCU) and forest (FS) sites is positively correlated ($P\leq0.05$) only between the middle and subsurface layers of forest site but there is insignificant correlation between the other layers in both the sites (Table 2.3).

Correlation coefficient (r) values of soil total nitrogen with various microbial population, biological, biochemical and other physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that soil moisture content is positively correlated with ambient temperature (0.544 at $P\leq 0.05$), soil temperature (0.557 at $P\leq 0.05$), fungal population (0.537 at $P\leq 0.05$) and phosphatase enzyme (0.507 at $P\leq 0.05$) respectively (Table 2.4). Soil total nitrogen in the middle layer of bun cultivation (Table 2.5) is positively correlated with soil pH (0.529 at $P\leq 0.05$) and negatively

correlated with soil potassium (–0.501 at $P \leq 0.05$). In the subsurface layer of bun cultivation, soil total nitrogen shows insignificant correlation (Table 2.6).

Correlation coefficient (r) values of soil total nitrogen with various microbial population, biological, biochemical and other physico-chemical properties of soil in forest (FS) site at surface (SL) layer (Table 2.7) is negatively correlated with soil temperature (-0.525 at $P \leq 0.05$). Soil total nitrogen in the middle (ML) layer of forest site is negatively correlated with ambient temperature (-0.637 at $P \leq 0.01$), soil temperature (-0.550 at $P \leq 0.05$) and organic carbon (-0.537 at $P \leq 0.05$) respectively (Table 2.8). In the subsurface layer of forest site, soil total nitrogen shows insignificant correlation (Table 2.9).

2.3.6. Soil available Phosphorus

Soil available phosphorus increases in June during first and second years at middle and surface layers in bun cultivation site. Whereas it increases in September during the first year of bun cultivation site. In forest site, soil available phosphorus dropped in June at subsurface layer and increases in August. In the middle layer of forest site, soil available phosphorus site dropped in September and increases in December in both the middle and subsurface layers. The soil available phosphorus dropped in the second year at the three depths in both bun cultivation and forest sites (Fig. 2.6).

In bun cultivation site, soil available phosphorus ranges from 0.0371 to 0.0016 µg-1 dry soil in August at subsurface layer and August at surface layer during the first year and it ranges from 0.015 to 0.0003 µg-1 dry soil in

June at surface layer and December at subsurface layer during the second year. Soil available phosphorus in forest site ranges from 0.0304 to 0.00096 µg-1 dry soil in September at surface layer and October at subsurface layer during the first year and it ranges from 0.0519 to 0.00013 µg-1 dry soil in June at surface layer and December at subsurface layer during the second year (Fig. 2.6)

The one-way analysis of variance (ANOVA) of soil available phosphorus at bun cultivation and forest sites shows significant variation ($P \leq 0.05$) between the two sites. The surface layers of the two sites show insignificant variation ($P \leq 0.05$). The middle layer and subsurface layers of bun cultivation site also shows insignificant variation ($P \leq 0.05$) with the middle and surface layer in forest site (Table 2.1).

The one-way analysis of variance (ANOVA) of soil available phosphorus at surface, middle and subsurface layers in bun cultivation and forest sites (Table 2.2) shows insignificant variation ($P \leq 0.05$).

The Correlation coefficient (r) values of soil available phosphorus at surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and forest (FS) sites is positively correlated ($P \leq 0.01$) between the different layers of the same site in both the forest and bun cultivation sites (Table 2.3).

Correlation coefficient (r) values of soil available phosphorus with various microbial population, biological, biochemical and other physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that soil available phosphorus is negatively correlated with soil

moisture content (-0.638 at $P \leq 0.01$), organic carbon (-0.517 at $P \leq 0.05$) and positively correlated with potassium (0.638 at $P \leq 0.05$), urease enzyme (0.843 at $P \leq 0.001$) respectively (Table 2.4). Soil available phosphorus in the middle layer of bun cultivation (Table 2.5) is positively correlated with potassium (0.546 at $P \leq 0.05$) and urease enzyme (0.710 at $P \leq 0.001$). In the subsurface layer of bun cultivation (Table 2.6), soil available phosphorus is negatively correlated with pH (-0.499 at $P \leq 0.05$) and positively correlated with organic carbon (0.555 at $P \leq 0.05$) and urease enzyme (0.794 at $P \leq 0.001$).

Correlation coefficient (r) values of soil available phosphorus with various microbial population, biological, biochemical and other physico-chemical properties of soil in forest (FS) site at surface (SL) layer (Table 2.7) is positively correlated with soil moisture content (0.486 at $P \leq 0.05$) and fungal population (0.573 at $P \leq 0.05$). Soil available phosphorus in the middle (ML) layer of forest site (Table 2.8) shows insignificant correlation ($P \leq 0.05$). In the subsurface layer of forest site (Table 2.9), soil available phosphorus is positively correlated with organic carbon (0.582 at $P \leq 0.05$) and fungal population (0.511 at $P \leq 0.05$).

2.3.7. Soil exchangeable potassium

The soil exchangeable potassium dropped in June at middle layer during the second year in bun cultivation site and first year in forest site. In bun cultivation site, soil exchangeable potassium increases in June at middle layer during the first year. The exchangeable potassium increases and dropped in August and September at surface layer during the second year.

In forest site, soil exchangeable potassium increases in October at the three depths during the first year whereas it increases in August at surface layer and September at middle layer during the second year (Fig. 2.7).

Soil exchangeable potassium ranges from 2.25 to 0.55% in May at surface layer and June at subsurface layer during the first year and it ranges from 3.15 to 0.75% in May at middle layer and November at subsurface layer during the second year of bun cultivation site. In forest site, soil exchangeable potassium ranges from 2.15 to 0.25% in October at subsurface layer and June at middle layer during the first year and it ranges from 1.65 to 0.35% in September at middle layer and May at subsurface layer during the second year (Fig. 2.7).

The one-way analysis of variance (ANOVA) of soil exchangeable potassium at bun cultivation and forest sites shows significant variation ($P \leq 0.05$) only at the surface layer of bun cultivation and middle layer of forest sites (Table 2.1).

The one-way analysis of variance (ANOVA) of soil exchangeable potassium at surface, middle and subsurface layers in bun cultivation and forest sites (Table 2.2) shows insignificant variation ($P \leq 0.05$).

The Correlation coefficient (r) values of soil exchangeable potassium is positively correlated ($P \leq 0.001$) between the same depths and different depths of the two sites (Table 2.3).

Correlation coefficient (r) values of soil exchangeable potassium with various microbial population, biological, biochemical and other physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer

shows that soil exchangeable potassium is positively correlated with organic carbon (0.596 at $P \leq 0.05$), available phosphorus (0.638 at $P \leq 0.01$) and fungal population (0.804 at $P \leq 0.001$) respectively (Table 2.4). Soil exchangeable potassium in the middle layer of bun cultivation (Table 2.5) is positively correlated with organic carbon (0.499 at $P \leq 0.05$), available phosphorus (0.546 at $P \leq 0.05$), fungal population (0.763 at $P \leq 0.001$) and negatively correlated with total nitrogen (-0.501 at $P \leq 0.001$). In the subsurface layer of bun cultivation, soil exchangeable potassium shows insignificant correlation (Table 2.6).

Correlation coefficient (r) values of soil exchangeable potassium with various microbial population, biological, biochemical and other physico-chemical properties of soil in forest (FS) site at surface (SL) shows positive correlation with soil temperature (0.584 at $P \leq 0.05$), organic carbon (0.607 at $P \leq 0.05$) and negatively correlated with soil moisture content (-0.476at $P \leq 0.05$) respectively (Table 2.7). Soil exchangeable potassium in the middle (ML) layer of forest site (Table 2.8) is significantly correlated with fungal population (0.509 at $P \leq 0.05$). In the subsurface layer of forest site (Table 2.9), soil exchangeable potassium is positively correlated with soil temperature (0.644 at $P \leq 0.01$).

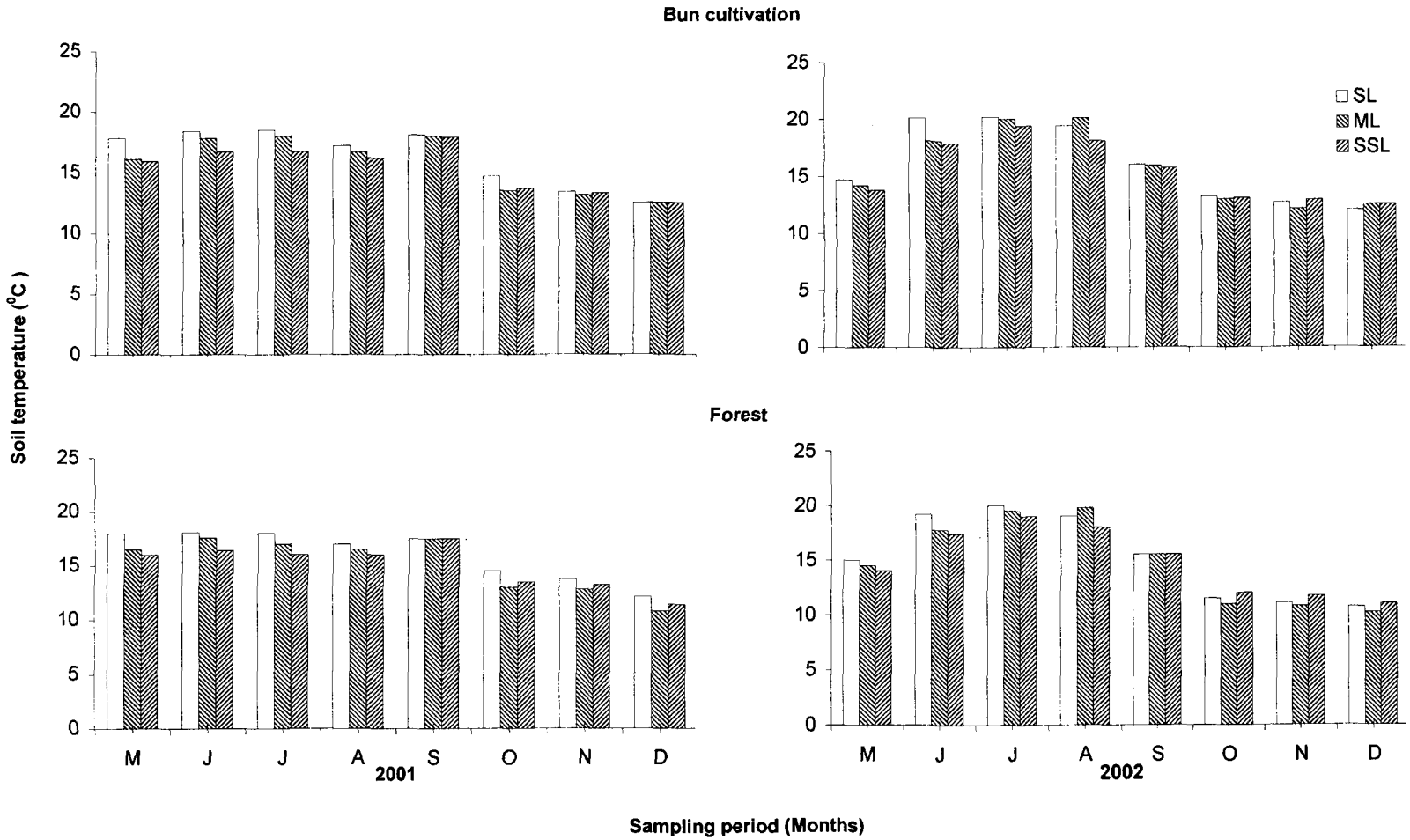

Fig 2.1. Soil temperature in Bun cultivation (BCU) and Forest (FS) sites at surface (SL), middle (ML) and subsurface (SSL) layers

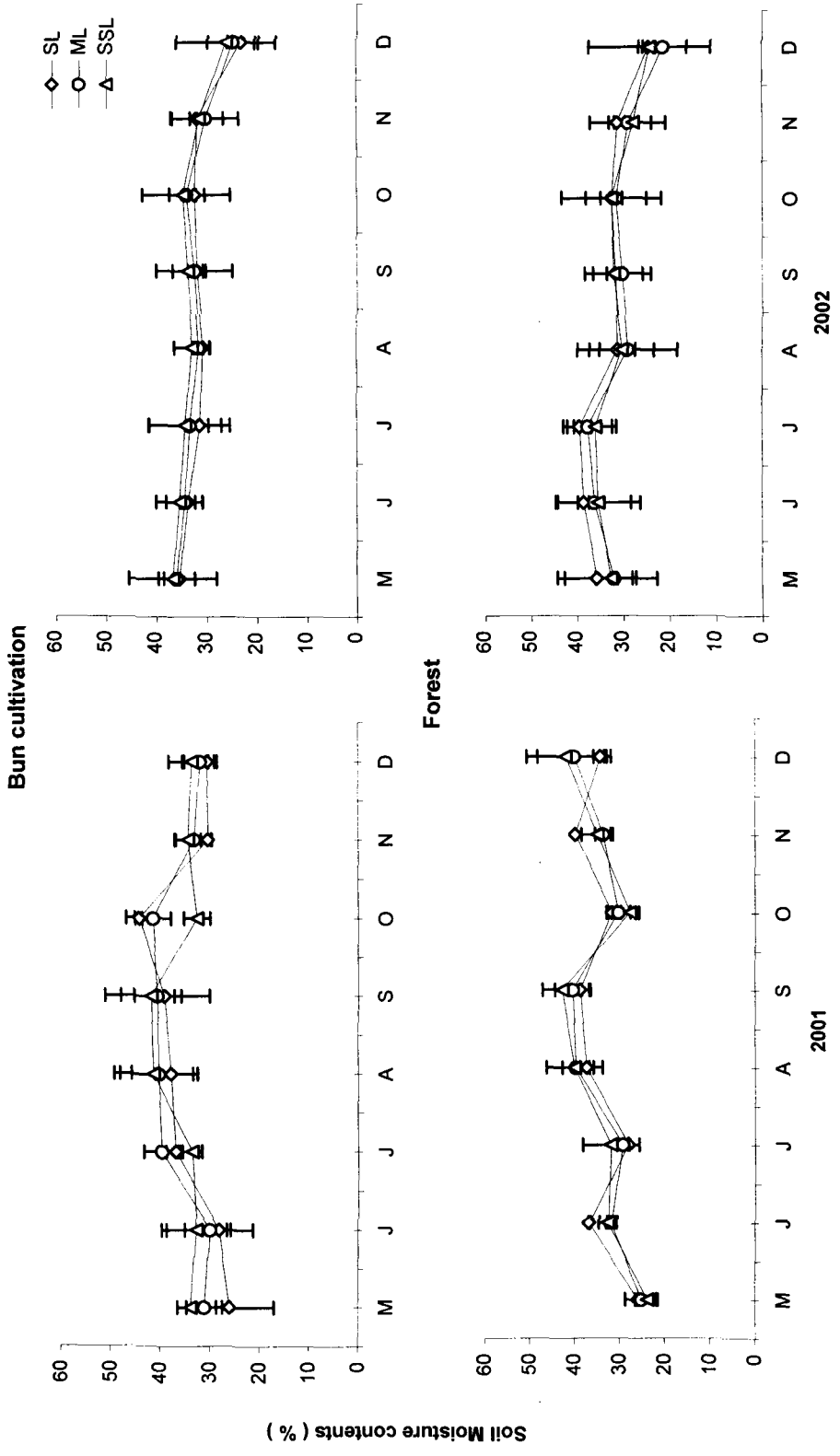

Fig 2.2. Soil moisture content of Bun cultivation (BCU) and Forest (FS) sites at surface (SL), middle (ML) and subsurface (SSL) layers.

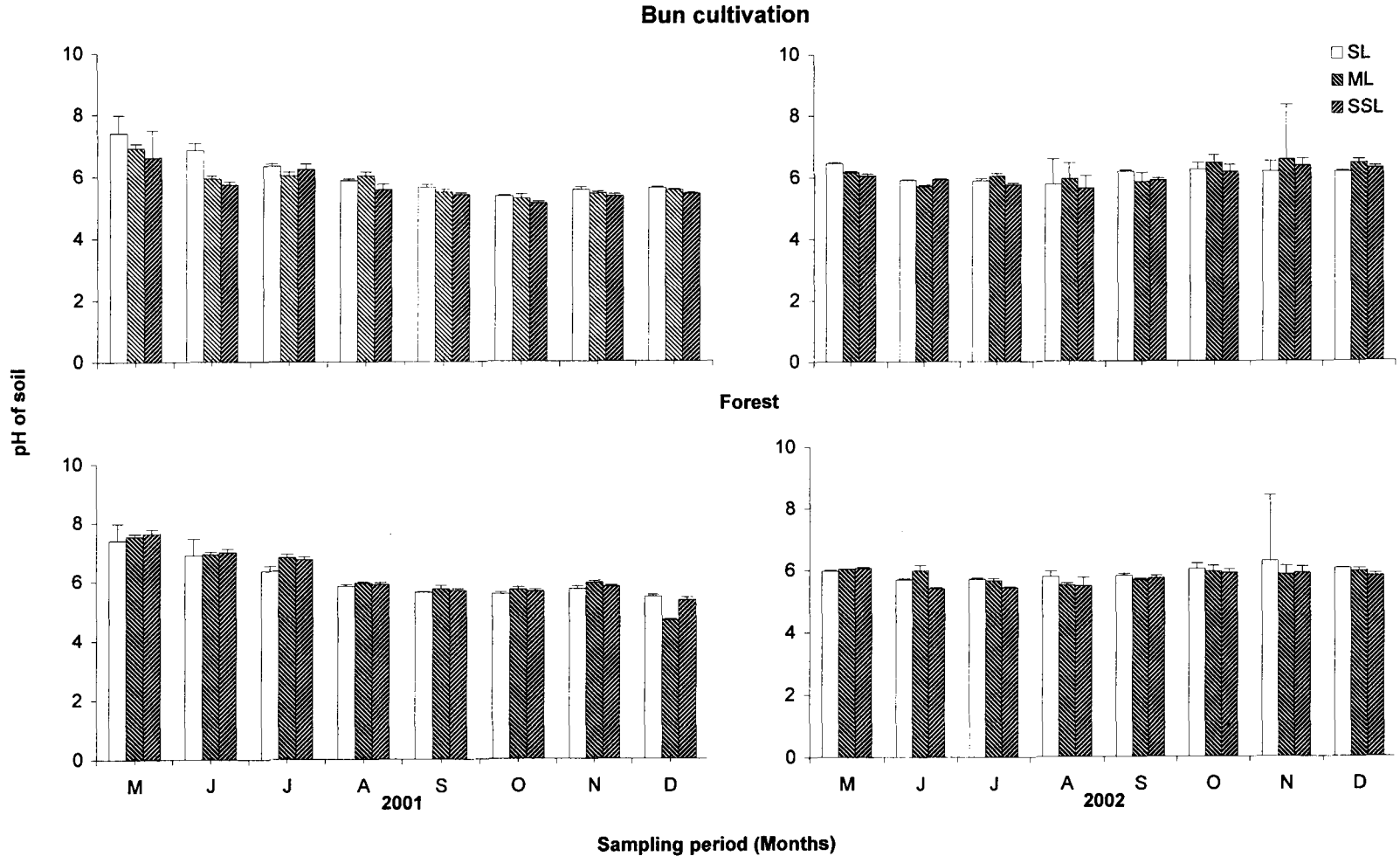

Fig 2.3. Distribution of soil pH at surface (SL), middle (ML) and subsurface layers (SSL) of Bun cultivation (BCU) and Forest (FS) sites.

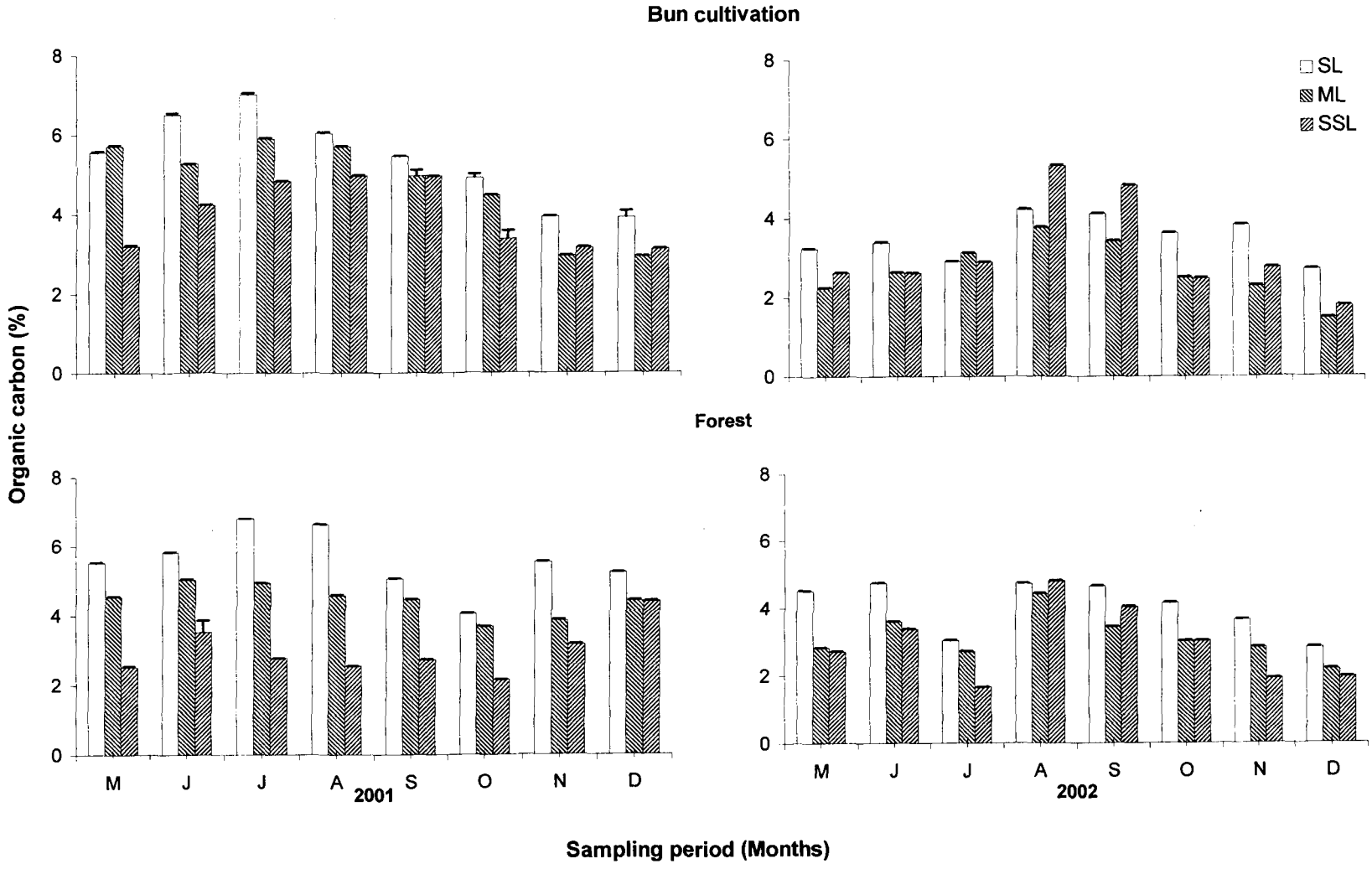

Fig 2.4. Distribution of soil organic carbon content of soil at surface (SL), middle (ML) and subsurface (SSL) soil layers of Bun cultivation (BCU) and Forest (FS) sites.

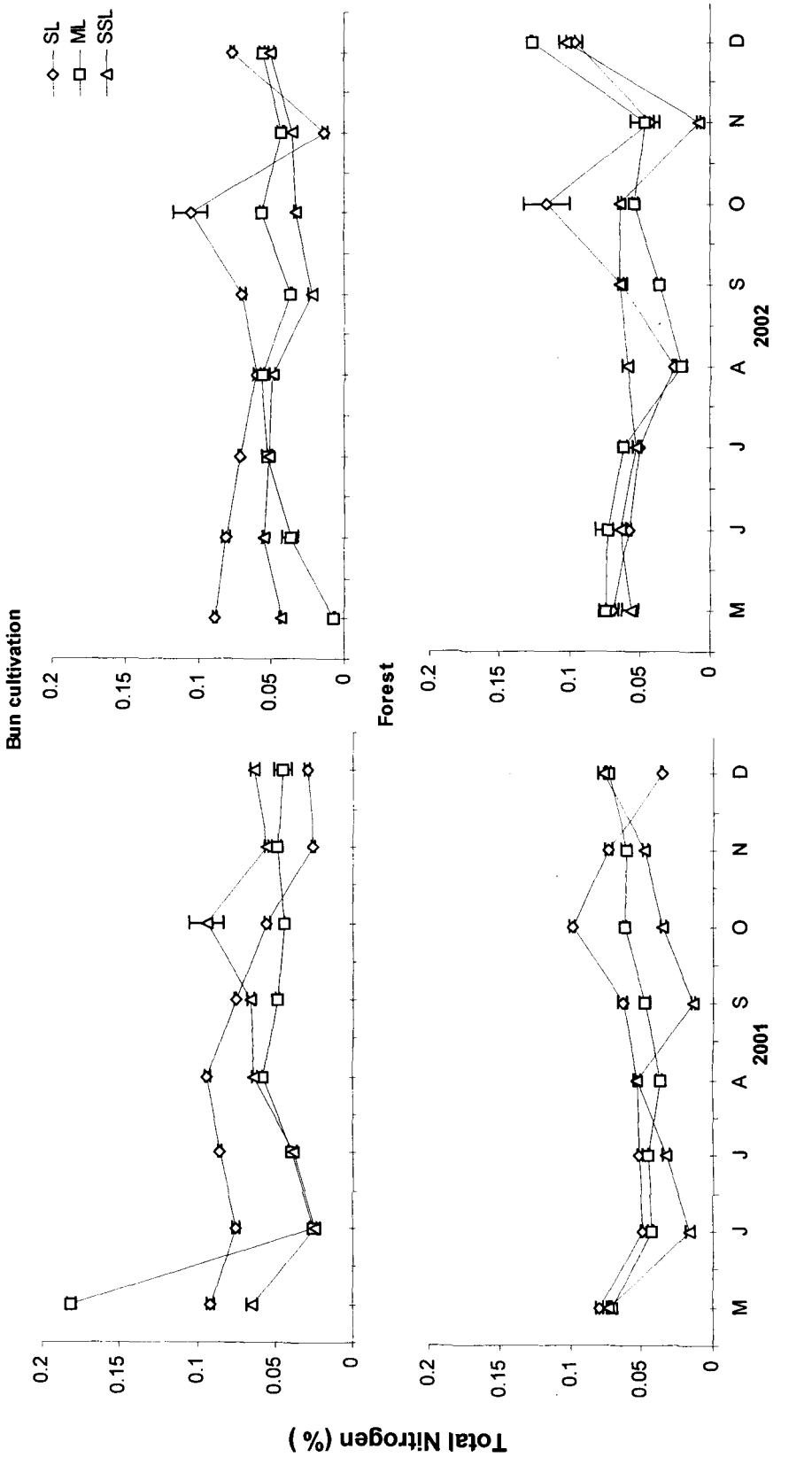

Fig 2.5. Distribution of soil total nitrogen of soil at surface (SL), middle (ML) and subsurface (SSL) soil layers of bun cultivation (BCU) and Forest (FS) sites.

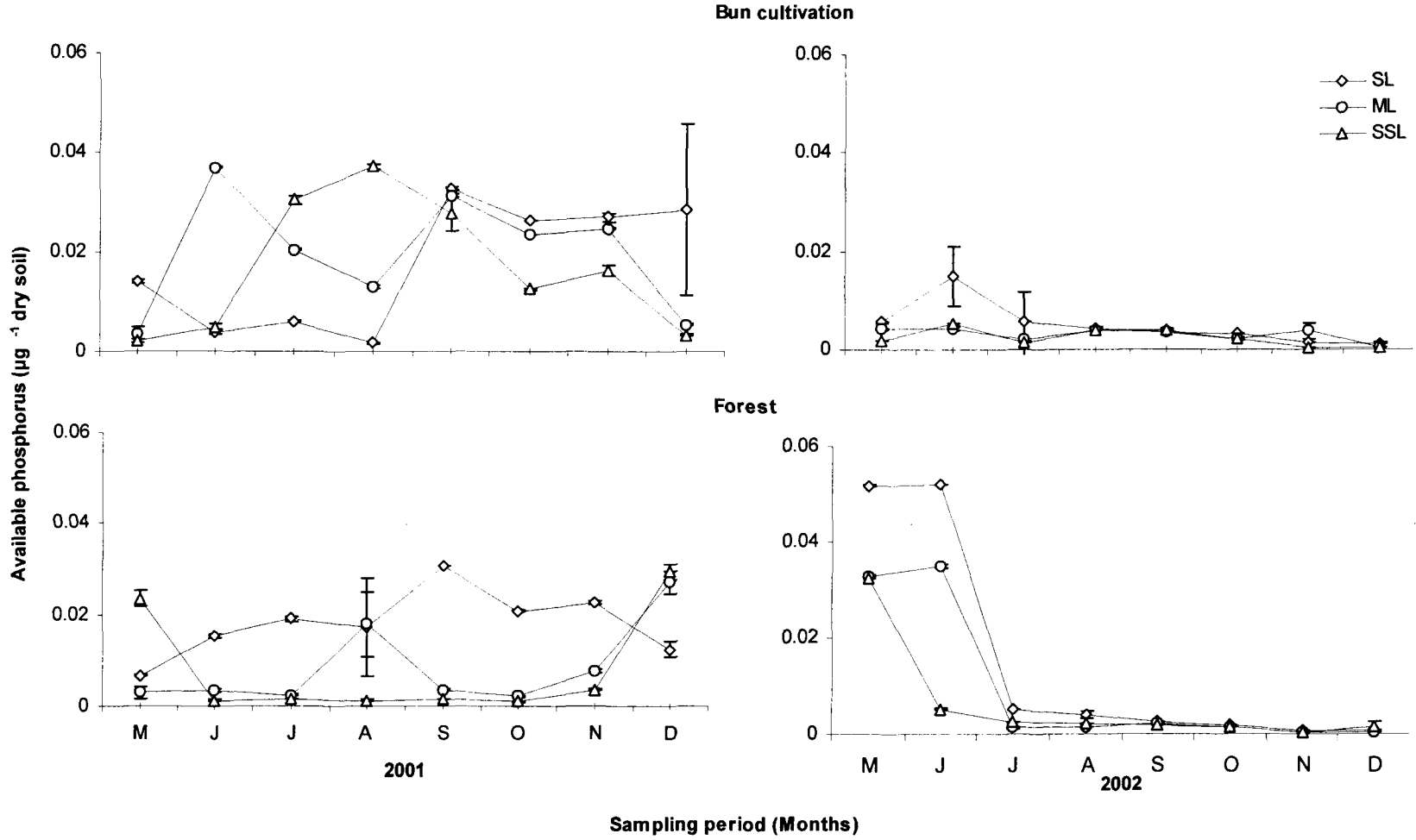

Fig 2.6. Distribution of soil available phosphorus content of soil at surface (SL), middle (ML) and subsurface (SSL) soil layers of Bun cultivation (BCU) and Forest (FS) sites.

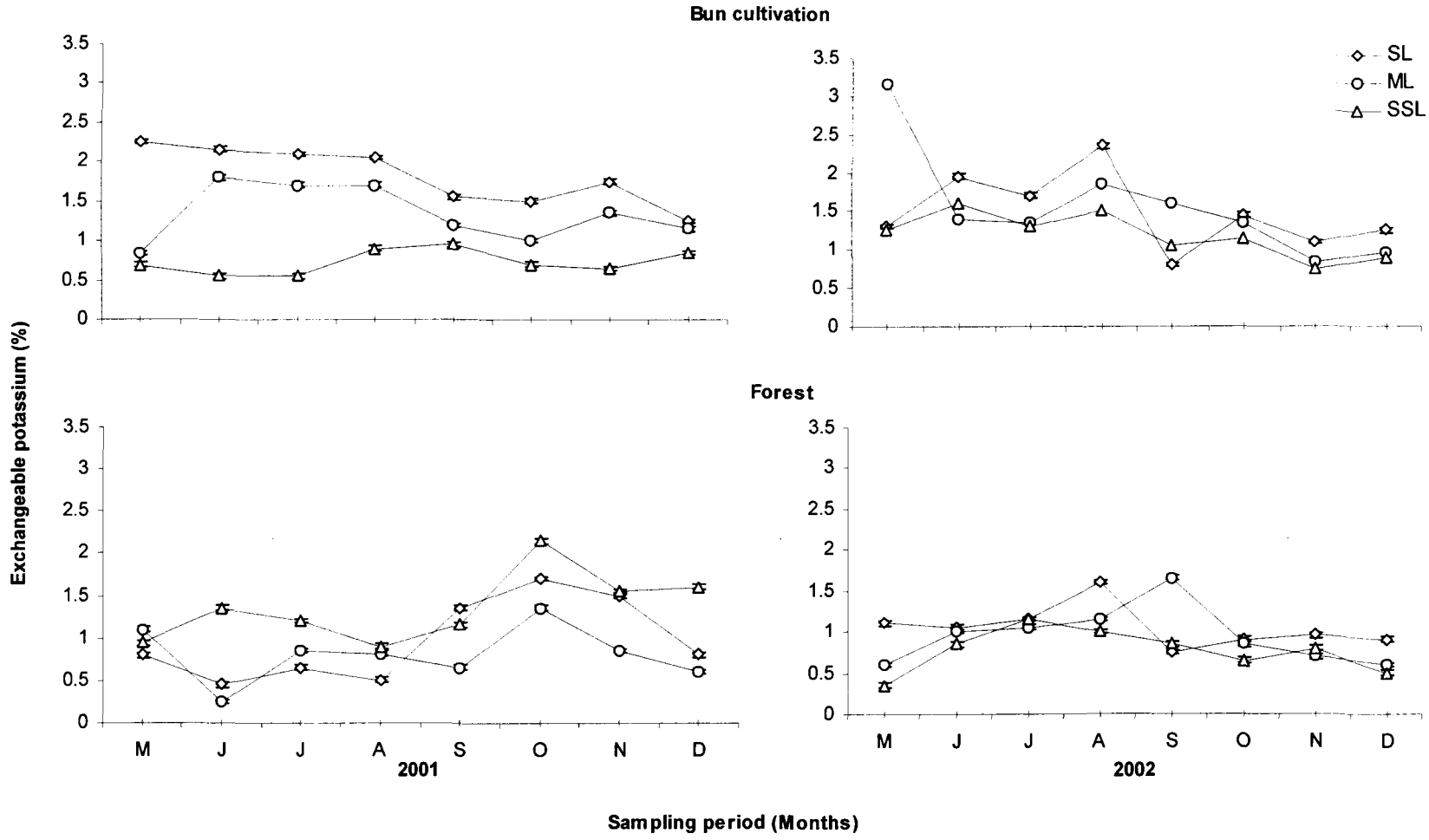

Fig 2.7. Distribution of soil total potassium of soil at surface (SL), middle (ML) and subsurface (SSL) soil layers of bun cultivation (BCU) and Forest (FS) sites.

2.4. Discussion

2.4.1. Soil temperature and soil moisture contents

The surface layer (0-10 cm) (SL) shows a higher soil temperature comparing to the middle (ML) layer (10-20 cm) and subsurface (SSL) layer (20-30 cm), which may be due to the increase of ambient temperature and lowering of the rainfall. The soil temperature is positively correlated to ambient temperature and rainfall in both bun cultivation (BCU) and forest (FS) sites. Soil temperature shows no correlation with soil moisture content in bun cultivation site but an observation has been made that there is positive correlation with the soil moisture content in the forest soil at all the three depths. A significant relationship between soil moisture and soil temperature and its consequent effect on N mineralization was observed by Cassman and Munns (1980) and Mogaddeghi *et al.,* (2000).

In the first year of experiment, soil moisture content increases in all the three depths and decreases in November and October that may be due to the decrease in the rainfall and increase in the ambient temperature at bun cultivation (BCU) and forest (FS) sites. A positive correlation was observed between soil moisture content and the ambient temperature throughout the experiments in both the sites at all the three depths. However, soil moisture content decreases in the second year at surface (0-10 cm), middle (10-20 cm) and subsurface (20-30 cm) layers in both the sites.

No significant variation ($P \leq 0.05$) in soil moisture content has been observed among the three depths but shows positive correlation in between the surface, middle and subsurface layers of bun cultivation and forest sites. A

negative correlation between soil moisture content and available phosphorus was observed in the surface layer at bun cultivation site. Soil moisture content shows a correlation with the microbial population in the middle layer (10-20 cm) of bun cultivation whereas in forest site it is correlated with the enzyme phosphatase in the surface (0-10 cm) and subsurface (20-30 cm) layers.

2.4.2. Soil pH

The soil surface layer (0-10 cm) of bun cultivation has a higher pH value than the inner depths but shows a uniform pH during the second year. This was similar to finding of Parham *et al.,* (2003) in which manure-treated soils shows almost uniform in the soil profiles from 0-10 cm depth (4.2-5.7) with that of untreated soil around pH 5. Soil pH of bun cultivation and forest sites shows a decrease in the pH during the first year but a slight increase was observed during second year in the surface (0-10 cm), middle (10-20 cm) and subsurface (20-30 cm) layers, which might be due to the effect of plant burning residues. Until now the detailed processes responsible for the change in soil pH after incorporation of plant material have not been completely understood. But the study reveals that addition of plant material may initially cause an increase in soil pH due to decomposition of organic anions and organic nitrogen. Soil pH may decrease if nitrification is involved. The concentrations of nitrogen and alkalinity of added plant material are decisive factors controlling soil pH change after incorporation of plant material. Manure (ashes from burning) application increased soil pH (Borie and Rubio, 2003).

No significant variation ($P \leq 0.05$) in soil moisture content has been observed among the three depths but shows positive correlation in between

the surface (0-10 cm), middle (10-20 cm) and subsurface (20-30 cm) layers of bun cultivation (BCU) and forest (FS) sites. Soil pH does not show any correlation with the soil enzyme activities whereas, Deng and Tabatabai (1997) reported that phosphatase activity, for instance, was not related to soil pH but manure addition to a soil may have resulted in changes in origin, states, and persistence of enzymes in the soil. The pH in the surface (0-10 cm) layer (SL) of bun cultivation shows a negative correlation with available phosphorus but has a positive correlation with ambient temperature in all the three depths. Bacterial population is positive correlation with soil pH at middle layer (10-20 cm) of bun cultivation but shows negative correlation (-0.476 at $P \leq 0.05$) in the forest site.

2.4.3. Soil organic C

Both the surface layer (0-10 cm) of bun cultivation and forest sites shows a very high soil organic carbon contents (7.21%) during the first year due to the continuous cropping of a field that has not lowered carbon contents beyond the levels of reached after some years of cropping in a plot abandoned in the previous year after crop rotation (Tiessen *et al.*, 1992). Robertson *et al.,* (1994) observed that soil organic C was higher in soils under vegetable in September, possibly due to more flows of C and N to soils. C input from crop roots, rhizosphere products and crop residues can have a large effect on organic C and N in soils. Soil organic carbon in the middle (10-20 cm) and subsurface (20-30 cm) layers decreases during the first year of experiment, which indicated the movement of soluble manure compounds from the surface to the lower soil profiles (Chardon *et al.,* 1997).

Soil organic carbon shows a significant variation ($P \leq 0.05$) in subsurface (20-30 cm) of bun cultivation (BCU) and surface (0-10 cm) layer of forest site (FS) but shows no variation between the other depths of both the sites. In case of forest site (FS), subsurface layer shows a significant variation ($P \leq 0.05$) with the surface (0-10 cm) and the middle (10-20 cm) layers. A positive correlation was observed among surface (0-10 cm), middle (10-20 cm) and subsurface (20-30 cm) layers of both bun cultivation (BCU) and forest (FS) sites.

Soil organic carbon has a negative correlations (-0.517 at $P \leq 0.05$) with available phosphorus in surface layer but shows a very high correlation with exchangeable potassium in all the three depths of bun cultivation (BCU) site. The correlation of soil organic carbon with soil enzymes (dehydrogenase, urease and phosphatase) and soil microbial population (bacterial population) in bun cultivation (BCU) site shows that soil organic carbon plays a vital role in the activity of soil microbial population and soil enzymes whereas in forest (FS) site, soil organic carbon is correlated with the soil microbial population (fungal) in the middle (10-20 cm) layer and in the subsurface (20-30 cm) layer with the soil enzyme (urease). Soil organic carbon is correlated with exchangeable potassium in the surface (0-10 cm) and middle (10-20 cm) layers of bun cultivation (BCU) where it shows correlation only in the surface (0-10 cm) layer of forest site. When forest ecosystems are converted to agriculture, there is generally a marked decline in the amounts of total and labile soil organic carbon (SOC). However, analysis of SOC changes induced by forest conversion to agriculture have generally been restricted to surface

horizons and to pools of total and microbial biomass SOC. Changes in water-soluble SOC, which is likely the most labile and mobile form of SOC. The result shows that, total SOC was higher in forest than in crop systems, but the difference was restricted to the surface layers (0-10 cm). Surprisingly, concentrations of water-soluble and bio-available SOC were higher in agricultural soils than in forest soils, again only in the surface layers. The difference in water-soluble and bio-available SOC between agricultural and wooded soils was largely caused by an increase in the soluble humic acid fraction in the agricultural soils (Boyer and Groffman, 1996).

Thus it supported the concept that changes in soil microbial biomass size and activity, which in turn affect rates of soil processes are controlled largely by the quality of available C. Short term effects of substrate inputs are likely to involve only a small part of the active biomass, whereas in response to long-term inputs the composition of the biomass as a whole may become modified by selection against organisms with slower metabolism (Anderson and Domsch, 1986; Lovell and Jarvis, 1998).

2.4.4. Soil total nitrogen

The surface layer shows very high total nitrogen content during the first and second year in both bun cultivation and forest sites, which reveals that distributions of soil total N contents over the soil profiles indicated movement of soluble manure compounds from the surface to lower soil profiles. Therefore, it is anticipated that a significant proportion of manure-P may also be found deeper in the soil profile.

The adverse decrease in the soil total nitrogen during the first year at bun cultivation (BCU) could be due to greater amount of nutrient limitations on cultivated sites that prevent organic matter maintenance or increase, unless carbon inputs are accompanied by substantial fertilization. Chardon *et al.* (1997) demonstrated that soil mineral N contents (NO_3 and NH_4) were higher under vegetation, lowest after cultivating a plot abandoned in the previous year after crop rotation and recovered with increasing fallow period following a pattern similar to that of the organic matter contents. Relatively greater amounts of NH_4 than NO_3 are found under native vegetation and re-growth and increases in mineral N in these plots are entirely due to increased NH_4 levels. The relatively slow rates of additions of organic matter as well as the slow rate of nutrient uptake by the re-establishing native vegetation are more effective in building up and maintaining soil organic matter levels than the periodic additions of crop residues under cultivation without fertilization. After several years of cultivation soil organic C and N were significantly higher in the monoculture than in the rotation with various crops and cabbage as the present crop (Omay *et al.*, 1997).

Soil total nitrogen has a significant variation ($P \leq 0.05$) among the surface (0-10 cm), middle (10-20 cm) and subsurface (20-30 cm) layers at bun cultivation site (BCU) where forest site (FS) shows no significant variation ($P \leq 0.05$) among the three depths but a positive correlation was observed between the middle (10-20 cm) and subsurface (20-30 cm) layers at forest site (FS). A positive correlation has been observed in the surface layer between soil total nitrogen and soil temperature at bun cultivation site but

shows a negative correlation at the forest site. Soil total nitrogen in the middle layer shows a negative correlation with available phosphorus of bun cultivation whereas forest site has a negative correlation (-0.559 at $P \leq 0.05$) of soil total nitrogen with bacterial population. Soil total nitrogen is positively correlated with fungal population in the surface layer of bun cultivation but shows a positive correlation in the subsurface layer of forest site.

2.4.5. Soil available phosphorus

The available phosphorus shows an increase in the month of September (0.043 μg^{-1}dry soil) during the first year of both the sites. This result is supported by Chardon *et al.* (1997) that available phosphorus contents increased with increasing soil depth. There is a mark increase in the available phosphorus in June and July at middle (10-20 cm) and subsurface (20-30 cm) layers of bun cultivation (BCU) site but no significant changes were observed in the forest (FS) site, which shows that there is greater phosphorus movement in manure-treated soils than in untreated soils. Borie and Rubio (2003) reported that available phosphorus levels are higher in manure-treated than untreated soils and cultivated soils presented higher amounts of available phosphorus and organic C than uncultivated soils.

In the second year of experiment, the soil available phosphorus decreases in July at all the three depths in forest site but shows no changes in the bun cultivation (BCU) site. There is a decrease in the soil available phosphorus during the second year in both the sites which shows that soil maturity enhance the loss of soil available phosphorus. The surface (SL) layer

(0-10 cm) of bun cultivation shows no significant variation of soil available phosphorus with the forest (FS) site.

The Bun cultivation (BCU) and forest (FS) sites show a significant variation between middle (ML) and subsurface (SSL) layers but has insignificant variation in the surface (SL) layer. The surface layer of bun cultivation (BCU) site has significant variation with the middle and subsurface layers of forest (FS) site.

2.4.6. Soil exchangeable potassium

The surface (SL) layer shows very high exchangeable potassium during the first year of bun cultivation and decreases with the increased of the soil depth. The content of available potassium and immediately available potassium is higher in uppermost soil layer than that in lower soil layers and it is close related to the organic matter content of the soil.

The exchangeable potassium increases in the month of July just after one month of burning at bun cultivation which shows that the effects of clearing and burning of soil within a month after burning, were increased in exchangeable potassium content, decreases in N and no change in P. Increases were the results of additions from ash and unburned organic matter leaching into the soil (Halenda and Christine, 1993; Rikhari and Palni, 1999).

Table 2.1. One-way analysis of variance (ANOVA) of the physico-chemical properties of soil in Bun cultivation (BCU) and Forest (FS) sites at surface (0-10 cm), middle (10-20 cm) and sub surface (20-30 cm) layers at $P<0.05$.

Soil properties	Source of variation	F-ratio	P-level
Soil temperature	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	-	-
	BCU (SL) x FS (SSL)	-	-
	BCU (ML) x FS (SL)	-	-
	BCU (ML) x FS (SSL)	-	-
	BCU (SSL) x FS (SL)	-	-
	BCU (SSL) x FS (ML)	-	-
Soil moisture	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	-	-
	BCU (SL) x FS (SSL)	-	-
	BCU (ML) x FS (SL)	-	-
	BCU (ML) x FS (SSL)	-	-
	BCU (SSL) x FS (SL)	-	-
	BCU (SSL) x FS (ML)	-	-
Soil pH	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	-	-
	BCU (SL) x FS (SSL)	-	-
	BCU (ML) x FS (SL)	-	-
	BCU (ML) x FS (SSL)	-	-
	BCU (SSL) x FS (SL)	-	-
	BCU (SSL) x FS (ML)	-	-
Organic Carbon	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	-	-
	BCU (SL) x FS (SSL)	-	-
	BCU (ML) x FS (SL)	-	-
	BCU (ML) x FS (SSL)	-	-
	BCU (SSL) x FS (SL)	11.917	0.137×10^{-2}
	BCU (SSL) x FS (ML)	-	-
Total Nitrogen	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	-	-
	BCU (SL) x FS (SSL)	-	-
	BCU (ML) x FS (SL)	-	-
	BCU (ML) x FS (SSL)	-	-
	BCU (SSL) x FS (SL)	-	-
	BCU (SSL) x FS (ML)	-	-
Available phosphorus	BCU x FS (SL)	-	-
	BCU x FS (ML)	4.638	0.037
	BCU x FS (SSL)	4.368	0.043
	BCU (SL) x FS (ML)	7.633	0.878×10^{-2}
	BCU (SL) x FS (SSL)	10.542	0.424×10^{-2}
	BCU (ML) x FS (SL)	-	-
	BCU (ML) x FS (SSL)	7.574	0.904×10^{-2}
	BCU (SSL) x FS (SL)	-	-
	BCU (SSL) x FS (ML)	-	-
Exchangeable potassium	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	5.548	0.023
	BCU (SL) x FS (SSL)	-	-
	BCU (ML) x FS (SL)	-	-
	BCU (ML) x FS (SSL)	-	-
	BCU (SSL) x FS (SL)	-	-
	BCU (SSL) x FS (ML)	-	-

Note: Insignificant values are denoted by '-' sign.

Table 2.2. One-way analysis of variance (ANOVA) of the physico-chemical properties of soil at surface (0-10 cm), middle (10-20 cm) and sub surface (20-30 cm) layers in Bun cultivation (BCU) and Forest (FS) sites at $P<0.05$.

Soil properties	Source of variation	F-ratio	P-level
Soil temperature	BCU (SL x ML x SSL)		
	FS (SL x ML x SSL)		
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	-	-
	FS (SL x SSL)	-	-
	FS (ML x SSL)	-	-
Soil moisture	BCU (SL x ML x SSL)	-	-
	FS (SL x ML x SSL)	-	-
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	-	-
	FS (SL x SSL)	-	-
	FS (ML x SSL)	-	-
Soil pH	BCU (SL x ML x SSL)	-	-
	FS (SL x ML x SSL)	-	-
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	-	-
	FS (SL x SSL)	-	-
	FS (ML x SSL)	-	-
Organic C	BCU (SL x ML x SSL)	-	-
	FS (SL x ML x SSL)	16.098	0.03×10^{-4}
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	9.531	0.376×10^{-2}
	FS (SL x SSL)	-	-
	FS (ML x SSL)	6.377	0.015
Total Nitrogen	BCU (SL x ML x SSL)	4.271	0.047
	FS (SL x ML x SSL)	-	-
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	-	-
	FS (SL x SSL)	-	-
	FS (ML x SSL)	-	-
Available phosphorus	BCU (SL x ML x SSL)	-	-
	FS (SL x ML x SSL)	-	-
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	-	-
	FS (SL x SSL)	-	-
	FS (ML x SSL)	-	-
Exchangeable potassium	BCU (SL x ML x SSL)	-	-
	FS (SL x ML x SSL)	-	-
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	-	-
	FS (SL x SSL)	-	-
	FS (ML x SSL)	-	-

Note: Insignificant values are denoted by '-' sign.

Table 2.3. Correlation coefficient (r) values among the physico-chemical properties of soil at surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and Forest (FS) sites ($P \leq 0.05$)

Source of variation	Soil temperature	Soil moisture	pH	Organic C	Total N	Available P	Exchangeable K
BCU x FS (SL)	0.826^{c}	0.472^{b}	0.857^{c}	0.798^{c}	-	-	0.806^{c}
BCU x FS (ML)	0.842^{c}	0.638^{b}	0.452^{a}	0.867^{c}	-	-	0.835^{c}
BCU x FS (SSL)	0.805^{c}	0.837^{c}	0.560^{b}	0.551^{a}	-	-	0.769^{c}
BCU (SL) x FS (ML)	0.805^{c}	0.575^{b}	0.787^{c}	0.867^{c}	-	-	0.721^{c}
BCU (SL) x FS (SSL)	0.834^{c}	0.558^{b}	0.835^{c}	-	-	-	0.704^{c}
BCU (ML) x FS (SL)	0.853^{c}	0.504^{b}	0.680^{c}	0.751^{c}	-	-	0.833^{c}
BCU (ML) x FS (SSL)	0.873^{c}	0.649^{b}	0.552^{b}	-	-	-	0.747^{c}
BCU (SSL) x FS (SL)	0.755^{c}	0.722^{c}	0.693^{c}	0.642^{b}	-	-	0.941^{c}
BCU (SSL) x FS (ML)	0.763^{c}	0.819^{c}	0.585^{b}	0.774^{c}	-	-	0.893^{c}
BCU (SL x ML)	0.981^{c}	0.947^{c}	0.775^{c}	0.941^{c}	-	0.904^{c}	0.733^{c}
BCU (SL x SSL)	0.964^{c}	0.700^{c}	0.791^{c}	0.767^{c}	-	0.892^{c}	0.841^{c}
BCU (ML x SSL)	0.982^{c}	0.801^{c}	0.890^{c}	0.776^{c}	-	0.698^{c}	0.865^{c}
FS (SL x ML)	0.983^{c}	0.885^{c}	0.897^{c}	0.830^{c}	-	0.888^{c}	0.905^{c}
FS (SL x SSL)	0.973^{c}	0.897^{c}	0.937^{c}	0.479^{a}	-	0.798^{c}	0.855^{c}
FS (ML x SSL)	0.974^{c}	0.950^{c}	0.935^{c}	0.598^{b}	0.596^{a}	0.922^{c}	0.900^{c}

Note: Values marked with a, b and c are significant at ($P \leq 0.05$), ($P \leq 0.01$) and ($P \leq 0.001$) respectively; insignificant values are marked with '-'.

Table 2.4. Correlation coefficient (r) values among physico-chemical properties of soil with various microbial population, biological and biochemical properties of soil in Bun cultivation (BCU) site at surface (SL) layer ($P \leq 0.05$).

Soil properties	ST	MC	pH	OC	TN	AP	K	AT	RF	FP	BP	C_{mic}	DHA	URA	PA
AT				0.579^a	0.544^a	-			0.692^b	0.561^a	0.656^b	-	0.556^a	-	0.567^a
ST		-	0.652^b	0.715^b	0.557^a	-	-	0.909^c	0.760^b	0.533^a	0.601^a	-	-	-	0.533^a
MC			-	-	-	-0.638^b	-	0.467^a	-	0.684^b	0.6683^a	-	0.278^a	0.666^b	0.657^a
pH				-	-	-	-	-	-	-	0.562^a		0.263^a	0.453^a	-
OC					-	-0.517^a	0.596^a		-	-	0.689^b	0.544^a	-	-	0.743^c
TN						-	0.569^a		-	0.537^a	-	-	-	-	0.507^a
AP							0.638^b		-	0.628^b	0.572^a	-	-	0.843^c	-
K										0.804^c	-	-	-	-	-

Table 2.5. Correlation coefficient (r) values among physico-chemical properties of soil with various microbial population, biological and biochemical properties of soil in bun cultivation (BCU) site at middle (ML) layer ($P \leq 0.05$).

Soil properties	ST	MC	pH	OC	TN	AP	K	AT	RF	FP	BP	C_{mic}	DHA	URA	PA
AT				0.508^a	-	-			0.692^b	0.541^a	0.646^b	-	0.626^b	-	0.541^a
ST		-	0.558^a	0.716^b	-	-	-	0.900^c	0.756^b	-	0.641^b	-	0.577^a	-	-
MC			-	0.622^b	-	-	-	0.538^a	-	-	-	-	-	-	0.533^a
pH				-	0.529^a	-	-	-	-	-	0.525^a	0.617^b	-	-	-
OC					0.582^a	-	0.499^a		-	0.626^b	-	-	-	0.528^a	0.680^b
TN						-	-0.501^a		-	-	-	-	-	0.554^b	-
AP							0.546^a		-	-	-	-	-	0.710^c	-
K										0.763^c	-	-	-	-	-

(**Note**: AT=ambient temperature, MC=moisture content, ST=soil temperature, pH=soil pH, OC=organic carbon, TN=total nitrogen, AP=available phosphorus, K=potassium, RF=rainfall, FP=fungal population, BP=bacterial population, C_{mic}=microbial biomass carbon, URA=urease, DHA=dehydrogenase, PA=phosphatase

Values marked with a, b and c are significant at *($P \leq 0.05$)*, *($P \leq 0.01$)* and *($P \leq 0.001$)* respectively; insignificant values are marked with '-'.

Table 2.6. Correlation coefficient (r) values among physico-chemical properties of soil with various microbial population, biological and biochemical properties of soil in Bun cultivation (BCU) site at subsurface (SSL) layer ($P \leq 0.05$).

Soil properties	ST	MC	pH	OC	TN	AP	K	AT	RF	FP	BP	C_{mic}	DHA	URA	PA
AT				0.572[a]	-	-			0.692[b]	0.540[a]	0.743[c]	-	-	-	0.540[a]
ST		-	-	-	-	-	-	0.891[c]	0.658[b]	-	0.640[b]	-	-	-	-
MC			-	0.562[b]	-	-	-	0.570[b]	-	-	-	-	-	-	-
pH				-	-	-0.499[a]	-	-	-	-	-	-	-	-	-
OC					-	0.555[a]	-		0.760[c]	0.602[b]	-	-	-	0.651[b]	-
TN						-	-		-	0.685[a]	-	-	-	-	-
AP							-		-	-	-	-	-	0.794[c]	-
K										-	-	-	-	-	-

Table 2.7. Correlation coefficient (r) values among physico-chemical properties of soil with various microbial population, *biological and biochemical* properties of soil in Forest (FS) site at surface (SL) layer *($P \leq 0.05$)*.

Soil properties	ST	MC	pH	OC	TN	AP	K	AT	RF	FP	BP	C_{mic}	DHA	URA	PA
AT				-	-	-			0.692[b]	-	0.762[c]	-	-	-	-
ST		0.526[a]	-0.705[a]	-	-0.525[a]	-	0.584a	0.844[c]	0.805[c]	-	0.898[c]	-	0.647[b]	-	-
MC			-	-	-	0.486[a]	-	0.464[a]	-	0.533[a]	-	-	-	-	0.523[a]
pH				-	-	-	-0.476[a]	0.458[a]	-	-	-0.613[a]	-	-	-	-
OC					-	-	0.607[a]		-	-	-	0.576[a]	-	-	-
TN						-	-		-	-	-	-	-	-	-
AP							-		-	0.573[a]	-	-	-	-	-
K										-	0.662[b]	-	-	-	-

(**Note**: AT=ambient temperature, MC=moisture content, ST=soil temperature, pH=soil pH, OC=organic carbon, TN=total nitrogen, AP=available phosphorus, K=potassium, RF=rainfall, FP=fungal population, BP=bacterial population, C_{mic}=microbial biomass carbon, URA=urease, DHA=dehydrogenase, PA=phosphatase

Values marked with a, b and c are significant at *($P \leq 0.05$)*, *($P \leq 0.01$)* and *($P \leq 0.001$)* respectively; insignificant values are marked with '-'.

Table 2.8. Correlation coefficient (r) values among physico-chemical properties of soil with various microbial population, biological and biochemical properties of soil in Forest (FS) site at middle (ML) layer *(P≤0.05)*.

Soil properties	ST	MC	pH	OC	TN	AP	K	AT	RF	FP	BP	C_{mic}	DHA	URA	PA
AT				-	-0.637[b]	-			0.692[b]	-	0.767[c]	-	-	-	-
ST		0.535[a]	-0.498[a]	0.624[b]	-0.550[a]	-	-	0.834[c]	0.735[b]	-	0.883[c]	-	-	-	-
MC			-	-	-	-	-	0.454[a]	-	-	0.504[a]	-	-	-	0.574[b]
pH				-	-	-	-	-	-	-	-	-	0.332[b]	-	-
OC					-0.537[a]	-	-		-	-	0.579[a]	-	-	-	-
TN						-	-		-	-	-0.559[a]	-	-	-	-
AP							-		-	-	-	-	-	-	-
K										0.509[a]	-	-	-	-	-

Table 2.9. Correlation coefficient (r) values among physico-chemical properties of soil with various microbial population, biological and biochemical properties of soil in Forest (FS) site at subsurface (SSL) layer *(P≤0.05)*.

Soil properties	ST	MC	pH	OC	TN	AP	K	AT	RF	FP	BP	C_{mic}	DHA	URA	PA
AT				-	-	-			0.692[b]	-	0.735[c]	-	-	-	-
ST		0.586[a]	-0.659[b]	-	-	-	0.644[b]	0.839[c]	0.775[c]	-	0.786[c]	-	-	-	-
MC			-	-	-	-	-	0.613[b]	-	-	0.722[b]	-	-	-	0.597[b]
pH				-	-	-	-	-	-	-	-	-	-	-	-
OC					-	0.582[a]	-		-	-	-	-	-	0.610[a]	-
TN						-	-		-	-	-	-	-	-	-
AP							-		-	0.551[a]	-	-	-	-	-
K										-	0.700[b]	-	-	-	-

(**Note**: AT=ambient temperature, MC=moisture content, ST=soil temperature, pH=soil pH, OC=organic carbon, TN=total nitrogen, AP=available phosphorus, K=potassium, RF=rainfall, FP=fungal population, BP=bacterial population, C_{mic}=microbial biomass carbon, URA=urease, DHA=dehydrogenase, PA=phosphatase

Values marked with a, b and c are significant at *(P≤0.05)*, *(P≤0.01)* and *(P≤0.001)* respectively; insignificant values are marked with '-'

Chapter- III

ENUMERATION OF FUNGAL AND BACTERIAL POPULATION IN BUN AND FOREST SOIL

3.1 Introduction

Microorganisms perform a vital role in the soil as they are responsible for the decomposition of the organic matter and they play an important role in the transformation of various nutrients from non-available to available forms that help in the nutrient cycling and functioning of soil ecosystem (Chaney *et al.,* 1978). Soil microorganisms are one of the most sensitive markers and the most useful agents for classifying disturbed or contaminated system. The regulated application of organics as a practice will energize the living microorganisms of the soil, involved in biochemical activity of importance to soil fertility and plant nutrition (Gaur *et al.*, 1990). Microbial activity is an important key to understanding biological processes in a soil. The variation in soil microbial activity in response to stress eventually effect plant productivity and ecosystem functioning. The size and activity of soil microbial population is critical to overall soil use and sustainability. Soil organisms contribute to the maintenance of soil quality through their control of many key processes such as decomposition, nutrient cycling and availability and soil aggregation. Some microorganisms may be suppressed and others may proliferate in the vacant ecological niches. This may in turn lead to successions in the microbial community and thus to altered activities at later point in time.

Soil microbial populations may act as early indicators of changes in soil quality as they can respond much more rapidly to perturbations than

other indicators such as soil C or N (Kennedy and Papendick, 1995). Ecosystem functioning before and after disturbance can be governed by soil microbial population dynamics (Kennedy and Smith, 1995).

Populations of soil microbes are generally carbon-limited and the addition of organic carbon to the soil stimulates the population growth of microbes until it eventually become limited by available nitrogen (Martin and Johnson; 1995, Garten *et al.,* 2000; Garten and Wullschleger, 2000). Soil microorganisms also process plant litter and residues into SOM, which improve soil quality by increasing soil aggregation and aeration and decreasing soil bulk density (Dominy and Haynes, 2002; Franzluebbers *et al.,* 1999; Spaccini *et al.,* 2002).

Microbial diversity encompasses a spectrum of microscopic organisms including bacteria and fungi. The agriculturally important microorganisms have their own, unique place and scope in the overall context of agro-biodiversity. They play an important role in nutrient acquisition for plants viz, N, P, K and other microelements. Microbial activity between 0.3 and 3 m depth contributed about 50% to the microbial activity in these soils, confirming the importance of the sub-soil in C cycling (Veldkamp *et al.,* 2003).

The plant growth stages also had an important impact on soil enzyme activities and microbial populations, with strong positive associations between soil microorganisms and enzyme activities (Li, *et al.,* 2002). The activity of soil microorganisms is strongly linked to the activity of enzymes, and soil managements (including crop rotations, fertilization, tillage and crop

residue placement) strongly influence the activity of soil enzymes (Miller and Dick, 1995; Deng and Tabatabai, 1994a, b; Klose *et al.,* 1999).

Microbial community structure is often based on detectable differences in terms of growth rate, substrate-use efficiencies, morphology and C/N content of biomass. These factors however differ markedly between fungi and bacteria and within groups of fungi and bacteria. Moreover cultivated microorganisms may reflect the natural microbial community because they represent only 0.001% to 0.3% of total direct counts (Amann *et al.,* 1995; Torsvik *et al.,* 1990). Increased soil pH in the acidic range caused a shift toward dominance of the bacterial community, while fungal populations were unaffected (Frostegard *et al.,* 1993; Pennanen, 2001). The growth of soil bacteria was inhibited at pH 4.6 and facilitated with increasing soil pH (Arao, 1999). Shifting of microbial community toward fungal dominance resulting from decreasing soil pH was attributed to reduced competition from the bacterial community.

3.2 Methodology

3.2.1 Enumeration of microbial populations

Isolation of fungal and bacterial populations was done by serial dilution plate method (Waksman, 1952; Parkinson *et al.,* 1971). One gram of soil sample was taken into the 250 ml conical flask containing 100 ml of sterilized distilled water to make 1:100 dilutions. The flask was swirled for 15 minutes to prepared homogenous solution. 10 ml of this solution was transferred to another flask containing 90 ml of sterilized distilled water by using sterilized pipette to make 1:1000 dilutions. Thereafter, 10 ml of this

solution was then transferred again to another flask containing 90 ml of sterilized distilled water to make the final 1:10000 dilution.

3.2.2 Fungal population

The study of fungal population was done by rose bengal agar medium (Martin, 1950). One milliliter of the soil dilution (1:1000) was transferred into a petridish containing rose bengal agar medium. The petridish were then rotated gently so as to disperse the suspension. Three replicates were maintained for each sample. The petridishes were incubated turning upside down in a BOD incubator at 25 ± 1^{0}C for 5 days. The colony forming unit (CFU) of fungi was estimated by counting the number of fungal colonies. The colony form unit of fungi per gram soil was calculated on the dry weight basis by using the formula below:

$$CFU\ of\ fungi\ /\ g\ D_w = \frac{Number\ of\ colony\ form\ x\ dilution\ factor\ x\ inoculums}{Dry\ weight\ of\ the\ soil\ (g)}$$

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

Fungal species were then identified based on their morphology and reproductive structures consulting the monographs by Subramaniam (1917), Barnet and Hunter (1972) and Domsch *et al.*, (1980). Determination of relative abundance of fungal species was done by using the formula below:

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

Rose Bengal Agar medium (Martin, 1950)

Agar	20 g
KH2PO4	10 g
MgSO4.7H2O	0.5 g
Peptone	5 g
Dextrose	10 g
Rose bengal	3.3 ml
Streptomycin	30 mg
Distilled water	1000 ml

3.2.3 Bacterial population

Isolation of bacterial species was done by using nutrient agar medium (Difco manual, 1953). 0.5 ml of the soil solution from 1:10000 dilution was transferred to a petridish containing nutrient agar medium. Three replicates were maintained for each sample. The plates were then rotated to allow dispersing the suspension uniformly. The inoculated plates were incubated upside down at 30 ± 1^0 C in bacteriological incubator. Colony form unit (CFU) of bacteria was estimated by counting the number of bacterial colonies. The CFU of bacteria per gram soil was then calculated on the dry weight basis.

$$CFU\ of\ bacteria\ /\ g\ D_w = \frac{Number\ of\ colony\ form\ x\ dilution\ factor\ x\ inoculum}{Dry\ weight\ of\ the\ soil\ (g)}$$

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

The following formula was used to determine the relative abundance of bacterial species:

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

Nutrient agar medium (Difco manual, 1953):

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

The final pH of the medium was adjusted to 7.3.

3.2.4 Statistical analysis

Data obtained from the above, the following indices of fungal and bacterial species are evaluated:

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

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

2). Index of dominance ©; (Simpson, 1949).

$$C = \sum(n_i / N)^2$$

3) Index of similarity; Sorensen (S) and Jaccard (J) (Krebs, 1989).

$$S = \frac{2C}{S_1 + S_2}$$

Where, S_1 = number of species in site 1.

S_2 = number of species in site 2.

C = number of species that are common in both site 1 and site 2.

$$J = j / r \, x \, 100$$

Where, j = number of species found in both of two sites.

R = number of species found in only one site or the other.

3.3. Results

3.3.1. Fungal population

The fungal population shows an increase in the first year of cultivation and a decreased of population in the second year in both the sites. In the first year, the population dropped in August at BCU and FS sites in all the three depths (Fig 3.1.1). In the second year, the population dropped in July in the middle and subsurface layers whereas at surface layer it dropped in October at both the sites.

At the surface layer (SL) of the bun cultivation the fungal population ranges from 41.6 x 10^3 and 14.4 x 10^3 CFU in August and September in the first year and 41.5 x 10^3 and 14.2 mg x 10^3 CFU in August and September in the second year. In forest site (FS), the population ranges from 70.2 x 10^3 and 26.4 x 10^3 CFU in September and November during the first year and 70.1 x 10^3 and 26.6 x 10^3 CFU in September and November during the second year (Fig 3.1.1).

In middle layer (ML) soil, the population ranges from 20.9 x 10^3 and 6.94 x 10^3 CFU in September and July during the first year and 20.8 x 10^3 and 5.4 x 10^3 CFU in September and August during the second year of bun cultivation (BCU).

In forest site (FS), the population ranges from 52.1 x 10^3 and 6.5 x 10^3 CFU in September and June during the first year and 51.9 x 10^3 and 6.1 x 10^3 CFU in September and June during the second year (Fig 3.1.1).

Fungal population at the subsurface layer (SSL) soil ranges from 11.7 x 10^3 and 1.53 x 10^3 CFU in May and August during the first year and 12.1 x 10^3 and 1.51 x 10^3 CFU in December and August during the second year of bun cultivation (BCU). In forest site (FS), the population ranges from 49.9 x 10^3 and 2.99 x 10^3 CFU in September and June during the first year and 49.2 x 10^3 and 4.63 x 10^3 CFU in September and July during the second year (Fig 3.1.1).

Shannon diversity index of fungal species during the first year ranges from 1.02 to 2.87 in subsurface layer in September and November at bun cultivation (BCU) site and it ranges from 1.01 to 2.61 in surface and middle layers in September and June at forest (FS) site respectively (Fig 3.1.2). Whereas in the second year, it ranges from 1.03 to 2.74 at surface layer in August and May at bun cultivation (BCU) site and it ranges from 1.02 to 2.75 in surface layer in October and December at forest (FS) site (Fig 3.1.2).

Simpson dominance index of fungal species during the first year ranges from 0.11 to 0.47 in subsurface layer in July and October at bun cultivation (BCU) site and it ranges from 0.12 to 0.46 in surface layer in September and June at forest (FS) site respectively (Fig 3.1.3). Whereas in the second year, it ranges from 0.11 to 0.42 in middle layer in October and August at bun cultivation (BCU) site and it ranges from 0.11 to 0.41 in middle and surface layers in December and October at forest (FS) site (Fig 3.1.3).

Sorenson and Jaccard similarity index of fungi shows a similarity distribution in the first and second years of both bun cultivation (BCU) and forest (FS) sites. Sorensen similarity index during the first year shows a similarity of 0.56(SL X ML), 0.64(SL X SSL), 0.69(ML X SSL) and Jaccard similarity index shows a similarity of 0.62(SL X ML), 0.48(SL X SSL), 0.78(ML X SSL) respectively at bun cultivation (BCU) site (Fig 3.1.4).

In forest site, Sorensen similarity index shows a similarity of 0.57(SL X ML), 0.51(SL X SSL), 0.64(ML X SSL) and Jaccard similarity index shows a similarity of 0.61(SL X ML), 0.57(SL X SSL) and 0.53(ML X SSL) respectively (Fig. 3.1.4).

In the second year, Sorensen similarity index shows a similarity of 0.64(SL X ML), 0.49(SL X SSL), 0.67(ML X SSL) and Jaccard similarity index shows a similarity of 0.49(SL X ML), 0.53(SL X SSL), 0.57(ML X SSL) respectively at bun cultivation (BCU) site (Fig. 3.1.4).

In forest site, Sorensen similarity index shows a similarity of 0.54(SL X ML), 0.61(SL X SSL), 0.57(ML X SSL) and Jaccard similarity index shows a similarity of 0.53(SL X ML), 0.67(SL X SSL), 0.57(ML X SSL) respectively (Fig 3.1.4).

The fungal species shows similar distribution in both bun cultivation and forest sites. The surface layer of both the sites shows a higher distribution of 76% in bun cultivation and 84% in forest sites. Subsurface layer shows the lowest distribution of 64% in bun cultivation site whereas middle layer shows lowest distribution of 68% in forest site (Fig 3.1.5).

The one-way analysis of variance (ANOVA) of the fungal population shows significant variation ($P \leq 0.05$) between the bun cultivation (BCU) and forest (FS) sites (Table. 3.3.1).

The middle (ML) layer of bun cultivation (BCU) site shows insignificant variation ($P \leq 0.05$) with surface (SL) layer of forest site. The subsurface (SSL) layer of bun cultivation (BCU) also shows insignificant variation ($P \leq 0.05$) with the surface (SL) and middle (ML) layers of forest site (Table. 3.3.1).

The one-way analysis of variance (ANOVA) of the fungal population shows insignificant variation ($P \leq 0.05$) in the different depths of forest and bun cultivation (Table. 3.3.2).

The Correlation coefficient (r) values of fungal population in surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and Forest (FS) sites at $P \leq 0.05$ is positively correlated between all the depths of the same site in both bun cultivation and forest sites but no significant correlation between the two sites of the three depths (Table 3.3.3).

Correlation coefficient (r) values of fungal population with various biological, microbial population and physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that fungal population is positively correlated with ambient temperature (0.561 at $P \leq 0.05$), soil temperature (0.533 at $P \leq 0.05$), total nitrogen (0.537 at $P \leq 0.05$) and exchangeable potassium (0.804 at $P \leq 0.001$) respectively (Table 3.3.4).

Fungal population in the middle layer of bun cultivation is positively correlated with ambient temperature (0.541 at $P \leq 0.05$), organic carbon (0.626 at $P \leq 0.01$) and exchangeable potassium (0.763 at $P \leq 0.001$). In the subsurface layer, fungal population is positively correlated with ambient temperature (0.540 at $P \leq 0.05$) and organic carbon (0.602 at $P \leq 0.01$) and negatively correlated with bacterial population (-0.613 at $P \leq 0.05$).

Correlation coefficient (r) values of fungal population with various biological, microbial population and physico-chemical properties of soil in forest (FS) site at surface (SL) layer shows that fungal population is positively correlated with soil moisture content (0.533 at $P \leq 0.05$), available phosphorus (0.573 at $P \leq 0.05$) and microbial biomass carbon (0.612 at $P \leq 0.05$) respectively (Table 3.3.5).

In the middle layer of forest site, fungal population is positively correlated with exchangeable potassium (0.509 at $P \leq 0.05$) and microbial biomass carbon (0.814 at $P \leq 0.001$). In the subsurface layer, fungal population is positively correlated with available phosphorus (0.551 at $P \leq 0.05$) and microbial biomass carbon (0.602 at $P \leq 0.01$).

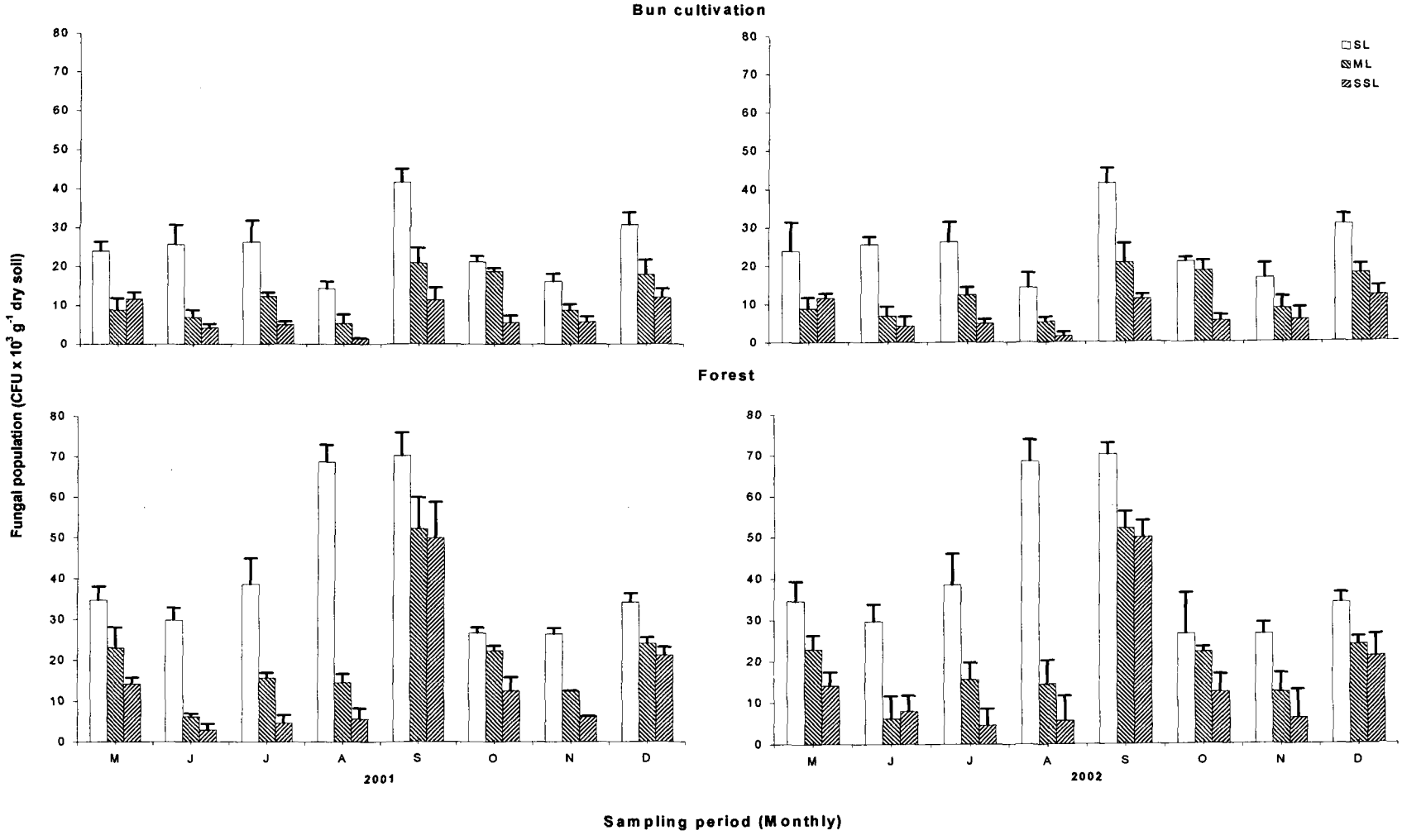

Fig. 3.1.1. Fungal populations of soil at surface (SL), middle (ML) and subsurface (SSL) soil layers of bun cultivation (BCU) and Forest (FS) sites.

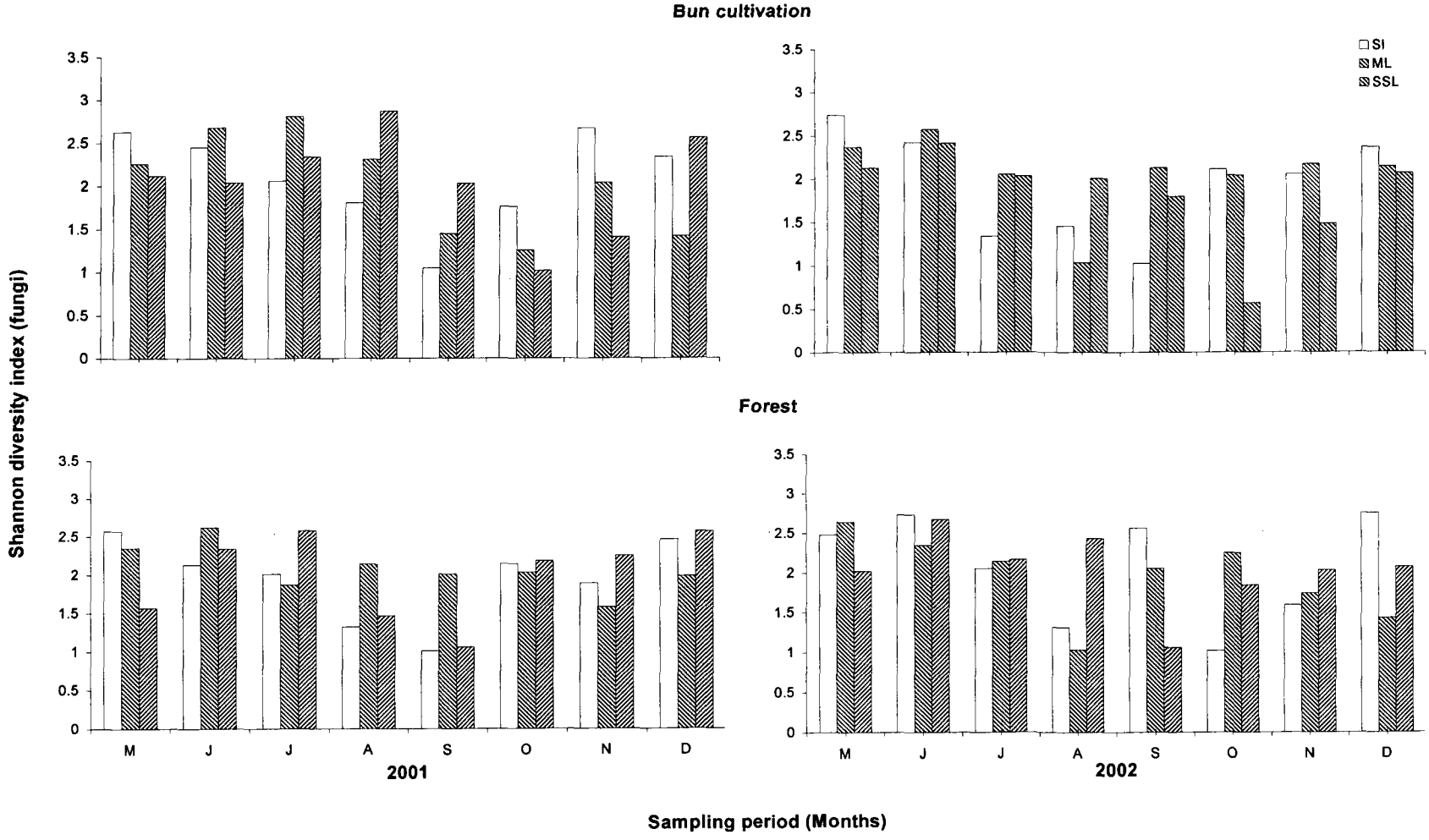

Fig. 3.1.2. Shannon diversity index of fungi at surface (SL), middle (ML) and subsurface (SSL) soil layers of bun cultivation (BCU) and Forest (FS) sites.

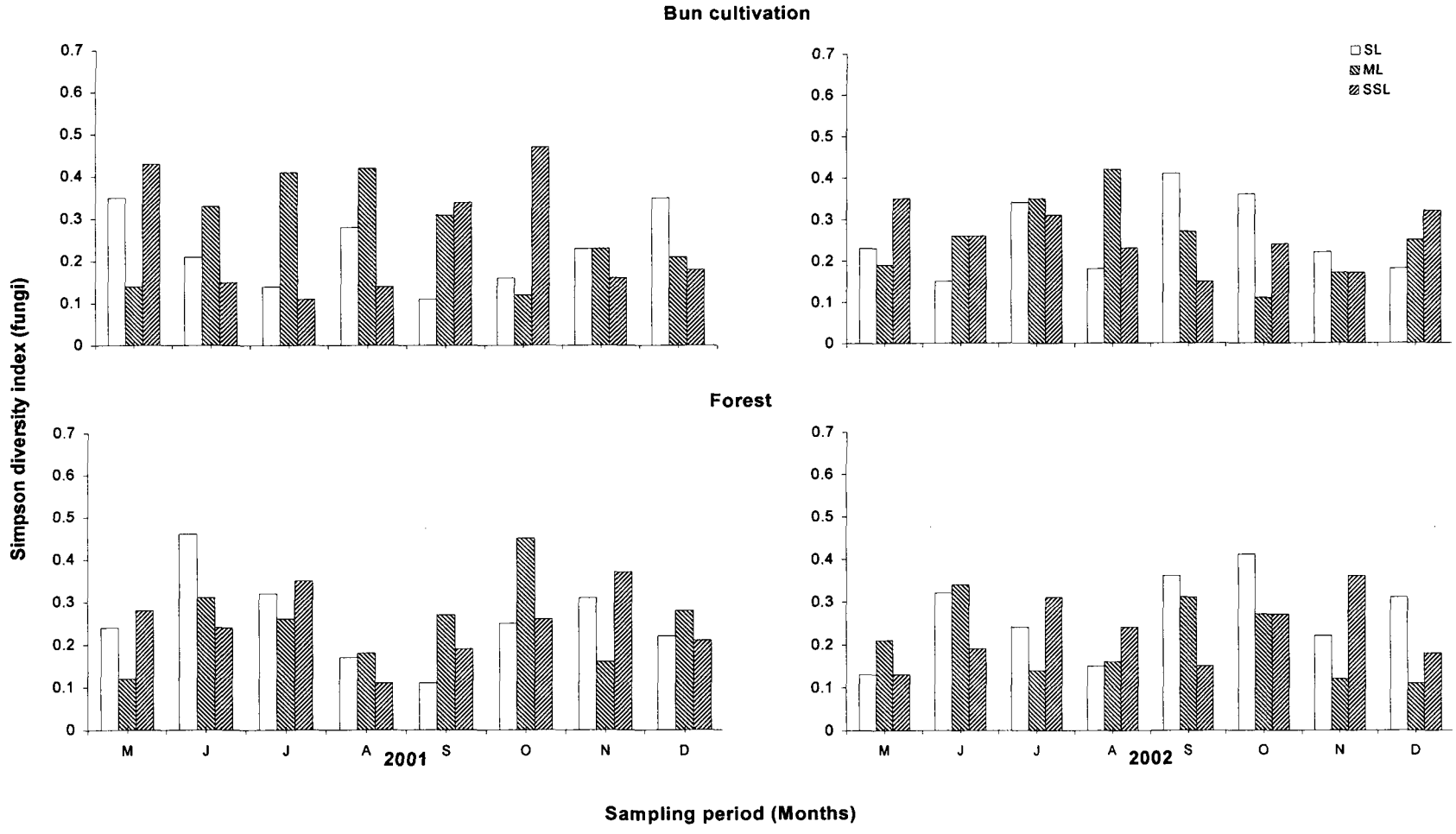

Fig. 3.1.3. Simpson diversity index of fungi at surface (SL), middle (ML) and subsurface (SSL) soil layers of bun cultivation (BCU) and Forest (FS) sites.

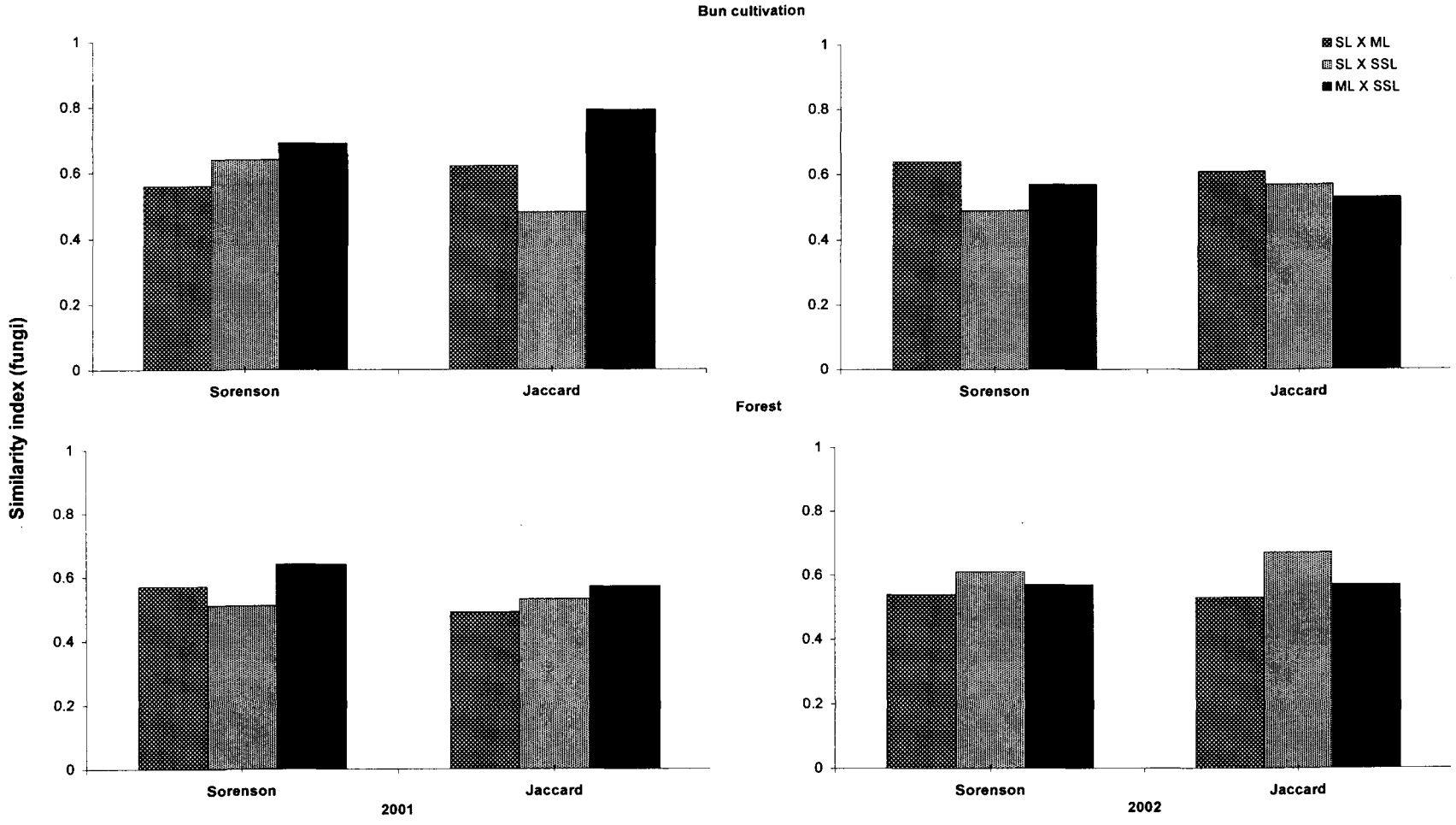

Fig. 3.1.4. Coefficient of Sorensen and Jaccard similarity index of soil fungi in paired layer of surface (SL), middle (ML) and subsurface (SSL) layers of forest (FS) and bun cultivation (BCU) sites.

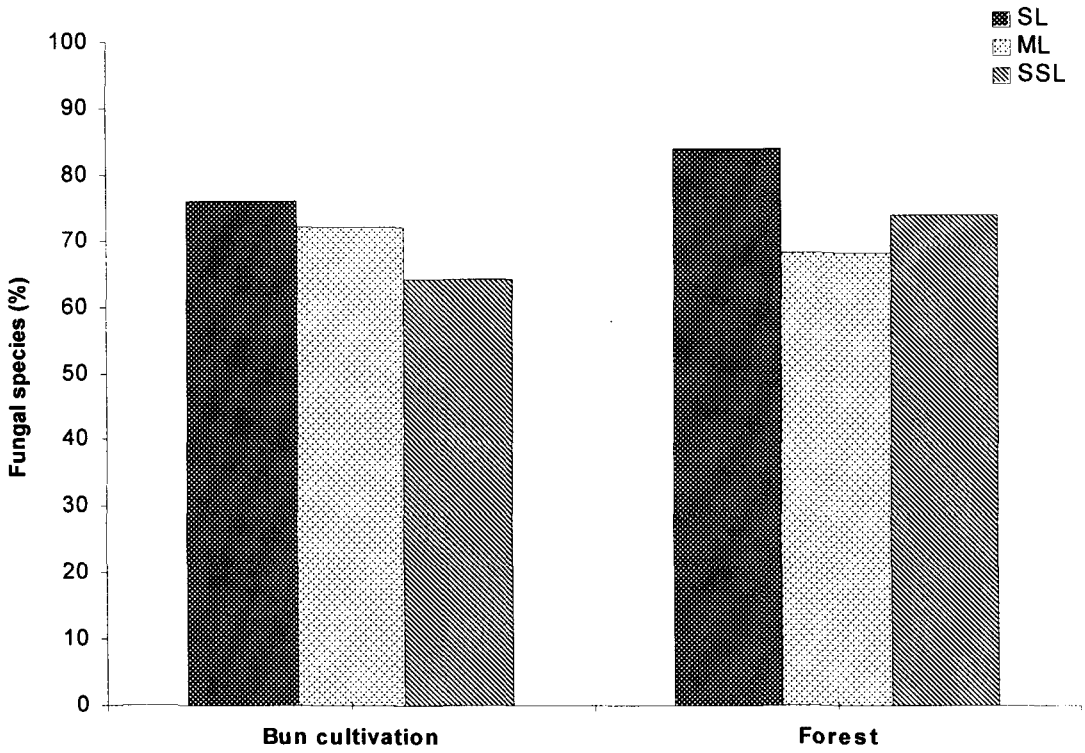

Fig. 3.1.5. Distribution of fungal species (%) at surface (SL), middle (ML) and subsurface (SSL) layers in bun cultivation (BCU) and forest (FS) sites.

Table 3.1.1. Monthly variation in the population of fungal species per gram dry soil x 10^3 in bun cultivation (BCU) site at surface (SL) layer (0-10 cm) depth.

Sl. No.	Fungal Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Acremonium murorum*	-	0.396 (0.801)	-	-	0.272 (0.549)	-	-	-	-	-	0.541 (1.092)	0.187 (0.377)	-	-	-	-
2	*Aspergillus aluteceus*	-	-	-	-	-	-	1.023 (2.066)	-	-	-	-	-	-	-	-	1.978 (3.995)
3	*A. clavatus*	-	2.341 (4.728)	-	-	5.252 (10.609)	-	-	1.212 (2.448)	0.742 (1.498)	3.245 (2.565)	-	5.873 (11.863)	-	-	2.458 (4.965)	-
4	*A. Flavus*	2.654 (5.361)	-	-	-	-	-	1.362 (2.751)	-	-	-	-	-	1.987 (4.013)	-	1.269 (2.563)	-
5	*A. niger*	-	-	-	1.236 (2.496)	-	-	-	-	-	-	-	-	-	2.357 (4.761)	-	-
6	*Cladosporium cladosporioides*	-	-	-	-	2.365 (4.777)	-	-	-	-	-	1.034 (2.088)	-	-	-	-	-
7	*Chaetomium globasum*	-	0.327 (0.660)	-	-	-	-	-	-	0.129 (0.260)	-	-	-	-	-	0.658 (1.329)	-
8	*Colletotrichum dematium*	-	-	-	2.347 (4.740)	-	-	-	-	-	-	-	-	-	-	-	-
9	*Cylindrocarpon magnusianum*	-	-	2.122 (4.286)	-	-	-	-	-	-	-	-	-	-	-	-	-
10	*C. olidum*	-	-	-	-	-	-	-	-	-	3.568 (7.207)	-	-	-	-	-	-
11	*Fusarium semitectum*	-	-	0.286 (0.577)	-	-	0.169 (0.341)	-	-	-	-	2.135 (4.316)	-	-	-	0.547 (1.106)	-
12	*F. sporotrichioides*	-	-	-	0.571 (1.154)	-	-	-	1.324 (2.677)	-	-	-	-	-	0.925 (1.870)	0.846 (1.710)	-
13	*Gongoronella butleri*	0.177 (0.357)	-	-	0.268 (0.541)	-	-	1.116 (2.256)	-	-	-	-	0.986 (1.993)	-	-	-	1.249 (2.525)
14	*Humicola fuscoatra*	-	2.676 (5.410)	-	-	2.928 (5.916)	-	-	0.987 (1.995)	-	-	-	-	2.248 (4.545)	-	-	0.557 (1.126)
15	*H. grisea*	-	-	-	0.884 (1.787)	-	1.854 (0.503)	-	-	-	0.627 (5.311)	-	-	-	-	1.725 (3.487)	-
16	*Idriella lunata*	0.521 (1.053)	-	-	-	-	0.249 (0.503)	-	2.369 (4.790)	0.721 (1.457)	-	-	-	1.436 (2.903)	-	-	0.992 (2.011)
17	*Mammaria echinobotryoides*	-	-	-	-	0.736 (1.488)	-	-	-	-	-	-	-	1.156 (2.337)	-	-	-
18	*M. vinacea*	-	-	-	1.261 (2.549)	-	-	-	0.154	-	-	0.882 (1.783)	-	-	-	-	-
19	*Mucor racemosus*	1.139 (2.303)	-	-	-	-	-	0.751 (1.518)	0.853 (0.311)	-	-	0.901 (1.821)	-	-	--	-	1.315 (2.658)
20	*Oidiodendron echinulatum*	-	-	-	-	-	3.456 (6.988)	-	-	2.487 (4.927)	-	-	-	-	-	0.946 (1.912)	-
21	*O. griseum*	0.218 (0.440)	-	-	-	-	-	-	1.159 (2.343)	-	-	-	-	0.987 (1.995)	-	-	-
22	*Penicillium artrovenetum*	-	-	-	2.368 (0.744)	-	-	3.482 (7.041)	4.365 (8.826)	-	3.128 (6.334)	4.218 (8.520)	-	-	-	-	1.958 (3.959)

23	*P. canescens*	-	-	0.758 (1.532)	-	-	-	-	-	-	-	-	-	3.648 (7.376)	-	-	-
24	*P. chrysogenum*	-	1.654 (3.344)	-	-	2.374 (4.801)	-	-	-	-	-	-	-	-	-	-	1.264
25	*P. frequentans*	-	-	-	-	-	-	3.265 (6.601)	-	-	-	-	0.784 (1.588)	-	-	-	-
26	*P. funiculosum*	-	-	2.573 (5.202)	-	-	0.142 (0.285)	-	-	1.417 (2.865)	-	-	-	-	-	-	-
27	*P. granulatum*	-	-	-	-	-	-	2.364 (4.780)	-	-	-	-	-	0.529 (1.065)	-	-	-
28	*P. jensenii*	1.445 (2.921)	-	-	-	-	-	-	1.025 (2.072)	-	-	2.149 (4.345)	-	-	-	-	-
29	*P. lanosum*	-	-	2.452 (5.746)	-	-	-	-	-	-	-	-	1.254 (2.535)	-	-	-	.943 (1.906)
30	*P. purpurogenum*	0.764 (1.544)	-	-	-	-	-	-	3.166 (6.401)	-	0.687 (1.389)	-	-	-	-	2.857 (5.776)	-
31	*P. restrictum*	-	-	-	1.184 (2.394)	-	-	-	-	2.545 (5.154)	-	-	-	-	-	-	-
32	*P. rubrum*	-	-	-	3.217 (6.504)	2.421 (4.895)	-	-	-	-	-	-	-	2.745 (5.550)	-	-	1.052 (2.127)
33	*P. waksmanii*	-	1.868 (3.777)	-	-	-	0.742 (1.501)	-	-	-	2.364 (4.781)	-	-	-	1.192 (2.412)	-	-
34	*Phoma eupyrena*	-	-	-	-	-	3.261 (6.593)	1.748 (3.543)	-	-	-	0.467 (0.944)	1.668 (3.372)	-	-	-	-
35	*P. medicaginis*	-	-	-	-	1.348 (2.725)	-	-	0.769 (1.554)	-	-	-	-	-	0.942 (1.904)	-	-
36	*Plectospaerella cucumerina*	-	-	-	2.239 (4.527)	-	-	-	-	2.594 (5.245)	-	-	-	-	-	-	-
37	*Pythium intermedium*	-	-	1.847 (2.734)	2.105 (4.256)	-	-	-	-	-	-	-	-	-	2.326 (4.703)	-	3.416 (6.907)
38	*Rhizopus oryzae*	-	-	-	-	-	-	2.421 (2.895)	1.056 (2.135)	-	3.446 (6.967)	0.874 (1.757)	-	-	-	-	-
39	*Staphylotrichum coccosporum*	-	2.714 (5.487)	-	-	-	0.974 (1.969)	-	-	-	-	-	3.153 (6.375)	1.386 (2.802)	-	1.759 (3.556)	-
40	*Trichoderma harzianum*	-	-	0.468 (0.946)	1.054 (2.131)	-	-	0.721 (4.998)	-	-	0.581 (1.174)	-	-	-	1.424 (2.879)	-	0.662 (1.338)
41	*T. polysporum*	-	0.992 (2.005)	-	-	-	2.472 (4.998)	-	-	2.328 (4.707)	-	-	-	-	-	1.837 (3.714)	-
42	*T. viride*	-	-	-	3.146 (6.361)	-	-	-	-	-	-	-	-	-	2.363 (4.777)	-	-
43	*Verticillium arbo-atrum*	-	-	-	1.649 (3.335)	-	-	2.934 (5.935)	-	3.459 (6.990)	-	-	-	-	2.084 (4.213)	-	-
44	*Green sterile mycelia*	-	-	3.752 (7.586)	-	-	-	1.904 (3.849)	2.662 (5.382)	-	3.648 (7.376)	1.387 (2.804)	-	-	-	-	4.116 (8.322)
45	*White sterile mycelia*	2.628 (5.313)	-	-	3.247 (3.565)	-	-	-	-	-	1.159 (2.343)	-	0.924 (1.868)	0.652 (1.318)	-	-	-

Note: Percentage relative abundance values are marked with parentheses.

Table 3.1.2. Monthly variation in the population of fungal species per gram dry soil x 10^3 in bun cultivation (BCU) site at middle (ML) layer (10-20 cm) depth.

Sl. No.	Fungal Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Acremonium murorum*	2.774 (5.609)	-	-	3.268 (6.607)	1.843 (3.726)	-	-	4.025 (8.138)	-	-	1.336 (2.701)	-	2.924 (5.912)	-	-	-
2	*Aspergillus aluteceus*	-	-	3.851 (7.786)	-	-	-	2.147 (4.341)	-	3.224 (6.518)	-	-	-	-	-	4.548 (9.196)	2.351 (4.753)
3	*A. clavatus*	-	-	-	-	2.784 (5.629)	3.649 (5.356)	-	1.472 (2.973)	-	0.748 (1.512)	-	0.936 (1.892)	-	-	-	1.736 (3.510)
4	*A. Flavus*	-	-	-	-	3.843 (7.772)	1.863 (3.766)	2.664 (5.386)	-	-	-	3.647 (7.374)	-	-	-	-	5.218 (10.550)
5	*A. niger*	-	4.289 (8.672)	-	-	-	-	0.843 (1.704)	-	-	-	-	-	-	0.972 (1.965)	-	-
6	*Cladosporium cladosporioides*	-	-	-	3.284 (6.640)	-	-	-	-	-	-	-	-	2.152 (4.351)	1.346 (2.721)	-	-
7	*Chaetomium globasum*	1.497 (3.026)	-	-	-	3.774 (7.631)	-	-	0.846 (1.710)	-	-	-	-	-	3.582 (7.242)	1.094 (2.212)	-
8	*Colletotrichum dematium*	-	-	-	-	-	3.594 (7.267)	-	.768 (1.552)	-	-	2.986 (6.037)	-	-	-	-	1.529 (3.091)
9	*Cylindrocarpon magnusianum*	-	-	-	3.463 (7.002)	1.871 (3.783)	-	-	1.985 (4.013)	-	2.749 (5.558)	-	-	-	1.874 (3.789)	0.821 (1.660)	-
10	*C. olidum*	-	-	2.673 (5.404)	-	-	-	1.697 (3.431)	-	2.743 (5.546)	-	-	3.418 (6.911)	-	-	-	-
11	*Fusarium semitectum*	-	2.342 (4.735)	-	-	1.914 (3.870)	-	-	-	-	-	-	-	-	3.539 (5.133)	1.386 (2.802)	-
12	*F. sporotrichioides*	-	-	-	-	-	-	2.647 (5.352)	1.495 (3.022)	-	2.536 (5.127)	-	-	-	-	-	2.048 (4.141)
13	*Gongoronella butleri*	-	-	-	-	1.684 (3.405)	-	-	-	2.498 (5.050)	-	-	3.762 (7.606)	-	-	-	-
14	*Humicola fuscoatra*	-	-	3.943 (7.972)	-	-	-	-	-	-	-	-	-	-	2.674 (5.406)	3.693 (7.467)	-
15	*H. grisea*	-	-	1.679 (3.394)	-	-	2.886 (5.835)	-	-	-	-	-	-	1.787 (3.613)	-	-	3.438 (6.951)
16	*Idriella lunata*	-	4.267 (8.672)	-	-	2.371 (4.794)	-	-	-	1.072 (2.167)	-	-	-	-	-	-	2.428 (4.909)
17	*Mammaria echinobotryoides*	-	-	2.405 (4.862)	-	-	-	-	-	3.182 (6.502)	-	-	1.365 (2.760)	-	-	-	3.418 (6.911)
18	*M. vinacea*	-	-	-	-	-	2.643 (5.344)	-	3.216 (6.502)	-	-	1.989 (4.021)	-	-	-	-	-
19	*Mucor racemosus*	0.847 (1.712)	-	-	-	2.563 (5.182)	-	-	-	-	-	-	-	-	1.224 (2.474)	2.145 (4.761)	-
20	*Oidiodendron echinulatum*	-	-	3.618 (3.715)	-	-	-	-	-	-	-	-	3.438 (6.951)	-	-	-	1.329 (2.687)
21	*O. griseum*	-	-	-	-	-	-	3.480 (7.036)	1.526 (3.085)	-	0.843 (1.704)	-	-	1.362 (2.753)	-	-	-
22	*Penicillium artrovenetum*	-	-	-	-	1.274 (2.576)	-	-	-	-	-	2.537 (5.125)	-	-	1.425 (2.889)	-	-

23	*P. canescens*	-	4.217 (8.526)	-	-	-	-	-	-	-	-	-	0.872 (1.763)	1.526 (3.085)	-	-	-
24	*P. chrysogenum*	-	-	-	3.261 (6.593)	-	4.228 (8.549)	-	-	2.686 (5.431)	-	-	-	-	-	-	-
25	*P. frequentans*	-	-	-	-	-	-	3.653 (7.386)	1.542 (3.117)	-	-	2.165 (4.377)	-	-	-	3.194 (6.458)	-
26	*P. funiculosum*	-	-	-	2.284 (4.618)	1.093 (2.214)	-	-	-	-	-	-	-	3.367 (6.802)	-	-	-
27	*P. granulatum*	1.226 (2.476)	-	-	-	-	-	-	2.627 (5.311)	-	-	-	3.249 (6.569)	1.896 (3.833)	-	-	4.384 (8.864)
28	*P. jensenii*	-	-	-	3.627 (7.339)	-	2.324 (4.699)	-	-	4.637 (9.376)	-	-	-	-	-	-	-
29	*P. lanosum*	-	-	2.765 (5.590)	-	-	-	2.524 (5.103)	-	-	-	-	-	-	-	2.773 (5.607)	-
30	*P. purpurogenum*	-	-	-	0.863 (1.744)	1.794 (3.627)	-	-	1.262 (2.551)	-	3.542 (7.157)	-	-	-	-	-	-
31	*P. restrictum*	-	3.538 (7.153)	-	-	2.498 (5.050)	-	-	-	-	-	-	-	3.625 (7.329)	-	2.947 (5.048)	-
32	*P. rubrum*	3.629 (7.337)	-	-	-	-	4.529 (9.157)	-	-	-	1.958 (3.959)	-	-	-	-	-	-
33	*P. waksmanii*	-	-	-	-	-	3.264 (6.599)	4.275 (8.644)	-	3.627 (7.334)	-	-	-	-	-	-	2.639 (5.336)
34	*Phoma eupyrena*	-	3.783 (7.649)	-	-	-	-	-	1.062 (2.147)	-	-	1.328 (2.685)	-	-	-	-	-
35	*P. medicaginis*	-	-	-	3.774 (7.631)	-	2.304 (4.658)	-	-	-	-	-	-	-	-	2.367 (4.786)	3.012 (6.091)
36	*Plectospaerella cucumerina*	-	-	-	-	2.192 (4.432)	-	-	-	-	-	-	1.843 (3.726)	0.762 (1.542)	-	-	-
37	*Pythium intermedium*	-	-	-	-	-	3.037 (6.141)	-	-	-	3.563 (7.204)	-	-	-	-	-	2.094 (4.234)
38	*Rhizopus oryzae*	1.438 (2.907)	-	-	-	-	-	5.124 (10.360)	-	-	-	3.827 (7.738)	-	-	-	-	-
39	*Staphylotrichum coccosporum*	-	3.349 (6.771)	-	-	-	-	-	-	-	-	-	-	-	2.452 (4.957)	1.237 (2.501)	-
40	*Trichoderma harzianum*	-	-	-	-	1.134 (2.292)	-	1.675 (3.386)	-	4.267 (8.627)	-	-	-	-	-	-	-
41	*T. polysporum*	-	-	-	-	2.554 (5.164)	-	-	-	-	-	4.613 (9.327)	2.491 (7.064)	-	-	-	-
42	*T. viride*	-	0.943 (1.906)	-	-	-	1.249 (2.525)	2.352 (4.755)	-	-	-	-	4.184 (8.460)	3.216 (6.502)	-	-	-
43	*Verticillium arbo-atrum*	-	-	-	3.964 (8.015)	3.625 (7.329)	-	1.273 (2.574)	-	-	2.692 (5.443)	-	-	-	-	-	0.891 (1.801)
44	*Green sterile mycelia*	0.629 (1.271)	-	-	-	2.054 (4.153)	-	-	-	-	-	-	-	3.258 (6.587)	-	-	-
45	*White sterile mycelia*	-	-	-	-	1.638 (3.312)	2.067 (4.179)	0.826 (1.672)	-	-	-	-	2.329 (4.709)	-	-	-	-

Note: Percentage relative abundance values are marked with parentheses.

Table 3.1.3. Monthly variation in the population of fungal species per gram dry soil x 10^3 in bun cultivation (BCU) site at subsurface (SSL) layer (20-30 cm) depth.

Sl. No.	Fungal Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Acremonium murorum*	-	3.249 (6.674)	-	-	1.862 (3.764)	-	-	-	-	-	2.438 (4.929)	-	-	5.104 (10.320)	-	2.326 4.703)
2	*Aspergillus aluteceus*	2.674 (5.406)	-	3.124 (6.316)	-	-	-	5.491 (11.102)	-	-	-	-	2.916 (5.896)	4.281 (8.656)	-	-	-
3	*A. clavatus*	-	-	2.145 (4.337)	3.279 (6.674)	-	-	-	-	-	4.061 (8.211)	-	-	3.283 (6.638)	-	-	4.327 (8.749)
4	*A. Flavus*	-	-	-	3.205 (6.480)	-	1.652 (3.340)	-	1.119 (2.262)	-	-	-	-	-	3.924 (1.868)	4.683 (9.469)	-
5	*A. niger*	-	2.942 (5.948)	-	-	-	1.976 (3.995)	-	-	3.862 (7.808)	1.724 (3.485)	-	-	1.781 (3.601)	-	-	-
6	*Cladosporium cladosporioides*	-	-	-	-	3.869 (7.823)	-	-	-	-	-	-	-	-	-	2.749 (5.558)	3.654 (7.388)
7	*Chaetomium globasum*	-	-	3.413 (6.901)	-	-	-	2.552 (5.180)	-	-	-	-	3.953 (7.992)	-	-	-	-
8	*Colletotrichum dematium*	-	4.672 (9.446)	-	-	-	-	1.926 (3.894)	-	-	-	3.873 (7.831)	-	-	-	-	-
9	*Cylindrocarpon magnusianum*	-	-	-	3.643 (7.366)	-	-	-	-	-	-	-	-	-	2.989 (6.043)	-	-
10	*C. olidum*	-	-	2.846 (5.026)	-	-	1.195 (2.416)	-	-	-	-	3.748 (7.578)	-	-	-	-	-
11	*Fusarium semitectum*	-	3.652 (7.384)	-	-	-	-	-	-	-	3.719 (7.519)	-	-	-	-	-	-
12	*F. sporotrichioides*	4.198 (8.488)	-	-	-	-	-	3.428 (6.931)	-	-	-	-	-	3.297 (6.267)	-	1.845 (2.719)	-
13	*Gongoronella butleri*	-	-	2.954 (7.994)	-	-	-	-	-	-	-	-	-	-	2.544 (5.143)	1.648 (3.332)	-
14	*Humicola fuscoatra*	-	-	-	2.668 (5.394)	-	-	-	-	-	-	-	-	-	-	-	-
15	*H. grisea*	-	-	-	-	3.242 (6.757)	1.938 (3.918)	-	-	-	-	3.563 (7.204)	-	-	-	-	-
16	*Idriella lunata*	-	-	3.228 (6.527)	-	-	-	-	1.294 (2.614)	-	-	-	-	2.625 (5.307)	-	-	-
17	*Mammaria echinobotryoides*	-	-	2.594 (5.892)	-	3.794 (7.671)	-	-	-	-	-	-	3.649 (7.378)	-	-	-	-
18	*M. vinacea*	-	-	-	-	-	3.298 (6.864)	3.136 (6.340)	-	-	-	2.437 (4.927)	-	-	-	-	-
19	*Mucor racemosus*	-	1.956 (3.955)	-	-	-	-	-	-	-	-	-	-	2.828 (5.718)	-	-	3.953 (7.992)
20	*Oidiodendron echinulatum*	-	-	-	3.827 (7.738)	2.416 (4.885)	-	-	-	3.622 (7.323)	-	-	-	-	-	-	-
21	*O. griseum*	-	4.254 (8.601)	-	-	-	-	-	3.683 (7.325)	-	-	-	3.472 (7.020)	-	-	-	-
22	*Penicillium artrovenetum*	-	-	-	-	-	3.124 (6.316)	-	-	-	-	-	-	-	-	-	2.228 (4.505)

23	*P. canescens*	-	3.491 (6.876)	-	-	-	-	2.836 (5.815)	-	-	-	-	-	3.207 (6.484)	-	-	-
24	*P. chrysogenum*	-	-	-	-	-	3.825 (7.734)	-	-	-	4.854 (9.814)	-	-	-	-	-	-
25	*P. frequentans*	-	2.194 (4.436)	-	-	-	-	-	-	-	-	-	-	-	-	-	-
26	*P. funiculosum*	-	-	-	3.882 (7.849)	2.927 (5.918)	-	-	-	-	-	-	-	-	-	2.848 (5.758)	-
27	*P. granulatum*	-	-	2.764 (5.588)	-	-	-	-	3.867 (6.808)	-	-	-	4.273 (8.640)	-	-	-	-
28	*P. jensenii*	-	-	3.257 (6.585)	-	-	3.365 (6.814)	-	-	-	1.197 (2.420)	2.186 (4.426)	-	-	-	-	-
29	*P. lanosum*	-	-	-	3.523 (7.123)	2.074 (4.193)	-	-	-	-	-	-	3.627 (7.334)	2.956 (5.977)	-	-	-
30	*P. purpurogenum*	-	-	-	-	3.193 (6.456)	-	-	-	-	-	-	-	-	2.594 (6.053)	-	-
31	*P. restrictum*	-	4.135 (8.360)	-	-	-	-	3.542 (7.168)	-	-	-	2.415 (4.883)	-	-	-	-	1.294 (2.616)
32	*P. rubrum*	3.538 (7.113)	-	-	-	-	-	-	1.836 (3.712)	-	-	-	-	-	2.747 (6.621)	-	-
33	*P. waksmanii*	-	-	-	-	-	2.268 (4.585)	3.214 (6.498)	-	-	3.742 (7.566)	-	-	-	-	-	-
34	*Phoma eupyrena*	-	-	-	3.846 (7.749)	-	-	-	-	1.952 (3.946)	-	-	-	-	-	3.827 (7.738)	-
35	*P. medicaginis*	-	-	-	-	-	4.213 (8.518)	3.127 (6.322)	-	-	-	-	3.632 (7.343)	-	-	-	-
36	*Plectospaerella cucumerina*	-	3.524 (7.125)	-	-	-	-	-	1.934 (3.910)	-	-	-	-	-	4.536 (9.171)	-	-
37	*Pythium intermedium*	-	-	-	3.842 (7.768)	-	-	-	-	-	-	-	-	-	-	-	2.487 (5.082)
38	*Rhizopus oryzae*	-	-	-	-	-	1.629 (3.293)	-	3.483 (7.042)	-	-	-	1.208 (2.463)	-	-	-	-
39	*Staphylotrichum coccosporum*	3.238 (6.547)	-	-	-	2.953 (5.970)	-	-	-	-	2.514 (5.083)	3.298 (6.682)	-	-	-	-	-
40	*Trichoderma harzianum*	0.926 (1.872)	-	-	-	-	-	3.682 (7.485)	-	-	-	-	-	4.874 (9.855)	-	-	-
41	*T. polysporum*	-	-	3.868 (7.821)	-	-	1.636 (3.307)	-	-	-	-	-	-	-	-	1.749 (2.990)	-
42	*T. viride*	-	4.219 (8.530)	3.653 (7.386)	-	-	-	2.423 (4.891)	-	-	-	-	2.844 (5.750)	-	-	-	-
43	*Verticillium arbo-atrum*	-	-	-	-	3.262 (6.597)	-	-	0.964 (1.949)	-	-	-	-	3.821 (7.726)	-	-	-
44	*Green sterile mycelia*	-	-	-	3.317 (6.706)	-	-	2.694 (5.486)	-	-	-	-	-	3.625 (7.329)	-	-	1.208 (2.445)
45	*White sterile mycelia*	-	-	-	-	4.195 (8.482)	3.244 (6.537)	-	-	-	-	0.796 (1.609)	-	-	-	-	-

Note: Percentage relative abundance values are marked with parentheses.

Table 3.1.4. Monthly variation in the population of fungal species per gram dry soil x 10^3 in forest (FS) site at surface (SL) layer (0-10 cm) depth.

Sl. No.	Fungal Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Acremonium murorum*	-	-	-	1.496 (3.024)	3.672 (7.424)	-	-	-	-	-	2.241 (4.531)	4.168 (8.427)	-	-	-	-
2	*Aspergillus aluteceus*	-	-	-	-	-	1.902 (3.845)	-	-	-	-	-	-	-	-	-	2.397 (4.846)
3	*A. clavatus*	-	3.263 (6.597)	-	-	4.258 (8.609)	-	-	3.241 (6.553)	1.654 (3.344)	2.324 (4.699)	-	4.827 (9.760)	-	-	3.224 (6.518)	-
4	*A. Flavus*	-	-	-	2.654 (5.366)	-	1.362 (2.753)	-	-	-	-	-	-		1.987 (4.017)	1.269 (2.565)	-
5	*A. niger*	-	-	-	1.842 (3.724)	-	-	-	-	-	-	-	-	-	2.735 (5.530)	-	-
6	*Cladosporium cladosporioides*	-	-	-	-	2.636 (5.329)	-	-	-	-	-	3.134 (6.326)	-	-	-	-	-
7	*Chaetomium globasum*	-		-	2.134 (4.314)	-	-	-	-	-	-	-	0.953 (1.926)	-	-	1. 865 (3.781)	-
8	*Colletotrichum dematium*	-	-	-	1.306 (2.640)	-	-	-	-	-	-	-	-	-	-	-	-
9	*Cylindrocarpon magnusianum*	-	-	2.645 (5.348)	-	-	-	-	-	-	-	-	-	-	-	-	-
10	*C. olidum*	-	-	-	-	-	-	-	-	-	2.568 (5.192)	-	-	-	-	-	-
11	*Fusarium semitectum*	-	-	1.325 (2.679)	-	-	1.398 (2.826)	-	-	-	-	2.425 (4.903)	-	-	-	2.651 (5.360)	-
12	*F. sporotrichioides*	-	-	-	1.329 (2.687)	-	-	-	1.964 (3.971)	-	-	-	-	-	2.130 (4.306)	3.426 (6.927)	-
13	*Gongoronella butleri*	-	1.645 (3.326)	-	-	-	-	-	-	2.362 (6.797)	-	-	-	-	-	-	1.227 (2.525)
14	*Humicola fuscoatra*	-	-	1.593 (3.227)	-	2.834 (5.730)	-	-	2.312 (4.674)	-	-	-	-	-	-	-	0.894 (1.807)
15	*H. grisea*	-	-	1.395 (2.820)	-	-	-	-	-	-	-	3.214 (6.498)	-	-	-	1.072 (2.167)	-
16	*Idriella lunata*	-	-	-	-	-	1.598 (3.231)	-	-	-	-	-	2.524 (5.103)	-	-	-	-
17	*Mammaria echinobotryoides*	-	-	-	-	3.412 (6.894)	-	-	-	-	-	-	-	-	2.874 (5.812)	-	-
18	*M. vinacea*	-	-	-	-	-	2.625 (5.307)	-	-	-	-	-	1.967 (3.958)	-	-	-	-
19	*Mucor racemosus*	-	2.754 (5.568)	-	-	-	0.859 (1.736)	-	-	-	-	-	-	2.568 (5.192)	--	-	-
20	*Oidiodendron echinulatum*	-	-	1.236 (2.498)	2.418 (4.889)	-	-	-	-	-	-	-	2.284 (4.618)	1.873 (3.793)	-	-	-
21	*O. griseum*	2.327 (4.705)	-	-	-	0.842 (1.702)	-	-	-	-	-	2.725 (5.509)	-	-	-	-	-
22	*Penicillium artrovenetum*	-	-	-	-	-	1.944 (3.930)	-	-	-	0.896 (1.811)	-	-	-	-	-	-

23	*P. canescens*	-	-	-	-	-	-	1.526 (3.085)	-	-	-	-	-	-	-	-	-
24	*P. chrysogenum*	-	-	3.427 (6.929)	-	-	1.694 (3.425)	-	-	-	-	-	-	1.483 (2.998)	-	-	1.264 (3.230)
25	*P. frequentans*	-	-	-	1.885 (3.811)	-	-	-	-	0.794 (1.605)	-	-	-	-	0.695 (1.405)	-	-
26	*P. funiculosum*	2.524 (5.103)	-	-	-	1.849 (3.738)	-	-	-	1.687 (3.412)	-	-	-	-	-	-	-
27	*P. granulatum*	-	-	3.162 (6.393)	-	-	-	-	1.469 (2.970)	-	-	-	2.061 (4.167)	-	-	-	-
28	*P. jensenii*	-	1.458 (2.948)	-	-	-	-	-	-	-	-	-	2.764 (5.582)	-	-	1.823 (3.686)	-
29	*P. lanosum*	-	-	-	-	1.297 (2.624)	-	-	-	-	-	1.182 (2.391)	2.426 (4.905)	-	-	-	-
30	*P. purpurogenum*	-	-	-	-	0.894 (1.806)	1.267 (2.561)	-	-	-	-	-	-	-	3.356 (6.785)	-	-
31	*P. restrictum*	-	-	-	-	-	1.983 (4.009)	-	-	-	2.445 (4.960)	-	-	-	1.874 (3.789)	-	-
32	*P. rubrum*	-	-	-	-	2.117 (4.401)	-	-	-	-	-	-	3.018 (6.102)	-	-	-	1.196 (2.418)
33	*P. waksmanii*	-	-	-	1.686 (3.409)	-	-	1.489 (3.010)	-	-	-	-	2.878 (5.819)	-	-	-	-
34	*Phoma eupyrena*	-	-	-	3.624 (7.327)	-	-	1.352 (2.734)	-	-	-	-	-	3.217 (6.504)	-	1.569 (3.172)	-
35	*P. medicaginis*	-	-	2.367 (4.786)	-	1.584 3.202)	-	-	1.294 (2.616)	-	-	1.453 (2.937)	-	-	-	3.218 (6.506)	-
36	*Plectospaerella cucumerina*	-	1.927 (3.896)	-	-	2.967 (5.985)	-	-	-	2.227 (4.502)	-	-	-	2.823 (5.708)	1.297 (2.623)	-	-
37	*Pythium intermedium*	-	-	-	-	-	2.425 (4.903)	-	-	-	-	-	-	1.276 (2.580)	3.126 (6.320)	-	-
38	*Rhizopus oryzae*	-	-	-	-	3.253 (6.576)	-	-	-	-	1.632 (3.297)	-	-	0.794 (1.605)	1.532 (3.097)	-	-
39	*Staphylotrichum coccosporum*	-	-	-	-	-	2.524 (5.103)	-	-	2.424 (4.901)	-	-	-	-	-	3.623 (7.325)	-
40	*Trichoderma harzianum*	-	-	-	4.127 (8.342)	-	-	2.205 (4.458)	-	-	-	3.854 (7.792)	-	-	-	-	1.593 (3.221)
41	*T. polysporum*	-	-	-	3.124 (6.358)	-	-	-	2.529 (5.113)	-	-	-	-	1.683 (3.403)	-	-	-
42	*T. viride*	0.567 (1.146)	-	-	-	-	3.412 (6.893)	-	-	-	-	-	2.849 (5.760)	3.821 (7.726)	-	-	-
43	*Verticillium arbo-atrum*	-	-	-	-	-	2.274 (4.598)	1.523 (3.085)	-	-	-	-	-	1.861 (3.762)	-	-	-
44	*Green sterile mycelia*	-	-	-	-	2.953 (5.243)	-	-	-	-	-	3.184 (6.438)	1.489 (3.010)	-	-	-	-
45	*White sterile mycelia*	-	-	-	2.524 (5.103)	3.538 (7.153)	-	-	-	-	-	-	2.424 (4.901)	-	-	-	0.916 (1.852)

Note: Percentage relative abundance values are marked with parentheses.

Table 3.1.5. Monthly variation in the population of fungal species per gram dry soil x 10^3 in forest (FS) site at middle (ML) layer (10-20 cm) depth.

Sl. No.	Fungal Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Acremonium murorum*	-	-	-	2.305 (4.670)	-	1.483 (2.998)	-	-	-	-	-	2.140 (4.327)	0.894 (1.807)	-	-	-
2	*Aspergillus aluteceus*	-	-	-	3.105 (6.278)	-	-	2.526 (5.107)	-	-	1.412 (2.854)	3.363 (6.797)	-	-	-	-	-
3	*A. clavatus*	-	-	2.825 (5.712)	-	-	-	-	-	-	-	-	3.106 (6.280)	-	-	-	2.447 (4.947)
4	*A. Flavus*	-	-	3.427 (6.929)	-	-	-	-	-	-	-	-	-	-	4.105 (8.301)	-	-
5	*A. niger*	-	0.898 (1.815)	-	-	-	3.221 (6.512)	-	-	1.162 (2.349)	-	-	-	-	-	2.627 (5.315)	-
6	*Cladosporium cladosporioides*	-	-	-	-	1.982 (4.017)	-	-	-	-	-	-	-	-	3.425 (6.925)	1.597 (3.249)	-
7	*Chaetomium globasum*	3.865 (7.815)	-	-	-	-	-	-	-	-	-	-	-	3.639 (7.358)	1.294 (2.613)	-	-
8	*Colletotrichum dematium*	-	-	-	-	-	4.129 (8.348)	-	-	-	3.628 (7.387)	-	-	-	-	-	-
9	*Cylindrocarpon magnusianum*	-	-	-	-	3.972 (8.031)	2.134 (4.314)	-	-	-	-	-	-	3.297 (6.629)	1.987 (3.208)	-	-
10	*C. olidum*	-	-	-	4.017 (8.124)	-	-	1.876 (2.781)	-	-	-	2.543 (5.141)	-	-	-	-	-
11	*Fusarium semitectum*	-	-	-	3.924 (7.934)	-	2.437 (4.927)	-	0.926 (1.872)	-	-	-	-	4.623 (9.347)	-	2.634 (5.406)	-
12	*F. sporotrichioides*	-	-	-	-	-	-	-	3.226 (6.574)	-	-	-	-	1.529 (3.085)	4.162 (4.815)	-	-
13	*Gongoronella butleri*	-	-	-	-	3.211 (6.492)	-	-	-	-	-	-	2.964 (5.956)	-	-	-	-
14	*Humicola fuscoatra*	-	-	-	-	2.624 (5.305)	-	3.104 (6.276)	-	-	-	-	-	-	2.863 (5.182)	-	-
15	*H. grisea*	-	-	-	-	-	3.657 (7.414)	-	-	-	-	-	-	-	3.983 (8.053)	1.274 (2.576)	-
16	*Idriella lunata*	-	-	-	-	1.529 (3091)	-	-	-	-	-	-	-	2.737 (5.534)	-	-	-
17	*Mammaria echinobotryoides*	-	-	3.215 (6.501)	-	-	-	-	-	0.994 (2.018)	-	-	2.638 (5.334)	-	-	-	-
18	*M. vinacea*	-	-	-	-	2.427 (4.907)	3.261 (6.593)	-	-	-	-	-	2.743 (5.546)	-	-	-	-
19	*Mucor racemosus*	-	-	-	-	-	1.845 (3.730)	-	-	-	-	-	-	-	3.636 (7.432)	-	-
20	*Oidiodendron echinulatum*	-	-	-	-	-	-	1.062 (2.147)	-	-	-	-	-	3.519 (7.115)	-	-	-
21	*O. griseum*	-	-	-	-	-	3.126 (6.324)	-	3.795 (7.673)	-	-	-	-	2.852 (5.761)	-	-	-
22	*Penicillium artrovenetum*	-	-	-	-	-	-	3.102 (6.272)	-	-	-	-	-	-	-	1.851 (2.734)	3.294 (6.673)

23	*P. canescens*	-	2.415 (4.883)	-	-	-	-	-	-	-	-	-	-	-	3.536 (7.149)	1.824 (3.685)
24	*P. chrysogenum*	-	-	-	1.967 (3.975)	-	-	-	2.375 (4.802)	-	-	-	-	5.193 (10.502)	-	-	-
25	*P. frequentans*	-	-	-	-	3.142 (6.353)	-	-	-	-	-	-	-	1.987 (4.017)	-	-	-
26	*P. funiculosum*	-	-	-	-	-	-	3.418 (6.912)	-	-	-	-	3.549 (7.176)	-	-	-	-
27	*P. granulatum*	-	-	-	-	-	-	3.637 (7.354)	1.845 (3.732)	-	-	-	-	-	3.686 (7.453)	-	-
28	*P. jensenii*	-	-	-	-	-	1.936 (3.914)	-	0.837 (1.692)	-	-	-	-	-	2.645 (5.348)	-	-
29	*P. lanosum*	-	-	-	-	-	-	4.182 (8.456)	0.794 (1.605)	-	-	1.732 (3.502)	-	-	-	-	1.431 (2.893)
30	*P. purpurogenum*	-	-	-	3.212 (6.494)	-	-	1.697 (3.431)	-	-	-	-	-	-	3.562 (7.202)	-	-
31	*P. restrictum*	-	1.639 (3.314)	-	-	-	-	3.427 (6.692)	-	-	-	-	-	-	2.853 (5.768)	-	-
32	*P. rubrum*	2.842 (5.746)	-	-	-	-	-	3.417 (6.909)	-	-	-	2.984 (6.035)	-	-	-	-	-
33	*P. waksmanii*	-	-	-	-	3.452 (6.974)	-	-	-	-	0.894 (1.807)	-	-	-	2.859 (5.780)	-	-
34	*Phoma eupyrena*	-	-	-	-	1.628 (3.291)	2.416 (4.885)	-	-	-	-	-	-	-	-	1.673 (3.382)	-
35	*P. medicaginis*	-	-	-	2.576 (5.208)	-	3.429 (6.936)	-	-	1.295 (2.618)	-	-	-	3.652 (7.384)	-	-	-
36	*Plectospaerella cucumerina*	-	-	-	-	-	2.873 (5.809)	-	1.537 (3.107)	-	-	-	3.297 (6.657)	2.874 (5.813)	-	-	-
37	*Pythium intermedium*	-	-	2.823 (5.708)	-	-	3.452 (6.972)	-	-	-	-	-	-	-	3.781 (7.645)	-	-
38	*Rhizopus oryzae*	-	1.846 (3.732)	-	-	-	-	2.458 (4.974)	-	-	-	-	-	-	3.629 (7.327)	-	0.936 (1.892)
39	*Staphylotrichum coccosporum*	-	-	-	-	3.951 (7.984)	-	-	2.716 (5.491)	-	-	-	-	4.106 (8.302)	2.487 (5.028)	-	-
40	*Trichoderma harzianum*	-	-	-	-	3.823 (7.731)	-	3.239 (6.658)	-	-	1.973 (3.989)	-	-	-	-	-	3.532 (7.141)
41	*T. polysporum*	-	-	-	-	-	4.219 (8.532)	2.836 (5.734)	-	-	-	-	3.128 (6.324)	2.865 (5.793)	-	-	-
42	*T. viride*	-	-	-	-	2.342 (4.735)	-	4.615 (9.331)	-	-	-	3.843 (7.772)	-	-	2.489 (5.026)	-	-
43	*Verticillium arbo-atrum*	-	-	-	0.983 (1.987)	2.658 (5.374)	-	-	-	-	3.145 (6.359)	2.573 (5.202)	-	-	-	-	-
44	*Green sterile mycelia*	-	1.762 (2.562)	-	-	-	3.564 (7.206)	-	-	0.872 (1.763)	-	-	-	4.251 (8.595)	-	-	-
45	*White sterile mycelia*	-	-	2.286 (4.625)	-	-	3.237 (6.634)	1.634 (3.317)	-	-	-	-	-	3.531 (6.745)	4.129 (8.348)	-	-

Note: Percentage relative abundance values are marked with parentheses.

Table 3.1.6. Monthly variation in the population of fungal species per gram dry soil x 10^3 in forest (FS) site at subsurface (SSL) layer (20-30 cm) depth.

Sl. No.	Fungal Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Acremonium murorum*	-	-	-	-	4.583 (9.267)	-	-	1.872 (3.785)	-	-	-	-	-	3.286 (6.434)	-	-
2	*Aspergillus aluteceus*	-	2.253 (4.524)	-	-	1.968 (3.972)	-	-	-	-	-	-	-	-	3.636 (7.351)	2.145 (4.346)	-
3	*A. clavatus*	-	-	-	-	-	2.745 (5.548)	-	-	-	-	-	2.649 (5.356)	-	-	-	-
4	*A. Flavus*	-	-	3.264 (6.594)	4.295 (8.684)	-	-	-	0.617 (1.247)	-	-	-	-	3.523 (7.123)	-	-	1.841 (3.725)
5	*A. niger*	-	-	-	-	-	2.346 (4.771)	-	-	1.137 (2.298)	-	-	-	-	3.284 (6.642)	-	-
6	*Cladosporium cladosporioides*	-	-	-	-	2.932 (5.224)	1.458 (2.948)	-	-	-	-	3.624 (7.327)	-	-	-	3.745 (7.572)	-
7	*Chaetomium globasum*	-	-	-	3.425 (6.925)	-	-	-	-	-	-	-	-	2.964 (5.983)	-	-	-
8	*Colletotrichum dematium*	-	-	-	-	4.152 (8.395)	-	2.114 (4.274)	3.628 (7.335)	-	-	-	-	-	3.563 (7.204)	-	-
9	*Cylindrocarpon magnusianum*	-	-	-	-	-	-	4.546 (9.192)	-	-	-	-	3.478 (7.032)	3.916 (7.683)	-	-	-
10	*C. olidum*	-	-	-	-	-	3.245 (6.561)	1.957 (3.957)	-	-	4.217 (8.526)	3.498 (7.072)	-	-	-	2.482 (5.018)	-
11	*Fusarium semitectum*	-	3.752 (7.586)	-	-	-	-	-	4.218 (8.528)	-	-	-	-	3.741 (7.564)	-	-	-
12	*F. sporotrichioides*	-	-	-	-	-	2.863 (7.814)	-	-	-	-	3.348 (6.769)	-	-	-	-	-
13	*Gongoronella butleri*	-	-	-	-	3.424 (6.923)	-	-	-	-	-	2.685 (5.426)	3.429 (6.937)	-	-	-	-
14	*Humicola fuscoatra*	2.562 (5.183)	-	-	-	-	1.853 (3.746)	-	-	-	-	-	-	3.625 (7.329)	4.214 (8.526)	-	-
15	*H. grisea*	-	-	-	-	-	-	4.592 (9.285)	-	-	-	-	-	-	3.461 (6.998)	-	-
16	*Idriella lunata*	-	-	-	-	-	2.884 (5.831)	3.367 (6.808)	-	-	-	1.859 (3.758)	-	-	-	-	-
17	*Mammaria echinobotryoides*	-	-	-	2.935 (5.934)	-	-	-	-	2.593 (5.243)	-	-	-	-	-	-	-
18	*M. vinacea*	-	-	-	-	3.226 (6.522)	4.153 (8.397)	-	-	-	-	-	-	2.743 (5.546)	-	-	-
19	*Mucor racemosus*	-	-	2.628 (5.313)	-	-	-	0.846 (1.715)	-	-	-	-	1.425 (2.872)	4.018 (8.124)	-	-	-
20	*Oidiodendron echinulatum*	-	-	-	-	2.874 (5.817)	-	-	-	-	-	-	3.526 (7.129)	-	-	-	0.972 (1.965)
21	*O. griseum*	-	-	-	-	-	2.534 (5.216)	-	3.182 (6.435)	-	2.519 (5.093)	-	-	3.627 (7.352)	-	-	-
22	*Penicillium artrovenetum*	-	-	-	4.182 (8.346)	-	-	-	-	-	-	-	-	3.632 (7.349)	-	-	-

23	*P. canescens*	-	-	-	-	-	2.584 (5.267)	-	-	-	-	-	-	3.142 (6.358)	3.283 (6.638)	-	-
24	*P. chrysogenum*	-	-	-	-	2.826 (5.714)	-	4.635 (9.371)	-	-	-	-	3.639 (7.358)	-	-	-	-
25	*P. frequentans*	-	-	-	-	3.524 (7.125)	2.852 (5.764)	-	-	-	-	-	-	3.364 (6.802)	-	-	-
26	*P. funiculosum*	-	-	-	4.218 (8.528)	-	-	2.529 (5.113)	-	-	-	-	-	2.638 (5.334)	-	-	-
27	*P. granulatum*	-	-	-	-	-	3.764 (7.615)	-	-	-	-	-	-	-	3.729 (7.542)	-	-
28	*P. jensenii*	-	-	-	-	-	-	1.864 (3.769)	-	-	3.625 (7.329)	-	-	-	2.429 (4.917)	-	-
29	*P. lanosum*	-	-	-	2.524 (5.103)	-	-	-	-	-	-	-	-	4.219 (8.536)	-	-	0.682 (1.379)
30	*P. purpurogenum*	-	1.427 (2.885)	-	-	-	-	-	0.893 (1.805)	-	-	-	3.485 (7.046)	-	-	-	-
31	*P. restrictum*	-	-	-	-	-	2.867 (5.797)	3.152 (6.373)	-	-	-	-	-	2.685 (5.374)	-	-	-
32	*P. rubrum*	-	-	-	-	4.526 (9.214)	1.694 (3.457)	-	-	-	-	3.563 (7.204)	-	-	-	4.527 (9.153)	0.914 (1.848)
33	*P. waksmanii*	-	-	-	-	-	4.523 (9.145)	-	-	-	-	-	-	-	3.874 (7.834)	-	-
34	*Phoma eupyrena*	-	-	4.715 (9.538)	-	-	-	-	3.637 (7.354)	1.568 (3.172)	-	-	-	-	-	-	-
35	*P. medicaginis*	-	-	-	-	-	-	1.761 (3.563)	3.859 (7.802)	-	-	-	-	-	-	4.516 (9.137)	-
36	*Plectospaerella cucumerina*	3.248 (6.567)	-	-	-	-	1.784 (3.534)	-	-	-	-	-	-	2.352 (4.756)	4.128 (8.346)	-	-
37	*Pythium intermedium*	-	-	-	3.387 (6.484)	2.563 (5.182)	-	-	-	-	-	-	2.429 (4.907)	-	-	-	-
38	*Rhizopus oryzae*	-	-	-	-	2.534 (5.126)	-	1.492 (3.016)	-	-	-	3.867 (7.819)	-	-	1.625 (3.285)	-	-
39	*Staphylotrichum coccosporum*	-	-	-	3.439 (6.953)	-	-	-	-	-	-	-	2.452 (4.957)	3.637 (7.354)	-	-	-
40	*Trichoderma harzianum*	-	-	4.846 (9.798)	-	-	-	-	-	-	-	-	-	-	2.783 (5.634)	-	-
41	*T. polysporum*	-	-	-	-	2.548 (5.152)	4.639 (9.387)	-	-	-	-	-	-	1.874 (3.749)	3.398 (6.871)	-	-
42	*T. viride*	-	-	-	-	-	4.129 (8.348)	-	-	-	-	-	-	-	3.532 (7.143)	-	1.749 (3.536)
43	*Verticillium arbo-atrum*	-	4.218 (8.528)	-	-	-	-	2.845 (5.752)	-	-	-	-	4.216 (8.524)	3.227 (6.524)	-	-	-
44	*Green sterile mycelia*	-	-	-	1.873 (3.787)	-	-	-	-	-	-	-	-	2.967 (5.984)	-	-	-
45	*White sterile mycelia*	-	-	2.453 (4.953)	3.617 (7.314)	-	-	-	-	-	-	-	4.539 (9.175)	1.987 (4.017)	-	-	-

Note: Percentage relative abundance values are marked with parentheses.

Table.3.1.7. List of fungi isolated from bun cultivation (BCU) and forest (FS) sites at surface (SL), middle (ML) and subsurface (SSL) layers.

Sl. No.	Fungal species	Bun cultivation			Forest		
		SL	ML	SSL	SL	ML	SSL
1	*Acremonium murorum*	+	+	-	+	-	+
2	*Aspergillus aluteceus*	+	+	+	+	+	+
3	*A. clavatus*	-	+	+	+	-	-
4	*A. Flavus*	+	+	+	+	+	+
5	*A. niger*	+	+	-	+	-	-
6	*Cladosporium cladosporioides*	+	+	-	+	-	+
7	*Chaetomium globasum*	+	+	+	+	+	-
8	*Colletotrichum dematium*	+	+	-	+	-	-
9	*Cylindrocarpon magnusianum*	+	-	-	+	+	-
10	*C. olidum*	-	+	-	+	-	-
11	*Fusarium semitectum*	+	+	+	+	+	-
12	*F. sporotrichioides*	+	-	-	+	-	-
13	*Gongoronella butleri*	+	+	-	-	+	-
14	*Humicola fuscoatra*	+	+	+	+	+	-
15	*H. grisea*	+	+	-	+	-	-
16	*Idriella lunata*	+	-	-	-	-	+
17	*Mammaria echinobotryoides*	+	+	+	+	-	-
18	*M. vinacea*	-	+	-	+	+	-
19	*Mucor racemosus*	+	+	+	+	-	-
20	*Oidiodendron echinulatum*	+	-	-	-	+	-
21	*O. griseum*	-	+	-	+	-	+
22	*Penicillium artrovenetum*	+	+	-	+	-	+
23	*P. canescens*	+	+	+	+	+	+
24	*P. chrysogenum*	+	-	-	+	+	-
25	*P. frequentans*	+	+	-	-	+	+
26	*P. funiculosum*	+	+	+	-	-	+
27	*P. granulatum*	-	+	-	-	-	+
28	*P. jensenii*	+	-	-	+	-	-
29	*P. lanosum*	+	+	-	+	-	-
30	*P. purpurogenum*	+	+	-	+	+	-
31	*P. restrictum*	-	+	+	+	-	-
32	*P. rubrum*	+	+	-	+	+	-
33	*P. waksmanii*	-	+	-	-	+	-
34	*Phoma eupyrena*	+	+	-	+	-	-
35	*P. medicaginis*	+	-	-	-	+	-
36	*Plectospaerella cucumerina*	-	+	+	-	-	+
37	*Pythium intermedium*	+	+	+	+	-	-
38	*Rhizopus oryzae*	+	-	-	+	-	-
39	*Staphylotrichum coccosporum*	+	-	-	+	-	-
40	*Trichoderma harzianum*	+	+	+	+	-	-
41	*T. polysporum*	-	+	+	-	-	+
42	*T. viride*	+	-	+	+	+	-
43	*Verticillium arbo-atrum*	-	+	+	+	-	-
44	*Green sterile mycelia*	+	-	-	-	+	-
45	*White sterile mycelia*	+	+	-	+	-	-

3.3.2 Bacterial population

The bacterial population shows an increase in the first year and dropped in the second year of both the sites. In the first year, the population dropped in August at BCU and FS sites in all the three depths (Fig 3.2.1). In the second year, the population dropped in July in the middle and subsurface layers whereas at surface layer it dropped in October at both the sites.

The bacterial population at the surface layer (SL) soil ranges from 9.69 x 10^3 and 2.78 x 10^3 CFU in July and December during the first year and 13.95 x 10^3 and 5.33 x 10^3 CFU in July and December during the second year of bun cultivation. In forest site (FS), the population ranges from 9.33 x 10^3 and 3.34 x 10^3 CFU in July and May during the first year and 8.30 x 10^3 and 1.47 x 10^3 CFU in July and December during the second year (Fig 3.2.1).

In middle layer (ML) soil, the bacterial population ranges from 9.98 x 10^3 and 3.06 x 10^3 CFU in June and December during the first year and 16.63 x 10^3 and 2.37 x 10^3 CFU in August and December during the second year of bun cultivation (BCU).

In forest site (FS), the population ranges from 7.59 x 10^3 and 2.44 x 10^3 CFU in August and May during the first year and 6.01 x 10^3 and 1.65 x 10^3 CFU in July and December during the second year (Fig 3.2.1).

Bacterial population at the subsurface layer (SSL) soil ranges from 7.33 x 10^3 and 1.68 x 10^3 CFU in July and December during the first year and 8.07 x 10^3 and 1.83 x 10^3 CFU in July and November during the second year of bun

cultivation (BCU). In forest site (FS), the population ranges from 5.37 x 10^3 and 1.12 x 10^3 CFU in June and December during the first year and 4.42 x 10^3 and 1.15 x 10^3 CFU in June and December during the second year (Fig 3.2.1).

Shannon diversity index of bacterial species during the first year ranges from 0.41 to 1.71 in surface layer in October and December at bun cultivation (BCU) site and it ranges from 0.87 to 1.74 in surface layer in September and June at forest (FS) site respectively (Fig 3.2.2). Whereas in the second year, it ranges from 0.42 to 1.74 in surface and middle layers in November and July at bun cultivation (BCU) site and it ranges from 0.51 to 1.75 in surface and middle layers in November and July at forest (FS) site (Fig 3.2.2).

Simpson dominance index of bacterial species during the first year ranges from 0.28 to 0.78 in surface layer in September and October at bun cultivation (BCU) site and it ranges from 0.29 to 0.69 in surface layer in July and August at forest (FS) site respectively (Fig 3.2.3). Whereas in the second year, it ranges from 0.26 to 0.61 in subsurface and middle layers in December and May at bun cultivation (BCU) site and it ranges from 0.26 to 0.57 in surface layer in September and august at forest (FS) site (Fig 3.2.3).

Sorenson and Jaccard similarity index of bacteria shows a similarity distribution in the first and second years of both bun cultivation (BCU) and forest (FS) sites. Sorensen similarity index during the first year shows a similarity of 0.79(SL X ML), 0.82(SL X SSL), 0.76(ML X SSL) and Jaccard similarity index shows a similarity of 0.82(SL X ML), 0.81(SL X SSL), 0.77(ML X SSL) respectively at bun cultivation (BCU) site (Fig 3.2.3). In forest site, Sorensen

similarity index shows a similarity of 0.78(SL X ML), 0.81(SL X SSL), 0.80(ML X SSL) and Jaccard similarity index shows a similarity of 0.76(SL X ML), 0.85(SL X SSL) and 0.84 (ML X SSL) respectively (Fig 3.2.4).

In the second year, Sorensen similarity index shows a similarity of 0.79(SL X ML), 0.81(SL X SSL), 0.80(ML X SSL) and Jaccard similarity index shows a similarity of 0.76(SL X ML), 0.85(SL X SSL), 0.84(ML X SSL) respectively at bun cultivation (BCU) site (Fig 3.2.4). In forest site, Sorensen similarity index shows a similarity of 0.81(SL X ML), 0.77(SL X SSL), 0.85 (ML X SSL) and Jaccard similarity index shows a similarity of 0.79(SL X ML), 0.84(SL X SSL) and 0.83(ML X SSL) respectively (Fig 3.2.4).

The bacterial species shows a distribution ranging from 86% to 95% in both bun cultivation and forest sites. The surface layer of both the sites show a higher bacterial species distribution of 91% in bun cultivation and 95% in forest sites. The middle layer shows the lowest bacterial species distribution of 86% in bun cultivation site whereas subsurface layer shows lowest distribution of 68% in forest site (Fig 3.2.5).

The one-way analysis of variance (ANOVA) of the soil bacterial population in bun cultivation (BCU) and forest (FS) sites shows significant variation ($P \leq 0.05$) in the different depths of forest and bun cultivation sites (Table 3.3.1). The subsurface (SSL) layer of bun cultivation shows insignificant variation ($P \leq 0.05$) with the surface (SL) and middle (ML) of forest site (Table. 3.3.1).

The one-way analysis of variance (ANOVA) of the soil bacterial population between surface (SL), middle (ML) and subsurface (SSL) layers bun cultivation

(BCU) and forest (FS) sites shows significant variation ($P \leq 0.05$) among the three depths of the two sites (Table 3.3.2). The surface (SL) layer of both bun cultivation (BCU) and forest (FS) sites shows insignificant variation ($P \leq 0.05$) with the middle (ML) layer (Table. 3.3.2).

The Correlation coefficient (r) values of bacterial population in surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and Forest (FS) sites at $P \leq 0.05$ is positively correlated between all the depths of the same site in both bun cultivation and forest sites. Bacterial population is also positively correlated between the two sites of all the three depths but no significant correlation between the middle (ML) and subsurface (SSL) layers at bun cultivation site (Table 3.3.3).

Correlation coefficient (r) values of bacterial population with various biological, microbial population and physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that bacterial population is positively correlated with ambient temperature (0.656 at $P \leq 0.01$), soil temperature (0.601at $P \leq 0.05$), organic carbon (0.689 at $P \leq 0.01$), dehydrogenase enzyme (0.767 at $P \leq 0.01$) and rainfall (0.715 at $P \leq 0.01$) respectively (Table 3.3.4).

Bacterial population in the middle layer of bun cultivation is positively correlated with ambient temperature (0.646 at $P \leq 0.01$), soil temperature 0.641 at $P \leq 0.01$), soil pH (0.525 at $P \leq 0.05$), rainfall (0.691 at $P \leq 0.01$) and dehydrogenase enzyme (0.824 at $P \leq 0.001$). In the subsurface layer, bacterial population is positively correlated with ambient temperature (0.743 at $P \leq 0.01$), soil

temperature (0.640 at $P \leq 0.01$), dehydrogenase enzyme (0.587 at $P \leq 0.05$) and rainfall (0.861 at $P \leq 0.05$).

Correlation coefficient (r) values of bacterial population with various biological, microbial population and physico-chemical properties of soil in forest (FS) site at surface (SL) layer shows that fungal population is positively correlated with ambient temperature (0.762 at $P \leq 0.01$), soil temperature (0.898 at $P \leq 0.001$), exchangeable potassium (0.662 at $P \leq 0.05$), dehydrogenase enzyme (0.589 at $P \leq 0.05$) and rainfall (0.786 at $P \leq 0.05$) but negatively correlated with pH (-0.613 at $P \leq 0.05$).

In the middle layer of forest site, bacterial population is positively correlated with ambient temperature (0.767 at $P \leq 0.01$), soil temperature (0.883 at $P \leq 0.001$), soil moisture content (0.504 at $P \leq 0.05$), soil organic content (0.579 at $P \leq 0.05$) and rainfall (0.650 at $P \leq 0.01$) but negatively correlated with total nitrogen (-0.559 at $P \leq 0.05$). In the subsurface layer, bacterial population is positively correlated with ambient temperature (0.735 at $P \leq 0.01$), soil temperature (0.786 at $P \leq 0.001$), soil moisture content (0.722 at $P \leq 0.01$), exchangeable potassium (0.700 at $P \leq 0.01$) and rainfall (0.773 at $P \leq 0.001$) respectively (Table 3.3.5).

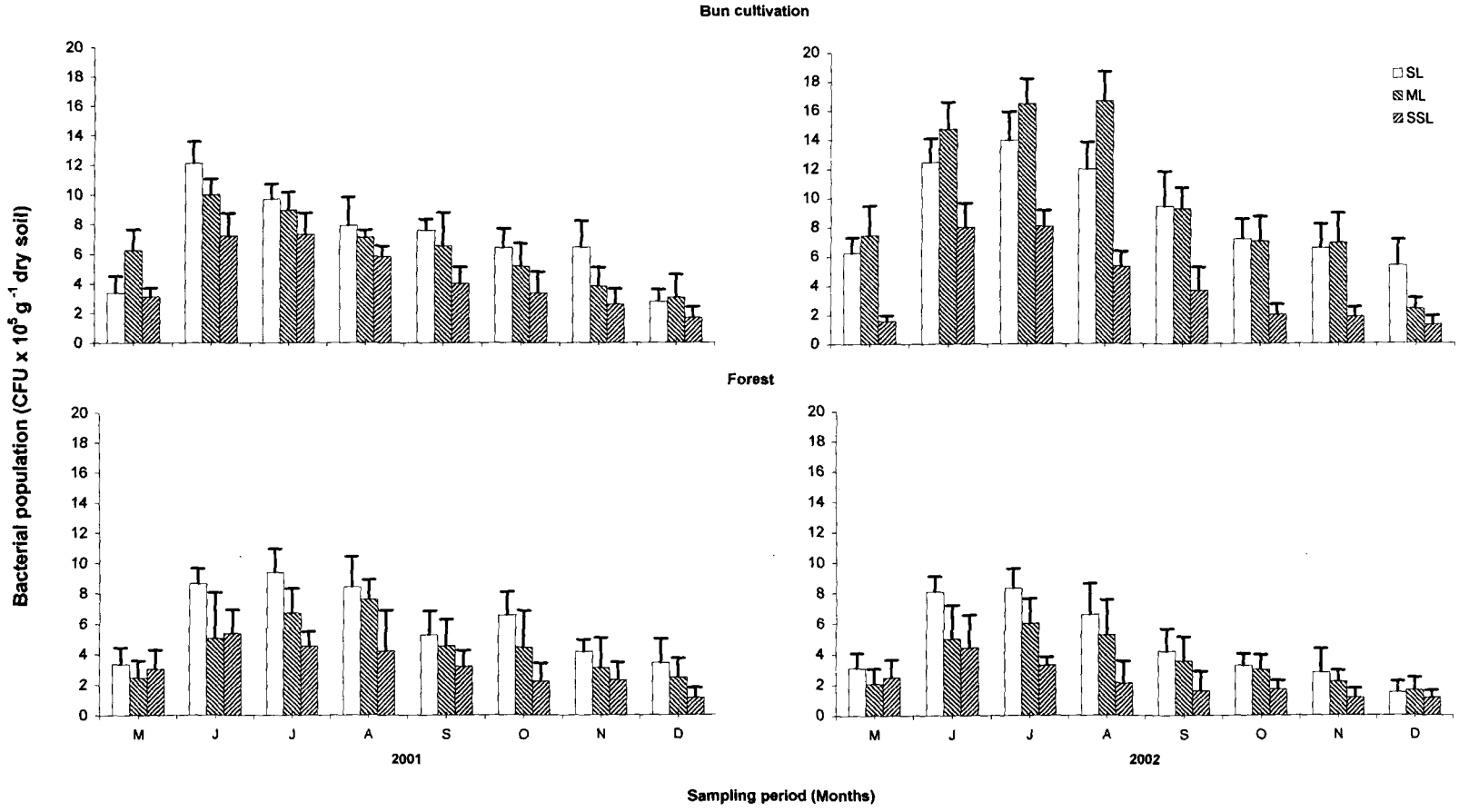

Fig. 3.2.1. Bacterial populations at surface (SL), middle (ML) and subsurface (SSL) soil layers of bun cultivation (BCU) and Forest (FS) sites.

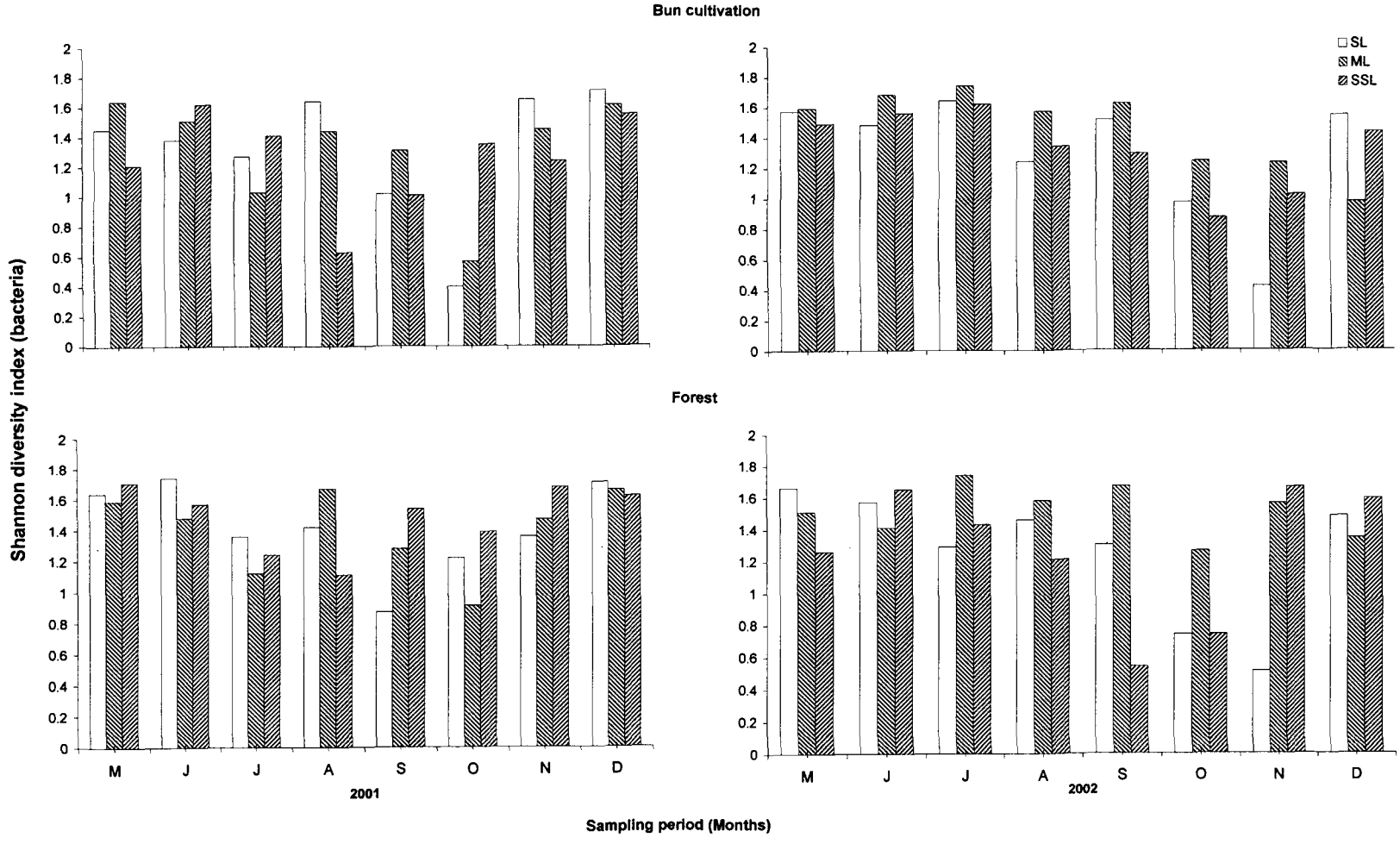

Fig. 3.2.2. Shannon diversity index of bacteria at surface (SL), middle (ML) and subsurface (SSL) soil layers of bun cultivation (BCU) and Forest (FS) sites.

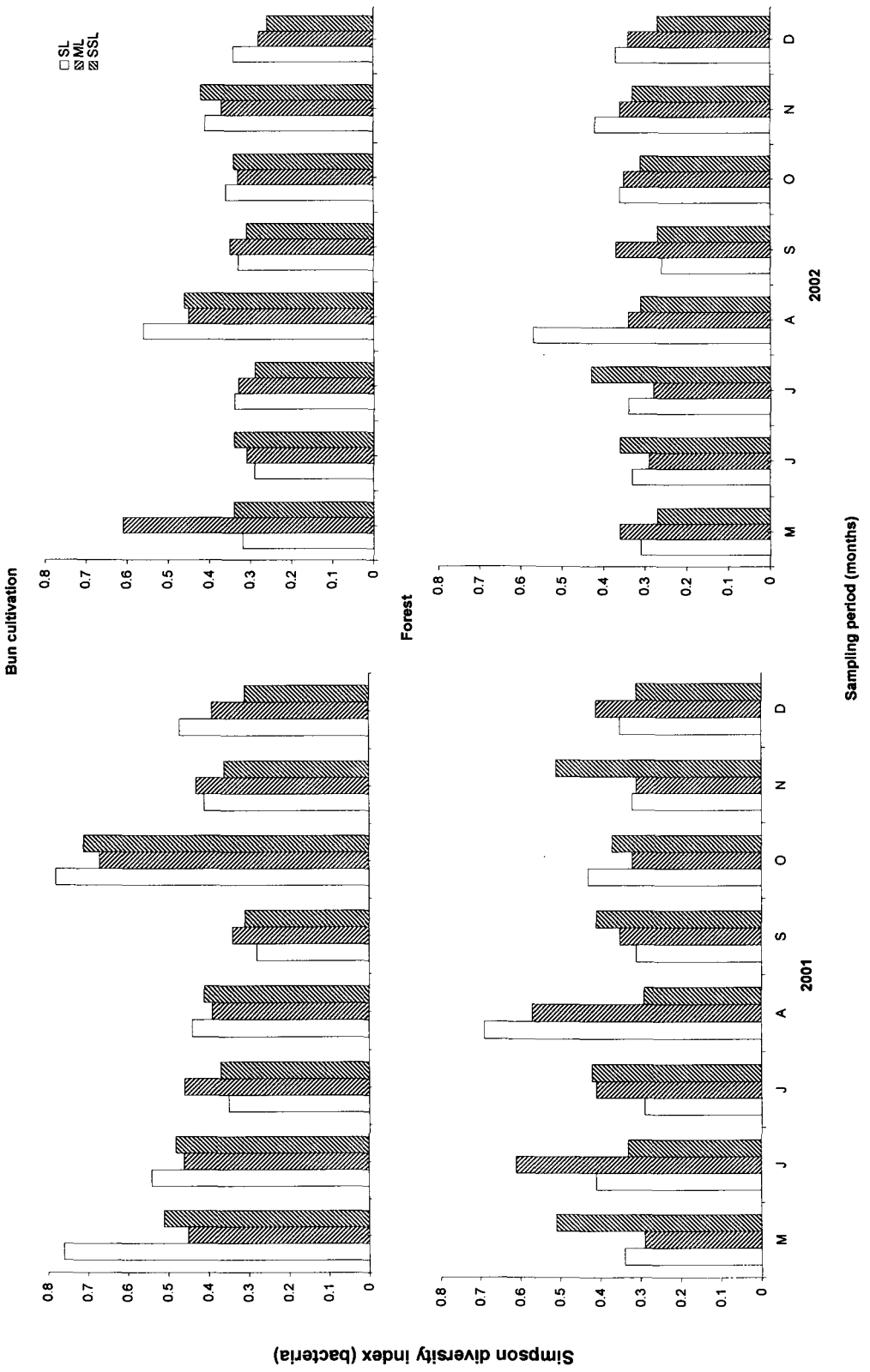

Fig. 3.2.3. Simpson diversity index of bacteria at surface (SL), middle (ML) and subsurface (SSL) soil layers of bun cultivation (BCU) and Forest (FS) sites.

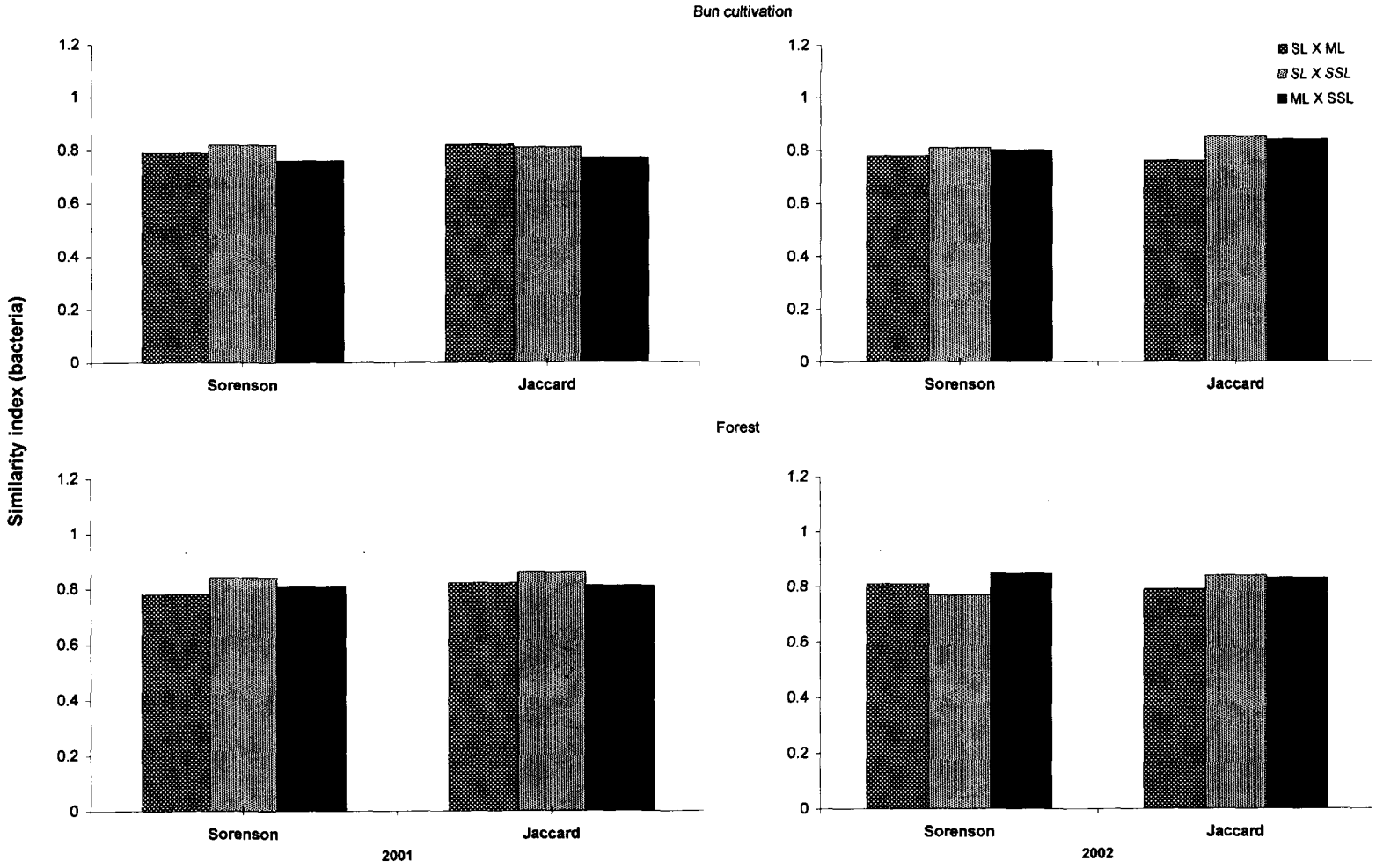

Fig. 3.2.4. Coefficient of Sorensen and Jaccard similarity index of soil bacteria in paired layer of surface (SL), middle (ML) and subsurface (SSL) layers of forest (FS) and bun cultivation (BCU) sites.

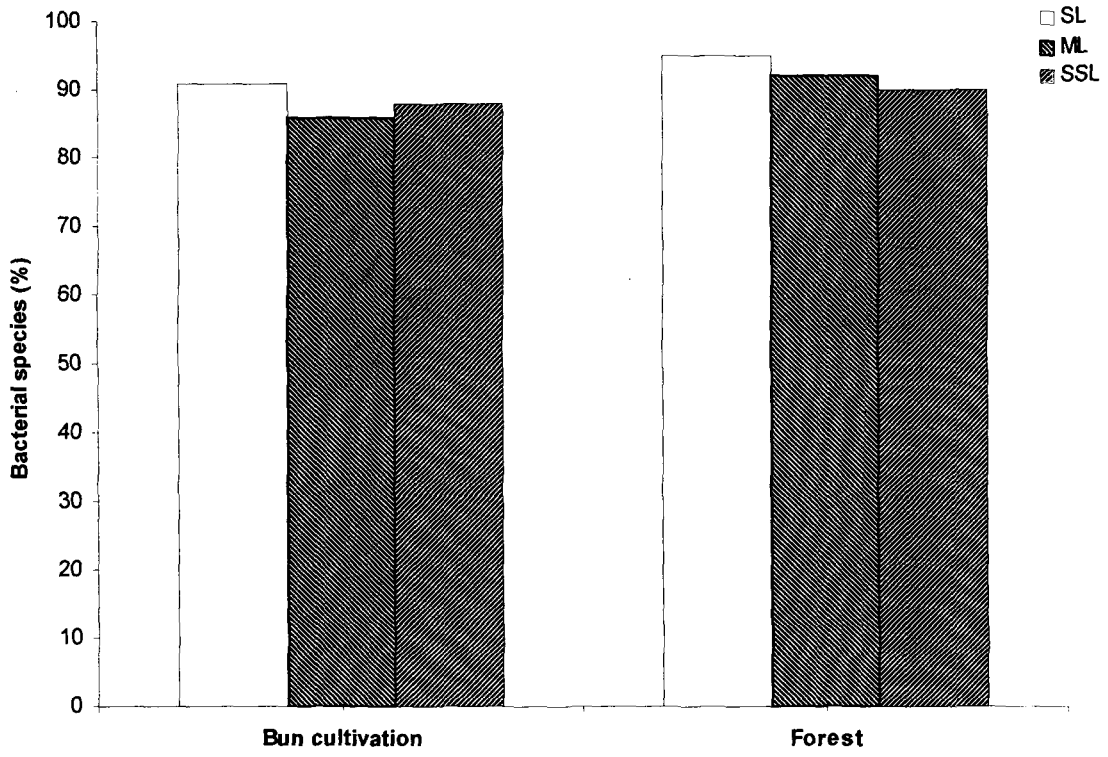

Fig. 3.2.5. Distribution of bacterial species (%) at surface (SL), middle (ML) and subsurface (SSL) layers in bun cultivation (BCU) and forest (FS) sites.

Table 3.2.1. Monthly variation in the population of bacterial species per gram dry soil x 10^3 in bun cultivation (BCU) site at surface (SL) layer (0-10 cm) depth.

Sl. No.	Bacterial Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Arthrobacter sp.*	0.023 (25.153)	-	0.146 (30.216)	2.436 (42.573)	1.845 (31.264)	8.138 (7.869)	-	-	-	0.045 (15.324)	2.427 (41.18)	0.675 (33.521)	1.934 (51.512)	7.826 (63.147)	4.625 (52.01)	-
2	*Azotobacter sp.*	-	-	0.593 (10.33)	0.725 (8.241)	1.275 (17.634)	0.362 (4.87)	0.529 (7.636)	0.67 (9.415)	0.237 (4.65)	-	0.597 (8.532)	1.025 (12.936)	0.942 (10.47)	1.348 (15.84)	0.674 (8.263)	0.214 (4.752)
3	*Baccillus cereus*	0.124 (3.925)	0.712 (10.54)	0.953 (14.925)	0.532 (9.451)	0.674 (12.326)	2.158 (30.473)	-	-	0.349 (6.225)	0.824 (12.627)	1.825 (25.634)	0.543 (11.245)	0.894 (16.853)	-	1.023 (12.697)	-
4	*Baccillus sp.*	0.124 (3.326)	0.638 (9.837)	1.429 (18.471)	1.046 (14.53)	0.937 (11.216)	2.725 (36.524)	0.71 (10.347)	0.211 (5.463)	1.243 (16.553)	0.319 (5.87)	0.853 (12.327)	1.86 (17.486)	1.149 (14.351)	0.462 (7.964)		0.436 (9.247)
5	*Baccilus subtilis*	-	0.627 (8.56)	0.769 (10.836)	-	1.253 (12.691)	0.876 (10.634)	-	-	0.302 (5.626)	0.884 (12.312)	-	1.212 (15.968)	-	0.874 (13.235)	-	-
6	*Micrococcus sp.*	-	1.203 (14.428)	0.723 (11.215)	-	-	0.638 (9.242)	0.329 (6.023)	-	-	-	0.243 (8.413)	0.862 (11.243)	1.241 (16.967)	-	0.349 (5.584)	-
7	*Pseudomonas sp.*	0.352 (7.639)	-	0.845 (12.635)	0.736 (15.637)	-	-	0.613 (9.854)	-	-	0.523 (7.843)	0.968 (15.367)	-	-	0.794 (12.526)	-	0.531 (8.427)

Note: Percentage relative abundance values are marked with parentheses.

Table 3.2.2. Monthly variation in the population of bacterial species per gram dry soil x 10^3 in bun cultivation (BCU) site at middle (ML) layer (10-20 cm) depth.

Sl. No.	Bacterial Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Arthrobacter sp.*	-	0.323 (6.835)	0.725 (12.63)	1.022 (14.632)	1.478 (21.572)	-	-	-	0.413 (7.582)	-	-	1.149 (18.643)	0.954 (13.635)	0.857 (10.238)	-	0.305 (8.584)
2	*Azotobacter sp.*	-	0.163 (6.748)	-	0.359 (9.625)	2.594 (35.261)		1.425 (23.214)	0.263 (5.796)	-	0.352 (7.632)	0.634 (11.845)	1.205 (28.462)	-	-	0.863 (12.638)	-
3	*Baccillus cereus*	0.312 (5.689)	-	0.483 (8.219)	-	1.264 (24.536)	1.042 (16.784)	-	-	-	0.925 (14.653)	0.529 (11.014)	1.682 (26.435)	-	-	-	0.523 (12.654)
4	*Baccillus sp.*	-	-	1.127 (24.872)	0.859 (12.746)	-	1.859 (18.461)	0.945 (16.636)	-	0.521 (14.452)	-	1.523 (18.475)	2.254 (32.56)	0.855 (15.342)	0.264 (9.415)	-	-
5	*Baccilus subtilis*	-	-	0.584 (11.628)	1.843 (29.114)	1.204 (24.475)	-	0.875 (16.598)	-	-	-	0.745 (13.24)	1.562 (18.596)	2.395 (28.546)	-	-	0.271 (7.486)
6	*Micrococcus sp.*	-	0.451 (10.835)	0.624 (15.447)	-	1.584 (21.127)	1.143 (15.432)	-	0.249 (8.864)	0.421 (9.957)	-	-	1.539 (20.536)	1.042 (15.648)	0.747 (13.235)	-	-
7	*Pseudomonas sp.*	0.253 (7.681)	0.889 (14.594)	-	1.425 (25.624)	1.127 (18.756)	-	0.539 (8.826)	-	-	-	0.538 (11.472)	-	1.274 (18.148)	1.732 (28.549)	0.493 (8.594)	-

Note: Percentage relative abundance values are marked with parentheses.

Table 3.2.3. Monthly variation in the population of bacterial species per gram dry soil x 10^3 in bun cultivation (BCU) site at subsurface (SSL) layer (20-30 cm) depth.

Sl. No.	Bacterial Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Arthrobacter sp.*	-	0.162 (4.958)	-	1.523 (18.742)	0.726 (12.523)	-	0.354 (8.46)	-	-	-	0.424 (6.493)	1.297 (24.328)	-	1.163 (18.315)	-	0.335 (7.473)
2	*Azotobacter sp.*	0.241 (6.582)	-	-	-	1.834 (20.143)	1.211 (16.493)	-	0.286 (6.356)	-	0.473 (9.228)	1.269 (21.43)	-	2.449 (36.546)	1.06 (18.941)	-	-
3	*Baccillus cereus*	-	0.562 (12.329)	-	1.253 (22.347)	-	0.965 (17.82)	0.543 (10.524)	-	0.701 (14.638)	1.43 (21.254)	-	1.842 (18.461)	-	2.051 (34.15)	1.073 (16.532)	-
4	*Baccillus sp.*	-	0.347 (7.847)	0.835 (14.2.03)	-	1.489 (22.536)	2.513 (34.265)	-	0.153 (6.284)	0.194 (3.274)	-	2.413 (37.86)	1.432 (26.337)	0745 (12.435)	-		0.524 (10.358)
5	*Baccilus subtilis*	0.392 (9.487)	-	1.116 (22.651)	0.924 (18.142)	-	-	0.356 (8.946)	-	-	0.625 (10.329)	1.109 (24.548)	-	0.847 (16.332)	-	0.126 (3.493)	-
6	*Micrococcus sp.*	-	-	0.587 (12.39)	-	1.213 (25.643)	1.106 (20.427)	-	-	-	0.562 (11.421)	0.825 (16.324)	-	1.94 (23.649)	1.143 (26.543)	-	-
7	*Pseudomonas sp.*	-	0.437 (10.863)	-	1.594 (20.785)	-	1.128 (14.781)	0.542 (11.635)	-	0.534 (12.352)	0.746 (15.623)	-	1.498 (21.625)	0.875 (15.426)	-	-	-

Note: Percentage relative abundance values are marked with parentheses.

Table 3.2.4. Monthly variation in the population of bacterial species per gram dry soil x 10^3 in forest (FS) site at surface (SL) layer (0-10 cm) depth.

Sl. No.	Bacterial Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Arthrobacter sp.*	-	0.426 (12.629)	-	0.867 (19.485)	1.476 (27.461)	1.065 (20.427)	-	0.324 (10.754)	-	-	0.368 (11.364)	0.812 (16.752)	1.329 (24.953)	-	0.927 (18.627)	-
2	*Azotobacter sp.*	0.241 (7.42)	-	0.632 (10.413)	1.022 (22.384)	2.34 (38.759)	-	0.862 (17.38)	-	-	0.468 (9.462)	-	0.891 (16.83)	1.435 (24.683)	0.752 (15.398)	-	0.392 (9.142)
3	*Baccillus cereus*	-	-	0.475 (10.668)	1.513 (20.361)	-	2.053 (34.256)	0.841 (14.529)	-	0.526 (10.243)	-	-	2.845 (38.765)	1.486 (27.694)	1.062 (24.3354)	-	0.846 (13.265)
4	*Baccillus sp.*	-	0.714 (15.652)	1.405 (24.749)	1.58 (28.942)	2.524 (42.153)	1.263 (25.239)	-	0.572 (10.387)	-	-	0.694 (13.068)	1.984 (27.483)	1.998 (34.263)	2.306 (41.267)	0.841 (20.113)	-
5	*Baccilus subtilis*	-	0.774 (16.25)	-	1.825 (34.173)	0.943 (20.758)	-	0.749 (14.962)	-	0.214 (9.458)	0.875 (18.451)	-	-	2.437 (31.526)	1.412 (16.245)	0.534 (13.228)	-
6	*Micrococcus sp.*	0.425 (11.204)	-	1.084 (24.75)	1.927 (28.596)	1.247 (26.348)	-	-	0.386 (12.45)	-	-	-	1.237 (15.943)	1.983 (25.364)	-	0.792 (13.235)	0.414 (10.352)
7	*Pseudomonas sp.*	-	-	0.761 (12.034)	1.234 (20.127)	2.413 (35.714)	-	-	0.425 (9.152)	-	1.12 (14.283)	0.842 (7.416)	-	-	2.438 (26.874)	-	0.405 (10.452)

Note: Percentage relative abundance values are marked with parentheses.

Table 3.2.5. Monthly variation in the population of bacterial species per gram dry soil x 10^3 in forest (FS) site at middle (ML) layer (10-20 cm) depth.

Sl. No.	Bacterial Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Arthrobacter sp.*	-	0.512 (10.457)	-	1.743 (25.846)	2.934 (33.642)	1.058 (21.427)	-	0.225 (9.295)	-	-	0.841 (16.562)	1.985 (20.427)	1.714 (29.348)	0.579 (12.419)	-	-
2	*Azotobacter sp.*	0.526 (11.204)	-	0.742 (15.249)	1.824 (32.491)	-	1.327 (21.047)	0.586 (14.721)	-	-	0.362 (9.279)	1.246 (16.428)	-	2.103 (35.264)	1.095 (22.142)	0.847 (14.795)	-
3	*Baccillus cereus*	-	-	0.217 (8.479)	0.843 (15.637)	1.835 (24.738)	-	-	0.586 (12.692)	0.215 (8.359)	0.743 (12.548)	-	2.163 (37.598)	1.143 (21.637)	-	0.948 (16.87)	0.614 (11.038)
4	*Baccillus sp.*	0.213 (7.489)	0.872 (12.352)	-	2.375 (20.413)	2.514 (42.697)	1.086 (23.547)	-	0.443 (13.52)	-	0.712 (14.825)	0.997 (18.714)	2.576 (40.328)	1.164 (22.415)	0.813 (17.485)	-	-
5	*Baccilus subtilis*	-	-	0.352 (9.452)	1.427 (17.394)	1.968 (25.637)	-	-	0.263 (8.274)	-	-	-	0.526 (10.275)	-	1.924 (21.308)	1.748 (24.738)	0.125 (7.394)
6	*Micrococcus sp.*	-	0.263 (8.462)	1.742 (16.589)	2.846 (40.215)	-	0.748 (17.247)	0.513 (10.235)	-	-	-	0.851 (12.349)	2.104 (34.827)	0.819 (14.967)	-	-	0.492 (10.948)
7	*Pseudomonas sp.*	-	0.632 (11.248)	0.967 (17.349)	-	1.718 (22.934)	1.236 (18.463)	-	-	0.126 (7.438)	0.695 (15.827)	-	2.087 (35.296)	-	1.243 (20.649)	-	-

Note: Percentage relative abundance values are marked with parentheses.

Table 3.2.6. Monthly variation in the population of bacterial species per gram dry soil x 10^3 in forest (FS) site at subsurface (SSL) layer (20-30 cm) depth.

Sl. No.	Bacterial Species	2001								2002							
		M	J	J	A	S	O	N	D	M	J	J	A	S	O	N	D
1	*Arthrobacter sp.*	-	-	0.437 (11.824)	1.741 (20.637)	-	0.784 (16.492)	1.43 (17.249)	-	-	0.374 (10.452)	-	1.438 (21.307)	1.143 (18.943)	0.564 (14.712)	-	0.237 (7.48)
2	*Azotobacter sp.*	-	0.478 (8.549)	1.524 (16.942)		0.842 (14.329)	-	-	0.214 (6.347)	0.127 (7.263)	-	-	-	1.239 (18.942)	2.109 (35.847)	0.472 (15.26)	-
3	*Baccillus cereus*	0.532 (10.935)	-	-	2.419 (32.627)	1.206 (22.347)	0.594 (16.493)	-	-	-	-	0.524 (14.325)	1.246 (13.428)	-	2.237 (31.526)	-	-
4	*Baccillus sp.*	0.224 (9.351)	0.846 (15.628)	-	2.415 (37.152)	1.273 (22.019)	1.032 (18.457)	0.237 (12.459)	-	-	0.524 (14.623)	1.245 (22.308)	0.867 (18.956)	-	1.214 (16.724)	-	0.342 (8.52)
5	*Baccilus subtilis*	-	0.341 (10.532)	0.846 (14.256)	-	-	-	0.844 (19.58)	0.127 (6.25)	-	-	-	0.523 (12.034)	0.84 (17.459)	-	-	-
6	*Micrococcus sp.*	-	0.345 (12.34)	-	0.946 (14247)	1.427 (24.528)	-	-	0.327 (10.274)	0.218 (9.453)	-	1.427 (21.438)	0.856 (16.537)	1.452 (28.946)	-	0.425 (15.286)	-
7	*Pseudomonas sp.*	-	-	0.253 (14.935)	-	-	0.947 (18.267)	0.843 (11.207)	-	-	-	-	1.053 (26.438)	0.476 (17.554)	-	0.828 (21.346)	-

Note: Percentage relative abundance values are marked with parentheses.

Table 3.2.7. List of bacteria isolated from bun cultivation and forest sites at surface (SL), middle (ML) and subsurface (SSL) layers.

Sl. No.	Bacterial species	Bun cultivation			Forest		
		SL	ML	SSL	SL	ML	SSL
1	*Arthrobacter sp.*	+	-	-	-	+	-
2	*Azotobacter sp.*	+	-	+	+	-	-
3	*Baccillus cereus*	+	+	+	+	+	+
4	*Baccillus sp.*	+	+	+	+	+	+
5	*Baccilus subtilis*	+	+	+	+	+	+
6	*Micrococcus sp.*	+	+	+	+	+	+
7	*Pseudomonas sp.*	+	+	+	+	+	+

3.4 Discussion

3.4.1 Soil fungal population

The fungal population increases at surface layer (0-10 cm) in the beginning of cultivation but a decline in the population was observed in August in the first and second year. Teuben and Roelofsma (1990) observed that soil microbial population depend on abiotic and biotic conditions of the experimental setup, i.e. soil temperature and moisture contents of the soil could be the factor in increasing the population of soil microorganisms. The decrease in the soil temperature and increase in the soil moisture content led to the sudden increase in the fungal population.

The colony forming unit of fungi shows a high variation in bun cultivation soil which could be due to the soil management practices and cropping system where agricultural practice could have large impact on the size and activity of soil microbial communities (Powlson *et al.,* 1987 and Kirchner *et al.,* 1993). Tilak *et al.,* (1995) also observed that various agricultural management practices such as cropping systems, cultivation practices and soil organic amendments can lead to the alteration of microbial dynamic in the agro-ecosystem. The inconsistent and higher variation in the fungal population in the cultivated soil could also be due to the product of transformation of soil properties, which have accumulated and become toxic to fungal populations during some months (Broadbent and Narashima, 1971).

Higher population was observed during the harvesting (end of September) of crop plants because when agricultural was abandoned an

increase in biomass and activity of microorganisms can be expected since a higher input of carbon from the resident vegetation and also an increase in root biomass occurs (Johnson *et al.,* 1991). It was hypothesized that this random distribution of peak population could be due to the enrichment of soil nitrogen and biological fixation and plant composition that affected the microbial diversity (Bardgett and Shine, 1999).

The least population was observed during August and October which is supported by the finding of Xiaoyun *et al.,* (2000) that burning reduced microbial diversity and most of enzyme activities as compared to the unburned in summer and spring. Zak *et al.,* (1994) also observed that the plant types have affected significant differences on functional diversity, which could be the reason for the least population during August in the first and second year of bun cultivation. The forest soil has no much inconsistent population through the investigation but has similarities in the increase of population in the surface (0-10 cm) layer with that of cultivated soil.

The abundance and activity of soil microorganisms are influenced by soil type, nutrient status, pH, moisture contents as well as plant factors (e.g. species, age, etc.). On the basis of the results, changes in temperature and water potential determine changes in microbiological activity, which in turn determine changes in the H^+ budget and its physicochemical consequences (Conyers, *et al.,* 1995).

The higher fungal population in the surface soil layer (0-10 cm) could be due to the high organic matter content, nutrient status and better aeration

in the top soil (Balasubramanium *et al.,* 1972) and soil moisture regimes (Selvaraj and Rangaswamy, 1978; Clarholm and Rosswall, 1980).

The decrease in the middle and subsurface layer could be due to the resource of soil management where nutrients are limited as the soil profile increases. However, shifts in microbial community structure linked to management have been attributed to changes in resource availability, particularly root exudates (Mawdsley and Bardgett, 1997). The higher fungal population could be due to the favourable soil environment viz. soil moisture content, relative high temperature and better availability of organic matter and mineral nutrients. Low population may link to run-off losses of fungal propagules along with plant materials from the hill slope due to heavy rainfall in the study area (Kshattriya *et al.,* 1992; Maithani, 1996).

Shannon-Weiner diversity index weight toward the species richness whereas Simpson diversity index was more influenced by abundance of most common species. The middle (10-20 cm) and the subsurface (20-30 cm) layers have more species richness at the beginning of the investigation both in bun cultivation and forest soils.

At the end of investigation in both first and second year, the surface layer (0-10 cm) at bun cultivation soil has maximum species richness whereas forest soil shows a similar level of species richness.

The result showed that surface layer has the maximum species richness and decreases as the soil depth increases at bun cultivation which could be due to biomass carbon where specific studies indicated that fungi are a dominant component of the total soil microbial biomass, accounting for

upto 90% of the total. Soil fungi are also the main decomposers of organic matter in soil and play and essential role in the processes that form humus at the surface soil profile (Christensen, 1989). Out of the total 45 fungal species isolated, 43 fungal species and 41 fungal species and 35 fungal species were isolated from the surface, middle and subsurface layers respectively.

The higher species diversity could be due to increase in soil nutrients and warming of the soils. This result was supported by Johansson *et al.,* (1999) that the microbes accumulated extra N and P only when soil inorganic N or P levels increased, suggesting that the soil microorganisms absorbed extra nutrients only in case of declining N and P sink strength in plants.

Warming may also affect the plants directly, if temperature controls growth processes, but also indirectly because warming is likely to accelerate litter and soil organic matter decomposition and nutrient mineralization (Nadelhoffer *et al.*, 1992).

The minimum species diversity in the middle and subsurface layers could be due to the decrease in the soil nutrients where there is growing evidence that the physical and chemical properties of soils can directly influence the structure, spatial distribution and activity of microbial populations and enzymes in soils (Schnurer *et al.*, 1985). Fliessbach *et al.,* (2000) also speculate that microbial diversity were generally highest at P impacted areas and decreased at un-impacted areas.

This decreasing trend occurred corresponding to decreases in soil P concentrations. It may be substrate limited, in addition to being limited by nutrients such as N or P (Amador and Jones, 1993).

Simpson diversity index shows a higher species abundant at middle and subsurface layers and minimum at surface layer, which are opposite to the results of Shannon diversity index because lesser the species richness higher is the species abundant.

The similarity index of Sorensen and Jaccard are observed in a paired of different layers both in bun and forest soils. The maximum similarity index was observed in the paired layers i.e. ML x SSL and ML x SSL in both bun cultivation and forest soils throughout the investigation which could be due to the higher distribution of microorganism in the surface layer. This result was supported by the Zhang *et al.,* (2002) that distribution of soil fungi in the upper surface was greater than that of the middle and innermost layers.

The minimum similarity index of Sorensen and Jaccard was observed in the paired layers of surface and middle layers (SL x ML) which could be due to the decrease in the distribution of soil microorganism with the increasing soil profiles and the physical properties of the soil. This was in agreement with the finding of Bardgett *et al.*, (1999), who showed that the proportion of fungi decreased along a soil profile gradient, which was created the limitation of N. The distribution of fungal species shows that the surface layer has the highest level of distribution (Fig. 3.1.4).

3.4.2. Soil bacterial population

The bacterial population increase at the beginning of the investigation similar to that of fungal population but a certain dropped in the population can be observed both in bun cultivation and forest soils. The bacterial population decrease as the increase of the soil profiles which was supported by the finding of Zhang *et al.*, (2002) that soil bacteria on upper layer soil were higher than that on the lower layer where the population exists in soil with single cell, easily adheres to soil particle, so it is easy to move with soil particle transport.

The microorganisms are especially abundant in the surface soil layers where they thrive of the large quantities of organic food made available in the form of dead root material (Ruark and Zarnoch, 1992; Garte, 2000 and Spaccini *et al.*, 2002).

The surface layer (0-10 cm) shows the highest bacterial population in both cultivated and forest soils which is supported by Staley and Konopka (1985) and Kuske *et al.*, (1997) that the majority of soil microorganisms for bacteria can amount to 99% of the total microbiota at the top soil profile.

The colony form unit of bacteria was markedly higher than that of fungi which could be due to the agricultural management practices and soil organic amendment where Tilak *et al.*, (1995) observed that various agricultural management practices such as cropping systems, cultivation practices and soil organic amendments could leads to the alteration of microbial dynamic in the agro-ecosystem. The diversity of colony morphotypes thus underestimates the total diversity (Atlas, 1984). The

culturally part of the microbial community represents only a minor fraction of the total community (Bakken, 1985) and only of the dominant bacteria.

The highest bacterial population was observed in the month of July and August which could be due to the increased in nutrient supply, pH, organic carbon and cation exchange capacity by the application of manures e.g. cow dung, pig slurry, etc. (Bache and Heathcote, 1969; Heathcote, 1970). The higher population during these months is because of the fact that the bacterial functioning diversity in agriculture would be affected by plant growing stages (Lahav and Steinberger, 2001).

The abundance and activity of soil microorganisms are also influenced by various environmental factors like soil type, nutrient status, soil pH and moisture content however shifts in microbial community structure linked to management have been attributed to changes in resource availability particularly root exudates (Mawdsley and Bardgett, 1997).

The increase in bacterial population in the middle layer during second year of investigation could be due to the physico-chemical properties which was supported by Bardgett and McAlister (1999) that neither cessation of fertilizer applications nor changes in cutting and grazing management significantly affected soil microbial biomass or the fungal: bacterial biomass ratio.

It is suggested that the lack of effects on the soil microbial community may be related to high residual fertility caused by retention of N in the soil. Higher bacterial population at the cultivated soil was essentially because of the low C: N ratio of the cow dung, a condition which would be more

conductive to N mineralization (Asiegbu, 1984). Sarathchandra *et al.,* (2001) also reported that the total N and available P had no effect on total bacteria and cellulolytic microbes.

Species capable of adapting to the agricultural practices will take the advantage of the situation and establish a new microbial community. The distribution of soil microorganism is very high at the bun cultivated (BCU) soil since bacteria do not specialize on a single substrate, but their diversity is the ability to use combinations of substrate under different physico-chemical conditions (Brock and Madigan, 1988; Paul and Clark, 1989).

The rhizosphere is a system exposed to environmental fluctuations due to shift on composition of root exudates, which has a marked influence on microbial communities (Lynch and Whipps, 1990).

If the application of manures results in a rhizosphere-like response, then the microbial community in the manure soils is likely to be more metabolically active which could be the reason for lesser population at the forest soil throughout the investigation. This was supported by Kirchner *et al.,* (1993) and Powlson *et al.,* (1987) that agricultural practices particularly input of cow dung as manure could possibly have large impacts on the size and activity of soil microbial communities. It has been observed that conventional agricultural management can promote bacterial and disrupt fungal parts of the microbial community (Moore, 1994).

The bacterial population at bun cultivated soil is double to that of the forest soil since the cultivated soil is likely to have higher levels of soluble organic C, therefore supporting higher levels of microbial activity. This

microbial community is made up by thousands of different species (defined as discernible genomes), of which only about 1% can presently be cultivated and thus characterized (Torsvik *et al.,* 1996). Hence the diversity of microorganisms is immense and how much of this diversity is required to guarantee the functioning of the destruction process of organic matter.

Bacteria have a large rhizosphere to bulk soil ratio indicating marked stimulation in the rhizosphere and nonsporulating rods, *Psuedomonas* and other Gram –ve bacteria are especially competitive in the rhizosphere. The genotypic microbial diversity may be as important as the functional diversity to understand the principles of soil characteristics, as shown by the elegant studies of Rainey *et al.*, (2000) on one *Psuedomonas species* at different diversities. *Pseudomonas, Flavobacterium, Acaligens* and *Agrobacterium* species has been stimulated particularly in the rhizosphere due to the release of exudates and lysates (Curl and Truelove, 1986).

The bacteria like *Azotobacter species* and *Arthrobacter species* are found very less in the bun cultivated soil which could be due to the lower pH and acidity of the soil. The result was similar to the observation of Ayanaba and Omayuli (1975) that acidity significantly influences microbial abundance and the higher acidity of the soil contains the lowest population of cellulolytic microbes.

The activity of many common bacteria is inhibited or suppressed by strong acidic conditions (pH < 4.0) in soils and relative abundance of bacteria goes down because of their lesser tolerance to acidity (Alexander, 1980).

The decrease in the bacterial population from surface layer (0-10 cm) to middle (10-20 cm) and subsurface (20-30 cm) layers could be due to the high organic matter, nutrient supply and better aeration in the surface layer (Balasubramanium *et al.,* 1972) and moisture regime (Selvaraj and Rangaswamy, 1978).

The soils differing most in diversity with respect to both culturable and nonculturable part of the microbial population also exhibit the greatest differences in sensitivity to the additional disturbance (Muller *et al.,* 2002).

The functional performance was generally unaltered by the transient disturbance, whereas it was slower and more sensitive to an additional disturbance in the presence of a permanent disturbance. The effect of a disturbance on microbial community function depends on its duration and specificity. The ratio of active to inactive biomass had altered with an increase proportion of microorganisms, which exhibit rapid rates of turnover in the rhizosphere which tend to be mainly bacteria (Bardgett *et al.,* 1999).

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). Microorganisms within functional group differ in their response to the environment as microorganisms are the fast growing, they can quickly fill out empty niches occurring when the environment is changing (Giller *et al.,* 1998).

The Shannon diversity index of bacteria shows that the surface layer shows the maximum species richness and decreases as the soil profile increases. This was supported by Alison and Killham (1988) and Tsai *et al.,*

(1997) in their study where fungal biomass increased after straw was incorporated into soil. Input of dung may perturb the soil processes causing changes in SMB composition and favoured microorganisms with lower metabolic activities.

Bacteria are quicker to respond to substrates from the dung inputs and may have capitalized on this situation. These circumstances could create a high degree of stability, but it is still not clear to what level of diversity is necessary to maintain stability (Wardle and Giller, 1996). Simpson diversity index is the reverse of Shannon diversity index and the least Shannon diversity index has higher is the Simpson dominance index.

The index of Sorensen and Jaccard shows that the paired layer of surface and subsurface (SL x SSL) has maximum similarity index of bacteria both in bun cultivation and forest soils. The minimum similarity index of bacteria is observed in the paired layer of middle and subsurface (ML x SSL) at forest soil the bun cultivated soil during the first year of investigation. The paired layer of surface and middle (SL x ML) does not show very high similarity index of bacteria both in bun cultivation and forest soil throughout the investigation.

Table 3.3.1. *One-way analysis of variance (ANOVA) of the microbial population of soil in Bun cultivation (BCU) and Forest (FS) sites at surface (0-10 cm), middle (10-20 cm) and sub surface (20-30 cm) layers at $P<0.05$.*

Soil properties	Source of variation	F-ratio	P-level
Fungal population	BCU x FS (SL)	5.309	0.028
	BCU x FS (ML)	5.020	0.032
	BCU x FS (SSL)	4.426	0.043
	BCU (SL) x FS (ML)	9.896	3.72×10^{-3}
	BCU (SL) x FS (SSL)	12.708	1.242×10^{-3}
	BCU (ML) x FS (SL)	-	-
	BCU (ML) x FS (SSL)	7.458	1.046×10^{-2}
	BCU (SSL) x FS (SL)	-	-
	BCU (SSL) x FS (ML)	-	-
Bacterial population	BCU x FS (SL)	6.641	0.015
	BCU x FS (ML)	12.527	1.33×10^{-3}
	BCU x FS (SSL)	4.271	0.047
	BCU (SL) x FS (ML)	18.825	1.49×10^{-4}
	BCU (SL) x FS (SSL)	36.886	1.01×10^{-6}
	BCU (ML) x FS (SL)	4.946	0.033
	BCU (ML) x FS (SSL)	23.1105	4.1×10^{-5}
	BCU (SSL) x FS (SL)	-	-
	BCU (SSL) x FS (ML)	-	-

Note: Insignificant values are denoted by '-' sign.

Table 3.3.2. One-way analysis of variance (ANOVA) of the microbial population of soil between surface (0-10 cm), middle (10-20 cm) and sub surface (20-30 cm) layers in Bun cultivation (BCU) and Forest (FS) sites at $P<0.05$.

Soil properties	Source of variation	F-ratio	P-level
Fungal population	BCU (SL x ML x SSL)	-	-
	FS (SL x ML x SSL)	-	-
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	-	-
	FS (SL x SSL)	-	-
	FS (ML x SSL)	-	-
Bacterial population	BCU (SL x ML x SSL)	7.113	2.06×10^{-3}
	FS (SL x ML x SSL)	7.553	1.48×10^{-3}
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	14.732	5.94×10^{-4}
	BCU (ML x SSL)	10.496	2.92×10^{-3}
	FS (SL x ML)	-	-
	FS (SL x SSL)	13.955	7.85×10^{-4}
	FS (ML x SSL)	5.583	0.024

Note: Insignificant values are denoted by '-' sign.

Table 3.3.3. Correlation coefficient (r) values among the microbial population of soil between surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and Forest (FS) sites ($P \leq 0.05$)

Source of variation	Fungal population	Bacterial population
BCU x FS (SL)	-	0.738[c]
BCU x FS (ML)	-	0.551[a]
BCU x FS (SSL)	-	0.830[c]
BCU (SL) x FS (ML)	-	0.663[a]
BCU (SL) x FS (SSL)	-	0.561[a]
BCU (ML) x FS (SL)	-	0.613[a]
BCU (ML) x FS (SSL)	-	-
BCU (SSL) x FS (SL)	-	0.938[c]
BCU (SSL) x FS (ML)	-	0.853[c]
BCU (SL x ML)	0.946[c]	0.886[c]
BCU (SL x SSL)	0.944[c]	0.846[c]
BCU (ML x SSL)	0.981[c]	0.757[c]
FS (SL x ML)	0.976[c]	0.937[c]
FS (SL x SSL)	0.964[c]	0.836[c]
FS (ML x SSL)	0.985[c]	0.728[c]

Values marked with a, b and c are significant at ($P \leq 0.05$), ($P \leq 0.01$) and ($P \leq 0.001$) respectively; insignificant values are marked with '-'.

Table 3.3.4. Correlation coefficient (r) values among biological properties of soil with various microbial population, biological and physico-chemical properties of soil in Bun cultivation (BCU) site at surface (SL), middle (ML) and subsurface (SSL) layers *(P*≤0.05)

Soil depths	Soil properties	BP	AT	ST	MC	pH	OC	TN	AP	K	C_{mic}	DHA	URA	PA	RF
SL	FP	-	0.561^{a}	0.533^{a}	0.684^{b}	0.561^{a}	-	0.537^{a}	0.628^{a}	0.804^{c}	0.724^{b}	-	-	0.643^{a}	0.665^{a}
	BP		0.656^{b}	0.601^{a}	0.683^{a}	0.562^{a}	0.689^{b}	-	0.572^{b}	-	0.651^{a}	0.767^{b}	0.452^{a}	0.687^{a}	0.715^{b}
ML	FP	-	0.541^{a}	-	-	-	0.626^{b}	0.592^{a}	-	0.763^{c}	-	-	-	-	-
	BP		0.646^{b}	0.641^{b}	-	0.525^{a}	-	-	-	-	-	0.824^{c}	-	0.536^{a}	0.691^{b}
SSL	FP	-0.613^{a}	0.540^{a}	-	-	-	0.602^{b}	-	-	-	-	-	-	-	-
	BP		0.743^{b}	0.640^{b}	-	-	-	-	-	-	-	0.587^{a}	-	-	0.861^{a}

(Note: AT=ambient temperature, MC=moisture content, ST=soil temperature, pH=soil pH, OC=organic carbon, TN=total nitrogen, AP=available phosphorus, K=potassium, RF=rainfall, FP=fungal population, BP=bacterial population, C_{mic}=microbial biomass carbon, URA=urease, DHA=dehydrogenase, PA=phosphatase)

Values marked with a, b and c are significant at (P≤0.05), (P≤0.01) and (P≤0.001) respectively; insignificant values are marked with '-'.

Table 3.3.5. Correlation coefficient (r) values among biological properties of soil with various microbial population, biological and physico-chemical properties of soil in Forest (FS) site at surface (SL), middle (ML) and subsurface (SSL) layers *(P*≤0.05).

Soil depths	Soil properties	BP	AT	ST	MC	pH	OC	TN	AP	K	C_{mic}	DHA	URA	PA	RF
SL	FP	-	-	-	0.533^{a}	-	-	-	0.573^{a}	-	0.612^{a}	-	-	0.568^{a}	-
	BP		0.762^{b}	0.898^{c}	-	-0.613^{a}	-	-	-	0.662^{a}	-	0.589^{a}	0.648^{a}	-	0.786^{a}
ML	FP	-	-	-	-	-	-	-	-	0.509^{a}	0.814^{c}	-	-	-	-
	BP		0.767^{b}	0.883^{c}	0.504^{a}	0.536^{a}	0.579^{a}	-0.559^{a}	-	-	-	-	-	-	0.650^{b}
SSL	FP	-	-	-	-	-	-	-	0.551^{a}	-	0.823^{c}	-	-	-	-
	BP		0.735^{b}	0.786^{c}	0.722^{b}	-	-	-	-	0.700^{b}	-	-	-	-	0.773^{c}

(Note: AT=ambient temperature, MC=moisture content, ST=soil temperature, pH=soil pH, OC=organic carbon, TN=total nitrogen, AP=available phosphorus, K=potassium, RF=rainfall, FP=fungal population, BP=bacterial population, C_{mic}=microbial biomass carbon, URA=urease, DHA=dehydrogenase, PA=phosphatase

Values marked with a, b and c are significant at (P≤0.05), (P≤0.01) and (P≤0.001) respectively; insignificant values are marked with '-'.

Chapter- IV

ESTIMATION OF MICROBIAL BIOMASS C (C_{mic}) IN BUN AND FOREST SOIL

4.1. Introduction

Microbial biomass is considered to be a transformation agent of soil organic materials and a labile reservoir of nutrients such as N, K, P and S (Garcia and Rice, 1994; Jenkinson and Ladd, 1981). It is an early indicator of changes that could occur in soil organic matter due to a particular management system (Powlson *et al.*, 1987; Chander *et al.*, 1995; Shibahara and Inubushi 1997; Kampichler *et al.*, 1998) and constituted 1 to 4% of the total soil C. The amount, activity and species composition of the microbial biomass are key factors controlling the amount of N mineralized and by the substrate level, water availability and temperature. Microbial biomass is linked primarily to the availability of substrates derived from crop residue which is affected by crop rotation, cultivation, residues management and fertilizer addition (Biederbeck *et al.*, 1984).

Microbial biomass is the characteristic of microorganisms, which participate in the biochemical cycles and are the live part of soil organic matter (Cengel, 1990; Srivastava, 1992). In agriculture soils, the soil microbial biomass is related to the soil organic matter content (Houot and Chaussod, 1995) and biomass represent 2-3% of soil organic C (Anderson and Domsch, 1989) whereas in semiarid areas soil has a very low microbial activity (Garcia *et al.*, 1994), low levels of microbial biomass and low organic matter content.

Soil fertility is strongly affected by microbial biomass, a small fraction of the soil's total organic matter content, which is both a source and sink of nutrients and controls soil organic matter mineralization. Addition of unsuitable soil amendments causes changes in the composition and activity of soil biomass and can damage soil fertility both in short and long-term (Brookes, 1995; Leita *et al.*, 1995; Leita, *et al.*, 1999).

The activity of microbial biomass is commonly used to characterize the microbiological status of soil (Nannipieri *et al.*, 1990) and to determine the effects of cultivation (Beyer *et al.*, 1991; Anderson and Domsch, 1993), field management (Perott *et al.*, 1992), on soil microorganisms. Soils under monocultures system contain significantly lower concentrations and qualities of soil organic matter, less soil structural stability and reduced amounts of microbial biomass and activities than system involving crop rotations (Robinson *et al.*, 1996).

4. 2. METHODOLOGY

4.2.1. Estimation of microbial biomass C (C_{mic})

Soil microbial biomass was estimated by chloroform fumigation incubation (FI) method of Anderson and Ingram (1993). The soil was sieved through 2 mm mesh sieve to remove stones, coarse roots and all visible litters. 10 g of each sample was weighed and taken in a beaker. The beakers were placed in a vacuum desiccator containing 30 ml of alcohol free chloroform in a shallow dish. The lid was closed and sealed to make it airtight. The vacuum was used till the last trace of chloroform evaporated and thereafter the desicator was kept in the dark for 5 days at 25^0 C. Another

10 g of each sample was weighed to proceed for the un-fumigated extraction (ct_2). The samples were then kept in watertight extraction bottle (125 ml) and extracted directly without any fumigation by adding 50 ml of 0.5 M K_2SO_4 and was shaken for 30 minutes. After 5 days, the fumigated soil (ct_1) sample was extracted like the un-fumigated soil sample.

The extracted soil was then filtered through Whatman filter paper No. 42. To this 4 ml filtrate, 1 ml of 0.0667 M potassium dichromate and 5 ml of concentrated sulphuric acid were added. The sample mixtures were then pre-heated at 150^0 C for 30 minutes. Two blanks were prepared, one pre-heated at 150^0 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 the titrated with acidified ferrous ammonium sulphate solution. The end point was a colour change from green/violet to red. Three replicates of the sample were maintained in each case. In case of 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

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

Where, M = Mortality of ferrous ammonium sulphate (=0.003M)

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

G = Dry soil mass (g)

E = Extraction volume (ml)

S = Digest sample volume (ml)

Microbial biomass C = (Extracted ct_1 – Extracted ct_2) X 2.46

Indicator solution:

1.485 g of O-phenanthroline monohydrate was mixed with 0.669 g of ferrous ammonium sulfate hexahydrate. 100 ml of distilled water was added to the solution.

4.3. Result

Soil microbial biomass carbon (C_{mic}) decreases in June during first and second years of bun cultivation (BCU) whereas; in the forest (FS) site, it decreases in June in the first year and increases in June during the second year. In the first year of bun cultivation, soil microbial biomass carbon dropped in June at all the three depth layers but increases in October at middle (ML) and subsurface (SSL) layers and in November at surface layer (SL). In the second year of bun cultivation soil microbial biomass carbon dropped in June in all the depths (Fig.4.1).

In the first year of forest (FS) site, soil microbial biomass carbon (C_{mic}) increases in June and dropped in September in all the depths whereas in the second year it dropped in June in all the depths but increase in July at surface layer (SL) and in August at middle (ML) and subsurface (SSL) layers (Fig.4.1).

Soil microbial biomass carbon (C_{mic}) at the surface layer (SL) soil ranges from 744.31 to 174.00 μg C g^{-1} dry soil in July and October during the first year and 981.00 to 377.13 μg C g^{-1} dry soil in May and August during the second year of bun cultivation (BCU) site. In forest (FS), site soil biomass carbon (C_{mic}) ranges from 752.67 to 127.01 μg C g^{-1} dry soil in

August and October during the first year and 1147.02 to 147.66 μg C g^{-1} dry soils in May and November during the second year (Fig.4.1).

At the middle layer (ML), soil microbial biomass carbon (C_{mic}) ranges from 833.01 to 323.04 μg C g^{-1} dry soil in October and June during the first year and 971.65 to 138.33 μg C g^{-1} dry soils in May and September during the second year of bun cultivation. In forest, the soil microbial biomass carbon (C_{mic}) ranges from 965.03 to 342.64 μg C g^{-1} dry soil in August and September during the first year and 1145.01 to 198.36 μg C g^{-1} dry soil in May and October during the second year (Fig.4.1).

Soil microbial biomass carbon (C_{mic}) at the subsurface layer (SSL) soil, ranges 976.66 to 373.56 μg C g^{-1} dry soil in November and August during the first year and 982.06 to 202.50 μg C g^{-1} dry soil in May and June during the second year of bun cultivation. In forest (FS) site, the soil microbial biomass carbon (C_{mic}) ranges from 844.33 to 383.34 μg C g^{-1} dry soil in June and May during the first year and 1111.02 to 203.64 μg C g^{-1} dry soil in May and September during the second year (Fig.4.1).

The one-way analysis of variance (ANOVA) of soil microbial biomass carbon (C_{mic}) at bun cultivation and forest sites at $P<0.05$ shows a significant variation in all the bun cultivation and forest ANOVA with the highest F-ratio (17.87) and P-level (1.11×10^{-4}) in the subsurface layer of bun cultivation and surface layer of forest sites. There is insignificant variation between the surface (SL), middle (ML) and subsurface layers of bun cultivation (BCU) and forest (FS) sites.

The one-way analysis of variance (ANOVA) shows significant variation ($P \leq 0.05$) in the different depths of forest and bun cultivation sites but insignificant variation ($P \leq 0.05$) in the same depths of the two sites (Table.4.1). Microbial biomass carbon (C_{mic}) in the surface, middle and subsurface layers of bun cultivation shows a significant variation ($P \leq 0.05$) between the three layers except in the case of surface and middle layer, whereas in the forest site there is significant variation ($P \leq 0.05$) between the three depths (Table.4.2).

The Correlation coefficient (r) values ($P \leq 0.05$) of microbial biomass carbon (C_{mic}) at surface layer of bun cultivation is positively correlated ($P \leq 0.001$) with the surface layer of forest site but no correlation with the other depths of bun cultivation and forest sites (Table.4.3).

Correlation coefficient (r) values of soil microbial biomass carbon (C_{mic}) show insignificant correlation ($P \leq 0.05$) with various microbial populations, biochemical and physico-chemical properties of soil in all the three depths of bun cultivation site (Table.4.4).

Correlation coefficient (r) values of soil microbial biomass carbon (C_{mic}) with various microbial population, biochemical and physico-chemical properties of soil in Forest site at surface shows significant correlation with fungal population (0.612 at $P \leq 0.05$). Soil microbial biomass carbon (C_{mic}) in the middle layer of forest site is significantly correlated with fungal population (0.814 at $P \leq 0.001$) and phosphatase enzyme (0589 at $P \leq 0.05$). In the subsurface layer of forest site, soil microbial biomass carbon (C_{mic}) is significantly correlation with fungal population (0.823 at $P \leq 0.001$).

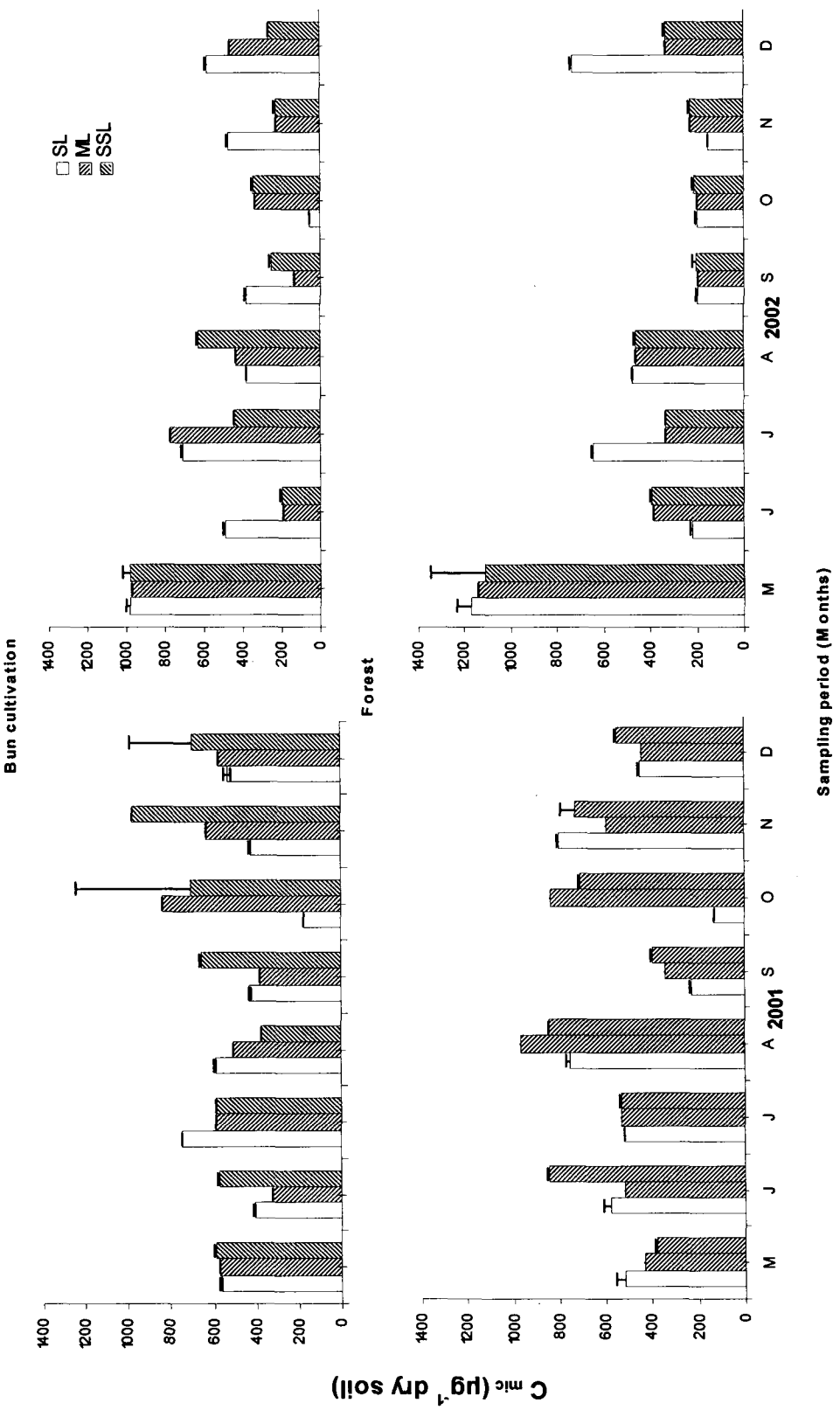

Fig. 4.1. Microbial biomass carbon (C_{mic}) of soil at surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and Forest (FS) sites.

4.4 Discussion

4.4.1. Soil microbial biomass

The microbial biomass carbon (C_{mic}) increases with the increase of soil depth after the burning at bun cultivation site and decreases in the second year, which could be due to the higher microbial population in the first year and lesser microbial population in the second year.

Specific studies indicated that fungi are a dominant component of the total soil microbial biomass, accounting for upto 90% of the total. Soil fungi are also the main decomposers of organic matter in soil (Polyanskaya *et al.*, 1997; Bardgett and McAlister, 1999). The decrease in soil microbial biomass carbon could also be due to the burning and addition of manure in the cultivated soil (Shiel, 1996).

It was noted that manure applications typically result in increased soluble organic carbon in soil and addition of manure also increased microbial biomass even when the organic carbon content of the soil is usually does not increased (Gregorich *et al.*, 1998; Liang *et al.*, 1998).

The various management practices differently affect microbial biomass levels in diverse soils (Srivastava and Lal 1994). Long-term soil management practices led to decreases in total microbial biomass (80-85% lower in spodosol and 20-55% lower in mollisol, which play a certain role in decreasing the contribution of microbial biomass carbon (C_{mic}) to the cultivated soil (Ananyeva *et al.*, 1999).

Microbial biomass carbon is higher in the surface layer in second year at bun cultivation site, which could be due to the decrease in carbon content of the soil but forest soil does not shows much variation in the microbial biomass content. The result is supported by the observation of Wright and Reddy (2001) where microbial biomass was highest at the detritus surface layer and microbial biomass carbon decreased with soil depth and was also higher at P impacted area than at the un-impacted area.

The soil microbial biomass shows no much change in the surface layer (0-10 cm) but increases in the middle (10-20 cm) and subsurface (20-30 cm) layers after four months at bun cultivation site with reduced pore space (Kaiser and Heinemeyer 1993; Lavahun *et al.,* 1996).

The higher accumulation of soil microbial biomass at the surface layer is due to higher microbial population where the analysis of variance shows no significant variation ($P \leq 0.05$) between the surface and middle layer (Table 4.1) at bun cultivation site (BCU).The higher microbial biomass C contents activity in cultivated soils at 20-30 cm depth than those at 10-20 cm suggest possibly higher microbial population and activities in the lower soil profiles.

The microbial biomass carbon at the surface layer (0-10 cm) of forest was significantly ($P \leq 0.05$) greater than that of middle and subsurface layers. The microbial biomass at the surface soil layer was attributed to the greater accumulation of litter and fine root biomass and presence of relatively dense growth of plants in forest (Maithani, 1996).

The data suggest that while agricultural soils generally have lower amounts of total and microbial C than forest soils, they may support equal or greater rates of microbial activity than forest soils due to increased production of water-soluble carbon (Boyer and Groffman, 1996).

Microbial biomass C contents were the highest in the 0–10 cm surface soils, with considerable variations due to sampling time. Microbial biomass C in the cultivation site soil taken in October and November was significantly higher than those in other months (Parham *et al.,* 2002). This trend, however, was not consistent in the forest site of samples taken.

Microbial biomass C contents decreased with increasing soil depth in all the soils tested. Interestingly, C_{mic} was higher in the 20–30 cm depth in both the sites soil than in the 10–20 cm depth soil.

Table 4.1. One-way analysis of variance (ANOVA) of the microbial biomass C (C_{mic}) of soil in Bun cultivation (BCU) and Forest (FS) sites at surface (0-10 cm), middle (10-20 cm) and sub surface (20-30 cm) layers at $P<0.05$.

Soil properties	Source of variation	F-ratio	P-level
Microbial biomass C (C_{mic})	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	6.644	.013
	BCU (SL) x FS (SSL)	16.164	2.66×10^{-4}
	BCU (ML) x FS (SL)	5.249	.026
	BCU (ML) x FS (SSL)	11.199	1.63×10^{-3}
	BCU (SSL) x FS (SL)	17.87	1.11×10^{-4}
	BCU (SSL) x FS (ML)	4.236	.045

Note: Insignificant values are denoted by '-' sign.

Table 4. 2. One-way analysis of variance (ANOVA) of the microbial biomass C (C_{mic}) of soil at surface (0-10 cm), middle (10-20 cm) and sub surface (20-30 cm) layers in Bun cultivation (BCU) and Forest (FS) sites at $P<0.05$.

Soil properties	Source of variation	F-ratio	P-level
Microbial biomass C (C_{mic})	BCU (SL x ML x SSL)	6.425	0.002
	FS (SL x ML x SSL)	12.061	3.2×10^{-5}
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	10.001	0.002
	BCU (ML x SSL)	11.199	0.001
	FS (SL x ML)	6.829	0.012
	FS (SL x SSL)	20.901	3.6×10^{-5}
	FS (ML x SSL)	6.447	0.014

Note: Insignificant values are denoted by '-' sign.

Table 4.3. Correlation coefficient (r) values among the Microbial biomass C (C_{mic}) of soil at surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and Forest (FS) sites ($P \leq 0.05$)

Source of variation	Microbial biomass C (C_{mic})
BCU x FS (SL)	0.7281^{c}
BCU x FS (ML)	0.7035^{b}
BCU x FS (SSL)	0.6901^{b}
BCU (SL) x FS (ML)	-
BCU (SL) x FS (SSL)	-
BCU (ML) x FS (SL)	0.6098^{a}
BCU (ML) x FS (SSL)	0.6089^{a}
BCU (SSL) x FS (SL)	0.5134^{a}
BCU (SSL) x FS (ML)	0.5974^{a}
BCU (SL x ML)	-
BCU (SL x SSL)	-
BCU (ML x SSL)	0.7196^{b}
FS (SL x ML)	0.6240^{b}
FS (SL x SSL)	0.6757^{b}
FS (ML x SSL)	0.9207^{c}

Values marked with a, b and c are significant at ($P \leq 0.05$), ($P \leq 0.01$) and ($P \leq 0.001$) respectively; insignificant values are marked with '-'.

Table 4. 4. Correlation coefficient (r) values among microbial biomass carbon (C_{mic}) of soil with various microbial population, biological and biochemical properties of soil in Bun cultivation (BCU) and Forest (FS) site at surface (SL), middle and subsurface layers ($P \leq 0.05$).

Sites	Soil depths	ST	MC	pH	OC	TN	AP	K	AT	RF	FP	BP	DHA	URA	PA
BCU	SL	-	-	-	0.549^{a}	-	-	-	-	-	0.724^{b}	0.651^{a}	0.648^{a}	-	-
	ML	-	-	-	0.617^{b}	-	-	-	-	-	0.535^{a}	0.682^{b}	-	-	-
	SSL	-	-	-	-	-	-	-	-	-	-	-	-	-	-
FS	SL	-	-	-	0.596^{a}	-	-	-	-	-	0.612a	0.527^{a}	-	-	-
	ML	-	-	-	-	-	-	-	-	-	0.814c	-	-	-	0.589^{a}
	SSL	-	-	-	-	-	-	-	-	-	0.823c	-	-	-	-

(**Note**: AT=ambient temperature, MC=moisture content, ST=soil temperature, pH=soil pH, OC=organic carbon, TN=total nitrogen, AP=available phosphorus, K=potassium, RF=rainfall, FP=fungal population, BP=bacterial population, C_{mic}=microbial biomass carbon, URA=urease, DHA=dehydrogenase, PA=phosphatase

Values marked with a, b and c are significant at *($P \leq 0.05$)*, *($P \leq 0.01$)* and *($P \leq 0.001$)* respectively; insignificant values are marked with '-'.

Chapter- V

ESTIMATION OF MICROBIAL ENZYME ACTIVITIES (DEHYDROGENASE PHOSPHATASE AND UREASE) IN BUN AND FOREST SOIL

5.1 Introduction

Enzymes in soils originate from animals, plants and microbial sources (Ladd, 1978; McKay, 1991) and are markedly dependent on pH, moistures regimes, temperatures and other environmental factors (Frenkenberger and Johanson, 1982; Dick and Tabatabai, 1983) but become stabilized in the soil by forming humus-enzyme or clay-enzyme complexes.

Soil enzymes play an important role in catalyzing reactions necessary for organic matter deposition and nutrient cycling. They are involved in energy transfer, environmental quality and crop productivity (Tabatabai, 1994a, b). Soil enzyme activities are greatly affected by organic matter content of the soil. Many studies have reported significant correlations among soil enzyme activities, microbial biomass and various soil properties (Ladd and Butler, 1972; Dalal, 1975; Tabatabai, 1977; Speir, 1977) and often are used as indices of microbial activity and soil fertility (Jha *et al.*, 1992; Dick and Tabatabai, 1992).

Soil enzyme activities are very sensitive to natural and anthropogenic disturbances, and show a quick response to the induced changes (Dick, 1997). Enzyme activities have been found to be very responsive to different agricultural soil conservation practices such as non-tillage (Bergstrom *et al.*, 1998), organic amendments (Perucci, 1992; Banerjee *et al.*, 1997), crop rotation (Dick, 1992; Miller and Dick, 1995) and organic cultivation (Beyer *et al.*, 1992). The activities of alkaline phosphatase and denitrifying enzymes

are mostly of microbial origin (Frenkenberger and Dick, 1983). Seasonal variation in enzyme activity appears to be dependent on soil aeration, soil moisture, soil temperature, vegetation and microbes (Blah, 1999). The soil nutrients were also the most important factor likely to regulate microbial activity (Wynn-Williams, 1982).

Enzyme assays are used as indicators of microbial biomass or microbial activity for monitoring the effects on soils of chemical disturbance or other environmental changes (Tabatabai, 1994c). Urease and phosphatase have been the most commonly studied soil enzymes along with dehydrogenase activity, which provides an index of total biological activity (Macfadyen, 1970).

Phosphatase is a broad group of enzymes that catalyze the hydrolysis of both esters and anhydrides of phosphoric acid (Schmidt and Laskowski, 1961). The rate of synthesis, release and stability of acid and alkaline phosphatases by soil organisms are dependent on the soil pH (Eivazi and Tabatabai, 1977; Juma and Tabatabai, 1977; Tabatabai, 1994b,c). Phosphatases are inducible enzymes where the intensity of their excretion by plant roots and microorganisms is determined by their requirement for orthophosphate that are affected by the soil pH (Skujins, 1976). The increase in the activity of alkaline phosphatase shows the effect of lime applications on the size of the soil microbial population as this enzyme is not present in higher plants and its activity is derived totally from microorganisms (Dick, *et al.,* 1983). Phosphatases are extracellular enzymes that catalyse the hydrolysis of organic phosphate to inorganic phosphates to inorganic

orthophosphates and important link between biologically unavailable phosphorus and available (Amador *et al.,* 1997).

The activity of dehydrogenase is considered an indicator of the oxidative metabolism in soils and thus of the microbiological activity (Skujins, 1973), because, being exclusively intracellular, it is linked to viable cells. However, the relationship between an individual biochemical properties and the total microbial activity is not always obvious, especially in the case of complex systems like soils, where the microorganisms and processes involved in the degradation of the organic compounds are highly diverse (Nannipieri *et al.,* 1990). Dehydrogenase activity has been used as an indicator of the microbiological activity in Mediterranean arid soils (Garcia, *et al.,* 1994) and in agricultural soils of more humid regions (Beyer, *et al.,* 1992). Dehydrogenase activity was lower in soils that had received the largest amounts of fertilizers and was further decreased in the absence of lime, which shows that dehydrogenase activity was highly sensitive to the inhibitory effects associated with large fertilizer additions (Simek *et al.,* 1999).

Urea has gradually become the most important nitrogen fertilizer in world agriculture. However, its efficiency is in general reduced due to excessive activity of a soil urease enzyme (Stefen and Simihian, 2002). Addition of nitrogenous fertilizers to soil can result in a series of biological and chemical reactions which affect its availability to plants and susceptibility to leaching and other losses, i.e., ammonium (NH_3) volatilization. When urea is applied to the soil, it is hydrolyzed to ammonium carbonate through the

action of an extra-cellular enzyme, urease. Enzyme activities and microbial biomass are related because transformations of the important organic elements occur through microorganisms (Frankenberger and Dick, 1983).

5.2. Methodology

5.2.1. Dehydrogenase

2-3-5-Triphenyl tetrazolium chloride (TTC) reduction technique (Casida, 1977) was used for the estimation of soil dehydrogenase activity. One gram fresh of 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 shaken till it mixed properly and plugged with a rubber stopper. The tubes are incubated at 30°C for 24 hours in an incubator. In each case three replicates were maintained. The resulting slurry was transferred on Whatman No. 1 and extracted with successive aliquots of concentrated methanol. The filtrate volume was maintained to 50 ml by continuous adding of methanol. The optical density of the filtrate was read at 485 nm on Hitachi Spectrophotometer (220) by using concentrated methanol as a blank. The activity was represented in terms of concentration of Formazan, which was calculated by a standard curve of triphenyl formazan (TPF) in methanol. Dehydrogenase activity per gram dry soil was expressed in terms of milligram formazan per gram dry soil per hour (mg TPF g^{-1} dry soil 24 h^{-1}).

5.2.2. Phosphatase

Phosphatase activity was measured by the method of Tabatabai and Bremner (1969). 0.1 g of air-dried ground sieved (0.2 mm) soil was taken into a 50 ml conical flask. Then, to it 4 ml of modified universal buffer (pH

6.5), 0.25 ml of toluene and I ml of 0.115 M p-nitrophenol phosphate (PNP) solution was added to the flask. The flasks were swirled for few seconds and are then incubated at 37^{0}C for one hour in an incubator. After incubation, 1 ml of 0.5 M calcium chloride ($CaCl_2$) and 4 ml of 0.5 M sodium hydroxide solution was added to the mixture. The soil suspension was filtered through Whatman filter No. 1. The Optical Density (OD) of the filtrate was measured at 430 nm in Hitachi (220) Spectrophotometer. A blank was maintained similarly without soil. The phosphate activity in terms of concentration of p-nitrophenol 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.

5.2.3. Urease

Urease activity was measured by the method of McGarity and Myers (1967). One gram of fresh soil was taken in 100 ml volumetric flask and to it 1 ml of toluene was added. This was then allowed to stand for 15 minutes to permit the complete penetration of toluene into the soil. Thereafter, 10 ml of buffer (pH 7.0 solution and 5 ml of 10% Urea solution were added. The flask was shaken for few minutes and incubated at 37^0 for 3 hours in an incubator. Whereas, in control, 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 inside the flask was mixed thoroughly and filtered through Whatman filter paper No. 5. Indophenol Blue Method was adopted for the measurement of ammonia released as a result of urease activity. Of this 0.5 ml of the filtrate was taken in a 25 ml volumetric flask and to it 5 ml of

distilled water was added. Then, the mixture in the flask was treated with 2 ml of phenolate solution and 1.5 ml of sodium hypochloride solution containing 5% of active chlorine. The final volume was made up to 25 ml by adding distilled water. The Optical Density (OD) of the solution 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 was taken and to it 20 ml of Caustic soda (Sodium hydroxide) solution was added. The whole volume was made up to 100 ml by adding distilled water. This was prepared fresh just before used.

Phenol solution

62.5 g of Phenol was dissolved in 20 ml of Methanol denatured alcohol and to it 18.5 ml of Acetone was added. The mixture was then made up to 100 ml by adding ethyl alcohol.

Caustic soda solution

27 g of Sodium hydroxide (NaOH) was dissolved in 100 ml of distilled water.

Both the phenolate solution and the caustic soda solution were kept in a freeze.

5.3. Results

5.3.1 Dehydrogenase

Dehydrogenase activity shows an increase in the first two months (May and June) and dropped in both the sites. The enzyme activity in the

first year of bun cultivation dropped in August at surface layer (SL) and in July at middle (ML) and subsurface (SSL) layers respectively, whereas in the second year the enzyme activity dropped in September. In the forest site (FS), dehydrogenase activity shows a dropped in June and August in the first year and second year (Fig. 5.1).

Dehydrogenase activity at the surface layer (SL) soil ranges from 0.33 to 0.13 mg TPF g^{-1} dry soil $24h^{-1}$ in October and May in the first year and 0.45 to 0.17 mg TPF g^{-1} dry soil $24h^{-1}$ in August and December in the second year of bun cultivation (BCU). In forest (FS), the enzyme activity ranges from 0.62 to 0.14 mg TPF g^{-1} dry soil $24h^{-1}$ in June and December in the first year and 0.69 to 0.19 mg TPF g^{-1} dry soil $24h^{-1}$ in July and May in the second year (Fig. 5.1).

Dehydrogenase activity at the middle layer (ML) soil ranges from 0.33 to 0.09 mg TPF g^{-1} dry soil $24h^{-1}$ in June and November during the first year and 0.38 to 0.11 mg TPF g^{-1} dry soil $24h^{-1}$ in August and May during the second year of bun cultivation. In forest, the enzyme activity ranges from 0.45 to 0.07 mg TPF g^{-1} dry soil $24h^{-1}$ in June and December during the first year and 0.34 to 0.05 mg TPF g^{-1} dry soil $24h^{-1}$ in June and May during the second year (Fig. 5.1).

Dehydrogenase activity at the subsurface layer (SSL) soil ranges from 0.31 to 0.06 mg TPF g^{-1} dry soil $24h^{-1}$ in June and November during the first year and 0.17 to 0.06 mg TPF g^{-1} dry soil $24h^{-1}$ in August and May in second year of bun cultivation. In forest, the enzyme activity ranges from 0.17 to 0.05 mg TPF g^{-1} dry soil $24h^{-1}$ in July and December in first year and 0.18 to

0.11 mg TPF g^{-1} dry soil $24h^{-1}$ in November and December in the second year (Fig. 5.1).

The one-way analysis of variance (ANOVA) shows significant variation ($P \leq 0.05$) in the different depths of forest and bun cultivation but insignificant variation ($P \leq 0.05$) in the same depths of the two sites (Table. 5.1). The surface (SL), middle (ML) and subsurface (SSL) shows a significant variation ($P \leq 0.05$) among the three layers except in the case of surface and middle layer of bun cultivation (BCU), whereas in the forest (FS) site there is significant variation ($P \leq 0.05$) among the three depths (Table. 5.2).

The Correlation coefficient (r) values ($P \leq 0.05$) of dehydrogenase enzyme at surface (SL) layer of bun cultivation was positively correlated with the subsurface layers of bun cultivation (BCU) while insignificant correlation with the other depths of bun cultivation and forest (FS) sites (Table. 5.3). The enzyme activity at middle and subsurface layers of bun cultivation are positively correlated ($P \leq 0.001$) and with all the depths of forest site.

Correlation coefficient (r) values of dehydrogenase enzyme with various microbial population, biological and physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that dehydrogenase enzyme are positively correlated with phosphatase (0.483 at $P \leq 0.01$), moisture content (0.278 at $P \leq 0.05$), rainfall (0.654 at $P \leq 0.01$), bacterial population (0.767 at $P \leq 0.001$) and negative correlation with pH (-0.263 at $P \leq 0.05$) respectively (Table 5.4). In the middle (ML) layer it positively correlated with ambient temperature (0.605 at $P \leq 0.01$), soil

temperature (0.622 at $P \leq 0.01$), bacterial population (0.628 at $P \leq 0.001$) and rainfall (0.741 at $P \leq 0.05$). Dehydrogenase enzyme is positively correlated with bacterial population (0.587 at $P \leq 0.05$) in the subsurface layer (SSL).

Correlation coefficient (r) values of dehydrogenase enzyme with various microbial population, biological and physico-chemical properties of soil in forest (FS) site at surface (SL) layer shows that dehydrogenase is positively correlated with phosphatase (0.641 at $P \leq 0.01$), soil temperature (0.524 at $P \leq 0.05$), bacterial population (0.582 at $P \leq 0.05$) and rainfall (0.606 at $P \leq 0.01$) respectively (Table 5.5). Dehydrogenase in the middle (ML) layer of forest is positively correlated with phosphatase (0.525 at $P \leq 0.05$) and pH (0.332 at $P \leq 0.01$). In the subsurface (SSL) layer it is negatively correlated with available phosphorus (-0.343 at $P \leq 0.01$).

5.3.2 Phosphatase

Phosphatase enzyme activity shows an increase in activity in first year and dropped in second year in both the sites. In the first year, the enzyme activity dropped in July and November at BCU and forest sites in all the depths (Fig 5.2). In the second year, the enzyme activity dropped in June at both the sites in all the depths. Phosphatase activity at the surface layer (SL) soil ranges from 95.66 to 12.66 µg PNP released g^{-1}dry soil h^{-1} in June and December in the first year and 77.33 and 5.66 µg PNP released g^{-1}dry soil h^{-1} in May and November in the second year of bun cultivation (BCU). In forest site (FS), the enzyme activity ranges from 61.66 and 11.50 µg PNP released g^{-1}dry soil h^{-1} in September and December in the first year and

55.50 and 11.00 µg PNP released g^{-1}dry soil h^{-1} in May and September in the second year (Fig. 5.2).

Phosphatase activity at the middle layer (ML) soil ranges from 84.00 and 12.33 µg PNP released g^{-1}dry soil h^{-1} in June and December during the first year and 57.33 and 5.50 µg PNP released g^{-1}dry soil h^{-1} in May and November in the second year of bun cultivation (BCU). In forest site (FS), the enzyme activity ranges from 51.83 and 9.66 µg PNP released g^{-1}dry soil h^{-1} in October and December in the first year and 45.33 and 7.33 µg PNP released g^{-1}dry soil h^{-1} in May and September in the second year (Fig. 5.2).

Phosphatase activity at the subsurface layer (SSL) soil ranges from 77.18 and 10.51 µg PNP released g^{-1}dry soil h^{-1} in June and December in the first year and 49.33 and 4.33 µg PNP released g^{-1}dry soil h^{-1} in May and November in the second year of bun cultivation (BCU). In forest site (FS), the enzyme activity ranges from 23.18 and 5.16 µg PNP released g^{-1}dry soil h^{-1} in September and December in the first year and 39.50 and 6.16 µg PNP released g^{-1}dry soil h^{-1} in May and September in the second year (Fig. 5.2). The one-way analysis of variance (ANOVA) shows significant variation ($P \leq 0.05$) in the different depths of forest and bun cultivation but insignificant variation ($P \leq 0.05$) in the same depths of the two sites (Table. 5.1 The surface (SL), middle (ML) and subsurface (SSL) layers shows insignificant variation ($P \leq 0.05$) among the three layers in both bun cultivation (BCU) and forest (FS) sites (Table. 5.2).

The Correlation coefficient (r) values of phosphatase enzyme at surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun

cultivation (BCU) and Forest (FS) sites is positively correlated ($P \leq 0.001$) in all the depths except between the middle (ML) and subsurface (SSL) layers of the forest site (Table 5.3).

Correlation coefficient (r) values of phosphatase enzyme with various microbial population, biological and physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that phosphatase is positively correlated with organic carbon (0.755 at $P \leq 0.001$) and available phosphorus (0.455 at $P \leq 0.05$). Phosphatase in the middle layer of bun cultivation is positively correlated with organic carbon (0.727 at $P \leq 0.001$) whereas there is no correlation in the subsurface layer (Table 5.4).

Correlation coefficient (r) values of phosphatase enzyme with various microbial population, biological and physico-chemical properties of soil in Forest (FS) site at surface (SL) and middle (ML) layers shows insignificant correlation (Table 5.5) whereas in the subsurface layer, phosphatase enzyme is positively correlated with urease enzyme (0.495 at $P \leq 0.05$).

5.3.3. Urease

Urease activity shows an increase in activity in the first year and dropped in the second year in both the sites. In the first year, the enzyme activity dropped in November and October at BCU and forest sites in all the three depths (Fig 5.3). In the second year, the enzyme activity dropped in July at both the sites in all the three depths.

Urease activity at the surface layer (SL) soil ranges from 1.42 and 0.45 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in October and July in the first year and 1.36 and 0.04 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in October and July in the second

year of bun cultivation. In forest site (FS), the enzyme activity ranges from 1.48 and 0.08 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in June and September in the first year and 1.56 and 0.04 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in June and September in the second year (Fig 5.3).

Urease activity at the middle layer (ML) soil ranges from 1.05 and 0.04 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in October and July in the first year and 1.25 and 0.32 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in September and December in the second year of bun cultivation (BCU). In forest site (FS), the enzyme activity ranges from 1.45 and 0.64 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in June and September in the first year and 1.439 and 0.102 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in June and September in the second year (Fig 5.3).

Urease activity at the subsurface layer (SSL) soil ranges from 0.85 and 0.38 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in October and July in the first year and 0.79 and 0.11 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in September and December in the second year of bun cultivation (BCU). In forest site (FS), the enzyme activity ranges from 1.40 and 0.087 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in June and September in the first year and 1.46 and 0.12 mg NH+4-N g^{-1} dry soil $3h^{-1}$ in June and September in the second year (Fig 5.4).

The one-way analysis of variance (ANOVA) shows significant variation ($P \leq 0.05$) in the different depths of forest and bun cultivation but insignificant variation ($P \leq 0.05$) in the same depths of the two sites (Table. 5.1). The surface (SL), middle (ML) and subsurface (SSL) layers shows insignificant variation ($P \leq 0.05$) among the three layers in both bun cultivation (BCU) and forest (FS) sites (Table. 5.2).

The Correlation coefficient (r) values of urease enzyme at surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and Forest (FS) sites at $P\leq 0.05$ is positively correlated but insignificant significant correlation between the middle (ML) layer of bun cultivation (BCU) with all the depths of forest (FS) site (Table 5.3).

Correlation coefficient (r) values of urease enzyme with various microbial population, biological and physico-chemical properties of soil in bun cultivation (BCU) site at surface (SL) layer shows that urease is positively correlated with soil temperature (0.834 at $P\leq 0.001$), moisture content (0.666 at $P\leq 0.001$) and organic carbon (0.453 at $P\leq 0.05$) respectively. Urease in the middle layer of bun cultivation is positively correlated with organic carbon (0.727 at $P\leq 0.001$) whereas there is insignificant correlation in the subsurface layer (Table 5.4).

Correlation coefficient (r) values of urease enzyme with various microbial population, biological and physico-chemical properties of soil in Forest (FS) site at surface (SL) and middle (ML) layers shows positively significant correlation with moisture content (0.523 at $P\leq 0.05$). In the middle layer of forest site, urease is positively correlated with moisture content (0.574 at $P\leq 0.01$) and microbial biomass carbon (0.588 at $P\leq 0.01$). Urease is positively correlated with phosphatase (0.495 at $P\leq 0.05$) and moisture content (0.597 at $P\leq 0.01$) in the subsurface layer (SSL) of forest (FS) site (Table 5.5).

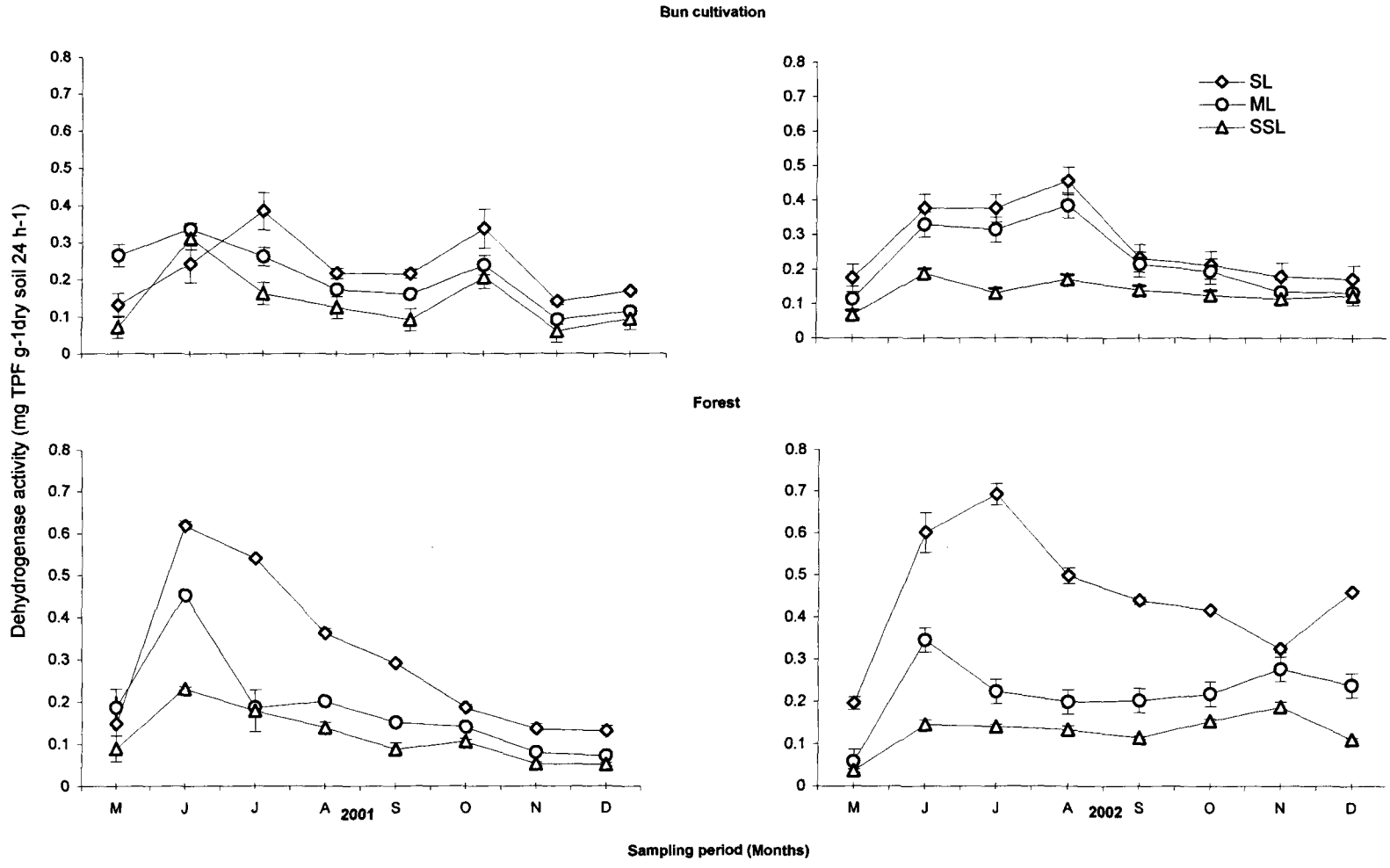

Fig 5.1. Dehydrogenase activity in soils of Bun cultivation (BCU) and Forest (FS) sites at surface (SL), middle (ML) and subsurface (SSL) layers.

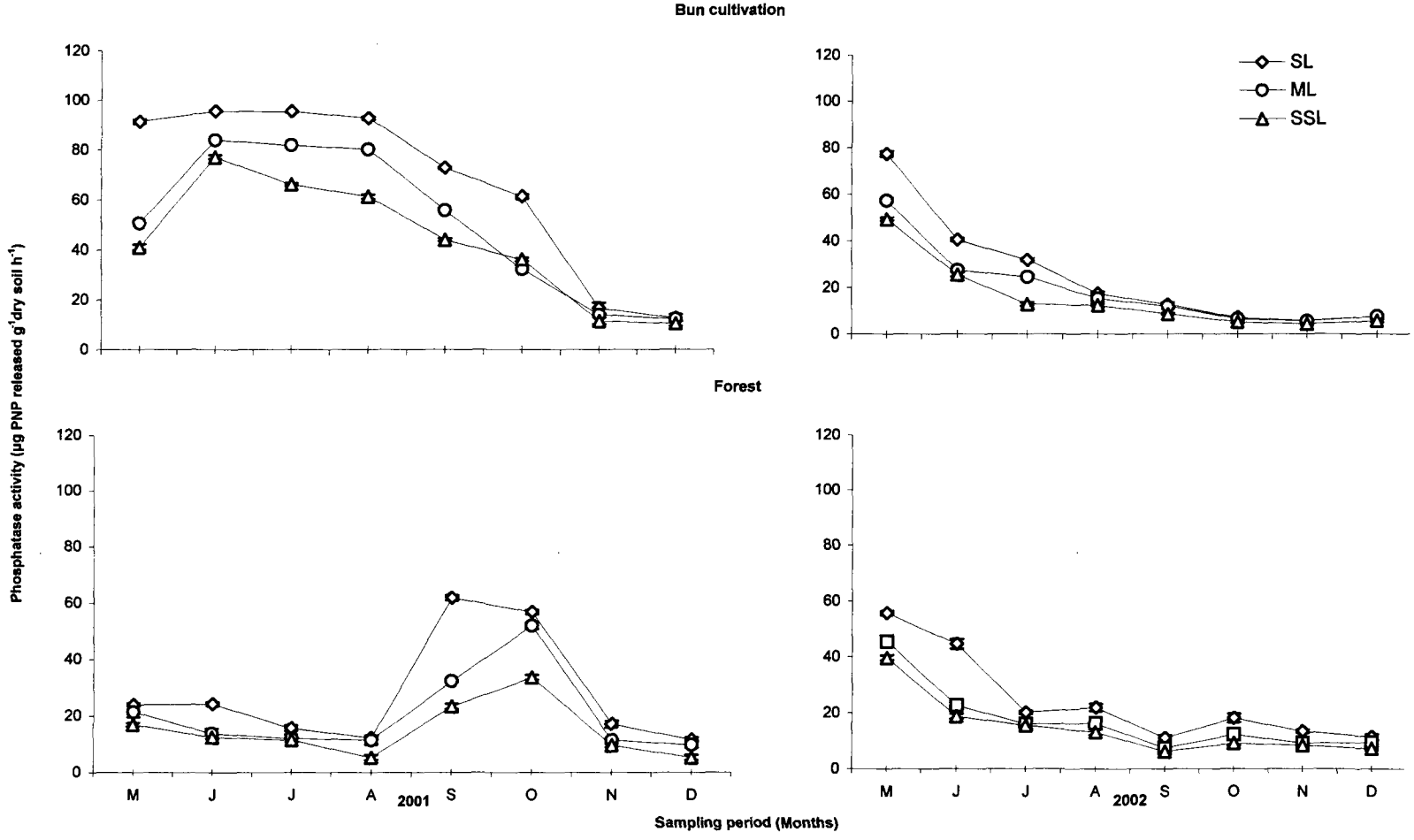

Fig 5.2. Phosphatase activity in soils of Bun cultivation (BCU) and Forest (FS) sites at surface (SL), middle (ML) and subsurface (SSL) layers.

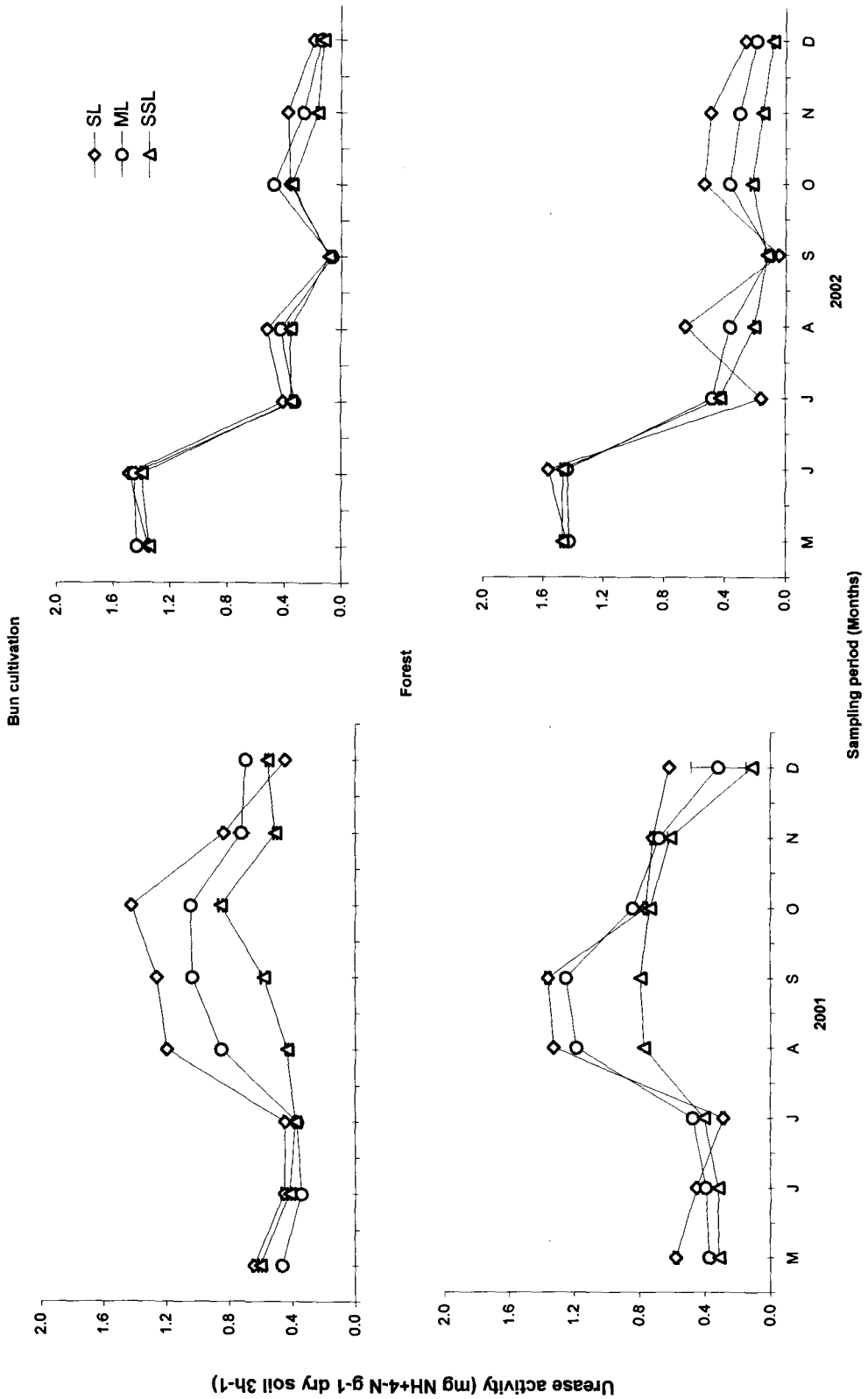

Fig 5.3. Urease activity in soils of Bun cultivation (BCU) and Forest (FS) sites at surface, middle and subsurface layers.

5.4 Discussion

5.4.1. Dehydrogenase enzyme activity

Dehydrogenase activity shows an increase at the beginning of bun cultivation but decrease in the activity with maturity of the cultivation. The activity is higher in the surface layer (0-10 cm) and decreases as the soil depth decreases in both bun cultivation and forest sites. This was supported by the observation of Parham *et al.,* (2002) that dehydrogenase enzyme activity was significantly higher at the surface and decreased with increasing soil depth, with the exception of manure-treated soils. Dehydrogenase activity showed a trend similar to that exhibited by C_{mic}, in that higher activity was detected in 20–30 cm depth (subsurface layer) soil than in the 10–20 cm depth (middle layer) soil at the beginning of the investigation.

Increase of dehydrogenase activity from dry (July, summer) to wet (October, autumn) season, shows a positive relationship between dehydrogenase enzyme and soil moisture content. The positive effect of increasing water content and nutrient addition on soil dehydrogenase has been already reported (Nannipieri *et al.,* 1990). Soil dehydrogenase enzyme activity was positively and significantly correlated with soil pH and soil moisture content in the surface (0-10 cm) layer at bun cultivation (BCU). On the other hand, no correlation was found between dehydrogenase enzyme activity and total soil nitrogen and organic carbon. Rainfall also has a certain role in the activity of soil dehydrogenase (Beyer *et al.*,1992).

The forest soil shows no significant and positive relationship but Leirós *et al.,* (2000) reported a clear positive relationship between soil dehydrogenase enzyme and soil C. Probably in these forest ecosystems soil microorganisms are nutrient rather than C limited, since dehydrogenase enzyme did not respond to the variation of C contents or to the C:N ratio.

The increase of dehydrogenase enzyme activity after three months of investigation could be due to the increase in soil microbial population where the activity of dehydrogenase is considered an indicator of the oxidative metabolism in soils and thus of the microbiological activity, especially in the case of complex systems like soils, where the microorganisms and processes involved in the degradation of the organic compounds are highly diverse (Skujins 1973; Nannipieri *et al.*, 1990). Beyer *et al.,* (1992) and Garcia *et al.,* (1994) reported that dehydrogenase activity (DHA) has been used as an indicator of the microbiological activity in arid soils and in agricultural soils.

Bun cultivation as soil management practice and the seasonal condition affects the soil enzyme activity (Batra *et al.,* 1997). Thus, the activity of soil microorganisms and soil management were strongly related to the soil enzyme (Deng and Tabatabai, 1997; Klose *et al.,* 1999; Consuelo *and* Teodoro., 2002).

Garcia *et al.,* (1994) also found that the rainy season enhanced the enzyme activity (DHA) of soils. Other also has attributed the increase in microbial activity in forest soils (Görres *et al.,* 1998; Banerjee *et al.,* 2000) and to higher soil moisture contents. The positive effect of increasing water

content and nutrient addition on soil DHA has been already reported (Nannipieri *et al.*, 1990).

The addition of manure saturated soil's capacity to retain manure-P and the soil P status might also influence microbial growth directly, through its effect on plant root growth and consequently influence the amount of C released by roots (James *et al.,* 1996). So any management practice that increase total C accumulation and showed that dehydrogenase activity significantly correlated with the physico-chemical properties of the soil (Buchanan, 1990).

The higher activity of dehydrogenase activity in the surface layer (0-10 cm) was due to presence of higher bacterial population and favourable soil moisture content (Das, 1980; Baruah and Mishra, 1984; and Tiwari *et al.,* 1987). The analysis of variance that showed a significant variation of dehydrogenase activity between surface (0-10 cm), middle (10-20 cm) and subsurface (20-30 cm) layers could be due to the differences in soil microbial population and rainfall. The higher dehydrogenase activity in bun cultivation soils at 20-30 cm depth than those at 10-20 cm depth suggest possibly higher microbial population and activities in the lower soil profiles.

Therefore, it can be concluded that dehydrogenase activity was significantly and positively correlated to the soil microbial population (bacteria) and soil management practice (bun cultivation). The environmental factor like soil moisture content and rainfall also has certain effects on the activity of the dehydrogenase enzyme.

5.4.2. Phosphatase enzyme activity

Phosphatase activities were always higher in the soil either by burning of the dried plants or by the application of animal manures. The phosphatase enzyme activity increases at the beginning and shows very high activity during the first year which may be due to the increase of the organic matter contents of the soil at bun cultivation site (Nilson and Eiland, 1980). There is declined of activity at the end of the investigation both in the first and second year of the study whereas forest soil shows no much variation in the activities of the phosphatase enzyme.

The surface layer (0-10 cm) at bun cultivation shows a very high phosphatase activity, which is supported by the result of Singh *et al.,* (1997) that enzyme activities involved in P cycling were generally more uniformly distributed over a 0–10 cm soil profile.

Considerable variations in phosphatase activities were significantly greater in the cultivated soil than that of forest soils. In general ashes from burning could show a greater impact over the 0–10 cm soil profile. This is strongly demonstrated by phosphatase activities over the soil profiles tested. Such a pattern of impact may result in a relatively long-term effect on crop production that could be the reason in the decrease of soil phosphatase enzyme activity in the second year.

Phosphatase activity decreases as the soil profile decreases at bun cultivation that could be due to the decrease of the soil nutrients and the application of ashes to the soil enriches the soil organic matter and increases total N contents. Tiwari (1996) reported significantly greater

(P0.005) activities of acid phosphatase in the soil that has high contents of nutrients (nitrogen and phosphorus).

The decrease of soil phosphatase enzyme from the top surface layer to the subsurface layer and a decline from first year to the second year is highly influenced by the soil microorganisms and the management practices where soil organisms and plant could utilize soil organic P by means of phosphatase enzyme (Pant *et al.,* 1994). Enzyme activities can be considered effective indicators of soil quality changes resulting from environmental stress or management practices. Enzyme activities have been found to be very responsive to different agricultural soil conservation practices such as non-tillage (Dick, 1992; Bergstrom *et al.*, 1998), organic amendments (Perucci, 1992; Banerjee *et al.*, 1997), crop rotation (Miller and Dick 1995), and organic cultivation (Beyer *et al.*, 1992). Soil phosphatases enzyme helps in the mineralization processes of organic phosphorus substrates.

Soil enzymes are used as indicators of microbial activity for monitoring the effects on soils of chemicals, disturbance, or other environmental changes (Tabatabai,1994c).The bun cultivation promoted biological and microbial activities, which accelerated the breakdown of organic substances in the soil. The enhanced biological activities in the cultivated soil are evidenced by relatively high phosphatase and dehydrogenase activities. One explanation is that some of the earlier conclusions were derived from studies with a much higher manure application rate (Agbenin and Goladi 1997).

The positive correlation between phosphatases and C and N is in good agreement with the results of Bonmati *et al.*, (1991); Chhonkar and Tarafdar (1984); Šarapatka and Kršková (1997). Chhonkar and Tarafdar (1984) described positive correlations of phosphatase activity with organic carbon, organic phosphorus and bacterial populations, and a negative relationship with soil pH.

It can be concluded that soil phosphatase enzyme activities significantly correlated with the soil organic C, total N (Bonmati *et al.*, 1991) and dehydrogenase activities. The availability of soil P in the soil generates the activities of microbial population that increases the activity of soil enzyme. Garcia *et al.,* (1998) found that restoration practices of degraded arid soils in marginal areas strongly influenced soil enzyme activities. Positive correlations were obtained between enzymatic activity and organic carbon, and with nitrogen; and between dehydrogenase activity and available phosphorus.

5.4.3. Urease enzyme activity

Urease activity decrease slightly at the beginning in the first year where enzyme activities decrease after decreasing total organic carbon (Beyer *et al.*, 1999) as can be seen in different intensively cultivated agricultural. Beri *et al.,* (1978) and Tiwari *et al.,* (1989) also reported that urease activity was principally associated with the organic matter of the soils. The studies from long-term experiments have shown that crop cultivation led to greater contents of organic C and N in soils which contributed to higher microbial activities (Bolton *et al.,* 1985). Aon and Colaneri (2001) described

strong relationships between organic carbon and total nitrogen with enzymatic activities.

The surface layer of bun cultivated soil shows the highest urease activity and decreases as the soil profile decreases. This was supported by the study of Angers *et al.,* (1993) that the enzyme activity could also be higher in the systems with minimum or no-tillage because of higher amounts of organic matter in the topsoil layer. The forest soil also has the similar types of urease activity but follows a very short increment in the activity of urease enzyme.

The urease activity increase in August after three months of investigation which could be due to the microbial activities and the soil nutrients where increasing the carbon, total nitrogen and phosphorus content could serve as a basis for increasing both biological and enzymatic soil activities. This is supported by Lioyd and Sheaffe (1973) that due to increment to ureolytic bacteria in soils, the urease activity might be increased.

The increased in the urease enzyme activity due to the presence of toluene where, toluene is known to enhance the activity of urease in nonfumigated soils, apparently by increasing the permeability of microbial membrane and this improves the contribution of the intracellular activity (Frankenberger and Johanson, 1986). It has also been reported that variation in urease activity was caused by changes in organic matter content of soils (Bremmer and Mulvaney, 1978). Tillage, residue management and cropping practices also have significant effects on urease activity in soils

(Dick, 1984; Bolton *et al.,* 1985; Deng and Tabatabai, 1996a,b; Burket and Dick, 1998).

The urease enzyme is positively correlated ($P \leq 0.05$) with soil temperature and soil pH where enzymes are markedly dependent on pH, moisture regimes, temperature and other environmental factors (Dick and Tabatabai, 1983). Long-term cropping systems can influence important soil properties such as soil structure and density, soil pH, the quantity, quality, and distribution of soil organic matter and of nutrient cycles within the soil profile (Tabatabai and Fu, 1992; Miller and Dick 1995; Omay *et al.*, 1997; Senwo and Tabatabai 1998).

The moisture contents of the soil also plays important role in the inclining of the urease activity. Pagliai and De-Nobili (1993) showed that the relationship of enzyme activity and the various pore size classes in the soil showed a positive common trend between the enzyme activity and the percentage area of pores.

In conclusion, the result indicates that enzyme activities are directly dependent on the content of organic substances in the soil (C, N forms), which may be influenced by farming activities. Soil enzyme activities are believed to be able to discriminate between soil management treatments (Dick, 1993)

Table 5.1. One-way analysis of variance (ANOVA) of the biochemical characteristics of soil in Bun cultivation (BCU) and Forest (FS) sites at surface (0-10 cm), middle (10-20 cm) and sub surface (20-30 cm) layers at $P<0.05$.

Soil properties	Source of variation	F-ratio	P-level
Dehydrogenase	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	6.644	.013
	BCU (SL) x FS (SSL)	16.164	2.66×10^{-4}
	BCU (ML) x FS (SL)	5.249	.026
	BCU (ML) x FS (SSL)	11.199	.001638
	BCU (SSL) x FS (SL)	17.87	1.11×10^{-4}
	BCU (SSL) x FS (ML)	4.236	.045
Phosphatase	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	3.727	.033
	BCU (SL) x FS (SSL)	5.145	.01
	BCU (ML) x FS (SL)	-	-
	BCU (ML) x FS (SSL)	3.77	.032
	BCU (SSL) x FS (SL)	-	-
	BCU (SSL) x FS (ML)	-	-
Urease	BCU x FS (SL)	-	-
	BCU x FS (ML)	-	-
	BCU x FS (SSL)	-	-
	BCU (SL) x FS (ML)	6.644	.013
	BCU (SL) x FS (SSL)	16.164	2.66×10^{-4}
	BCU (ML) x FS (SL)	5.249	.026
	BCU (ML) x FS (SSL)	16.316	2.01×10^{-4}
	BCU (SSL) x FS (SL)	17.870	1.11×10^{-4}
	BCU (SSL) x FS (ML)	4.236	.045

Note: Insignificant values are denoted by '-' sign.

Table 5.2. One-way analysis of variance (ANOVA) of the biochemical characteristics of soil at surface (0-10 cm), middle (10-20 cm) and sub surface (20-30 cm) layers in Bun cultivation (BCU) and Forest (FS) sites at $P<0.05$.

Soil properties	Source of variation	F-ratio	P-level
Dehydrogenase	BCU (SL x ML x SSL)	6.425	.002
	FS (SL x ML x SSL)	12.061	3.2×10^{-5}
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	10.001	0.002
	BCU (ML x SSL)	11.199	0.001
	FS (SL x ML)	6.829	0.012
	FS (SL x SSL)	20.901	3.6×10^{-5}
	FS (ML x SSL)	6.447	0.014
Phosphatase	BCU (SL x ML x SSL)	-	-
	FS (SL x ML x SSL)	-	-
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	-	-
	FS (SL x SSL)	-	-
	FS (ML x SSL)	-	-
Urease	BCU (SL x ML x SSL)	-	-
	FS (SL x ML x SSL)	-	-
	BCU (SL x ML)	-	-
	BCU (SL x SSL)	-	-
	BCU (ML x SSL)	-	-
	FS (SL x ML)	-	-
	FS (SL x SSL)	-	-
	FS (ML x SSL)	-	-

Note: Insignificant values are denoted by '-' sign.

Table 5.3. Correlation coefficient (r) values among the biochemical properties of soil at surface (SL), middle (ML) and subsurface (SSL) layers of soil in Bun cultivation (BCU) and Forest (FS) sites ($P \leq 0.05$)

Source of variation	Dehydrogenase	Urease	Phosphatase
BCU x FS (SL)	-	0.499[a]	0.832[c]
BCU x FS (ML)	0.783[c]	-	0.911[c]
BCU x FS (SSL)	0.800[c]	0.494[a]	0.833[c]
BCU (SL) x FS (ML)	-	0.478[a]	0.909[c]
BCU (SL) x FS (SSL)	0.445[a]	0.452[a]	0.909[c]
BCU (ML) x FS (SL)	0.646[c]	-	0.906[c]
BCU (ML) x FS (SSL)	0.670[c]	-	0.911[c]
BCU (SSL) x FS (SL)	0.845[c]	0.445[a]	0.816[c]
BCU (SSL) x FS (ML)	0.889[c]	0.453[a]	0.833[c]
BCU (SL x ML)	-	0.917[c]	0.859[c]
BCU (SL x SSL)	-	0.910[c]	0.872[c]
BCU (ML x SSL)	0.650[c]	0.977[c]	0.916[c]
FS (SL x ML)	0.786[c]	0.920[c]	0.934[c]
FS (SL x SSL)	0.813[c]	0.859[c]	0.934[c]
FS (ML x SSL)	0.735[c]	0.962[c]	-

Values marked with a and c are significant at ($P \leq 0.05$) and ($P \leq 0.001$) respectively; insignificant values are marked with '-'.

Table 5.4. Correlation coefficient (r) values among biochemical properties of soil with various microbial population, biological and physico-chemical properties of soil in Bun cultivation (BCU) site at surface (SL), middle (ML) and subsurface (SSL) layers ($P \leq 0.05$).

Soil depths	Soil properties	PA	URA	AT	ST	MC	pH	OC	TN	AP	K	FP	BP	C_{mic}	RF
SL	DHA	0.483^{a}	-	-	-	0.278^{a}	-0.263^{a}	0.615^{a}	-	-	-	0.643^{a}	0.678^{c}	0.648^{a}	0.654^{b}
	PA		-	-	-	0.657^{a}	-	0.755^{c}	0.507a	0.455^{a}	-	-	0.687^{a}	-	.
	URA	-		-	0.834^{c}	0.666^{b}	0.453a	0.745^{b}	0.638^{b}	-	-	-	0.452^{a}	-	-
ML	DHA	-	-	0.605^{b}	0.622^{b}	-	-	-	-	-	-	-	0.826^{c}	-	0.741^{a}
	PA		-	-	-	-	-	0.727^{c}	-	-	-	-	0.536^{a}	-	-
	URA	-		-	-	0.533^{a}	-	0.528^{a}	0.554^{b}	-	-	-	-	-	-
SSL	DHA	-	-	-	-	-	-	-	-	-	-	-	0.587^{a}	-	-
	PA		-	-	-	-	-	-	-	-	-	-	-	-	-
	URA	-		-	-	-	-	-	0.528^{a}	-	-	-	-	-	-

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

Table 5.5. Correlation coefficient (r) values among biochemical properties of soil with various microbial population, biological and physico-chemical properties of soil in Forest (FS) site at surface (SL), middle (ML) and *subsurface (SSL) layers (P≤0.05).*

Soil depths	Soil properties	PA	URA	AT	ST	MC	pH	OC	TN	AP	K	FP	BP	C_{mic}	RF
SL	DHA	0.641[b]	-	-	0.524[a]	-	-	-	-	-	-	-	0.589[a]	-	0.606[b]
	PA		-	-	-	-	-	-	-	-	-	-	0.568[a]	-	-
	URA	-			-	0.523[a]	-	-	-	-	-	0.648[a]	-	-	-
ML	DHA	0.525[a]	-	-	-	-	0.332[b]	-	-	-	-	-	-	-	-
	PA		-	-	-	-	-	-	-	-	-	-	-	-	-
	URA	-			-	0.574[b]	-	-	-	-	-	-	-	0.588[b]	-
SSL	DHA	-	-	-	-	-	-	-	-	-0.343[b]	-	-	-	-	-
	PA		0.495[a]	-	-	-	-	-	-	-	-	-	-	-	-
	URA	0.495[a]			-	0.597[b]	-	-	-	-	-	-	-	-	-

Note: AT=ambient temperature, MC=moisture content, ST=soil temperature, pH=soil pH, OC=organic carbon, TN=total nitrogen, AP=available phosphorus, K=potassium, RF=rainfall, FP=fungal population, BP=bacterial population, C_{mic}=microbial biomass carbon, URA=urease, DHA=dehydrogenase, PA=phosphatase

Values marked with a, b and c are significant at *(P≤0.05)*, *(P≤0.01)* and *(P≤0.001)* respectively; insignificant values are marked with '-'.

Chapter-VI

GENERAL DISCUSSION

The investigation shows an increases in the microbial population (fungi and bacteria) at soil surface layer (0-10 cm) at the beginning of cultivation that can be attributed to the effect of soil temperature and moisture content (Teuben and Roelofsma, 1990; Kuske *et al.*, 1997). Johnson *et al.*, (1991) also stated that higher microbial population could be due to increase in biomass since a higher input of carbon from the resident vegetation and increase in soil biomass. The decreases on microbial population could be due to soil management practices and cropping system where the product of transformation of soil properties which have accumulated and become toxic to fungal populations during some months affects the population (Broadbent and Narashima, 1971; Powlson *et al.*,1987; Kirchner *et al.*, 1993 and Tilak *et al.*, 1995). The least population observed in August and October was due to burning which reduced soil microbial diversity and most enzyme activities (Zak *et al.*, 1994; Xiaoyun *et al.*, 2000). Forest soil has no much inconsistent population but has similarities in the increase of population in the surface (0-10 cm) layer when compared with that of cultivated soil.

The surface layer has maximum fungal species richness and decreases as the soil depth increases at bun cultivation which could be due to biomass carbon where studies indicated that fungi are dominant component of the total soil microbial biomass (Christensen, 1989; Dick, 1994). The higher fungal population in the surface soil layer (0-10 cm) could

be due to the high organic matter content, nutrient status and better aeration in the top soil (Balasubramanium *et al.,* 1972) and soil moisture regimes (Selvaraj and Rangaswamy, 1978; Clarholm and Rosswall, 1980). The colony form unit of bacteria was markedly higher than that of fungi which could be due to the agricultural management practices and soil organic amendment which could leads to the alteration of microbial dynamic in the agro-ecosystem (Tilak *et al.,* 1995).

Higher population observed at the end of September is of the fact that the bacterial functioning diversity in agriculture would be affected by plant growing stages (Lahav and Steinberger, 2001). Higher bacterial population at the cultivated soil was essentially because of the low C: N ratio of the cow dung, a condition which would be more conductive to N mineralization (Asiegbu, 1984). The microbial diversity may be as important as the functional diversity to understand the principles of soil characteristics, as shown by the elegant studies of Rainey *et al.,* (2000) on *Psuedomonas species* at different diversities. The bacteria like *Azotobacter species* and *Arthrobacter species* are found very less in the bun cultivated soil due to the lower soil ph and acidity of the soil. The decrease in the bacterial population from soil surface layer (0-10 cm) to middle (10-20 cm) and subsurface (20-30 cm) layers could be due to the high organic matter, nutrient supply and better aeration in the surface layer (Balasubramanium *et al.,* 1972) and moisture regime (Selvaraj and Rangaswamy, 1978; Clarholm and Rosswall, 1980; Muller *et al.,* 2002). 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). The Shannon diversity index of fungal and bacteria shows that the surface layer has maximum species richness and decreases as the soil profile increases that are supported by Allison and Killham, (1988) and Tsai *et al.,* (1997) in a study where microbial biomass increased after straw was incorporated into soil.

The maximum similarity index of fungi was observed at a paired layer of ML x SSL and minimum similarity index of fungal was observed in the paired layer of SL x ML in both bun cultivation and forest soils throughout the investigation. The higher distribution of microorganism in the surface layer and limitation of soil N at the upper soil profiles favours the fungal population (Zhang *et al.,* 2002). The index of Sorensen and Jaccard shows that the paired layer of surface and subsurface layer (SL x SSL) has maximum similarity index of bacteria both in bun cultivation and forest soils. The minimum similarity index of bacteria is observed at a paired layer of middle and subsurface (ML x SSL) layers at forest and bun cultivation soils during the first year of investigation. The paired layer of surface and middle (SL x ML) does not show very high similarity index of bacteria both in bun cultivation and forest soil throughout the investigation. The bacterial population shows negative correlation ($P \leq 0.05$) with soil pH at forest soil which is supported by the study of Shaw *et al.,* (1990) that due to the indirect influence on soil pH and increase in microbial N mineralization may be important effect of the microbial population.

The surface (0-10 cm) layer (SL) shows a higher soil temperature and soil moisture content that could be due to the increase of ambient

temperature and lowering of the rainfall in both bun cultivation sites. A significant relationship between soil moisture and soil temperature and its consequent effect on net N mineralization was observed by Cassman and Munns (1980) and Mogaddeghi *et al.,* (2000). The soil surface layer (0-10 cm) of bun cultivation has a higher pH value than the inner depths but shows a uniform pH during the second year. This was similar to finding by Parham *et al.,* (2003) in which manure-treated soils shows almost uniform in the soil profiles from 0-10 cm depth (4.2 - 5.7) with that of untreated control soil at pH 5.0. Bun and forest sites shows a decrease in the pH during the first year but a slight increase was observed in the second year at all the three depths due to effect of plant burning residues. However, the finding reveals that addition of plant material may initially cause an increase in soil pH due to decomposition of organic anions and organic nitrogen. The first year of cultivation shows very high percentages of soil organic carbon contents (7.21%) at surface layer due to continuous cropping of field, that has not lowered carbon contents beyond the levels of reached after some years of cropping in a plot abandoned in the previous year after crop rotation (Tiessen *et al.*, 1992). Robertson *et al.,* (1994) observed that soil organic C was higher in soils under vegetation in September, possibly due to more flows of C and N to soils. In case of forest site (FS), subsurface layer shows a significant variation ($P \leq 0.05$) with the surface (0-10 cm) and the middle (10-20 cm) layers. The correlation of soil organic with soil enzymes (Urease and phosphates) and soil microbial population (bacterial population) in bun cultivation (BCU) site shows that soil organic plays a vital role in the activity

of soil microbial population and soil enzymes whereas in forest (FS) site soil organic carbon is correlated with the soil microbial population (fungal) in the middle (10-20 cm) layer and in the subsurface (20-30 cm) layer with the soil enzyme (urease). The result shows that, total SOC was higher in forest than in crop systems, but the difference was restricted to the surface layers (0-10 cm). The total nitrogen content shows very high at the surface layer during the first and second year of investigation in both bun cultivation and forest sites due decreasing distributions of soil total N contents form the surface to lower soil profiles. The adverse decrease in the soil total nitrogen during the first year at bun cultivation (BCU) is the effect of nutrient limitations on cultivated sites that prevent organic matter maintenance or increase, unless carbon inputs are accompanied by substantial fertilization. Chardon *et al.,* (1997) demonstrated that soil mineral N contents (NO_3 and NH_4) were higher under vegetation, lowest after cultivating a plot abandoned in the previous year after crop rotation and recovered with increasing fallow period following a pattern similar to that of the organic matter contents. Soil total nitrogen has a significant variation ($P \leq 0.05$) among the surface (0-10 cm), middle (10-20 cm) and subsurface (20-30 cm) layers at bun cultivation site (BCU) where forest site (FS) shows no significant variation ($P \leq 0.05$) among the three depths. Soil total nitrogen in the middle layer shows a negative correlation with available phosphorus at bun cultivation site whereas forest site has a negative correlation (-0.559 at $P \leq 0.05$) of total nitrogen with bacterial population.

The available phosphorus shows an increase in the month of September (0.04 μg $^{-1}$dry soil) during the first year of both the sites. This result is supported by Chardon *et al.* (1997) that dissolved organic phosphorus contents increased with increasing soil depth. The mark increase in the available phosphorus in the month of June and July at middle (10-20 cm) and subsurface (20-30 cm) layers of bun cultivation (BCU) site could be due to greater phosphorus movement in manure-treated soils than in untreated soils(Borie and Rubio,2003). The second year of experiment shows decrease of soil available phosphorus in July at all the three depths in forest site but shows no changes in the bun cultivation (BCU) site. There is a decrease in the soil available phosphorus during the second year in both the sites which shows that soil maturity enhance the loss of soil available phosphorus. The surface (SL) layer (0-10 cm) of bun cultivation shows no significant variation of soil available phosphorus with the forest (FS) site. The Bun cultivation (BCU) and forest (FS) sites show a significant variation between middle (ML) and subsurface (SSL) layers but has insignificant variation in the surface (SL) layer.

Exchangeable potassium is very high exchangeable potassium during the first year of bun cultivation and decreases with the increased of the soil depth. The content of available potassium is always higher in uppermost soil layer than that in lower soil layers and it has close relation to the organic matter content of the soil. The exchangeable potassium increases in July just after one month of burning at bun cultivation site which indicates that the effects of clearing and burning on the soil within a month after burning were

increase in exchangeable potassium content, decreases in N and no change in P. Increases of soil exchangeable potassium were the results of additions from ash and unburned organic matter leaching into the soil (Halenda, 1993; Rikhari and Palni, 1999).

The microbial biomass carbon (C_{mic}) increases with the increase of the soil depth after the burning at bun site and decreases in the second year of investigation, which could be due to the higher and lesser microbial population in the first and second year respectively. Specific studies indicated that fungi are a dominant component of the total soil microbial biomass, accounting for upto 90% of the total and are also the main decomposers of organic matter in soil (Polyanskaya *et al.,* 1997; Bardgett and McAlister, 1999). The decrease in the soil microbial biomass carbon could also be due to the burning and addition of the manure to the cultivated soil (Shiel, 1996). Soil management practices lead to decrease in total microbial biomass that plays a certain role in decreasing the contribution of microbial (Ananyeva *et al.,* 1999). The higher C_{mic} at surface layer during second year at bun cultivation site could be due to the decrease in carbon content of the soil whereas forest soil does not shows much variation in the microbial biomass content throughout the investigation. Wright and Reddy (2001) also observed that microbial biomass was highest at the detritus surface layer and microbial biomass carbon decreased with soil depth and was also higher at P impacted area than at the un-impacted area. The higher accumulation of soil microbial biomass at the surface layer is due to higher microbial population where the analysis of variance shows no significant

variation ($P \leq 0.05$) between the surface and middle layer at bun cultivation site (BCU). The microbial biomass carbon at the soil surface layer (0-10 cm) of disturbed forest was significantly ($P \leq 0.05$) greater than that of the middle and subsurface layers due to greater accumulation of litter and fine root biomass and presence of relatively dense growth of plants in the forest floor (Maithani, 1996). The data suggest that agricultural soils generally have lower amounts of total and microbial C than forest soils and may support equal or greater rates of microbial activity than forest soils due to increased production of water soluble carbon (Boyer and Groffman, 1996). Microbial biomass C contents were highest in the 0–10 cm surface soils with considerable variations due to sampling time. Microbial biomass C obtained in October and November in bun cultivation was significantly higher than in other months (Parham *et al.,* 2002). This trend, however, was inconsistent in the forest site. Microbial biomass C contents decreased with increasing soil depth in all the soils analysed.

The dehydrogenase activity increases at the beginning of bun cultivation but decrease in the activity towards maturity of the cultivation where surface layer (0-10 cm) has higher activity (Parham *et al.,* 2002). Dehydrogenase activity showed a trend similar to that exhibited by C_{mic}, where higher activity was detected in 20–30 cm depth (subsurface layer) soil than in the 10–20 cm depth (middle layer) soil at the beginning of the investigation. Increase of soil dehydrogenase activity from dry (July, summer) to wet (October, August) season, shows a positive relationship between dehydrogenase enzyme and water content (soil moisture content

and rainfall). The increase of soil dehydrogenase enzyme activity after three months of investigation could be due the increase in soil microbial population where the activity of dehydrogenase is considered as an indicator. The positive effect of increasing water content and nutrient addition on soil dehydrogenase has been already reported (Nannipieri *et al.,* 1990). The forest soil shows no significant and positive relationship of dehydrogenase with other factors but Leirós *et al.,* (2000) reported a clear positive relationship between soil dehydrogenase enzyme and soil C. Beyer *et al.,* (1992) and Garcia *et al.,* (1994) reported that dehydrogenase activity (DHA) has been used as an indicator of the microbiological activity in arid soils and in agricultural soils. Bun cultivation as soil management practice and the seasonal condition affects the soil enzyme activity (Batra *et al.,* 1997). Thus, the activity of soil microorganisms and soil management were strongly related to soil enzymes (Deng and Tabatabai, 1997; Klose *et al.,* 1999; Consuelo and Teodoro, 2002). Garcia *et al.,* (1994) also found that the rainy season enhanced the enzyme activity (DHA) of soils. Other authors also attributed the increase in microbial activity in forest soils (Görres *et al.,* 1998; Banerjee *et al.,* 2000) and to higher soil moisture contents. The positive effect of increasing water content and nutrient addition on soil DHA has been already reported (Nannipieri *et al.*, 1990). So management practice which increase total C accumulation and shows that dehydrogenase activity was significantly correlated with the physico-chemical properties of the soil (Buchanan, 1990).

The analysis of variance showed a significant variation of dehydrogenase activity between surfaces (0-10 cm), middle (10-20 cm) and subsurface (20-30 cm) layers which could be due to the differences in soil microbial population and rainfall. Dehydrogenase activity was significantly correlated positively with soil microbial population (bacteria) and soil management practice (bun cultivation).

Phosphatase activities were always higher at the top soil either by burning residues of the dried plants or by application of manures. The phosphatase enzyme activity increases at the beginning of the experiment and shows very high activity during the first year of investigation which may be due to the increase of the organic matter contents of the soil at bun cultivation site (Nilson and Eiland, 1980). There is a great decrease at the end of the investigation both during the first and second year of the study whereas forest soil shows no much variation in the activities of the phosphatase enzyme. The surface layer (0-10 cm) at bun cultivation shows a very high phosphatase activity, which is supported by the result of Singh *et al.*, (1997) and Tarafdar and Rao, (1990) that enzyme activities involved in P cycling were generally more uniformly distributed over a 0–10 cm soil profile. Considerable variations in phosphatase activities were significantly greater in the cultivated soil than that of the forest soils. In general, ashes from burning could show a greater impact over the 0–10 cm soil profile. This is strongly demonstrated by phosphatase activities over the soil profiles tested. Such a pattern of impact may result in a relatively long-term effect on crop production that could be the reason in the decrease of soil phosphatase

enzyme activity during the second year of investigation. Phosphatase activity decreases as soil profile decreases at bun cultivation that could be due to the decrease of soil nutrients and application of ashes to the soil enriches the soil organic matter and increases total N contents. Tiwari (1996) reported significantly greater ($P \leq 0.05$) activities of acid phosphatase in soil that has high contents of nutrients (nitrogen and phosphorus). The decrease of soil phosphatase enzyme from top surface layer to the subsurface layer and a decline from the first year to the second year of investigation is highly influenced by the soil microorganisms and the management practices where soil organisms could utilize soil organic P by means of phosphatase enzyme (Pant *et al.,* 1994). Enzyme activities have been found to be very responsive to different agricultural soil conservation practices such as non-tillage (Bergstrom *et al.*, 1998), organic amendments (Perucci, 1992; Miller and Dick 1995; Banerjee *et al.*, 1997), crop rotation (Dick 1992; Miller and Dick 1995), and organic cultivation (Beyer *et al.,* 1992). Soil phosphatase enzyme helps in the mineralization processes of organic phosphorus substrates. Enzymes are used as indicators of microbial activity for monitoring the effects on soil chemicals, disturbance, or other environmental changes (Tabatabai, 1994a). The reason is that bun cultivation promoted biological and microbial activities, which accelerated the breakdown of organic substances in the soil. The enhanced biological activities in the cultivated soil are evidenced by relatively high phosphatase and dehydrogenase activities. One explanation is that some of the earlier conclusions were derived from short-term experiments (Suri and Puri, 1997) and from studies

with a much higher manure application rate (Agbenin and Goladi, 1997). The effect of cultivation induced significant changes in the quality, chemical composition and molecular size of organic matter which in turn influenced the activities of enzymes involved in the C, N and P cycles. The positive correlation between phosphatases and C and N is in accordance with the results of Chhonkar and Tarafdar (1984); Bonmati *et al.*, (1991); Šarapatka and Kršková (1997). Chhonkar and Tarafdar (1984) described positive correlations of phosphatase activity with organic carbon, organic phosphorus and bacterial populations, and a negative relationship with soil pH. The investigation shows that soil phosphatase enzyme activities significantly correlated with the soil organic C, total N (Bonmati *et al.*, 1991) and dehydrogenase activities. The availability of soil P in the soil generates the activities of microbial population that increases the activity of soil enzyme. Garcia *et al.,* (1998) found that restoration practices of degraded arid soils in marginal areas strongly influenced soil enzyme activities. Positive correlations were found between enzymatic activity and organic carbon, nitrogen; and between dehydrogenase activity and available phosphorus.

The urease enzyme activity decrease slightly at the beginning in the first year where enzyme activities decrease after decreasing total organic carbon (Beyer *et al.,* 1999) as can be seen in cultivated soil. Beri *et al.,* (1978) and Tiwari *et al.,* (1989) also reported that urease activity was principally associated with the organic matter of the soils. The studies from long-term experiments have shown that crop cultivation led to greater contents of organic C and N in soils which contributed to higher microbial

activities (Bolton *et al.,* 1985). Aon and Colaneri (2001) described strong relationships between organic carbon and total nitrogen with soil enzymatic activities. The surface layer of bun cultivated soil shows the highest urease activity and decreases as the soil profile decreases. This was supported by the study Angers *et al.,* (1993) that the enzyme activity could also be higher in the systems with minimum or no-tillage because of higher amounts of organic matter in the topsoil layer. The forest soil also has the similar types of urease activity but follows a very short increment in the activity of urease enzyme. The urease activity increase in August after three months of investigation which could be due to the microbial activities and the soil nutrients where increasing carbon, total nitrogen and phosphorus content could serve as a basis for increasing both biological and enzymatic soil activities. This is supported by Lioyd and Sheaffe (1973) that due to increasing of ureolytic bacteria in soils, the urease activity increased. The increased in the urease enzyme activity due to the presence of toluene where, toluene is known to enhance the activity of urease in non-fumigated soils, apparently by increasing the permeability of microbial membrane and this improves the contribution of the intracellular activity (Skujins, 1976; Frankenberger and Johanson, 1982). It has also been reported that variation in urease activity was caused by changes in organic matter content of soils (Bremmer and Mulvaney, 1978). Increases in molecular size and bonding complexity in the cultivated soil were, in the evaluation of Schulten *et al.*, (1995), accompanied by decreases in the activities of enzymes involved in C, N and P contents due to which there are a high decline in the activity of

urease enzyme during the second year at cultivated soil. The investigations show that tillage, residue management and cropping practices have significant effects on urease activity in soils (Dick, 1984; Bolton *et al.,* 1985; Deng and Tabatabai, 1996a; Burket and Dick, 1998). The urease enzyme is positively correlated ($P \leq 0.05$) with soil temperature and pH of the soil where enzymes are markedly dependent on soil pH, moisture regimes, temperature and other environmental factors (Dick and Tabatabai, 1983). Long-term cropping systems can influence important soil properties such as soil structure and density, soil pH, quantity, quality, and distribution of soil organic matter and of nutrient cycles within the soil profile (Tabatabai and Fu, 1992; Miller and Dick, 1995; Omay *et al.*, 1997; Senwo and Tabatabai, 1998). The soil moisture contents also plays important role in the inclining of the urease activity. Pagliai and De-Nobili (1993) showed that the relationship of enzyme activity and the various pore size classes in the soil showed a positive common trend between the enzyme activity and the percentage area of pores. The results indicate that enzyme activities are directly dependent on the content of organic substances in the soil (C, N forms) which may be influenced by farming activities. Soil enzyme activities are believed to be able to discriminate between soil management practices (Anwarzay *et al.,* 1990; Dick, 1993).

Chapter-VII

Summary

The practice of shifting cultivation is well known amongst the hill tribes of India especially in the Northeast region. Although shifting cultivation is understood by many workers from various fields of studies but in the context of the Khasi of Meghalaya, it exists in various forms. Besides the generally known slash and burn practice of jhum cultivation, there is a special type of shifting cultivation practiced particularly in the Khasi and Jaintia Hills of Meghalaya known as "bun" strips cultivation for potato and other crops. It is practice in three different ways according to the land distribution: i) Syiem system, where the king, owner of the land distributes land, ii) Village head system, where the leader of the clan earmarks land for the village and iii) Individual owner, who have the land in the name of single or joint families. The first and second systems of cultivation practice are orthodox and never go for permanent cultivation but the third category is adaptable. Nearly 38.4 percent of the land utilization area is under the Neolithic agricultural practice of 'BUN' cultivation.

The research investigation focus on the enumeration and isolation of soil microorganisms and the effect of 'BUN' cultivation on the soil microbial community (fungal and bacterial population), soil microbial biomass C (C_{mic}), physico-chemical properties (moisture content, pH, organic carbon, N, P and K) and the soil enzyme activities.

The investigation was carried out at Mawlali village of Nongkrem area (Altitude of 1785 m above the sea level; $25^0 35^l$ N latitude and $91^0 47^l$ E longitude), Shillong, Meghalaya on cabbage crop cultivation for two consecutive cropping seasons in the month of May 2001 to December 2002. The whole valley of the Nongkrem area is covered with agricultural land, where bun cultivation is the main agricultural practice that is extensively practiced by the local people.

The average annual rainfall in the year 2001 and 2002 is 1200 cm with average temperature of 18-20°C. The climate of the study area has been very much controlled by the south-west monsoons and north-east winter winds which are neither too warm in summer nor too cold in winter. The year are divided into four seasons spring, summer, autumn and winter season respectively. The cabbage crop cultivation is conducted on the basis of local cultivation practices called 'BUN" cultivation where a bun strips are made and burn. The adjacent forest site has also been chosen for the comparative study as control where *Myrica esculenta* and *Pinus khasiana* are the dominant species.

The experimental fields were divided into two sites as bun cultivation site (BCU) and forest site (FS) with three replicates each at different depths of the soil. Soil samples were collected from surface (0-10 cm) layer (SL), middle (10-20 cm) layer (ML) and subsurface (20-30 cm) layer (SSL) respectively. The soil shows different types of textural class at each level of the soil. The surface layer (0-10 cm) of both forest and bun cultivation soil are sandy loam whereas the middle layer (10-20 cm) of forest soil is loamy

and bun cultivation middle layer soil is sandy loam. Both forest and bun cultivation subsurface layer (20-30 cm) soil has the sandy loam type of soil.

At the beginning of the experiments microbial population (fungi and bacteria) increases both in bun cultivation and forest sites. Fungal population is higher at the surface layer (0-10 cm depth) in both the sites and decreases as the soil depth decreases. The middle layer (10-20 cm depth) of bun cultivation has higher bacterial population during the second year. In general, the surface layer recorded the maximum microbial population. The highest fungal population is recorded at bun cultivation site whereas highest bacterial population was recorded at forest site. The middle and subsurface layers (20-30 cm depth) of bun cultivation maintained a very low fungal population.

The optimum species diversity of fungi was observed at bun cultivation site at middle and subsurface layers whereas surface layer of forest site contributed the highest fungal population. Bacterial species diversity was highest at middle layer at both bun cultivation and forest sites. In general, peak microbial species diversity was noted at middle layers in both the sites. The maximum similarity index of fungal species was observed at a paired layers of middle and subsurface layers (ML x SSL) during the first year at both bun cultivation and forest sites, whereas during the second year maximum fungal species similarity index is observed at a paired layers of surface and middle layers (SL x ML) at bun cultivation site and surface and subsurface layers (SL x SSL) at forest site. The minimum similarity index of fungal species was observed at a paired layers of surface and subsurface

(SL x SSL) at bun cultivation site and surface and middle layers (SL x ML) at forest site. The maximum similarity index of bacterial species was noted at paired layers of surface and subsurface layers (SL x SSL) at both bun cultivation and forest sites. Minimum similarity index of bacterial species was observed at a paired layers of middle and subsurface layers (ML x SSL) at bun cultivation site and surface and subsurface layers (SL X SSL) at forest site.

The maximum distribution of fungal and bacterial species was noted at surface layer at forest site. Minimum fungal species distribution was observed at subsurface layer and minimum bacterial distribution was noted at middle layer of bun cultivation. In general, microbial distributions are higher at the forest site. The significant variation ($P \leq 0.05$) of microbial population and positive correlation ($P \leq 0.05$) among the surface, middle and subsurface soil layers were observed. Insignificant variation of fungal population was observed between the surface, middle and subsurface soil layers. Fungal population shows insignificant correlation ($P \leq 0.05$) between surface, middle and subsurface soils layers at different sites.

Bacterial population has negative correlation ($P \leq 0.05$) with fungal population at subsurface soil layer at bun cultivation site. A total of 45 fungal species has been isolated and identified, out of which *Aspergillus sp.*, and *Pennicillium sp.* are the dominant species. A fungal species like *Penicillium granulatum, P. Jensenii, Rhizopus oryzae* etc. and *Penicillium waksmanii, Phoma eupyrena, Colletotrichum dematium* etc. are some of the species that are isolated only at a single layer at bun cultivation and forest sites. A total of

7 bacterial species were isolated where *Arthrobacter sp.* and *Bacillus sp.* are the dominant species. *Azotobacter sp.* was isolated at middle soil layer at forest site and *Arthrobacter sp.* at surface soil layer at forest site.

In general bun cultivation site shows higher soil temperature range than the forest site. Soil moisture content was higher during the cropping season and decreases at the post harvest at bun cultivation site. There is no much different in soil moisture content at surface, middle and subsurface layers in both the sites.

The soil pH decreases from pre-sowing to post harvest. Higher pH was noted at surface layer at bun cultivation site and middle layer at forest site. Surface layer contributed higher soil organic carbon and significant variation ($P \leq 0.05$) between surface, middle and subsurface layers was observed at both bun cultivation and forest sites.

Burning increases the soil total nitrogen while decreases during the post harvest. Surface layer of bun cultivation site has higher soil nitrogen content as compared to forest site. There is a significant variation ($P \leq 0.05$) of soil nitrogen content between surface, middle and subsurface layers at bun cultivation site and positive correlation ($P \leq 0.05$) of soil nitrogen content between middle and subsurface layers at forest site was observed.

The peak soil nitrogen content was observed at bun cultivation site during pre-sowing and inconsistent distribution of soil nitrogen in each monthly sampling was noted at surface, middle and subsurface layers at both the sites. The inconsistent distribution of soil phosphorus content was observed at both bun cultivation and forest sites during first year whereas

significant variation ($P \leq 0.05$) between surface, middle and subsurface layers was also observed. Higher increases in soil phosphorus content was noted at the middle and subsurface layers of bun cultivation site during the first year and higher decreases in soil phosphorus was noticed at surface layer at forest site during the second year. The surface layer showed highest soil exchangeable potassium and decreases as the soil depth decreases at bun cultivation site inconsistent distribution of soil exchangeable potassium was observed at forest site.

The soil microbial biomass carbon (C_{mic}) shows slight increases at the initial stage where higher microbial biomass carbon (C_{mic}) was observed during post harvest at both bun cultivation and forest sites. Bun cultivation has higher microbial biomass carbon (C_{mic}) accumulation at soil subsurface layer whereas middle layer showed high accumulation at forest site. Inconsistent distribution of soil microbial biomass carbon (C_{mic}) during monthly soil sampling was noticed at both bun cultivation and forest sites and insignificant variation between same layers of different sites was also observed. The subsurface layer at bun cultivation site showed insignificant correlation of soil microbial biomass carbon (C_{mic}) with surface and middle layers but there is a positive correlation of soil microbial biomass carbon (C_{mic}) with soil microbial population and dehydrogenase enzyme.

There is an inconsistent distribution of soil dehydrogenase enzyme during the monthly soil sampling at bun cultivation site whereas surface soil layer has maximum activities at forest site and decreases as the soil depth

decreases. Soil phosphatase enzyme increases rapidly during pre-sowing and decreases as the maturity of the soil at bun cultivation site.

Higher soil phosphatase enzyme activity was recorded during the first year and the peak soil phosphatase enzyme activity was observed at the surface soil layer at both bun cultivation and forest sites. Soil urease enzyme activity increases during the cropping season and the highest soil urease enzyme activity was observed at the surface soil layer in both the sites. Soil urease enzyme showed an inconsistent accumulation during the monthly sampling at forest site.

The significant variation of soil enzymes (dehydrogenase, urease and phosphatase) between surface, middle and subsurface layers at bun cultivation and forest sites was observed. The surface soil enzymes (dehydrogenase, urease and phosphatase) showed a positive correlation with middle and subsurface soil enzymes in both the sites.

Local cropping management (BUN) enhances the soil nutrient availability, microbial biomass carbon (C_{mic}) and increases the soil microbial (fungal and bacterial) population.

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9. List of seminar and workshop attended

Sl. No.	Names	Organized By	Place	Year	Participation
		Seminars			
1.	*National Roving Seminar on Patenting in Biotechnology.*	Department of Biotechnology, Ministry of Science & Technology, New Delhi.	Shillong	26th Oct. 2002	Participant
2.	*Intellectual Property Rights.*	North Eastern Hill University, Shillong.	Shillong	24-25th May 2002	Participant
3.	*72nd Annual Session of NASI and Symposium.*	The National Academy of Sciences, India.	Shillong	25-27th Oct. 2002	Participant
4.	*Atomic Energy & Societal Development in India.*	Department of Atomic Energy, Mumbai.	Shillong	18th Sept, 2002	Participant
		Workshops			
1.	*Evolving Strategies for Conservation and Commercialization of Medicinal and aromatic Plants for North East India.*	North Eastern Council, Shillong & NEBRC, NEHU, Shillong.	Shillong	15th July 2003	Participant
2.	*Conservation and Sustainable Utilization of Medicinal Plants.*	State Level Planning Committee, Meghalaya.	Shillong	1st April 2003	Participant
3.	*Workshop on Conservation of Biodiversity and Community Participation.*	Indian Environmental Society, New Delhi.	New Delhi	14-15th March 2000	Paper Presented