NITROGEN DYNAMICS IN A CHICKPEA-WHEAT ROTATION IN A HUMMOCKY FIELD

A Thesis Submitted to the College of

Graduate Studies and Research

in Partial Fulfillment of the Requirements

for the Degree of Doctor of Philosophy

in the Department of Soil Science

University of Saskatchewan

Saskatoon

By

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Spring 2000

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ABSTRACT

A study was initiated in 1996 to investigate N dynamics in a chickpea (*Cicer arietinum* L.)-wheat (*Triticum aestivum* L.) versus a wheat-wheat rotation, as influenced by landscape position, in Saskatchewan, Canada. Symbiotic N_2 fixation, decomposition of crop residue, and the N and non-N effects of chickpea were investigated.

Percentage of N derived from the atmosphere (%Ndfa) ranged from 29 to 97% in shoulders and from 38 to 95% in footslopes. According to an analysis of semivariance, only 28% of the variance in %Ndfa, measured at 0.3-m intervals, could be accounted for by spatial correlation.

Wheat grown in the second year recovered 2.2% and 3.3% of the chickpea residue N, and 2.1% and 1.7% of the wheat residue N in shoulders and footslopes, respectively. Landscape position significantly influenced N recovery from chickpea residue but not wheat residue.

In shoulders, approximately 35% of the chickpea residue N was recovered in the soil microbial biomass (SMB), whereas 13% and 30% was recovered in light (LF) and heavy fraction soil organic matter (HF), respectively. In footslopes, approximately 11% of the chickpea residue N was recovered in the SMB, whereas 29% and 44% was recovered in LF and HF, respectively. In contrast, approximately 13%, 22%, and 38% of wheat residue N was recovered in the SMB, LF and HF, respectively, in shoulders. Approximately 15%, 25%, 33% of wheat residue N was recovered in the SMB, LF and HF, respectively, in footslopes.

The influence of chickpea and wheat residue on the added N interaction (ANI) generally was low. The ANI of chickpea residue (1.1 kg ha^{-1} N) was higher than wheat

residue (-0.8 kg ha^{-1} N) in footslopes, whereas there was no detectable difference between chickpea (-1.2 kg ha^{-1} N) and wheat residue (1.3 kg ha^{-1} N) in shoulders.

The grain yield of wheat grown on chickpea stubble was 8% greater than that of wheat grown on wheat stubble in shoulders and 43% greater in footslopes. The *A* value explained 52% of the yield variation suggesting that the N effect was as important as the non-N effect.

ACKNOWLEDGEMENTS

I sincerely would like to thank Dr. F. Walley (supervisor) and Dr. C. van Kessel (former and co-supervisor) for their valuable guidance, help and patience. My appreciation is also extended to the members of the advisory committee, Dr. E. de Jong (committee chair), Dr. D.J. Pennock, Dr. A.E. Slinkard, and Dr. K.C.J. van Rees for their advice. Especially, I would like to thank Dr. D.J. Pennock for his valuable help in many aspects, Dr. K.C.J. van Rees for his advice on the studies associated with crop roots, and Dr. E. de Jong for measuring soil physical properties. The assistance from Dr. H.H. Janzen and Ms. Y. Bruinsma on the study of soil organic matter fractionation is highly appreciated. I also thank Dr. G.P. Lafond for acting as the external examiner for my dissertation defense.

My thanks also go to Dr. R. E. Farrell and Dr. J. D. Knight, and my fellow graduate students (F.C. Stevenson, A. Matus, S.P. Mooleki and M.P. Solohub) of the Stable Isotope Laboratory. Technical help from G.R. Parry, B.A. Miller, Q. Chen, R.F. Anderson, D. K. Elliott, J. Bennett, and B.L. McCann is greatly appreciated.

The study was made possible through the financial support of Saskatchewan Agriculture and Food (ADF), and Maurice Hanson Sr. Postgraduate Scholarship and Barbara and Frank Pavelich Postgraduate Scholarship provided by the College of Agriculture, University of Saskatchewan.

Last, but not least, I am very indebted to my family and close friends, especially my parents (Zaogeng Fu and Yuanxiu Zheng) and my wife (Xiaomei Jin), for their love and confidence in me throughout my Ph. D. program.

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LIST OF ABBREVIATIONS

ANI	Added nitrogen interaction
EC	Electrical conductivity
EDA	Exploratory data analysis
FC	Field capacity
FRV	Fertilizer replacement value
HF	Heavy fraction of soil organic matter
IQR	Interquartile range
LF	Light fraction of soil organic matter
MIT	Mineralization-immobilization turnover
Ndfs	Nitrogen in samples derived from soil
PWP	Permanent wilting point
SMB	Soil microbial biomass
SOM	Soil organic matter
%Ndfa	Percentage of N in samples derived from symbiotic N_2 fixation
%Ndff	Percentage of N in samples derived from ^{15}N-labeled fertilizer
%Ndfr	Percentage of N in samples derived from ^{15}N-labeled residue

1. INTRODUCTION

An important characteristic of legumes is their ability to fix atmospheric N in symbiosis with nodule-forming *Rhizobium* bacteria. Symbiotic N_2 fixation can be highly variable in field soils (Reichardt et al., 1987; Androsoff et al., 1995; Stevenson et al., 1995) and inherent variability of the soil can be a problem in the interpretation of results involving N uptake and symbiotic N_2 fixation (Reichardt, 1990). Symbiotic N_2 fixation is a dynamic process and is controlled by soil properties. It is expected that symbiotic N_2 fixation will respond to the spatial variability of soil properties. Stevenson et al. (1995) found that mean estimates of percentage of N derived from symbiotic N_2 fixation (Ndfa%) in pea (*Pisum sativum* L.), using a natural ^{15}N abundance and *A* value approach, were 72% and 84% in the footslope and the shoulder element complexes, respectively. Androsoff et al. (1995) observed a landform effect on %Ndfa in pea using an enriched ^{15}N dilution approach, but not when a natural ^{15}N abundance approach was used. Neither Stevenson et al. (1995) nor Androsoff et al. (1995) detected a correlation between %Ndfa estimated by two different estimation approaches. These authors hypothesized that symbiotic N_2 fixation was partially controlled at the landscape scale, but that strong micro-scale control may ultimately regulate symbiotic N_2 fixation.

Part of the symbiotically-fixed N in a legume crop is available to subsequent crops through the decomposition and mineralization of the legume residues. The legume residues can supply more mineral N to the succeeding crops than cereal residues due to their relatively high N content and relatively low C:N ratio as compared to cereal

residues. Research indicates, however, that the N in legume residues is only partially available to plants during the first growing season (Wagger, 1989; Stevenson and van Kessel, 1997). Crop residues added to the soil must pass through a microbial biomass that partly mineralizes them and partly converts them into new products (van Veen et al., 1984). The residue C and N remaining in the soil are gradually transferred from labile pools to more stabilized pools (Hassink and Dalenberg, 1996). The information regarding the transfer of residue N into soil organic matter (SOM) fractions, however, is limited.

As a direct N source for the microbial biomass and the succeeding crops, the incorporated residues also influence the availability of soil N via a 'priming effect' or an 'added nitrogen interaction' (ANI) process. Yaacob and Blair (1980) and Azam et al. (1993) investigated the impact of residue on the ANI. These studies, however, were conducted only under laboratory conditions and not under field conditions. Differences in soil properties and soil N pools at different landscape positions might cause diverse degrees of pool substitution and mineralization and immobilization turnover (MIT), suggesting possible landscape controls on the ANI process, specifically, on the N dynamics of the incorporated crop residues.

The benefits associated with the inclusion of a legume in a crop rotation can be partitioned into the N effect and the non-N effect (Bullock, 1992; Stevenson and van Kessel, 1996a). The N effect is defined as the yield advantage associated with the extra soil N available to the succeeding crop attributable to the symbiotic N_2 fixation in the legume (Pierce and Rice, 1988; Stevenson and van Kessel, 1996a). The non-N effect of a legume in a legume-cereal rotation is that portion of the yield increase not explained

by the extra N accumulated by the succeeding crop and that cannot be compensated for by N fertilization (Hesterman et al., 1987; Bullock, 1992). The common methods used to quantify the N effect involve the determination of the fertilizer replacement value (FRV) of the legume or the N availability from the ^{15}N-labeled legume residue to the succeeding crops.

Using the FRV method, the yield of a cereal crop following a legume is compared to the yield of the same cereal crop with various rates of N fertilization in continuous cereal monoculture. The FRV is the quantity of N fertilizer required to produce a yield of the cereal crop following a cereal crop equivalent to that produced following a legume (Bullock, 1992; Stevenson and van Kessel, 1996b). Stevenson and van Kessel (1996b) found that the FRV of pea in a pea-wheat (*Triticum aestivum* L.) rotation was 150 kg ha^{-1} N. The ^{15}N methodology provides a direct measurement of the N contribution from the legume residue to the succeeding cereal crop (e.g., Ladd et al., 1983; Harris and Hesterman, 1990). Using this method, Stevenson and van Kessel (1996b) found that pea residue contributed only 6 to 14 kg ha^{-1} N more than wheat residue to the succeeding wheat crop.

Apparently, a gap exists between the FRV and ^{15}N methodology for estimating the N effect. The reason for the gap may be due to the fact that the FRV method actually measures the overall benefits (i.e., the N effect plus the non-N effect) of growing a legume crop in the rotation with a cereal crop. In contrast, the ^{15}N method measures only the N contribution from the incorporated legume residue to the succeeding cereal crops (Bullock, 1992).

The direct measurement of the N contribution from the legume residue to the succeeding cereal crop, using the ^{15}N method, is the first step in gaining a more accurate estimate of the N effect of a legume crop. Nitrogen contribution from other sources, such as N release from microbial biomass and the SOM pools, the MIT effect on the release of N from the legume residue, and the increased availability of soil N due to the incorporation of legume material (i.e., the 'priming effect'), might, however, significantly contribute to the N effect (Harris and Hesterman, 1990). In addition, Ladd et al. (1981) observed 72 to 78% of the added legume ^{15}N in the soil organic fractions after one cropping season and suggested that building up the SOM was the main benefit of growing a legume crop. Consequently, the N effect, measured by the ^{15}N methodology, might be underestimated.

The result of using ^{15}N methodology also might be influenced by pool substitution, i.e., added ^{15}N can take the place of unlabeled native soil mineral N that would otherwise have been immobilized or removed from the soil mineral N pool (Jenkinson et al., 1985). This process conserves the added ^{15}N through pool substitution and MIT, and makes it less available to the succeeding crops.

In order to optimize the utilization of the legume residue N by succeeding crops and understand the mechanisms of the N effect of a legume crop in the crop rotation, it is necessary to improve our understanding of the symbiotic N_2 fixation process, dynamics of the N turnover of legume residues incorporated into the soil, the effect of legume residue incorporation on the mineralization of native SOM, and the contribution of legume residues to SOM maintenance. Because N-cycling processes, such as symbiotic N_2 fixation and mineralization of crop residues, respond to the spatial variability of soil

properties, a landscape approach would be more valuable than a small-plot approach to help understand the mechanism of the N-cycling processes.

A chickpea (*Cicer arietinum* L.)-wheat (*Triticum aestivum* L.) and wheat-wheat crop rotation study was established in a hummocky field in the Dark Brown soil zone in Saskatchewan in order to:

(i) estimate symbiotic N_2 fixation in chickpea at both the landscape scale and the micro scale;

(ii) measure the availability of N in chickpea residue and wheat residue to the succeeding wheat crop, and investigate the influence of added chickpea residue and wheat residue on the availability of native soil N;

(iii) study the transfer of the chickpea residue N and wheat residue N into SOM fractions; and

(iv) determine the N effect and the non-N effect of chickpea in a crop rotation.

2. LITERATURE REVIEW

2.1 Decomposition and Mineralization of Crop Residues

2.1.1 Nitrogen contribution from legume crops to soil via symbiotic dinitrogen fixation

Grain legumes have emerged as important components of arable systems due to the attractive financial returns from their harvested grain. When grown in a rotation with cereal crops, however, legume crops can also provide many other benefits related to the process of symbiotic N_2 fixation.

It has been suggested that legume crops will add N to the soil system via symbiotic N_2 fixation, if the total quantity of N symbiotically fixed by the legume crop is greater than the quantity of N removed in harvested grain (Evans et al., 1989; Doughton et al., 1993). This concept, however, ignores the N gains from atmospheric deposition and non-symbiotic fixation, as well as losses from plant and soil volatilization, runoff, leaching and denitrification (Doughton et al., 1993). Stevenson and van Kessel (1997) observed that the N contribution by pea to the soil N pool via symbiotic N_2 fixation was 45 kg ha^{-1} in shoulder element complexes and 63 kg ha^{-1} in footslope element complexes in a hummocky field in Saskatchewan. In Australia, Evans et al. (1989) found that narrow-leaf lupin (*Lupinus angustifolius* L.) contributed an average of 38 kg ha^{-1} N to the soil N pool, whereas pea contributed 18 kg ha^{-1} N. Armstrong et al. (1994) reported that the average N contribution to the soil N pool by field pea was 26 kg ha^{-1} in Australia. Legume crops, however, may not always make a positive N contribution to the soil in which they grow.

For example, Evans et al. (1989) demonstrated a large range in the N contribution from legume crops (i.e., - 41 to + 135 kg ha^{-1} for narrow-leaf lupin and – 32 to + 96 kg ha^{-1} for pea). Larson et al. (1989) found that N contribution to the soil system by white lupin (*Lupinus albus* L.) ranged from – 68 to – 23 kg ha^{-1} in the United States.

The N contribution of legume crops to the soil system likely is dependent on the symbiotic N_2-fixing activity, growth and N harvest index of the legume crops, which in turn, are controlled by soil and environmental conditions. Doughton et al. (1993) demonstrated that chickpea provided a positive N contribution to the soil N pool when the symbiotic N_2 fixation rates were high and a negative N contribution when the symbiotic N_2 fixation rates were low. In addition, a close negative relationship (r^2 = 0.85) existed between soil nitrate measured at the time of establishment of chickpea and the subsequent N contribution to the soil N pool.

Evans et al. (1989) and Armstrong et al. (1994) noticed that the N contribution by pea to the soil N pool usually was greatest at sites having the highest available soil moisture. They concluded that this relationship was due to lower N harvest indices and increased symbiotic N_2 fixation by pea. Larson et al. (1989) attributed the negative N contribution to the soil N pool by white lupin to high N harvest indices, which ranged from 0.80 to 0.91. When working with the narrow-leaf lupin in Australia, however, Herridge (1982) found that the N removed with harvested grain was less than the quantity of N fixed (i.e., a positive N contribution) when the N harvest indices ranged from 0.37 to 0.42.

Legumes may use less soil mineral N than cereal crops because legume crops use atmospheric N as their major N source (Evans et al., 1989). Thus, legumes might increase the plant-available N in the soil. Higher concentrations of soil mineral N result from the

conservative use of N (i.e., 'N sparing') by the preceding legume crop, and the release of mineral N from the legume residues incorporated to the soil (Doughton and McKenzie, 1984).

2.1.2 Availability of crop residue nitrogen to the succeeding crops via decomposition and mineralization

The N contribution of crop residues to succeeding crops is of particular importance with legume residues. Most estimates of the N effect of legume residues have been based on the uptake of ^{15}N from ^{15}N-labeled legume residues by the succeeding crop. Previous investigations in Australia (Ladd et al., 1983), Finland (Muller and Sundman, 1988), and USA (Harris and Hesterman, 1990), which used ^{15}N-labeled legume residues as a green manure, indicated that 10 to 34% of the legume N was recovered in the succeeding barley (*Hordeum vulgare* L.) or wheat crop. At three sites on the Canadian prairies (i.e., Lethbridge, Swift Current and Saskatoon), wheat recovered an average of 14% of residue ^{15}N applied when lentil (*Lens culinaris* cv. Indianhead) and Tangier flatpea (*Lathyrus tingitanus* cv. Tinga) were used as green manures (Janzen et al., 1990). Bremer and van Kessel (1992) found that 5.5% of the ^{15}N added as labeled lentil straw was assimilated by the succeeding wheat crop. These studies illustrate that climate, residue type and soil influence the decomposition of soil-incorporated legume residues.

Moore (1974) observed that, following small additions of high N-concentration crop residues (i.e., legume residues), the availability of N to the succeeding crops declined progressively with each growing season. Working in Denmark, Jensen (1994b) found

that the recovery of N from ^{15}N-labeled mature pea residue in successive crops of spring-sown barley, oilseed rape (*Brassica napus oleifera* L.) and wheat was 6%, 2% and 2%, respectively. The author suggested that the rate of residue N mineralization declined rapidly after one year of decomposition, due to stabilization of the labeled residue N in the slowly decomposable pools of SOM. Results of similar studies in uncropped soil revealed that after three years of decomposition, 45% of the residue ^{15}N input was present in organic forms in the topsoil, whereas only 1 to 2% of the residual organic ^{15}N was potentially mineralizable after two years of decomposition (Jensen, 1994a). Ta and Faris (1990) also observed that in the first, second and third years following application of alfalfa (*Medicago sativa* L.) residue, residual alfalfa N provided an average value of 15%, 6% and 5% of the total N content in the succeeding barley crops and that the N use efficiency from labeled alfalfa residue was 11%, 4% and 3%, respectively. In Canada, Janzen et al. (1990) noticed that the second succeeding wheat crop recovered only 1 to 2% of the N from Tangier flatpea residue and lentil residue. They suggested that the primary advantage of green manure production may be the long-term replenishment of stable organic C and N reserves in the soil.

Although these studies suggest that decomposition of legume residues occurs mainly in the first year after application, a large portion of the N in the legume residue apparently is unavailable to the first succeeding crop. This raises questions with regards to the dynamics of N from legume residues. A number of studies have demonstrated that most of the N from legume residues or cereal residues is assimilated by microbes and subsequently transferred into more passive fractions of the SOM (Wagger et al.,

1985; Harris and Hersterman, 1990; Bremer and van Kessel, 1992). Meanwhile, a portion of the N in the crop residue can be lost from the soil system by leaching (Jensen, 1994a) and denitrification (Harris and Hersterman, 1990; Aulakh et al., 1991) when soil moisture is excessive. The observation that residue N recovery generally is low also may be due to the asynchronous mineralization of N from residues with the N demand of the succeeding crop. The asynchronous N mineralization and uptake could promote leaching and denitrification losses of mineral N derived from crop residues (Aulakh et al., 1991).

Legume residues and cereal residues differ in decomposition rates due to differences in their chemical composition. Cereal residues, such as grain sorghum (*Sorghum bicolor* L.) and corn (*Zea mays* L.), generally have higher C:N ratios (or low N contents) and may require additions of exogenous N in order for decomposition to proceed (Herman et al., 1977). Consequently, the decomposition rates of legume residues are higher than those of cereal residues because the critical C:N ratio for net mineralization to occur would be attained more quickly by the legume residues as compared to the cereal residues (Janzen and Kucey, 1988). Residues from corn, rice (*Oryza sativa* L.), and wheat with high C:N ratios typically cause N immobilization to occur initially (Hargrove et al., 1982; Christensen, 1985; Power et al., 1986). In an incubation study, using ^{15}N-labeled sorghum, wheat and alfalfa residues, Smith and Sharpley (1993) found that less than 28% of the sorghum residue N and wheat residue N was mineralized at the end of a 168-d incubation period, whereas 35% of the alfalfa residue N was mineralized. Norman et al. (1990) also studied the mineralization of different

crop residues and found that 3% and 11% of the N from rice and soybean (*Glycine max* L.) residues, respectively, were available to the succeeding crop.

Differences in N recovery from legume and cereal residues are not always detected. For example, Bremer and van Kessel (1992) observed that about 5.5% of the ^{15}N added in lentil straw and wheat straw was assimilated by the succeeding wheat crop. Their result may have been attributable to the similar C:N ratio in wheat straw (43:1) and lentil straw (31:1).

Because the recovery efficiency of residue N depends on many factors, including crop management, soil properties (e.g., N concentration, soil temperature and moisture), residue quality, and uptake ability of succeeding crops, diverse results for this value are expected from experiments at different sites or in different years. In addition, the non-N effect may influence the estimation of the N contribution from legume residues to the succeeding cereal crop in a legume-cereal rotation (Hesterman et al., 1987).

2.1.3 Effect of residues on soil organic matter maintenance

Soil organic matter is the major source of N, P, S and many micronutrients in soils, is essential for the maintenance of soil structure and contributes to the ability of the soil to retain nutrients and moisture (Allison, 1973; Stevenson, 1985). Recently, SOM has assumed an even greater importance as a source or potential sink for atmospheric CO_2 (Post and Mann, 1990). As a result, preserving or augmenting SOM content is justified from both an agronomic and an environmental perspective.

Crop residues, including roots, are the primary source of organic material additions to soil in many cropping systems. All crop residues incorporated into the soil must pass

through a microbial community that partly mineralizes them and partly converts them into new products (van Veen et al., 1984). The residue C and N remaining in the soil are gradually transferred from labile pools to more stabile pools (Hassink and Dalenberg, 1996). Degradation of crop residues releases approximately 55 to 70% of the C to the atmosphere as CO_2, whereas 5 to 15% of the C is incorporated into the microbial biomass, and the remaining C (15 to 40%) is partially stabilized in the soil as new humus (Jenkinson, 1971; Stott and Martin, 1989). In an incubation study using ^{14}C-labeled rye (*Secale cereale* L.) shoots, Hassink and Dalenberg (1996) found that most of the label was present in the soluble and light macro-organic matter fractions two days after application. They also observed that the newly synthesized microbial biomass fed on the labeled components of the fractions.

In addition to providing a direct N source to the succeeding crops, Ladd et al. (1981) suggested that building up the SOM was the main benefit of legume residues. They observed 72 to 78% of added legume ^{15}N was in the soil organic fractions after one cropping season. After 8 years, 31 to 38% of the added legume ^{15}N was still in the organic fractions of the soil (Ladd et al., 1985). Research by Wagger et al. (1985) and Stevenson and van Kessel (1997) also indicates that the N in legume residue is only partially available to plants during the first growing season, whereas the beneficial effect of these residues is due largely to the fact that they increase the long-term fertility of the soil (Ladd et al., 1983; Palm and Sanchez, 1991). The increase in the long-term soil fertility is likely due to the conversion of a portion of the biologically-fixed N in residues into stable humus which directly and indirectly improves soil fertility.

2.2 Factors Controlling Decomposition and Mineralization of Crop Residues

Crop residues, which are incorporated into the soil as particulate organic matter, are colonized by the soil microbes, adsorbed by mineral particles, and reduced in particle size by the feeding activity of decomposing microbes (Swift et al., 1979; Golchin et al., 1994). Microbial decomposition of crop residues is controlled largely by soil moisture and temperature (Paul and Clark, 1996), exogenous N supply (Swift et al., 1979), and the availability of substrate (Stott et al., 1990), both in terms of the physical placement of the residue and the chemical composition of the residue (Christensen, 1986; Janzen and Kucey, 1988).

2.2.1 Soil moisture and temperature

Soil moisture influences not only the quantity of moisture available to the decomposing soil microorganisms but also the soil aeration status, the quantity of soluble nutrients, osmotic pressure, and soil pH (Paul and Clark, 1996). The optimum soil moisture potential for residue decomposition occurs between soil water potentials of – 0.03 and – 0.1 MPa (Paul and Clark, 1996). Bacterial respiration declines rapidly as the soil water potential declines below – 0.3 MPa, whereas fungal activity may continue to decline to a soil water potential of – 4 to – 5 MPa (Wilson and Griffin, 1975). Schomberg et al. (1994) found that the decomposition rate coefficients of both alfalfa residue and grain sorghum residues increased linearly with the amount of water applied in a field study in Texas.

In an incubation study of mineralization of winter rye (*Secale cereale* cv. Halo) and oilseed radish (*Raphanus sativus* cv. Pegletta) shoot material (5-wk old) incorporated

into the soil, van Dam and Fu (1996) found that mineralization of plant shoot material generally increased with temperatures from 0 to 20^{0}C; however, 45 to 60% of the plant shoot N was present as mineral N in soil after an 8-wk incubation period at 0^{0}C, compared to 62 to 72% at 20^{0}C. Roper (1985) and Stott et al. (1990) also observed that significant microbial decomposition of wheat straw can occur at temperatures as low as 0^{0}C; however, the maximum decomposition rate occurred near 30 to 35^{0}C. Douglas and Rickman (1992) developed a model, which included cumulative degree days, N coefficient based on initial residue N content, and water coefficient based on a combination of residue and field management, to predict the decomposition of cereal residues. They found that the relationship between cereal residue decomposition and cumulative days was the same at nearly all locations evaluated. Thus, they suggested that cereal residue decomposition could be estimated using cumulative degree days computed with air temperature, if residue placement and initial N content of the residue were known.

Several scientists have detected complex interactions between soil moisture and temperature that influence the decomposition rates of residues (Bunnell et al., 1977; Gilmour et al., 1977; Hunt, 1977). It is difficult to interpret the interactions involving soil moisture and temperature because stress factors seldom act independently in nature. In some instances, the effects may be additive, in others, multiplicative (Paul and Clark, 1996).

2.2.2 Residue application methods

Adoption of zero-tillage or minimum tillage systems, which maintain crop residues at or near the soil surface, continues to increase (Hargrove et al., 1991). The advantages of such systems include reduced soil erosion, less on-farm energy use, more available soil moisture, and greater soil particle aggregation (Unger and McCalla, 1980; Martin et al., 1989; Carter, 1992). Adoption and increased usage of these systems, however, have created a concurrent requirement for more detailed information regarding N availability in the presence of surface-applied crop residues. Leaving crop residues on the soil surface may increase mineral N levels in percolate and/or surface water runoff (Smith et al., 1991), and improve N use efficiency of crop residues (Phillips et al., 1980).

Christensen (1986) found that during the first month after burial, the average weight loss of barley straw was 35% when the straw was enclosed in a mesh bag and buried, whereas the average weight loss was 13% when the barley straw was enclosed in a mesh bag and placed on the soil surface. Parker (1962) observed that half the corn stalk residue placed on the soil surface had decomposed in 8 wk, compared to 5 wk for incorporated corn stalk residue. Hargrove et al. (1991) also observed that the decomposition rates of four different crop residues were significantly greater when residues were incorporated into the soil as compared to when they were maintained on the soil surface. The higher decomposition rate of buried residues, as compared to that of surface-applied residues, is attributable to greater soil-residue contact, a more favorable and stable microenvironment for decomposition, and increased availability of exogenous N for decomposing microorganisms (Unger and Parker, 1968; Brown and Dickey, 1970).

2.2.3 Chemical composition of the residue

In their model of plant residue decomposition, van Veen et al. (1984) assumed that the plant substrate consists of three fractions: (1) easily decomposable sugars and amino acids; (2) slowly decomposable cellulose and hemicellulose; and (3) resistant lignin. Each constituent of the crop residue has its own specific decomposition rate. For example, the first-order decay constants of lignin and hemicellulose were 0.003 and 0.03 day^{-1}, respectively, under laboratory conditions (Paul and Clark, 1996), whereas rapid loss of simple sugars and amino acids may occur within a few hours to a few days (Stott and Martin, 1989).

The chemical composition of crop residues plays an important role in determining the rates of residue decomposition. The concentrations of N, polyphenols and lignin in crop residues are the generally recognized plant factors controlling the N mineralization rate of residues incorporated into the soil (Haynes, 1986). Several researchers found that the N concentration or the C:N ratio of incorporated crop residues was the best predictor of N mineralization rates (e.g., Molina et al., 1983; Janzen and Kucey, 1988).

The C:N ratios of legume residues, which are reported in the literature, range from 10:1 in vetch (*Vicia villosa* Roth.) residue (Azam et al., 1993) to 60:1 in chickpea residue (Hooda et al., 1986; Manna et al., 1997); whereas the C:N ratios of cereal residues ranged from 16:1 in corn residue (Azam et al., 1993) to 120:1 in wheat residue (Stevenson and van Kessel, 1996b). The C:N ratios of crop residues likely are dependent on the crop genotype, length of growing season, growth medium and the environmental conditions.

The N concentration must be greater than a critical level of 15 to 25 g kg^{-1} or the C:N ratio should reach approximately 30:1 or less during decomposition before net mineralization will occur (Stevenson, 1985; Power et al., 1986). The underlying biological principle for this observation is that the soil microorganisms have an average C:N ratio of 8:1, suggesting that soil microorganisms must take up eight parts of C for every part of N (Brady and Weil, 1996). Because about only one-third of the C metabolized by microorganisms is incorporated into their cells (the remainder is respired and lost as CO_2), the microorganisms must find about 24 parts of C for every part of N assimilated (Brady and Weil, 1996). Thus, if the C:N ratio of crop residues incorporated into the soil exceeds approximately 25:1, the soil microorganisms will take up mineral N from the soil solution. As a result, the incorporation of residues with a high C:N ratio will deplete the supply of mineral N from the soil, and consequently cause net N immobilization during the initial stages of mineralization (Hargrove et al., 1982; Power et al., 1986). The decomposition of crop residues will be retarded if sufficient mineral N to support microbial growth is neither present in the crop residue undergoing decomposition nor available in the soil solution.

It has been demonstrated that when crop residue contains high concentrations of lignin or polyphenols, little mineralization of residue N will occur, even though the N concentration may be considerably greater than the critical level (Muller et al., 1988; Palm and Sanchez, 1991). Likely, residues with similar C:N ratios can have different decomposition rates because of variations in the chemical constituents (Stott and Martin, 1989). Consequently, the use of critical C:N ratios and initial N contents in crop

residues to predict residue N immobilization and mineralization patterns has been criticized because they are site specific and species specific.

Melillo et al. (1982) and Muller et al. (1988) found that the lignin concentration of crop residue was a much better predictor of crop residue decomposition rates than N concentration. Frankenberger and Abdelmagid (1985) demonstrated that N mineralization of residue was most closely correlated with residue N concentration, but the lignin concentration controlled the mineralization rate. Studies where lignin had no effect on N mineralization rate were generally conducted with crop residues containing low lignin concentrations (e.g., Iritani and Arnold, 1960; Janzen and Kucey, 1988). Palm and Sanchez (1991) demonstrated that the best predictor of N mineralized in 8 wk from ten tropical legumes and rice straw was the polyphenol:N ratio in the residue. Fox et al. (1990) observed that (lignin+polyphenol):N ratio of the legume was significantly correlated with N mineralization in an incubation study.

It is evident that the chemical composition of crop residue is the important factor controlling residue decomposition. The best predictor of net N mineralization, however, will vary with the experimental methods used and the method of measuring the net mineralization (i.e., N uptake by a growing crop or periodic subsampling of incubated soil). Moreover, single studies probably are not sufficient to develop a general relationship.

2.2.4 Soil mineral nitrogen

Lueken et al. (1962) and Swift et al. (1979) observed that mineral N in soil (native or added) enhanced the mineralization of crop residues. In a laboratory incubation

experiment using six Mollisols from Illinois, Azam et al. (1993) found that mineralization of N from soybean residue and corn residue was most rapid in the soil having the highest content of mineral N and potentially mineralizable N.

In order to examine the effect of soil mineral N on the decomposition of maize residues, Recous et al. (1995) incubated maize residues in soils with five initial mineral N concentrations (i.e., 10, 30, 60, 80, and 100 mg kg^{-1} N) and found that N immobilization was much lower in the two lowest N treatments because decomposition was slow and microbial N immobilization per unit of mineralized C was reduced. Christensen (1986) also noticed that the capacity of the soil to supply mineral N controlled the amount of N immobilized during the decomposition of buried straw.

2.3 Influence of Crop Residues on the Availability of Soil Nitrogen

2.3.1 Added nitrogen interaction

Nitrogen fertilizer, labeled with ^{15}N, has been used to investigate N recovery and the fate of fertilizer N because it is possible to distinguish between soil-derived N (i.e., unlabeled) and fertilizer-derived N (i.e., labeled) in such experiments. Many studies demonstrated that addition of fertilizer N promoted the mineralization of soil N (e.g., Woods et al., 1987; Azam et al., 1991; Rao et al., 1991). Hauck and Bremner (1976) referred to the increase in N derived from the soil following N fertilization as the 'priming effect'. In their discussion of the 'priming effect', Jenkinson et al. (1985) introduced the term 'added nitrogen interaction', or 'ANI', to describe any increase (i.e., positive ANI) or decrease (i.e., negative ANI) in the mineralization of native soil N following N fertilization. The reasons for the ANI occurrence are diverse and have been

comprehensively discussed by Jansson (1958), Broadbent (1965) and Jenkinson et al. (1985).

According to Jenkinson et al. (1985), ANI can be 'real', or 'apparent'. A real ANI occurs if fertilizer N increases the volume of soil explored by roots and, thus, increases N uptake. Similarly, if the soil microbial populations are limited by low concentrations of soil mineral N, a real priming effect may accompany the increased microbial activity that results from mineral N additions. An apparent ANI may be caused by pool substitution or by isotope displacement reactions. Pool substitution is the process by which added labeled N takes the place of unlabeled native soil mineral N that would otherwise have been immobilized or removed from the soil mineral N pool (Jenkinson et al., 1985; Hart et al., 1986). Moreover, ANI processes can conserve applied ^{15}N through MIT, pool substitution or biological exchange reactions (Jenkinson et al., 1985).

The ^{15}N isotopic technique has been used by many scientists to determine the N recovery efficiency by direct measurement of N from ^{15}N-labeled fertilizers or from crop residues taken up by the crop (e.g., Yaacob and Blair, 1980; Rao et al., 1991; Azam et al., 1993; Hamid and Ahmad, 1995). The interpretation of results from this methodology, however, may be influenced by pool substitution of ^{15}N for ^{14}N, which can result in erroneous estimations of N recovery efficiency when pool substitution is not accounted for quantitatively. The labeled ^{15}N acts as a substitute for unlabeled soil ^{14}N that otherwise would have been removed from the mineral N pool during processes such as immobilization and denitrification (Jenkinson et al., 1985). This substitution and the exchange of ^{15}N for ^{14}N during MIT, i.e., the continuous cycling of N between

organic and mineral products of microbial activities, could result in low apparent N recovery efficiency because a portion of the applied ^{15}N was not accessible to the crops (Jansson and Persson, 1982). Consequently, estimates of mineralized N from crop residues, based on the availability of residue N to the succeeding crop, might be underestimated (Janzen et al., 1990).

Using an incubation study in which $^{15}NO_3^-$ was added to the soil along with unlabeled corn residue, Blackmer and Green (1995) found that, although most of the NO_3^- immobilized early in the study was labeled, most of the N subsequently mineralized was non-labeled, suggesting that little of the N incorporated into the microbial biomass during residue decomposition was mineralized to NO_3^- during the study period. They concluded that this sequential immobilization and mineralization should be recognized as a potential source of error in ^{15}N-tracer studies because sequential processes violate the commonly held assumption that mineralization and immobilization occur simultaneously.

Most studies on ANI were conducted using ^{15}N-labeled N fertilizers. Both positive ANI (Rao et al., 1991) and negative ANI (Leitch and Vaidyanathan, 1983) have been reported. When fertilizer additions become too high for the plant to take up, the ANI decreases and can even become negative (Bigeriego et al., 1979). Hart et al. (1986) observed a positive ANI for N fertilizers in a pot experiment, but not in a field experiment. Azam et al. (1991) reported a significant enhancement in mineralization of native soil N due to applied N and the effect was positively correlated with the rate of application in an incubation study. Hamid and Ahmad (1995) found that ammonium nitrate, urea and ammonium sulphate resulted in 59%, 43%, and 26% more ANI,

respectively, when the fertilizers were broadcast and worked-in as compared to band placement. Most investigators, who have compared NH_4^+-N and NO_3^--N, have observed larger ANI with NH_4^+-N than with NO_3^--N due to the preferential immobilization of applied NH_4^+-N (Walker et al., 1956; Rennie and Rennie, 1973; Steele et al., 1980).

2.3.2 Impact of crop residues on added nitrogen interaction

Few reports are available regarding the interaction of crop residue N with native soil N. Yaacob and Blair (1980) used ^{15}N-labeled soybean residue and siratro (*Macroptillium atropurpureum* L.) residue in a pot study and found that the addition of either residue stimulated the release of native organic N (i.e., positive ANI) on all soils tested. In a laboratory incubation study, Azam et al. (1993) observed that ^{15}N-labeled soybean residue and corn residue incorporated into the soil resulted in a negative ANI, whereas ^{15}N-labeled vetch residue incorporated into the soil resulted in a positive ANI. In a field study, Jensen (1994b) observed that the incorporation of ^{15}N-labeled pea residues increased the accumulation of non-labeled soil N in autumn-sown crops by 6 and 2%, when harvested in December and at maturity, respectively.

Such diverse ANI findings from studies with labeled N fertilizers and labeled crop residues undoubtedly reflect the variability in experimental conditions, soil tested, test crop, the nature of added materials, and the approaches of the study.

2.4 Fractionation of Soil Organic Matter

2.4.1 Identifying labile components of soil organic matter

Soil organic matter is highly heterogeneous. It is generally accepted that SOM contains fractions with a rapid turnover rate (i.e., labile fractions) and fractions with a slower turnover rate (i.e., passive fractions) (Campbell et al., 1967). Labile SOM refers to a heterogeneous pool of living and dead organic material that is readily circulated through biological processes and pools. The equilibrium between decay and renewal processes in this labile pool controls nutrient availability and SOM status, i.e., determines whether organic matter qualities are improving or degrading (Wander et al., 1994).

The various labile fractions are very dynamic and account for much of the organic matter fluctuations over time, although they represent only a small proportion of the total SOM (Cambardella and Elliott, 1992). Motavalli et al. (1994) suggested that the methods commonly used for measuring and characterizing SOM, such as levels of organic C and humic acids, may be of limited use to understanding the link between SOM dynamics and nutrient availability because they do not measure biologically active SOM.

Most of the methods used in identifying and quantifying the labile SOM can be categorised into two groups: bioassay methods and fractionation methods (Biederbeck et al., 1994). The bioassay methods typically use incubation studies to estimate the quantity of potentially mineralizable SOM by analyzing the end products, such as CO_2 or NO_3^-. The results provide a direct estimate of the quantity of decomposable SOM in the soil. These results, however, provide little information about the chemical or physical nature of the labile SOM. Fractionation methods, on the other hand, isolate specific SOM constituents with different turnover rates and physically divide SOM into

pools differing in composition and biological functions (Christensen, 1992). In addition, the density fractionation methods, used for the physical separation of the labile fraction of SOM, are straightforward, reliable and reproducible (Gregorich and Ellert, 1993).

Using density fractionation techniques for physical separation, SOM can be divided into two broad components: (1) mineral-free and partly-decomposed plant debris, i.e., the light fraction (LF); and (2) organic matter adsorbed onto mineral surfaces (or deposited on them by microflora) and sequestered within organo-mineral microaggregates, i.e., the heavy fraction (HF) (Strickland and Sollins, 1987). The mineral-free debris is lighter and can be separated from the bulk soil by flotation on water or a denser solution (1.2 to 2.0 g cm^{-3}), such as NaI at 1.7 g cm^{-3} (Gregorich and Ellert, 1993; Bremer et al., 1994). Various heavy liquids have been used in densimetric fractionation procedures, including bromoform (Greenland and Ford, 1964), carbon tetrachloride (Scheffer, 1977), and tetrabromomethane/benzene (McKeague, 1971). Use of inorganic media, such as NaI in density separation techniques, obviates the problems with toxicity (i.e., to humans), carbon contamination, and coagulation of suspended particles associated with the use of organic solvents (Gregorich et al., 1994).

2.4.2 Biological activity associated with the light and heavy fractions

Spycher et al. (1983) found that major components of the LF in a forest soil, identifiable from scanning electron micrographs, were dead root fragments, hyphae, charcoal and pumice - all with adsorbed or entrapped colloidal particles. The LF material is intermediate between plant residue and humified SOM with regard to carbohydrate composition, amino acid composition, and C:N ratio (Turchenek and

Oades, 1979). The C:N ratio of the LF is usually wider than that of the bulk soil and of the particle-size fractions, reflecting the dominant influence of crop materials on this pool of SOM (Greenland and Ford, 1964). The LF has a relatively narrow C:N ratio (Molloy and Speir, 1977) and a high ash content (Malone and Swartout, 1969; Spycher et al., 1983) as compared to crop residues, suggesting that the LF pool has undergone some decomposition and/or humification. Ladd et al. (1977) showed that fumigation of soils resulted in a significant decrease in the N content of the LF, indicating that the soil microbial biomass contributed significantly to this fraction.

The LF has a much higher turnover rate than bulk SOM (Skjemstad et al., 1986). The higher turnover rate of the LF was due to its high concentration of oligosaccharides, polysaccharides and hemicelluloses (Trumbore, 1993). Dalal and Mayer (1986) observed that loss of organic C from the LF following cultivation was 2 to 11 times faster than that from the HF in fine-textured soils, which was probably related to the labile nature of its constituents and to the lack of protection by soil colloids (Spycher et al., 1983).

The LF organic matter generally accounts for 0.1 to 4.0% of the total weight of cultivated soils, but it has up to 15 times more C and 10 times more N than the bulk soil (Gregorich et al., 1994). In a study of long-term crop rotations in three environments in Canada, Janzen et al. (1992) found that the LF of the surface soil (0- to 7.5-cm depth) accounted for 2.0 to 5.4%, 3.3 to 7.1%, and 7.1 to 17.5% of the soil organic C at Indian Head, Melfort, and Scott, SK, respectively. Bremer et al. (1994) reported that the LF organic matter accounted for 9 to 24% of the soil organic C and 2 to 17% of the soil organic N in the surface layer (0- to 7.5-cm depth) in a soil at Lethbridge, SK.

A large portion of the microbial population and enzyme activity in the soil is associated with the LF (Kanazawa and Filip, 1986), and soil respiration rates are correlated with the LF content (Janzen et al., 1992). In a study of three sites, i.e., a cornfield, a pine stand and a maple stand, Boone (1994) found that the LF represented 11% (corn), 13% (pine) and 2% (maple) of the N mineralization potential (by the anaerobic incubation method) for the whole mineral soil. These results suggest that the LF was not the primary N source in coarse-textured mineral soils because the LF represents only a small proportion of the SOM, even though the LF is relatively labile. Sollins et al. (1984) also observed that the net N mineralization during anaerobic incubation was greater from the HF than the LF in five of six soils tested. The results of these studies demonstrated that a considerable part of the N mineralized in soil must have originated from dying microbial biomass and a more stabilized organic matter fraction. The HF is not only a pool of older and recalcitrant organic matter, but it apparently includes a significant portion of non-protected and active organic matter that can be a major source of mineralizable N (Boone, 1994).

Labile SOM is more sensitive to changes in management or environmental conditions than total SOM (McGill et al., 1988; Bremer et al., 1994). Theoretically, the ratio of labile SOM to total SOM changes considerably after a shift in management or environmental conditions, and then gradually returns to that of the initial soil. Short-term changes in the labile SOM may be useful for predicting long-term changes in the SOM. Ford and Greenland (1968) found that the LF content of a soil under an extended rotation including pasture was higher than that under continuous wheat, which in turn, was higher than that under fallow-wheat. Janzen (1987) similarly observed that the LF

content was inversely proportional to the frequency of summer fallow in various spring wheat rotations. The LF content was more sensitive to cropping practices than total organic C or N concentration in the soil in these studies. Bremer et al. (1994) noticed that the LF organic matter was the most robust indicator of management-induced effects on SOM.

2.5 Soil Variability at the Landscape Scale

In order to understand the N-cycling processes in a natural landscape, it should be realised that soils are anisotropic natural bodies (Schlichting, 1982), i.e., their properties vary with direction in space. Soil anisotropy is the result of pedogenic horizonation, sedimentation, geologic structure and compaction (Hall and Olson, 1991). The variability of soil physical, chemical and biological properties is a phenomenon common to all soils in a non-level landscape (Pennock et al., 1987; Pennock et al., 1994; Androsoff et al., 1995; Stevenson et al., 1995).

Rowe (1984) suggested that a landscape included the interaction of climate, soil, vegetation, and landform. The major factors controlling soil variability at the landscape scale are surface topography, water redistribution and soil type (Parkin, 1993).

Topography plays a critical role in modifying both the microclimate and the hydrological conditions within a landscape (Rowe, 1984). Zaslavsky and Sinai (1981) found that the slope gradient and form controlled the water flow pathways. Heterogeneous distribution of water across a landscape may occur as a result of the differences in topography and soil across the landscape. Water is redistributed to convergent areas such as footslopes and lower complexes in a hummocky terrain

(Pennock et al., 1987), and the flow of water in response to the surficial substance and shape of landform is an important factor controlling landscape-scale variability (Zaslavsky and Sinia, 1981; Pennock et al., 1987).

Richardson et al. (1992) observed that the long-term direction of the water flow altered the type of soil. Pennock et al. (1987) similarly found that soil morphological observations associated with landform element could be explained by the patterns of water movement and distribution on hillslopes.

Because each landform element has its own distinctive hydrological and pedological regime (Pennock et al., 1987), soils in a given landscape position will exhibit similar morphological and chemical characteristics. As a result, many landscape properties and processes are predictable (Hall and Olson, 1991). For example, according to Pennock et al. (1994), water typically diverges from shoulder landform complexes, which limits downward water movement and results in the development of Regosols and Rego Chernozems in this position. In the water-receiving positions, such as the footslope element complexes, the greater soil water availability results in an increase in the rate of soil pedogenesis and soils with a deep solum, such as Chernozems, develop (Pennock et al., 1987; Pennock et al., 1994).

Higher soil moisture levels typically occur in footslope positions as compared to the shoulder positions (Pennock et al., 1987; Androsoff et al., 1995; Stevenson et al., 1995). Many soil properties, such as mineral N content (Androsoff et al., 1995; Stevenson et al., 1995), organic C content (Aguilar and Heil, 1988; Pennock et al., 1994) and total N content (Honeycutt et al., 1990), and biological processes, such as denitrification (Pennock et al., 1992) and soil N mineralization (Parkin, 1993) follow the same spatial

pattern as soil moisture content. Clearly, these soil properties and biological processes are controlled primarily by soil moisture status.

Hall and Olson (1991) categorised soil variability into two groups, i.e., systematic and random. As the sampling distance decreases, the randomness of the landscape decreases. The error in predicting the variability at the landscape scale can be understood and explained if enough samples are taken (Wilding and Drees, 1983). Furthermore, Parkin (1993) proposed that the spatial variability can occur at different scales, i.e., micro scale, plot scale, field or landscape scale, and regional scale. Each scale is associated with its own implications regarding the interpretation of study results.

2.6 Impact of Soil Variability on Nitrogen-Cycling Processes

2.6.1 Symbiotic dinitrogen fixation

Symbiotic N_2 fixation is a dynamic process which is controlled by soil factors, such as soil nutrients (Munns, 1977; Sprent and Minchin, 1983; Danso et al., 1993), moisture (Sprent, 1972), pH (Hera, 1978), salinity (Munns, 1977) and temperature (Roponen et al., 1970). These soil factors are controlled by topographic variation in non-level landscapes (e.g., Gregorich and Anderson, 1985; Trangmar et al., 1987; Honeycutt et al., 1990; Cahn et al., 1994). Consequently, symbiotic N_2 fixation likely will respond to the spatial variation of soil properties. For example, Mahler et al. (1979) found that the percentage of N derived from symbiotic N_2 fixation (%Ndfa) in dry pea was 22% in the ridgetops and 33% in the bottomlands, using the C_2H_2 reduction estimation approach. Androsoff et al. (1995) found that the %Ndfa in field pea was 69% on the divergent

footslope complexes and 28% on the convergent shoulder complexes, using an enriched ^{15}N dilution approach for estimating symbiotic N_2 fixation in a landscape-scale study in Saskatchewan, whereas the %Ndfa, estimated using a natural ^{15}N abundance approach, did not follow landform patterns. Stevenson et al. (1995) demonstrated that the *A* value approach and a natural ^{15}N abundance approach gave similar mean estimates of %Ndfa in pea at maturity, with values of approximately 72% and 84% for footslope complexes and shoulder complexes, respectively. The estimates of %Ndfa probably are dependent on site, environmental conditions, estimating approaches and test legume crops.

Spatial variability inherent in soils could be a major problem for the interpretation of results from field experiments involving N uptake and symbiotic N_2 fixation. In addition, variability of soil properties can occur at different scales (Parkin, 1993). For example, soil organic C was characterised by small-scale spatial variation nested within large-scale spatial variation in central Illinois (Cahn et al., 1994). Wendroth et al. (1992) found that the %Ndfa changed up to 25% across a 1.5-m distance. Androsoff et al. (1995) observed that the %Ndfa in pea ranged from 0 to 93% within a 2-ha area. Neither Androsoff et al. (1995) nor Stevenson et al. (1995) found a strong correlation between the symbiotic N_2 fixation (i.e., %Ndfa) estimating approaches that they used. They hypothesized that symbiotic N_2 fixation was partially controlled at the landscape scale, whereas strong micro-scale control may have existed that ultimately regulated symbiotic N_2 fixation. Direct evidence from field studies, however, is required to understand the micro-scale variability of soil variables and symbiotic N_2 fixation.

2.6.2 Other nitrogen-cycling processes

Information on the spatial variability and spatial patterns of N-cycling processes, such as residue N decomposition, the ANI, and the transfer of residue N into the labile SOM fractions, is limited. Stevenson and van Kessel (1997) found that the recovery of ^{15}N from labeled pea residue in the microbial biomass in spring was greater in the footslope complexes (71%) than in the shoulder complexes (51%), and the result was related to the greater soil water content in the footslope complexes. However, the N contribution by pea residue to the succeeding wheat crop, as estimated by the quantity of N that the wheat crop derived from the labeled pea residue, was 11% and similar among landform element complexes.

The ANI can be affected by quantity of N added, SOM, soil C:N ratio, and microbial biomass (Rao et al., 1991). Differences in soil properties, the size of the soil N pool and the size of the microbial N pool in different landscape positions might cause diverse degrees of pool substitution, MIT, denitrification and biological exchange reactions across a landscape. Observed differences in these processes in a non-level landscape suggest the possibility of landscape-scale controls on the ANI.

As a non-humified fraction of organic matter, the size of the LF pool is a balance between residue inputs and decomposition which are controlled mainly by the soil and environmental conditions. Crop residues are the primary source of organic material added to soil in many cropping systems and also are the major source of the LF (Boone, 1994). Residue input typically is higher in the footslope complexes than in the shoulder complexes as a consequence of higher crop residue yields in the footslope complexes (Androsoff et al., 1995; Stevenson and van Kessel 1997). Meanwhile, the relatively high moisture and mineral N levels in the footslope complexes could increase the

microbial activity, thus enhancing the mineralization and decomposition of crop residues (Parkin, 1993) and, thereby, increasing the input from crop residue to the LF pool. These processes suggest the possibility of higher LF content and more transfer of residue N into the LF pool in the footslope complexes as compared to the shoulder complexes.

2.6.3 Study approach

Due to the response of N-cycling processes to the spatial variability of soil properties, the landscape-scale approach for investigating the residue N dynamics and rotation benefits of legumes is of greater value than the traditional small-plot experimental approach. The small-plot approach is based on only a small portion of the field which typically is relatively level. The landscape-scale approach encompasses a larger field area and covers all of the landform element complexes. Thus, it can be used to evaluate and explain the spatial variability and landscape controls of the examined processes. Water redistribution in a hummocky terrain and its effects on soil properties, crop growth and N-cycling processes do not influence results when the experiment is conducted in a small-plot experimental approach. Small-plot studies can not be used to examine major processes affecting soil properties, crop production and N-cycling processes in the field. For example, Stevenson and van Kessel (1996a) observed that the rotation benefit of pea, based on the landscape-scale approach, was larger than when it was based on a small-plot approach in the same field. Consequently, they argued that the rotation benefit of pea and the diseases of wheat that occurred at the landscape-scale level might be confounded among the small plots in the small-plot approach.

3. MATERIALS AND METHODS

3.1 Experimental Approach

3.1.1 Study objectives

A landscape-scale rotation study was initiated in 1996 to examine the impact of including chickpea in a cereal rotation on a hummocky field near Biggar, SK. A chickpea-wheat rotation and a wheat-wheat rotation were established. Within each rotation, ^{15}N-labeled microplots were established at each grid cell and ^{15}N-labeled crop residue was collected in the first phase of the rotations. The ^{15}N-labeled chickpea residue and ^{15}N-labeled wheat residue were used in the second phase of the rotations to study the dynamics of residue N. The design of the study allowed the following experiments to be conducted simultaneously.

(i) Estimation of symbiotic N_2 fixation in chickpea. The specific objectives of this study were to investigate the landscape-scale and micro-scale variability of symbiotic N_2 fixation in chickpea with emphasis on the micro-scale variability, and to estimate the N contribution to the soil system by chickpea via symbiotic N_2 fixation;

(ii) Determination of the percentage and amount of N derived from the labeled residues in the subsequent crops and the influence of crop residue N on the availability of soil N. The specific objectives of this study were to estimate the amount of N in the chickpea residue and the wheat residue that was taken up by the succeeding crops, to measure the ANI magnitude of the chickpea residue and the wheat residue, and to investigate the landscape controls on residue N mineralization and the ANI;

(iii) Estimation of the variability of LF organic matter and the transfer of residue N into the SOM fractions. The specific objectives of this study were to investigate the temporal, horizontal and vertical variability of LF organic matter, to measure the transfer of chickpea residue N and wheat residue N into SOM fractions, and to estimate the availability of N in SOM fractions; and

(iv) Determination of the N effect and the non-N effect in the rotation benefit of chickpea. The specific objectives of this study were to assess the N effect and the non-N effect of chickpea in a chickpea-wheat rotation, and to investigate the landscape-scale controls on the N effect and the non-N effect of chickpea.

These studies will be described in detail in the following sections.

3.1.2 Site description

The study site was located in the Bear Hills near Biggar, Saskatchewan ($107^{0}59'W$, $52^{0}04'N$) in the Dark Brown soil zone. The site was characterized by a hummocky surface with slope gradients ranging from 10 to 15%, which is typical of landscapes in the Bear Hills.

The soils developed under grassland vegetation in a medium to moderately fine textured, moderately calcareous and silty glacio-lacustrine deposit, and belong to the Elstow Association (Acton and Ellis, 1978). Within this association, soil texture ranges from sandy loam to silty clay. Distinctive soil profiles were associated with specific positions in the landscape. Soil profiles on the shoulders were shallow and had very thin or no A horizons. The soils on the shoulders were classified as Orthic Regosols. Soil profiles on midslopes were deeper, better developed and had both Chernozemic A and

thick B horizons. The soils on the midslopes were classified as Orthic Dark Brown. Soil material likely had been transported from higher slope positions to lower slope positions in the field. The soil profiles on the lower slopes were deep and characterized by eluvial features. The soils on the lower slopes were classified as Eluviated Dark Brown.

The site was located on a commercial field belonging to Mr. John Bennett. The field has been farmed using a minimal disturbance direct seeding system since 1990. No food legumes had been grown previously in the study field. The cropping history of the site from 1992 to 1995 was barley-wheat-canola-wheat. In general, lack of precipitation and a somewhat limited moisture holding capacity are the principal factors affecting agricultural suitability in the area (Acton and Ellis, 1978).

3.1.3 Sampling design and field preparation

An area of the landscape was selected for study (Appendix A). A regularly-spaced sampling grid with grid points located at 14-m intervals was laid out on the landscape surface. The grid was laid out as six strips, each comprising 14 grid cells. The grid cells and the surrounding fringe area were surveyed using a Sokkisha SET 5 Total Station in April 1996. The survey data were used to derive a Digital Elevation Model with a cell resolution of 5 × 5 m (Fig. 3.1). The grid cells subsequently were classified into four landform element complexes: shoulders, backslopes, footslopes and levels (Pennock et al., 1987; Pennock et al., 1994). No backslopes were identified. Because only four grid cells were classified as levels, they were subjectively regrouped as either shoulders or footslopes. These quantitatively-defined grid cells were used as the sampling units to characterize the spatial variability at the landscape-scale level within the study field.

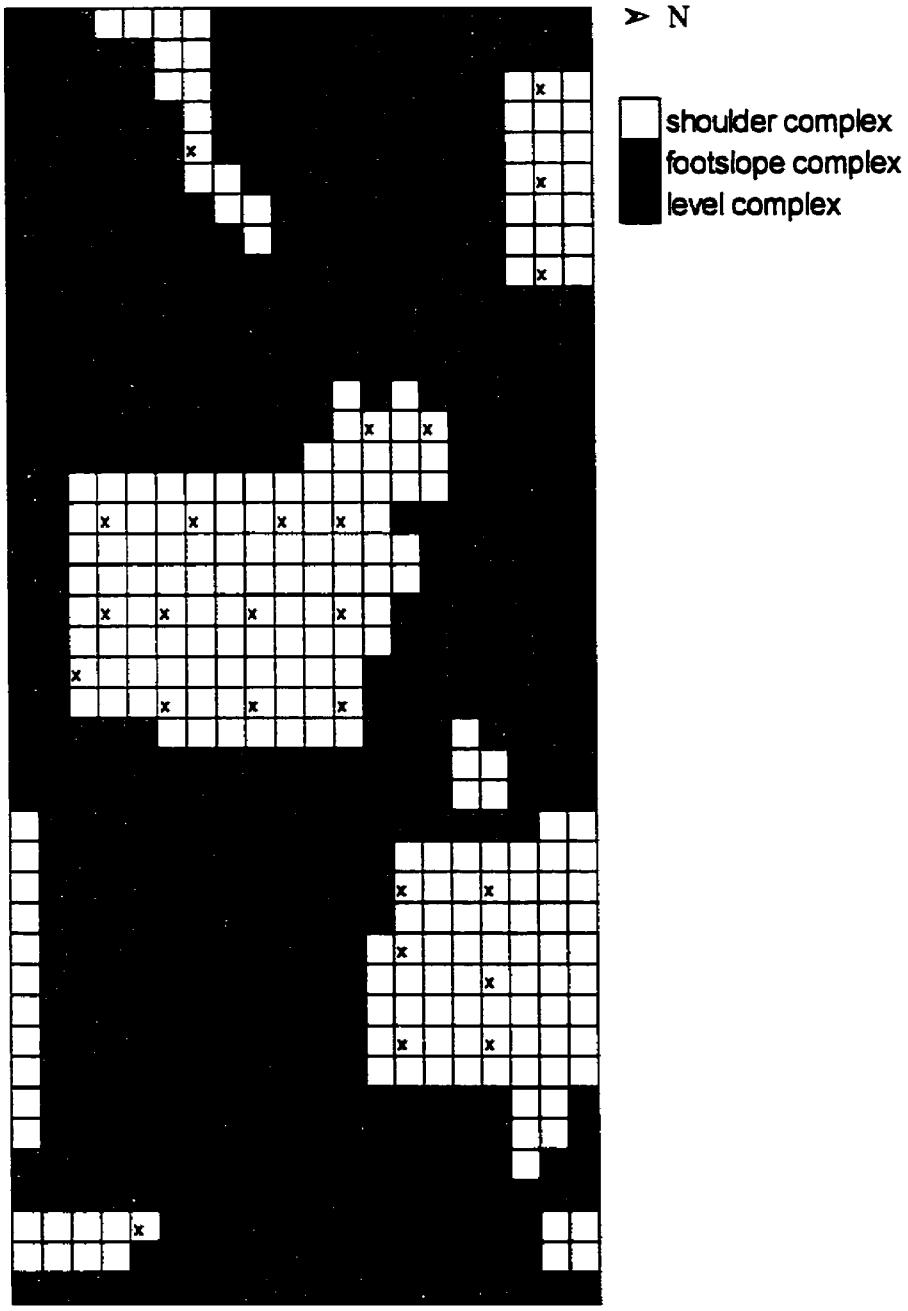

Figure 3.1. Landform element complexes of the study field in the Bear Hills near Biggar, SK. The x indicates the position of sampling grid cell.

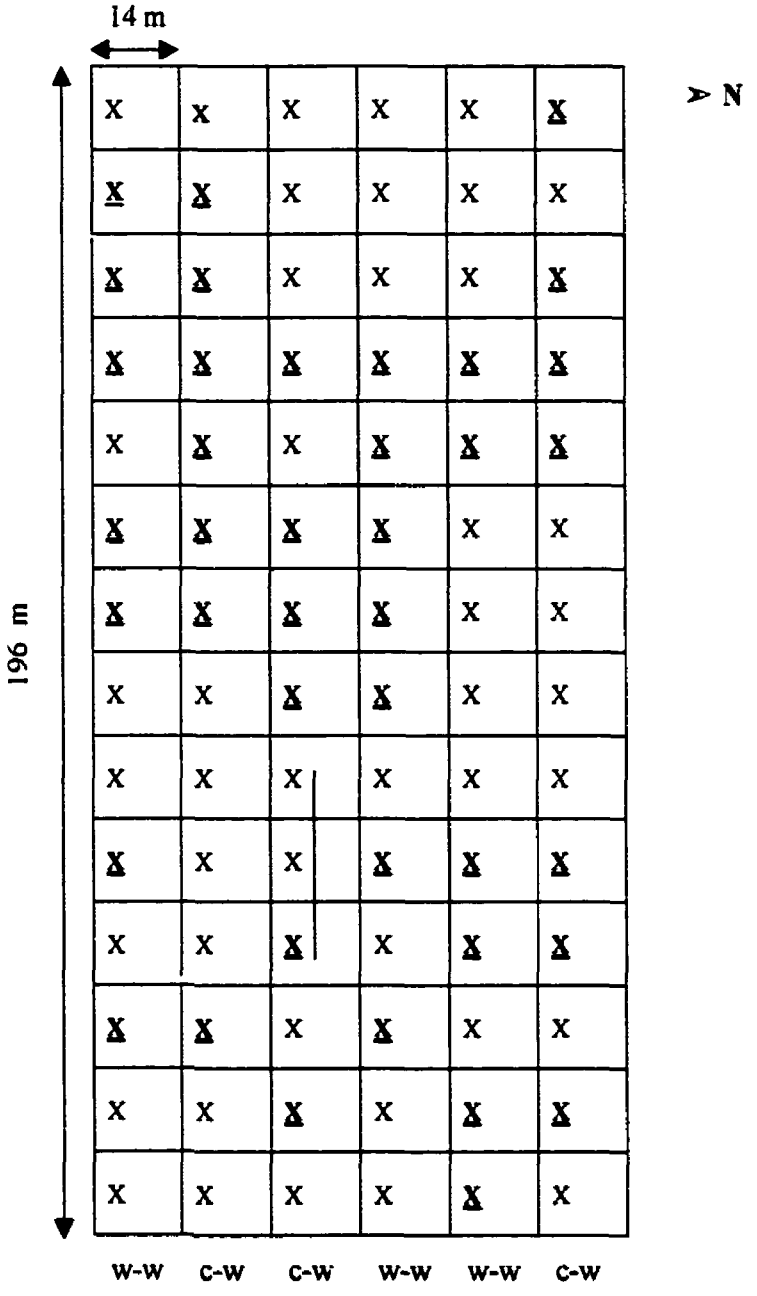

Figure 3.2. The layout of the chickpea-wheat and wheat-wheat rotations and the sampling design used at the experimental site in the Bear Hills near Biggar, SK (w = wheat, c = chickpea, X = the center of grid cell). The underlined X indicates the grid cell for root yield determinations and the light fraction study. The black line indicates the position of the transect used for estimating symbiotic N_2 fixation at the micro scale.

Seeding was accomplished with a 14-m-wide commercial air seeder. The seeding was conducted along the sampling strips in such a way that the grid points lay midway within each seeding pass, i.e., the study field was divided into six adjacent 14-m by 196-m strips (Fig. 3.2).

Two crop rotations, i.e., chickpea-wheat and wheat-wheat, were randomly arranged among the six strips (Fig. 3.2). The research area was sown to chickpea or wheat in 1996, to wheat in 1997, and then to canola in 1998.

In the first week of May 1996 (hereinafter referred to as the first phase of the rotation) prior to seeding, ethalfluralin [N-ethyl-N-(2-methyl-2-propenyl)-2,6-dinitro-4-(trifluoromethyl)benzenamine] and 2,4-D [(2,4-dichlorophenoxy) acetic acid] were applied in the chickpea and the wheat strips, respectively. On 26 May, kabuli chickpea (*Cicer arietinum* L. cv. Sanford), inoculated with a peat-based, self-stick inoculant containing *Rhizobium cicer* (MicroBio RhizoGen. Corp., Saskatoon, SK), was sown at a rate of 190 kg ha^{-1}. Chickpea strips were fertilized with 4.4 kg ha^{-1} N and 9.6 kg ha^{-1} P, applied as monoammonium phosphate. On the same day, wheat (*Triticum aestivum* L. cv. Katepwa) was sown at a rate of 90 kg ha^{-1}. Wheat strips were fertilized with 44 kg ha^{-1} N and 9 kg ha^{-1} P, applied as a fertilizer blend (37-17-0).

In the second week of May 1997 (hereinafter referred to as the second phase of the rotation), bromoxynil (3,5-dibromo-4-hydroxybenzonitrile), MCPA [(4-chloro-2-methylphenoxy)acetic acid], and fenoxaprop {(±)-2-[4-[(6-chloro-2-benzoxazolyl)oxy]phenoxy]propanoic acid} were applied across the field. Wheat (*Triticum aestivum* L. cv. Katepwa) was sown at a rate of 80 kg ha^{-1} across the entire field on 30 May 1997 and 9 kg ha^{-1} P was applied as triple superphosphate.

The research area was sown to canola (*Brassica napus* L. cv. Excel) at a rate of 6.5 kg ha^{-1} on 5 June 1998 (hereinafter referred to as the third phase of rotation), and 39 kg ha^{-1} N, 5 kg ha^{-1} P, 9 kg ha^{-1} K and 11 kg ha^{-1} S were applied as a fertilizer blend (35-10-10-10). On 5 July 1998, glufosinate [2-amino-4-(hydroxymethylphosphinyl)butanoic acid] was sprayed on the field for weed control.

3.1.4 Soil characterization

On 2 May 1996, soil samples were collected to a depth of 60 cm, using a Dutch auger at the center of each grid cell, and separated into 0- to 15-, 15- to 30- and 30- to 60-cm increments. Twenty grams of field moist soil for each depth was added to 200 mL of 2 *M* KCl solution and shaken for 1 h. The solution was filtered through a Whatman No. 40 filter paper, and mineral N in the extract was determined, using a Technicon AutoAnalyzer II System (Labtronics Inc., Tarrytown, NY).

Gravimetric soil moisture content was determined (105^0C, 24 h) for each depth, using approximately 30 g of field soil. Soil samples remaining after the determination of mineral N and moisture content were air dried and ground (< 2 mm). Percentage C and N in the air-dried soil were determined for each depth, using a LECO-CNS-2000 analyzer (LECO Corp., St. Joseph, Michigan). Soil pH and electrical conductivity (EC) (0- to 15-cm depth) were measured in a 1: 1 soil-water paste, using a PHM 82 Standard pH meter (Radiometer Copenhagen, Denmark) and an ES-12E conductivity meter (Horiba Ltd., Japan), respectively. Potentially mineralizable N in the soil (0- to 15-cm depth) was estimated, using the hot-KCl extraction method as described by Jalil et al. (1996). The

details of the procedure are outlined in Section 3.4.4. Soil mineral N, total C content and total N content are reported for the combined upper 60-cm depth.

3.2 Estimation of Symbiotic Dinitrogen Fixation in Chickpea

3.2.1 Symbiotic dinitrogen fixation at the landscape scale

Fourteen grid cells were established within each of the three chickpea strips (Fig. 3.2) for the estimation of symbiotic N_2 fixation, using a natural ^{15}N abundance approach (Shearer and Kohl, 1986). This approach provides a time-integrated estimation of symbiotic N_2 fixation without disturbing the soil system because ^{15}N-labeled fertilizers are not required (Rennie and Rennie, 1983).

Canola was used as the reference crop. Seven days after sowing chickpea, one 1-m^2 microplot was established at a distance of 0.5 m from the center of each grid cell. Canola (*Brassica napus* cv. Bounty), treated with Vitavax (carbathiin 20%, thiram 28.9%, lindane 18.7%), was hand-seeded into the microplots at a rate of 10 kg ha^{-1}. Chickpea seedlings, that emerged within the canola microplots, were removed by hand.

At physiological maturity, the aboveground portion of the canola plants within the canola microplots and chickpea plants around the center of the grid cell were sampled. The sampling distance between the chickpea plants and the canola plants was 0.5 to 1 m. Plant samples were dried at 40^{0}C in a forced-air oven to a constant weight, separated by hand into residue and grain, and then ground in a cyclone mill (0.4-mm screen). A portion of the sample was ground further in a rotating ball-bearing mill. Percentage N and atom % ^{15}N in a 2.00±0.20 mg ground subsample were determined using a 20-20 Mass Spectrometer interfaced with an ANCA-GSL sample converter (Europa Scientific, Crewe, UK).

Unlabeled pea grain with an atom % ^{15}N of 0.3675 and standard deviation of 0.0001 was included as a working standard.

A representative 1-m^2 microplot of chickpea was harvested near the center of each grid cell for yield determination. Samples were dried at 40^{0}C in a forced-air oven to a constant weight, then threshed and separated into residue and grain for yield determination.

3.2.2 Symbiotic dinitrogen fixation at the micro scale

An experiment to examine the variability of symbiotic N_2 fixation at the micro scale was conducted in the first phase of the rotation. One week after sowing chickpea, a 33-m transect was identified in one of the three chickpea strips (Appendix A and Fig. 3.2). The transect consisted of an existing chickpea row and a row of wheat established adjacent to this chickpea row. The wheat was hand seeded as a reference crop for estimating symbiotic N_2 fixation, using the natural ^{15}N abundance approach (Shearer and Kohl, 1986). Wheat (*Triticum aestivum* L. cv. Katepwa) was seeded one week after sowing chickpea at a distance of 0.15 m parallel to the chickpea row. It was necessary to grow the reference plant in close proximity to the N_2-fixing legume and to sample the reference and legume crop in pairs in order to assess the spatial variability of symbiotic N_2 fixation (Shearer and Kohl, 1986; Sutherland et al., 1991).

The average distance between chickpea plants was 0.3 m; therefore, 0.3 m was the smallest possible sampling scale that could be used. Prior to seeding wheat, soil samples (0- to 15-cm depth) were collected at an interval of 0.3 m along the micro-scale transect

between the chickpea plant transect and wheat plant transect. Mineral N, moisture, total C, total N, pH and EC were determined using the procedures described in Section 3.1.4.

At harvest, the aboveground portion of the chickpea plants and the wheat plants was sampled in pairs at an interval of 0.3 m along the micro-scale transect. A total of 110 samples were used for each soil measurement and plant measurement. Webster (1985) argued that for a single transect, it is advisable to have at least 100 sampling points for the analysis of semivariance.

The plant samples were dried, ground and analyzed, using the procedures described in Section 3.2.1.

3.2.3 Calculations

The proportion of N derived from the atmosphere via symbiotic N_2 fixation (%Ndfa) in chickpea was calculated according to Shearer and Kohl (1986):

$$\%Ndfa = \left(\frac{\delta^{15}N_{reference} - \delta^{15}N_{chickpea}}{\delta^{15}N_{reference} - c}\right) \times 100 \qquad (3.1)$$

where $\delta^{15}N$ is:

$$\delta^{15}N = \left(\frac{\text{atom \% } {}^{15}N_{sample} - \text{atom \% } {}^{15}N_{atmosphere}}{\text{atom \% } {}^{15}N_{atmosphere}}\right) \times 1000 \qquad (3.2)$$

and c is the $\delta^{15}N$ value of chickpea straw grown in N-free medium (Shearer and Kohl, 1986). The c value was determined by growing chickpea plants in Leonard jars (Vincent, 1970) in a growth chamber under the following conditions: 50% relative humidity, 20^{0}C during the day and 18^{0}C during the night. The photoperiod was a 16-h day and an 8-h

night. The c value was −1.71±0.25‰. Atom % ^{15}N of atmosphere was 0.3663% (Mariotti, 1983).

The quantity of N derived from symbiotic N_2 fixation, expressed as kg ha^{-1} N, was calculated as follows:

$$\text{kg ha}^{-1}\text{ Ndfa} = \%\text{Ndfa} \times \text{N}_{\text{plant}} \tag{3.3}$$

where N_{plant} is the total N accumulated in the plant expressed as kg ha^{-1} N.

Subtraction of the amount of harvested grain N from the amount of symbiotically-fixed N in chickpea was used to estimate the potential N contribution of chickpea to the soil N pool via symbiotic N_2 fixation (Doughton et al., 1993):

$$\begin{aligned}\text{N contribution} &= (\text{kg ha}^{-1}\text{ Ndfa}_{\text{residue}} + \text{kg ha}^{-1}\text{ Ndfa}_{\text{grain}}) - \\ &\quad (\text{kg ha}^{-1}\text{Ndfa}_{\text{grain}} + \text{kg ha}^{-1}\text{Ndfs}_{\text{grain}}) \\ &= \text{kg ha}^{-1}\text{ Ndfa}_{\text{residue}} - \text{kg ha}^{-1}\text{ Ndfs}_{\text{grain}}\end{aligned} \tag{3.4}$$

where kg ha^{-1} $Ndfa_{residue}$ is the quantity of N from symbiotic N_2 fixation in chickpea residue; kg ha^{-1} $Ndfa_{grain}$ is the quantity of N from symbiotic N_2 fixation in chickpea grain; kg ha^{-1} $Ndfs_{grain}$ is the quantity of N derived from soil in the chickpea grain and was calculated as follows:

$$\text{kg ha}^{-1}\text{ Ndfs}_{\text{grain}} = \text{kg ha}^{-1}\text{ N}_{\text{grain}} - \text{kg ha}^{-1}\text{ Ndfa}_{\text{grain}} \tag{3.5}$$

where kg ha^{-1} N_{grain} is the quantity of N accumulated in the chickpea grain.

3.3 Availability of Residue Nitrogen and Its Influence on the Availability of Soil Nitrogen

3.3.1 Residue labeling and treatment design

Selected chickpea plants and wheat plants were labeled with ^{15}N-enriched fertilizer in 1996 in order to supply ^{15}N-labeled residues for the 1997 growing season. After sowing chickpea and wheat in 1996, one unconfined 1-m^2 microplot was established near the center of each grid cell. A solution of $^{15}NH_4{}^{15}NO_3$, labeled with 10 atom % ^{15}N, was applied on the soil surface of the microplot. The solution was applied on two separate dates (i.e., 24 June and 9 July) at a rate of 10 kg ha^{-1} N, thereby ultimately supplying N at rate of 20 kg ha^{-1} to each microplot.

The aboveground portion of the plants in each ^{15}N-labeled microplot was harvested at maturity (23 September 1996). Each sample was dried in a forced-air oven (40^{0}C) to a constant weight and separated into residue and grain, by hand, in order to avoid cross contamination. The ^{15}N-labeled chickpea residue yield and the ^{15}N-labeled wheat residue yields were then determined. The residue portion subsequently was coarsely ground in a Wiley mill without a screen.

On 23 October 1996, a second 1-m^2 microplot was established in each grid cell and the residue within the microplot after harvest was removed by hand (Fig. 3.3). Each coarsely ground labeled residue sample was returned to the prepared microplot. The residue was returned to its original sampling grid cell and spread evenly on the soil surface to simulate the minimal disturbance direct seeding system practiced in the study field. The ^{15}N-labeled residue microplots were used to follow the fate of ^{15}N from labeled residues in the microbial biomass and the succeeding wheat crop in 1997, and to calculate the ANI.

On the same day that labeled residues were applied, one 1.5-m by 1.5-m microplot was established as a control microplot (i.e., without residue) in each grid cell (Fig. 3.3).

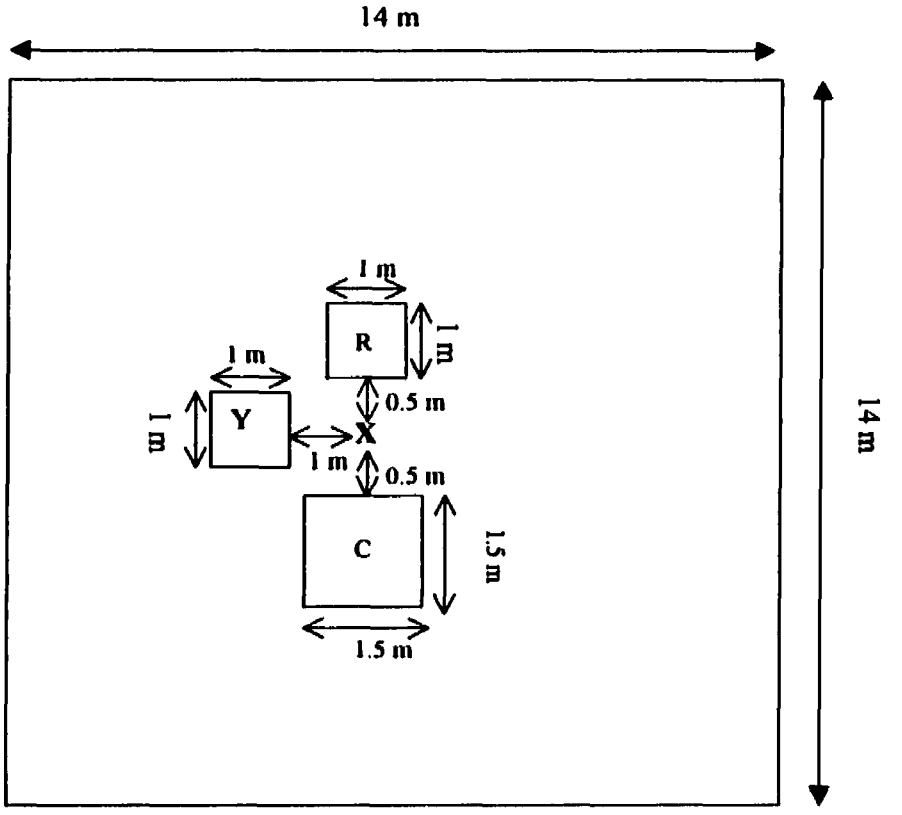

Figure 3.3. The layout of microplots for the ANI study at each grid cell in 1997 (R = labeled chickpea or wheat residue microplot, C = control microplot (i.e., no residue), Y = unlabeled microplot used for yield determination, X = the center of grid cell).

Crop residues left within the microplot after harvest in 1996 were removed by hand from the control microplot. The control microplots were used to calculate ANI.

A small subsample of coarsely ground ^{15}N-labeled chickpea or ^{15}N-labeled wheat residue was finely ground in a rotating ball-bearing mill and analyzed for percentage C and percentage N, and atom % ^{15}N in a 2.00±0.20 mg subsample, using a Tracer Mass Spectrometer interfaced with a RoboPrep sample converter (Europa Scientific, Crewe, UK). The working standard was ^{15}N-enriched pea residue with an atom % ^{15}N of 0.6013 and standard deviation of 0.0007.

3.3.2 Microbial biomass ^{15}N analysis

On 4 April 1997 (prior to sowing) and 22 September 1997 (after harvest), soil samples (0- to 15-cm depth) were taken from within each labeled chickpea and each labeled wheat residue microplot using a Dutch auger. The chloroform-fumigation extraction method was used to assess soil microbial N content (Voroney et al., 1993). For each soil sample, 50 g of field soil was extracted with 100 mL of 0.5 *M* K_2SO_4 solution. A second 50-g sample of field soil was transferred into a 100-mL beaker and then fumigated in a desiccator, using ethanol-free chloroform (VWR Company, Ontario) under vacuum at room temperature for 24 h. The chloroform vapor that had diffused into the soil was removed by repeated evacuation. Soil samples subsequently were extracted with 100 mL of 0.5 *M* K_2SO_4 solution. Gravimetric soil moisture content was determined on a 25-g field subsample (105^0C, 24 h).

Organic N in the K_2SO_4 extract was converted to NH_4^+ by Kjeldahl digestion, followed by steam distillation (Voroney et al., 1993). Mineral N in the distillate was

determined, using a Technicon AutoAnalyzer II system (Labtronics Inc., Tarrytown, NY). Microbial biomass N content was calculated, according to Voroney et al. (1993):

$$\text{Microbial biomass N} = \Delta N / k_{EN} \quad (3.6)$$

where ΔN is the increase in extractable N after fumigation, and k_{EN} is the efficiency of N extraction from the microbial biomass. A k_{EN} of 0.54 was assumed (Joergensen and Mueller, 1996).

Fumigated and unfumigated extracts were dried at 60°C and dissolved in deionized H_2O to obtain an aliquot containing approximately 30 μg of N. The aliquot was then transferred to an 8 × 5 mm tin capsule (Europa Scientific Inc., Ohio) and dried at 60°C prior to ^{15}N analysis. Samples were analyzed, using a Tracer Mass Spectrometer interfaced with a RoboPrep sample converter (Europa Scientific, Crewe, UK). The working standard was unlabeled $(NH_4)_2SO_4$ with an atom % ^{15}N value of 0.3668 and standard deviation of 0.0029.

3.3.3 Plant ^{15}N analysis and crop yield determination

On 26 August 1997, the aboveground portion of the wheat plant was sampled from within the center of the labeled residue microplot and control microplot in each grid cell. Samples were dried in a forced-air oven (40°C) to a constant weight and subsequently separated into straw and grain, by hand, in order to avoid cross contamination. Both straw and grain samples subsequently were ground in a cyclone mill (0.4-mm screen). A subsample of each was ground further in a rotating ball-bearing mill. Percentage N and atom % ^{15}N in a 2.00±0.20 mg ground subsample from each labeled residue microplot were determined, using a Tracer Mass Spectrometer

interfaced with a RoboPrep sample converter (Europa Scientific, Crewe, UK). The working standard was ^{15}N-enriched pea residue with an atom % ^{15}N of 0.6013 and standard deviation of 0.0007. Percentage N and atom % ^{15}N in a 2.00±0.20 mg ground subsample from the control microplots were determined, using a 20-20 Mass Spectrometer interfaced with an ANCA-GSL sample converter (Europa Scientific, Crewe, UK). The working standard was unlabeled pea grain with an atom % ^{15}N of 0.3675 and standard deviation of 0.0001.

At each grid cell, yields, representative of the ^{15}N-labeled residue microplots, were estimated by harvesting an unlabeled companion 1-m^2 microplot, located adjacent to the ^{15}N-labeled microplot (Fig. 3.3). The yield for the control treatment was determined by sampling a 1-m^2 area within the control microplot. All samples were dried, threshed and weighed to determine the yield of straw and grain.

On 12 August 1998 (i.e., the third phase of the rotation), the canola was sampled from within each labeled chickpea residue or wheat residue microplot. Unfortunately, the producer had cut the canola crop prior to sampling due to a perceived crop failure. Thus, only the standing stubble was sampled and determination of total canola yield was not possible. However, the %Ndfr of the stubble could still be determined. Stubble samples were dried, ground and analyzed for percentage N and atom % ^{15}N, as described earlier.

3.3.4 Calculations

The percentage of N derived from ^{15}N-labeled residue (%Ndfr) in the plant tissue or the microbial biomass was determined according to Hauck and Bremner (1976):

$$\%Ndfr = \left(\frac{\text{atom \% } ^{15}N \text{ excess}_{\text{sample}}}{\text{atom \% } ^{15}N \text{ excess}_{\text{labeled residue}}} \right) \times 100 \quad (3.7)$$

where atom % ^{15}N excess $_{sample}$ denotes the atom % ^{15}N excess in straw, grain or microbial biomass samples. The atom % ^{15}N excess of these samples was determined by subtracting the atom % ^{15}N in natural abundance samples collected from unlabeled residue microplots. The %Ndfr was then used to calculate the recovery of ^{15}N of labeled residues (Stevenson and van Kessel, 1997):

$$\text{Residue } ^{15}N \text{ recovery (\%)} = \left(\frac{\text{kg ha}^{-1} \text{ Ndfr}}{\text{kg ha}^{-1} \text{ N}_{\text{labeled residue}}} \right) \times 100 \quad (3.8)$$

where kg ha^{-1} N $_{labeled\ residue}$ is N content of the labeled residue, expressed as kg ha^{-1} N, and kg ha^{-1} Ndfr is the quantity of N derived from labeled residue in samples, expressed as kg ha^{-1} N. These values were calculated as follows:

$$\text{kg ha}^{-1} \text{ Ndfr} = \%Ndfr \times N_{uptake} \text{ (kg ha}^{-1}) \quad (3.9)$$

where N_{uptake} is the quantity of N accumulated in the straw, grain or microbial biomass from within the labeled residue microplots, expressed as kg ha^{-1} N.

Nitrogen derived from the soil (Ndfs) in wheat plants grown within the labeled residue microplots, expressed as kg ha^{-1} N, in the second phase of the rotation was calculated as follows:

$$\text{Ndfs (kg ha}^{-1} \text{ N)} = (1 - \%Ndfr) \times N_{uptake} \text{ (kg ha}^{-1}) \quad (3.10)$$

The Ndfs in wheat plants grown in the no-residue microplot was calculated as follows:

$$\text{Ndfs (kg ha}^{-1} \text{ N)} = N_{uptake} \text{ (kg ha}^{-1}) \quad (3.11)$$

The ANI is quantitatively measured as the difference in the uptake of soil-derived N between a labeled and a control treatment (Jenkinson et al., 1985; Azam et al., 1993). Thus, ANI was calculated as follows:

$$ANI = Ndfs_{\text{labeled residue treatment}} - Ndfs_{\text{no residue treatment}} \quad (3.12)$$

where $Ndfs_{\text{labeled residue treatment}}$ is the quantity of N derived from the soil in the plants grown within the labeled residue microplot expressed as kg ha^{-1} N, and $Ndfs_{\text{no residue treatment}}$ is the quantity of N derived from the soil in the plant grown in the control microplot, expressed as kg ha^{-1} N.

3.4 Variability of the Light Fraction Organic Matter and Transfer of Residue Nitrogen into Soil Organic Matter Fractions

3.4.1 Soil sampling

Ten shoulder element complexes (hereinafter referred to as shoulders) and ten footslope element complexes (hereinafter referred to as footslopes) were randomly selected as sampling grid cells from within both the chickpea-wheat and the wheat-wheat rotations (Fig. 3.2). On 23 October 1996 (after harvest and prior to the application of labeled residues), 4 April 1997 (before sowing), 29 June 1997, and 22 September 1997 (after harvest), five soil cores (0- to 5-cm depth) were collected from within each labeled chickpea residue or labeled wheat residue microplot and combined as one sample for each grid cell. To facilitate the investigation of the vertical distribution of the LF content, soil cores also were collected to the depths of 5- to 15-, 15- to 30-, and 30- to 60-cm on 22 September 1997 at the 20 gird cells within the

chickpea-wheat rotation (i.e., 10 shoulders and 10 footslopes). Soil samples were air dried, ground (< 2 mm), and stored in vials at room temperature for further analysis.

3.4.2 Determination of the light fraction content

Separation of the LF from bulk soil was conducted according to the procedures described by Janzen et al. (1992) and Gregorich and Ellert (1993). Approximately 25 g of an air-dried soil sample was weighed into a 120-mL sample container. After adding a 50-mL aliquot of NaI solution (1.70 g cm^{-3}), the suspension was dispersed by shaking for 1 h. Approximately 10-mL NaI solution was used to wash down the sides of each sample container. The containers were covered and the suspension was allowed to stand undisturbed for 48 h at room temperature. The suspended material (i.e., LF) was then removed under suction and transferred directly to a Millipore filtration unit (Millipore Corp., Bedford, MA) equipped with MSI Nylon magna filter paper (47 mm diameter). The material remaining in the sample container was kept for further analysis. The LF was then washed under suction, with three successive aliquots (approximately 100 mL) of 0.01 *M* $CaCl_2$ solution and three successive aliquots (approximately 100 mL) of deionized water. The LF attached to the filter funnel was scraped and collected using a rubber policeman. The filter paper and the LF on the filter paper were then removed from the filter holder. The filter paper and LF were placed in an aluminum dish and put in an oven to dry at 60^{0}C for 24 h. The LF was subsequently scraped from the filter paper, weighed and stored in vials at room temperature for further analysis.

The retained samples were resuspended by adding approximately 20 mL of NaI solution and the above procedures were repeated. The first and second LF extractions were combined as the total LF for each soil sample.

The NaI solution was recycled during use. All the soil samples from the same sampling date were analyzed as one set of soil samples. Before the separation for each new set of samples, the density of NaI solution was readjusted to 1.70 g cm^{-3}.

After the second separation, NaI in the residual material was removed by washing the material 3 to 6 times with deionized water followed by centrifugation at 1200 g for 10 min until no visual evidence of NaI was detected. The remaining material, designated the HF, was then dried in the oven at 60^0C for 24 h and stored in vials at room temperature for further analysis.

3.4.3 Light fraction and heavy fraction ^{15}N analysis

In order to avoid cross contamination of ^{15}N in the LF samples and HF samples, the NaI solution was not recycled during the separation of LF for ^{15}N analysis. In all other aspects, the procedures for the separation of LF from ^{15}N-labeled samples were the same as described in Section 3.4.2.

A portion of the dried LF sample was finely ground using an amalgamator (Model 3110-3A, Crescent Dental Mfg. Co., USA) due to the small size of the LF sample. A portion of the dried HF sample was finely pulverized, using a rotating ball-bearing mill. Percentage C and percentage N, and atom % ^{15}N in a 2.00±0.20 mg subsample of the LF and a 10.00±2.00 mg subsample of the HF were determined using a Tracer Mass Spectrometer interfaced with a RoboPrep sample converter (Europa Scientific, Crewe,

UK). The working standard for the LF samples was unlabeled pea grain with an atom % ^{15}N of 0.3675 and a standard deviation of 0.0001. The working standard for HF samples was an unlabeled soil sample with an atom % ^{15}N of 0.3690 and a standard deviation of 0.0021.

3.4.4 Assessment of potentially mineralizable nitrogen in the soil organic matter fractions

Potentially mineralizable N in the LF and in the HF was estimated using the hot-KCl extraction method as described by Jalil et al. (1996). Each unground LF (0.10 g) or HF (3.0 g) subsample was suspended in 20 mL of 2 *M* KCl solution in 250-mL digestion tubes. The tubes were heated at 100^0C for 4 h in a digestion block. The tubes were covered with rubber stoppers during heating in order to minimize moisture loss. The tubes were then removed from the digester and cooled at room temperature. The NH_4^+ present in the extracts was analyzed using a Technicon Auto-Analyzer II system (Labtronics Inc., Tarrytown, NY).

In addition to the hot-KCl extraction, a cold-KCl extraction also was performed. The LF (0.10 g) and KCl (20 mL) mixture, or HF (3.0 g) and KCl (20 mL) mixture was allowed to sit at room temperature for 4 h without heating. The NH_4^+ in the extracts was determined using the same procedure as used for the hot-KCl extraction. It was found that NH_4^+ in the cold-KCl extracts was negligible. Thus, only the quantity of NH_4^+ extracted by the hot KCl will be reported.

A 3.0-g bulk soil sample (i.e., without LF separation) also was extracted with 2 *M* KCl using the same procedures as described for the LF samples and the HF samples.

3.4.5 Calculations

The soil LF content is reported as a percentage of soil weight (105^0C).

The quantity of N in the LF as a percentage of the quantity of N in the bulk soil was calculated as follows:

$$\text{LF N as a percentage of soil N} = \left[\frac{\text{LF content (\%)} \times \text{N\%(LF)}}{\text{N\%(soil)}}\right] \times 100 \qquad (3.13)$$

where N%(LF) is the N content of the LF expressed as a percentage, and N%(soil) is the N content of the soil expressed as a percentage.

The quantity of C in the LF as a percentage of the quantity of C in the soil was similarly calculated as follows:

$$\text{LF C as a percentage of soil C} = \left[\frac{\text{LF content (\%)} \times \text{C\%(LF)}}{\text{C\%(soil)}}\right] \times 100 \qquad (3.14)$$

where C%(LF) is the C content of the LF expressed as a percentage, C%(soil) is the C content of the soil expressed as a percentage.

The quantity of hot-KCl extractable NH_4^+ in the LF (LF NH_4^+) as a percentage of the quantity of hot-KCl extractable NH_4^+ in the soil (soil NH_4^+) was calculated as follows:

$$\text{LF } NH_4^+ \text{ as a percentage of soil } NH_4^+ = \left[\frac{\text{LF } NH_4^+ \text{ (mg kg}^{-1}) \times \text{LF content (\%)}}{\text{soil } NH_4^+ \text{ (mg kg}^{-1})}\right] \times 100 \qquad (3.15)$$

The atom % ^{15}N excess in the LF or HF was used to calculate the percentage of N derived from labeled residue (%Ndfr) in the LF or HF, according to Hauck and Bremner (1976):

$$\%\text{Ndfr}_{\text{LF or HF}} = \left(\frac{\text{atom \%}^{15}\text{N excess}_{\text{LF or HF}}}{\text{atom \%}^{15}\text{N excess}_{\text{labeled residue}}}\right) \times 100 \qquad (3.16)$$

A representative background atom % ^{15}N of the LF or the HF samples from unlabeled soil within the study field was subtracted from each measurement to calculate atom % ^{15}N excess. The %Ndfr was then used to calculate the recovery of ^{15}N from labeled residues in the LF or HF:

$$\text{Residue } ^{15}\text{N recovery}_{\text{LF or HF}}(\%) = \left(\frac{\text{kg ha}^{-1}\ \text{Ndfr}_{\text{LF or HF}}}{\text{kg ha}^{-1}\ \text{N}_{\text{labeled residue}}}\right) \times 100 \qquad (3.17)$$

where kg ha^{-1} $Ndfr_{LF\ or\ HF}$ is the quantity of N derived from the labeled residue in the LF or HF, expressed as kg ha^{-1} N, and was calculated as follows:

$$\text{kg ha}^{-1}\ \text{Ndfr}_{\text{LF or HF}} = \%\ \text{Ndfr}_{\text{LF or HF}} \times \text{N}_{\text{LF or HF}}\ (\text{kg ha}^{-1}) \qquad (3.18)$$

where $N_{LF\ or\ HF}$ is the quantity of total N accumulated in the LF or HF, expressed as kg ha^{-1} N.

3.5 Nitrogen Effect and Non-Nitrogen Effect of Chickpea

3.5.1 Soil sampling and analysis

On 4 April 1997 (i.e., the second phase of the rotation), soil samples were collected with 0- to 15-, 15- to 30- and 30- to 60-cm increments using a Dutch auger before sowing at each grid cell. Mineral N and gravimetric moisture content were determined, using the procedures described in Section 3.1.4.

3.5.2 Determination of *A* value

After sowing wheat in 1997, one unconfined 1-m^2 microplot was established at each grid cell. A solution of $^{15}NH_4{}^{15}NO_3$, labeled with 10 atom % ^{15}N, was applied to the microplots on the soil surface at a rate of 5 kg ha^{-1} N, on 10 June 1997. The

aboveground portion of plant subsamples was taken from each microplot at maturity (26 August). The samples were dried in a forced-air oven (40^0C) to a constant weight and separated into residue and grain, by hand, in order to avoid cross contamination. The plant samples were then coarsely ground in a cyclone mill (0.4-mm screen). A small portion of the coarsely ground samples was further ground in a rotating ball-bearing mill. Finely ground subsamples (2.00±0.20 mg) were analyzed for percentage N and atom % ^{15}N, using a Tracer Mass Spectrometer interfaced with a RoboPrep sample converter (Europa Scientific, Crewe, UK). The working standard was ^{15}N-enriched pea residue with an atom % ^{15}N of 0.6013 and standard deviation of 0.0007.

The atom % ^{15}N of wheat residue was used to calculate the *A* value according to Fried and Broeshart (1975):

$$A \text{ value} = \left(\frac{100 - \% \text{ Ndff}}{\% \text{ Ndff}} \right) \times Q \qquad (3.19)$$

where Q is the quantity of N (kg ha^{-1} N) in the applied ^{15}N-enriched fertilizer and %Ndff is the percentage of plant N derived from ^{15}N-enriched fertilizer and calculated as follows according to Hauck and Bremner (1976):

$$\% \text{ Ndff} = \left(\frac{\text{atom \% } ^{15}\text{N excess}_{\text{residue}}}{\text{atom \% } ^{15}\text{N excess}_{\text{fertilizer}}} \right) \times 100 \qquad (3.20)$$

A representative background atom % ^{15}N of unlabeled wheat residue was subtracted from each atom % ^{15}N of labeled residue to calculate atom % ^{15}N excess of residues. Natural ^{15}N abundance of the atmosphere, i.e., 0.3663%, was subtracted from atom % ^{15}N of labeled fertilizer to calculate the atom % ^{15}N excess of the labeled fertilizer.

3.5.3 Wheat diseases

On 29 July 1997 (i.e., in the second phase of the rotation), common root rot (*Cochliobolus sativus* Ito & Kurib.) lesions on the subcrown internode of the wheat plants were rated using a 0 to 4 scale (i.e., 0 = 0%, 1= 1 to 25%, 2 = 26 to 50%, 3 = 51 to 75%, and 4 = 76 to 100%) (Stevenson and van Kessel, 1996a) in each grid cell for both rotations. Approximately 20 plants were randomly excavated around the center of the grid cell. A mean of the ratings from the 20 plants was used to obtain a single rating for each grid cell. Leaf disease (tan spot: *Pyrenophora tritici-repentis* [Died.] Drechs., and septoria leaf blotch: *Septoria avenae* Frank f. sp. *triticea*, Johns., *Septoria tritici* Rob. in Desm., and *Septoria nodorum* [Berk.] Berk.) lesions were assessed at each grid cell. The lesions on the flag and upper leaves were rated using the 0 to 11 scale developed by McFadden (1991). The higher rating indicates higher disease severity.

3.5.4 Root sampling and nitrogen analysis

On 23 September 1996 (after harvest), 10 shoulders and 10 footslopes were randomly selected for root sampling from within each rotation (Fig. 3.2). A soil core with a 10-cm diameter was taken, using a punch truck, from within the crop row to a depth of 30 cm from an area close to the center of the grid cell. The soil core was separated into 0- to 15- and 15- to 30-cm increments. Samples were stored at 4^0C before root separation. The roots were separated from the soil by flotation in water. Plant debris other than roots was removed by hand and the roots were then rinsed in deionized water. Immediately after rinsing, the roots were dried in an oven at 60^0C for 48 h and weighed (Bohm, 1979). The dried samples were finely ground in a rotating ball-bearing mill. The

root samples, collected in 1996, were not ^{15}N-enriched. Percentage N and atom % ^{15}N in a 2.00±0.20 mg ground subsample were determined, using a 20-20 Mass Spectrometer interfaced with an ANCA-GSL sample converter (Europa Scientific, Crewe, UK). Unlabeled pea grain with an atom % ^{15}N of 0.3675 and standard deviation of 0.0001 was included as a working standard.

On 15 September 1997 (after harvest), wheat root yields were determined by sampling the same 10 shoulders and 10 footslopes as in 1996 (Fig. 3.2). A soil core with a 10-cm diameter was obtained, using a punch truck, from within the crop row to a depth of 60 cm in the labeled residue microplots (i.e., the root samples were ^{15}N-enriched). The soil core was separated into 0- to 15-, 15- to 30- and 30- to 60-cm increments. The procedures for root separation and root sample preparation were the same as described earlier. Percentage N and atom % ^{15}N in a 2.00±0.20 mg subsample were determined using a Tracer Mass Spectrometer interfaced with a RoboPrep sample converter (Europa Scientific, Crewe, UK). The working standard was ^{15}N-enriched pea residue with an atom % ^{15}N of 0.6013 and standard deviation of 0.0007.

The atom %^{15}N excess of roots, taken from within the labeled residue microplot in 1997, was used to calculate % Ndfr (percentage of root N derived from labeled residue) in the roots (Hauck and Bremner, 1976):

$$\% \text{ Ndfr} = \left(\frac{\text{atom \% } ^{15}\text{N excess}_{\text{root}}}{\text{atom \% } ^{15}\text{N excess}_{\text{labeled residue}}} \right) \times 100 \qquad (3.21)$$

A representative background atom % ^{15}N of unlabeled root samples obtained from within the study field was subtracted from each measurement to calculate atom % ^{15}N excess in roots.

The %Ndfr was then used to calculate the recovery of ^{15}N from labeled residues in roots:

$$\text{Residue } ^{15}\text{N recovery (\%)} = \left(\frac{\text{kg ha}^{-1}\text{ Ndfr}}{\text{kg ha}^{-1}\text{ N}_{\text{labeled residue}}}\right) \times 100 \qquad (3.22)$$

where kg ha^{-1} Ndfr is the quantity of N in roots derived from labeled residue expressed as kg ha^{-1} N and was calculated as follows:

$$\text{kg ha}^{-1}\text{ Ndfr} = \%\text{Ndfr} \times N_{uptake}\ (\text{kg ha}^{-1}) \qquad (3.23)$$

where N_{uptake} is the quantity of N accumulated in the roots grown within the labeled residue microplots expressed as kg ha^{-1} N.

3.6 Climate

A meteorological station was installed in the study field on 29 May 1996. Average daily soil temperature (0- to 10-cm depth) and total daily rainfall were monitored throughout the 1996 growing season. Total daily precipitation was measured using a tipping rain bucket gauge (Model RG2510, Sierra Misco Inc., Berkeley, CA) and daily average soil temperature was measured, using thermocouples. The rain gauge and thermocouples were connected to a Campbell Scientific CR10 data logger. The data were regularly downloaded from the data logger to a laptop computer for further analysis.

It was relatively dry in August 1996 (Fig. 3.4). Total rainfall from June to September in 1996 was approximately 10 mm less than the average of the previous 76-yr period (193 mm) (Environment Canada, 1993).

In 1997, total daily precipitation, soil temperature (0- to 10-cm depth) and soil moisture (0- to 10-cm depth) were monitored on a daily basis in a shoulder and a footslope throughout the 1997 growing season. Daily average soil temperature was measured using thermocouples. Sixteen measuring points were used in both the shoulder and the footslope. The daily average soil resistance (0- to 10-cm depth) was measured, using parallel Cu wires 10 cm apart. Twelve measuring points were used in both the shoulder and the footslope. The thermocouples and Cu wires were connected to two multiplexers, which in turn, were connected to a Campbell Scientific CR10 data logger.

Daily maximum, minimum and average temperature were recorded. The average values of the 16 measuring points in the shoulder or the footslope were used to represent the average daily soil temperature in the shoulder or the footslope (Fig. 3.5). The daily average soil temperatures in the shoulder and the footslope were similar. During the 1997 growing season, soil samples (0- to 10-cm depth) were taken at ten different dates, using a Dutch auger around each soil resistance measuring point (i.e., 12 in the shoulder and 12 in the footslope) and gravimetric soil water content was determined (105^0C, 24 h). Gravimetric soil water content at each measuring point was used to calibrate the soil resistance to gravimetric water content.

The average values of the 12 measuring points in the shoulder or the footslope were used to represent the mean daily soil water content in the shoulder or the footslope (Fig.

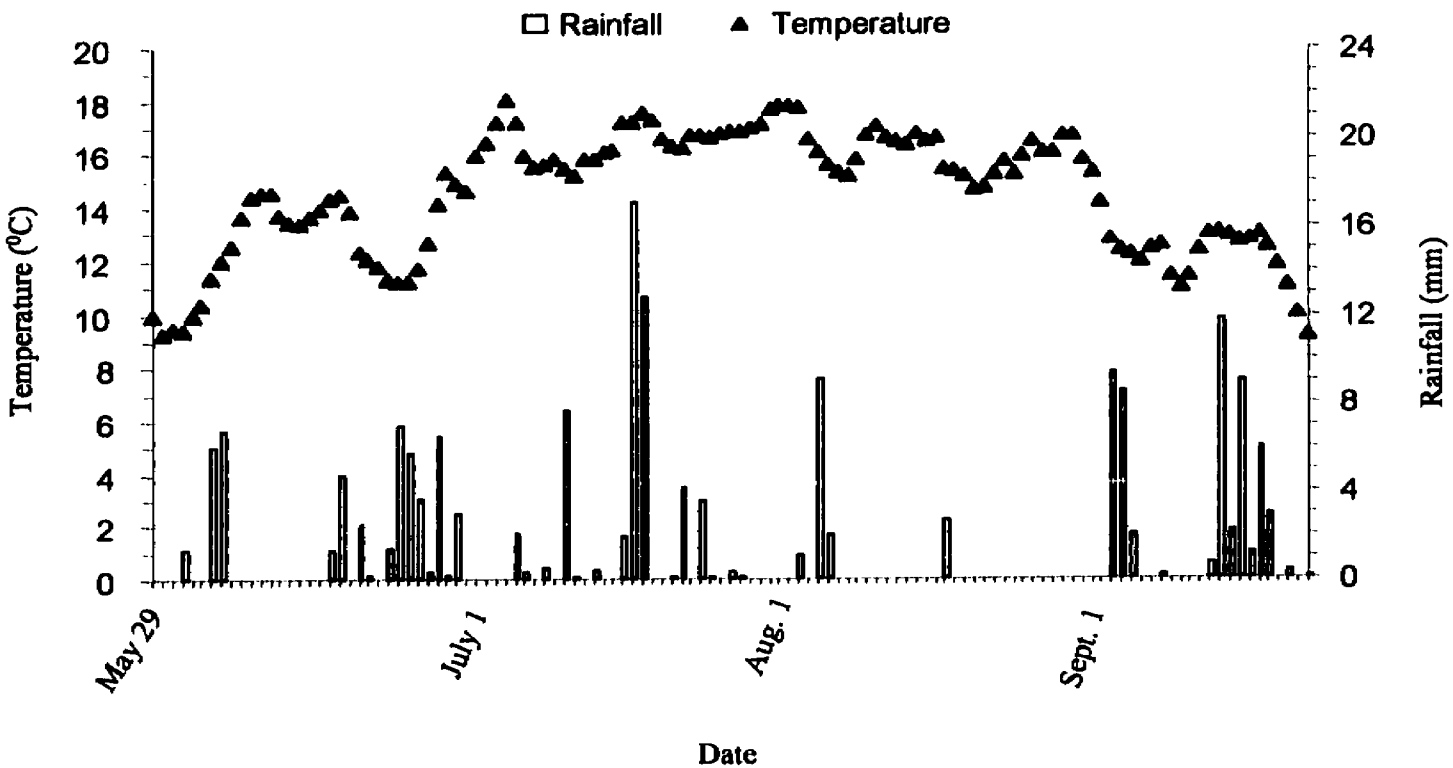

Figure 3.4. Average daily soil temperature (0- to 10-cm depth) and daily rainfall during the growing season at the study site in the Bear Hills near Biggar, SK, in 1996.

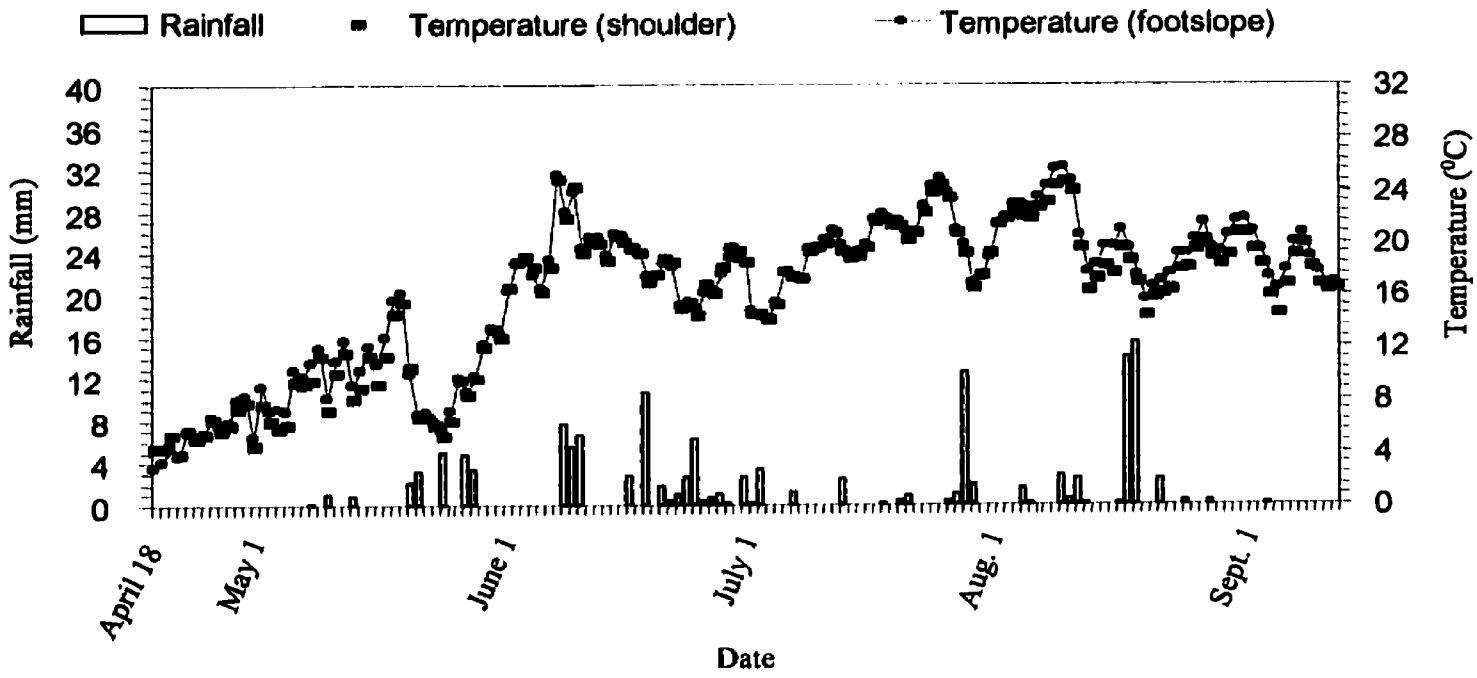

Figure 3.5. Average soil temperature (0- to 10-cm depth) in the shoulder and footslope, and daily rainfall during the growing season at the study site in the Bear Hills near Biggar, SK, in 1997.

3.6). Soil water contents (0- to 5-cm depth) at field capacity (FC) and permanent wilting point (PWP) were determined under a suction of 0.3 bar and 15 bar, respectively. Soil samples (0- to 5-cm depth) for water retention measurement at FC and PWP were from 10 shoulders and 10 footslopes.

In 1997, little precipitation occurred from early July to the end of the growing season. Total rainfall from May to September was approximately 40 mm less than the average of the previous 76-yr period (236 mm) (Environment Canada, 1993).

The temporal variability of soil water content (0- to 10-cm depth) in 1997 followed the pattern of precipitation (Fig. 3.5 and 3.6). The water content in the footslope was slightly higher than in the shoulder before 1 July. The water content in the shoulder and the footslope, however, was similar after 1 July. The soil moisture content was lower than the PWP during more than half of the growing season (Fig. 3.6).

3.7 Statistical Analysis

Assumptions inherent to the use of parametric statistics include normality, randomness, homogeneous variance and independence of observations (Steel et al., 1997). Many soil properties and soil landscape analysis data sets, however, do not have a normal distribution (Pennock et al., 1992). Thus, non-parametric statistics should be used to summarize data of this nature (Pennock et al., 1994; Walley et al., 1996). Non-parametric statistical methods are based on ranked data rather than the original data values. The use of ranked data eliminates some of the problems associated with highly skewed distributions.

Exploratory data analysis (EDA), as described by Pennock et al. (1992), was used in the first stage of the statistical analysis. According to the EDA, some measured soil and plant

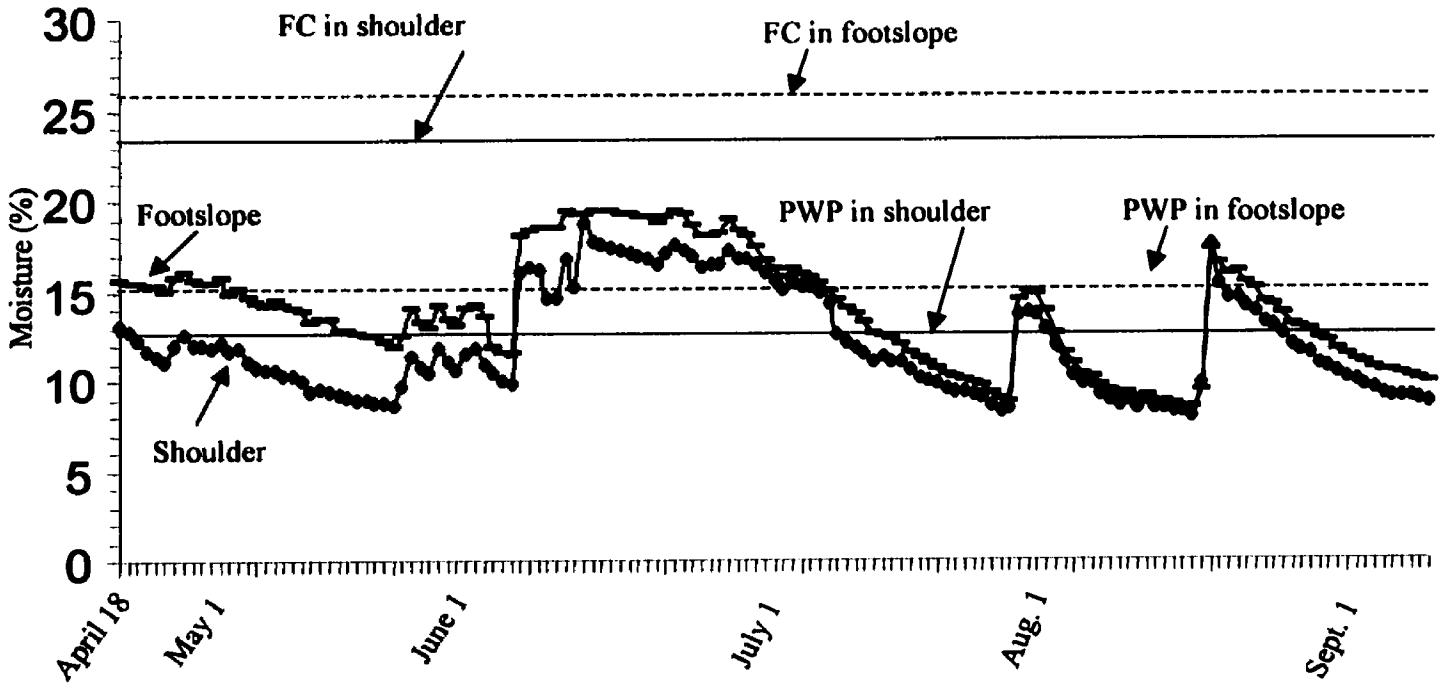

Figure 3.6. The temporal variability of gravimetric soil moisture content (percentage in the 0- to 10-cm depth, calibrated from the soil resistance) in the shoulder and footslope during the growing season at the study site in 1997.

variables were not normally distributed. To facilitate consistent statistical analysis, all of the data were analyzed using non-parametric statistics. An index of variability (i.e., interquartile range / median × 100) was included as a non-parametric measurement of the relative amount of variation for populations with different medians (Androsoff et al., 1995). The interquartile range (IQR) was included as a measure of dispersion for the data. The Mann-Whitney U test or multiple comparison extension of the Kruskal-Wallis H test was used to test differences in soil and plant variables among the treatments. The significance level (α) was chosen as 0.20. Pennock et al. (1994) and Walley et al. (1996) proposed that an α of 0.20 is most appropriate for landscape-scale studies in order to minimize Type II errors (i.e., failing to reject H_0 when it is not true) (Peterman, 1990). Spearman's coefficient of rank correlation was used to test correlation among measured variables. All the mentioned statistical analyses were accomplished using a statistical package of SPSS for Windows (SPSS Inc., 1993).

Geostatistics, originally used in the mining industry (Matheron, 1963), has proven useful in soil science for characterizing and mapping spatial variation of soil variables. In addition, geostatistical analysis of within-field variation of important soil nutrients and plant growth parameters can help identify cause-effect relationships among these variables (Tabor et al., 1984). Analysis of semivariance was used to detect the spatial structure of soil variables and symbiotic N_2 fixation data from the micro-scale study, using a GEO-EAS package (Englund and Sparks, 1988). Characteristics of the semivariogram, such as nugget (y-intercept, residual and random variation, not removed by close sampling), sill (point of leveling off, maximum variance) and range (the distance across which data are spatially correlated), can be useful in explaining the structure of spatial dependence and

correlation in the field (West et al., 1989). Details of the application of the analysis of semivariance are presented in Appendix B.

4. RESULTS AND DISCUSSION

4.1 Landscape-Scale and Micro-Scale Variability of Symbiotic Dinitrogen Fixation in Chickpea

4.1.1 Soil variability at the landscape scale and micro scale

When soil was sampled at 14-m interval, the measured soil characteristics were highly variable (Table 4.1). Total N content of the soil had the largest index of variability (78%), whereas pH had the smallest index of variability (8%). The distributions of soil EC, total C and total N content were skewed and not normally distributed, suggesting that the use of non-parametric statistics was appropriate.

Variability of physical, chemical and biological properties is a phenomenon common to all soils in natural landscapes (Pennock et al., 1987; Pennock et al., 1994; Androsoff et al., 1995; Stevenson et al., 1995). Soil moisture content was higher in the footslopes than in the shoulders (Table 4.2) because water is redistributed to the convergent areas, such as footslope and lower level complexes, in a hummocky terrain (Pennock et al., 1987; Stevenson et al., 1995). According to Pennock et al. (1987), moisture redistribution is the dominant process of soil formation and the major factor controlling many soil properties. Thus, differences in soil moisture content across the landscape also have been linked to the differences in pedogenesis and the accentuation of pedogenic differences due to accelerated erosion brought on by water, wind, and cultivation. It is not surprising, then, that soil variables, such as mineral N, and total C

Table 4.1. Descriptive statistics for selected soil variables measured at the landscape scale at the experimental site prior to sowing in 1996.

Variable	Unit	Median	Mean	Min.	Max.	IQR†	Index of variability (%)‡	Skewness
Moisture§ (0 – 60 cm)	kg kg^{-1}	0.28	0.27	0.13	0.39	0.09	31	-0.18
Mineral N (0 – 60 cm)	kg ha^{-1}	71.4	70.3	22.6	114.1	27.0	38	-0.07
Hot-KCl N (0 – 15 cm)	kg ha^{-1}	32.8	37.0	15.6	62.9	22.7	67	0.51
pH (0 – 15 cm)		8.0	7.9	6.9	8.7	0.65	8	-0.33
EC (0 – 15 cm)	mS cm^{-1}	0.28	0.29	0.11	0.83	0.10	36	2.54
Total C (0 – 60 cm)	Mg ha^{-1}	113.6	126.8	74.7	259.7	70.8	62	1.10
Total N (0 – 60 cm)	Mg ha^{-1}	9.4	10.9	5.2	24.7	7.4	78	1.04

† IQR stands for interquartile range.
‡ Index of variability = (IQR / median) × 100.
§ The sample points were 42 for each variable.

Table 4.2. Median values and results of the Mann-Whitney U test for selected soil variables measured at the landscape scale at the experimental site prior to sowing in 1996.

Landform element complex	n	Moisture ($kg\ kg^{-1}$) 0-60 cm	Mineral N ($kg\ ha^{-1}$) 0-60 cm	Hot-KCl N ($kg\ ha^{-1}$) 0-15 cm	pH 0-15 cm	EC ($mS\ cm^{-1}$) 0-15 cm	Total C ($Mg\ ha^{-1}$) 0-60 cm	Total N ($Mg\ ha^{-1}$) 0-60 cm
Shoulder	15	0.22	56.4	29.7	8.2	0.29	99.7	7.2
Footslope	27	0.29	76.1	40.0	7.7	0.27	118.8	11.6
Shoulder vs. footslope†		< 0.01	< 0.01	< 0.01	< 0.01	0.60	0.02	< 0.01

† The *P* value associated with the comparison between the shoulders and the footslopes.

content and total N content were influenced by the landform in a manner similar to soil moisture (Table 4.2).

Hot-KCl extractable N was used as an index of the availability of soil N (i.e., potentially mineralizable N). Hot-KCl extractable N was greater in the footslopes as compared to the shoulders (Table 4.2), suggesting that soils in the footslopes could supply more available N to crops than soils in the shoulders. Qian and Schoenau (1995) made similar observations in landscape-scale studies in Saskatchewan, using an anion exchange membrane technique.

Downward movement of $CaCO_3$ in the soil profile will decrease the pH of surface horizons. Leaching of carbonates occurs in portions of the landscape such as north slopes, lower slopes and depressions where accumulation of moisture favors downward flow of water (Anderson, 1987). As a result, soil pH was higher in the shoulders as compared to the footslopes.

When soil was sampled at 0.3-m interval along a 33-m micro-scale transect, a high degree of variability was detected in measured soil characteristics (Table 4.3). For example, EC values ranged from 0.05 to 0.32 mS cm^{-1}. The EC had the largest index of variability (35%), whereas pH had the smallest index of variability (2%). The pH and total N content of the soil were not normally distributed, based on the 110 sampling points in the transect (Table 4.3).

The semivariogram curves of soil moisture, total C and total N content were of similar shape; their range values also were similar (Fig. 4.1 and Table 4.4). Moreover, soil moisture, total C and total N content were significantly correlated to each other ($P < 0.01$). The results suggest that these variables were intimately integrated and would

Table 4.3. Descriptive statistics for selected soil variables (0- to 15-cm depth) measured along a 33-m transect (0.3-m sampling interval) at the experimental site prior to sowing in 1996.

Variable	Unit	Median	Mean	Min.	Max.	IQR	Index of variability (%)†	Skewness
Moisture‡	kg kg^{-1}	0.28	0.28	0.17	0.34	0.03	13	-0.77
Mineral N	kg ha^{-1}	23.8	24.1	11.6	38.6	6.7	28	0.10
pH		7.2	7.2	6.8	8.2	0.2	2	1.69
EC	mS cm^{-1}	0.23	0.22	0.05	0.32	0.08	35	-0.63
Total C	Mg ha^{-1}	71.7	70.6	47.5	84.7	9.3	13	-0.83
Total N	Mg ha^{-1}	6.8	6.7	4.6	7.8	0.8	12	-0.98

† Index of variability = (IQR / median) × 100.
‡ The sample numbers were 110 for each soil variable.

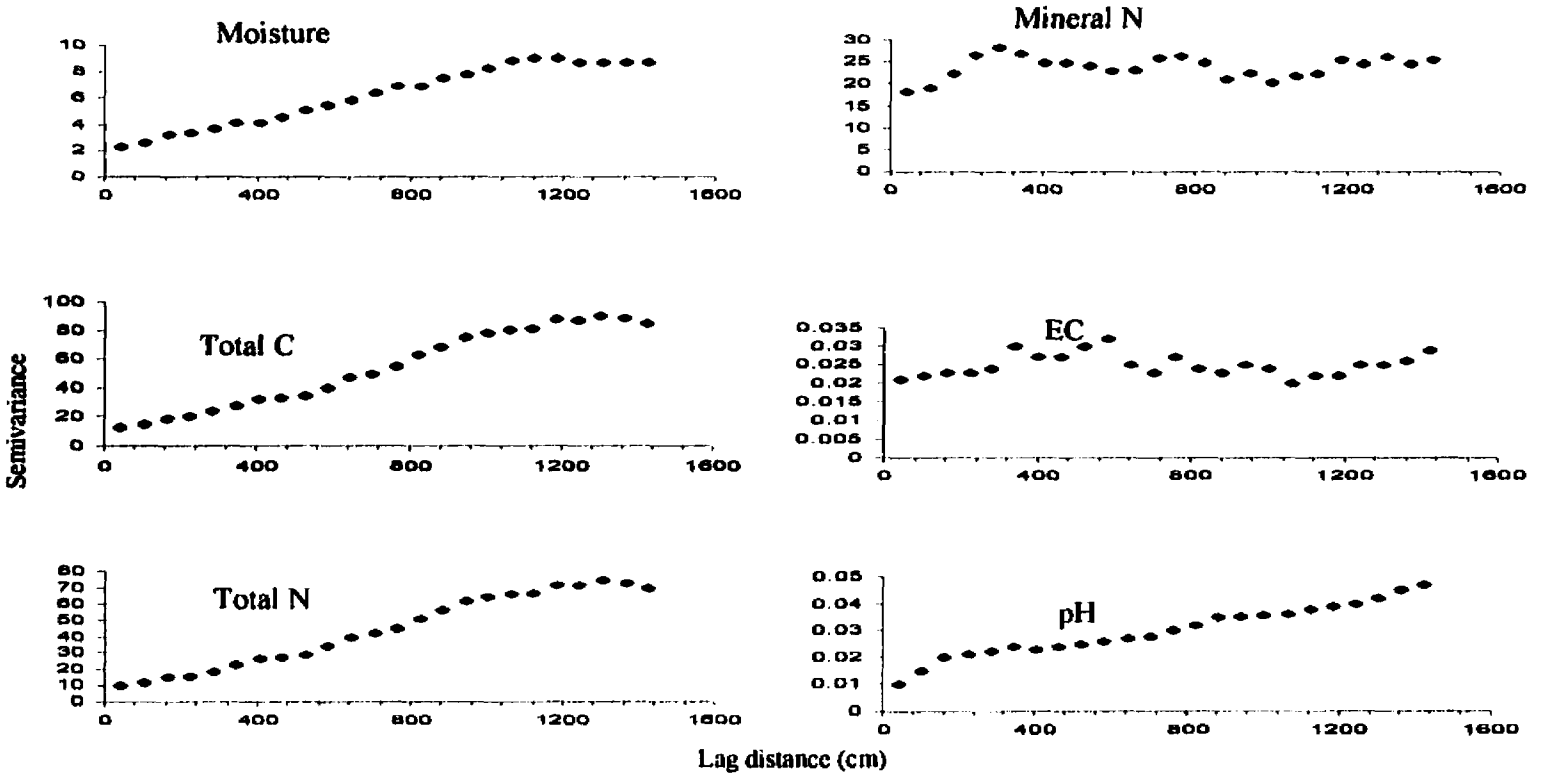

Figure 4.1. The semivariograms of soil variables measured along the 33-m transect (0.3-m sampling interval) at the study site in the spring in 1996.

affect soil biochemical processes in concert. The significant correlation of soil moisture and total N content with total C content suggested that the spatial distribution of soil moisture and N was largely associated with SOM. Soil moisture, total C content and total N content also had a relatively low percentage of sill value (Table 4.4), indicating that the majority of the variance associated with these variables could be accounted for by the spatial correlation.

Table 4.4. The summary of results of semivariograms of selected soil variables (0- to 15-cm depth) measured along a 33-m transect (0.3-m sampling interval) at the experimental site in the spring in 1996.

Variable	Nugget	Sill	Percentage of sill†	Range (m)
Moisture	2.3	9.0	26	11.8
Total C	13.0	90.3	14	13.0
Total N	10.2	74.7	14	13.0
Mineral N	18.3	28.4	64	2.8
EC	0.02	0.03	67	3.4
pH			no pattern	

† Percentage of sill = (nugget / sill) × 100.

The range of mineral N was approximately 3 m (Fig. 4.1 and Table 4.4). Cahn et al. (1994) similarly observed that the spatial correlation of nitrate was less than 5 m, whereas PO_4^- and K^+ had a range of 30 to 50 m, based on analysis of semivariance in

central Illinois. These authors suggested that the different ranges of spatial correlation for soil nutrients might be related to the mobility of the nutrient ions in the soil.

Nitrate can be highly variable due to the influence of the climate and tillage practices, including residue management. In addition, the local (or micro-scale) variability of surface roughness, soil C and texture might cause micro-site variability of runoff and soil water storage, and consequently cause micro-site variability of microbial activity and N mineralization. Evidence of the micro-scale variability is found in the relatively high nugget and percentage of sill value of the mineral N, i.e., 64% of the variance of mineral N was present at a scale less than the sampling interval (i.e., 0.3 m), and only 36% of the variance could be accounted for by the spatial correlation.

Mineral N and EC had similar values for percentage of sill, which were larger than those of soil moisture, total C and total N content (Table 4.4). No spatial pattern was evident for pH. It did not reach a sill (Fig. 4.1), indicating that pH was highly heterogenous and if any spatial pattern did exist, it occurred on a larger scale.

4.1.2 Symbiotic dinitrogen fixation at the landscape scale and micro scale: Impact of soil variability

The contribution of symbiotic N_2 fixation to the N accumulated (i.e., %Ndfa) in the chickpea residue ranged from 29.1 to 96.7%, whereas the %Ndfa in the chickpea grain ranged from 45.1 to 74.7% when estimated at 14-m interval (Table 4.5). Although landform effect was present for the measured soil variables except EC (Table 4.2), no landform effect on symbiotic N_2 fixation (%Ndfa) was detected (Table 4.5). Likewise, the total quantity of N derived from symbiotic N_2 fixation (kg ha^{-1} Ndfa) in the chickpea

Table 4.5. Descriptive statistics and the results of the Mann-Whitney U test for %Ndfa and Ndfa (kg ha^{-1}) in chickpea residue and grain measured at the landscape scale at the experimental site at harvest in 1996.

Landform element complex	Median	Mean	Min.	Max.	IQR	Index of variability (%)†	Skewness
				Residue %Ndfa			
Shoulder	74.3	68.8	29.1	96.7	21.9	29	-0.92
Footslope	73.7	70.1	37.8	94.8	19.1	26	-0.53
Shoulder vs. footslope‡	0.82						
				Grain %Ndfa			
Shoulder	63.5	63.3	45.1	73.3	10.1	16	-0.96
Footslope	62.6	63.2	50.2	74.7	14.9	24	-0.11
Shoulder vs. footslope	0.88						
				Residue Ndfa (kg ha^{-1})			
Shoulder	7.3	8.8	3.2	17.8	8.2	112	0.69
Footslope	10.9	12.8	5.3	25.5	5.5	50	1.06
Shoulder vs. footslope	0.01						
				Grain Ndfa (kg ha^{-1})			
Shoulder	36.4	37.4	26.5	49.6	18.5	51	0.19
Footslope	37.9	37.9	14.3	63.9	19.3	51	0.27
Shoulder vs. footslope	0.96						

† Index of variability = (IQR / median) × 100.

‡ The *P* value associated with the comparison between the medians of the shoulder and the footslope.

grain did not differ between the shoulders and the footslopes. The accumulation of symbiotically-fixed N in the chickpea residue (kg ha^{-1} Ndfa) was higher in the footslopes than in the shoulders (Table 4.5).

Ammonia has a repressive effect on the production of new nitrogenase in both free-living and symbiotic N_2 fixers (Paul and Clark, 1996). Nitrate, when present in the soil, similarly represses nodulation and symbiotic N_2 fixation (Paul and Clark, 1996). Thus, a negative correlation between soil mineral N and symbiotic N_2 fixation would be expected (Doughton et al., 1993; Stevenson et al., 1995). In the current study, a negative correlation between %Ndfa in chickpea residue and soil mineral N was detected ($r = -0.45$, $P < 0.01$). The landform had an effect on the soil mineral N content (Table 4.2). Landform, however, had no effect on residue %Ndfa or grain %Ndfa (Table 4.5). The data indicated that factors other than mineral N likely controlled symbiotic N_2 fixation and masked the landform effect on symbiotic N_2 fixation. If soil mineral N directly controlled symbiotic N_2 fixation in the field, the median value of %Ndfa should have been higher in the shoulders than in the footslopes. Wendroth et al. (1992) suggested that variability among available plant nutrients other than N or soil chemical parameters also may have masked detectable landform effect on symbiotic N_2 fixation.

Androsoff et al. (1995) did not detect a landform effect on symbiotic N_2 fixation in pea, using a natural ^{15}N abundance approach, in a landscape under minimum tillage in Saskatchewan. In contrast, Stevenson et al. (1995) observed that %Ndfa in pea, using a natural ^{15}N abundance approach, was higher in the footslopes than in the shoulders in a conventionally tilled landscape in Saskatchewan. In the current study, a minimum disturbance direct seeding system was practiced. It is probable that surface placement of

the residue influenced water redistribution, residue decomposition, and soil N dynamics across the landscape during the growing season in ways that were different from those in the conventionally tilled fields. For example, leaching and denitrification losses of N may increase because surface-placed residues can result in increased water infiltration and reduced evaporation rates (Schomberg et al., 1994).

Other researchers, using a landscape-scale approach in Saskatchewan, reported that soil moisture always was higher in the footslopes than in the shoulders at early spring sampling times, due to the water redistribution from shoulders to convergent footslopes (e.g., Pennock et al., 1994; Stevenson et al., 1995). In a minimum tillage field, water redistribution may be different as compared to fields under conventional tillage, which in turn, may influence other dynamic soil processes, such as mineralization of soil N. Thus, the landform effect on symbiotic N_2 fixation may be masked in the minimum tillage fields.

According to Bremer et al. (1988), symbiotic N_2 fixation in lentil, pea and fababean (*Vicia faba* L.) declined concurrently with a decline in soil moisture, suggesting that plant water usage and symbiotic N_2 fixation were interrelated. The effect of soil moisture on symbiotic N_2 fixation probably is dependent on the pattern of precipitation, i.e., the timing of precipitation events during the growing season. In the current study, 102 mm rainfall occurred in June and July 1996, which likely enhanced early chickpea growth. The enhanced chickpea growth could quickly deplete soil mineral N, thereby, making atmospheric N_2 the dominant N source for chickpea. Precipitation, however, was limited in August 1996 (total 15 mm). The difference in soil moisture content between the shoulders and the footslopes probably was minimal during this time. The droughty conditions

experienced across the landscape likely inhibited chickpea growth and symbiotic N_2 fixation, further masking the landform effect on symbiotic N_2 fixation.

The landform effect on symbiotic N_2 fixation may also be site specific. Topographic differences in symbiotic N_2 fixation probably are more distinct at a site with steeper slopes, mainly through amplification of differential hydrologic-pattern controls on soil factors (e.g., soil moisture and mineral N) that regulate symbiotic N_2 fixation. For example, in a study to evaluate the influence of slope position on growth and symbiotic N_2 fixation in pea in Washington, Mahler et al. (1979) observed higher yield and greater symbiotic N_2 fixation in pea growing on the bottomland than the ridgetop, and they suggested that this result was apparently related to greater root penetration and removal of water on the bottomland.

Estimates of %Ndfa in the chickpea residue within the 33-m micro-scale transect were highly variable (Table 4.6). For example, the maximum %Ndfa value in the chickpea residue was approximately three times greater than the minimum %Ndfa value. Point-to-point variability in symbiotic N_2 fixation was evident along the micro-scale transect (Fig. 4.2). The smallest difference in %Ndfa between two neighboring sampling points (i.e., 0.3 m apart) was 0.4%, whereas the largest difference in %Ndfa between two neighboring sampling points was 37.8%.

Analysis of semivariance demonstrated that the range for estimates of %Ndfa in the chickpea residue was 1.6 m (Fig. 4.3), indicating that estimates of symbiotic N_2 fixation measured in close proximity to each other were correlated. Estimates of %Ndfa in the chickpea residue had relatively large nugget and percentage of sill values (Fig. 4.3), indicating that the majority of variance (72%) in %Ndfa existed at a scale smaller than

Table 4.6. Descriptive statistics for %Ndfa in chickpea residue and grain measured along a 33-m transect (0.3-m sampling interval) at the experimental site at harvest in 1996.

Variable	n	Median	Mean	Min.	Max.	IQR	Index of variability (%)†	Skewness
Residue %Ndfa	110	55.6	55.4	21.9	80.4	12.9	23	-0.51
Grain %Ndfa	110	55.1	54.6	36.0	69.7	7.6	14	-0.51

† Index of variability = (IQR / median) × 100.

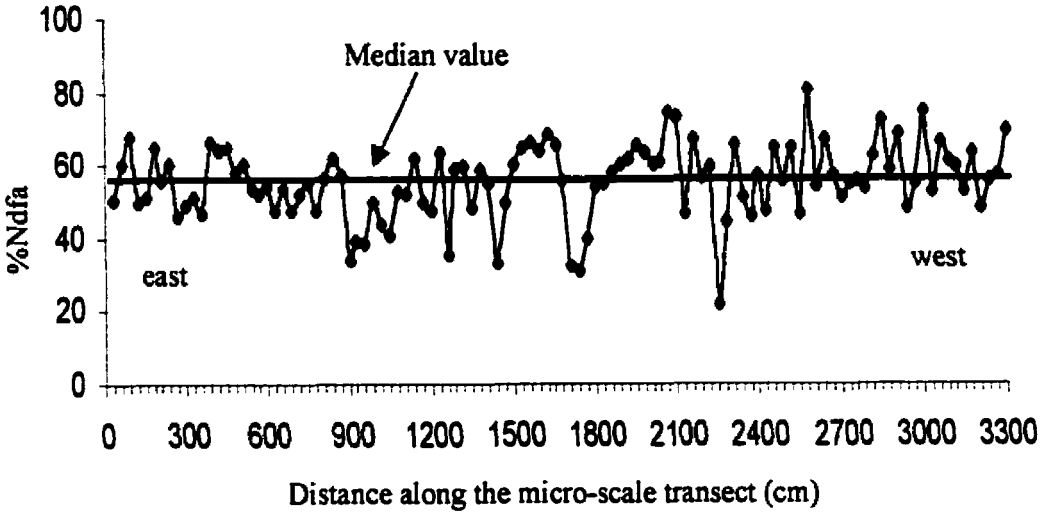

Figure 4.2. Horizontal point-to-point variation of chickpea residue %Ndfa along the 33-m micro-scale transect (0.3-m sampling interval) at the study site at harvest in 1996. There are a total 110 samples.

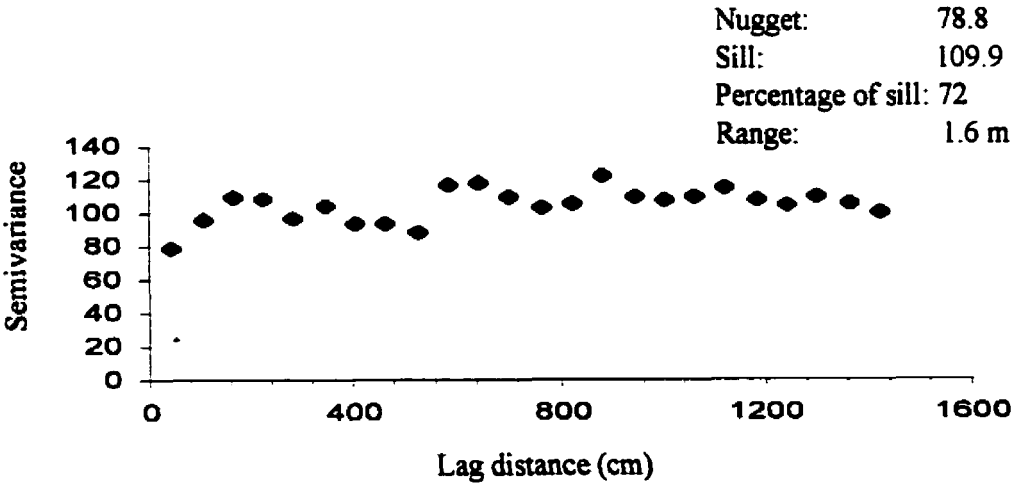

Figure 4.3. The semivariogram for %Ndfa in chickpea residue along the 33-m transect (0.3-m sampling interval) at the study site at harvest in 1996.

the sampling interval and only a small portion of variance (28%) in %Ndfa in the chickpea residue could be attributable to spatial correlation.

The variability in symbiotic N_2 fixation at the micro scale suggests that symbiotic N_2 fixing activity was highly variable in the field. The analysis of semivariance found a spatial pattern for all measured soil variables except pH (Table 4.4). The spatial pattern of symbiotic N_2 fixation was similar to that of mineral N and dissimilar to all other soil variables (Fig. 4.1 and 4.3), suggesting that the spatial distribution of %Ndfa was associated with soil mineral N. The range of %Ndfa was less than the ranges of soil variables, probably because chickpea roots proliferated within a considerable soil volume and integrated soil variability at all directions, thereby reducing or mitigating the effects of long distance variability of soil variables on chickpea growth and symbiotic N_2 fixation.

Micro-scale variability in symbiotic N_2 fixation may be attributable to the micro-site variability of drainage conditions, native *Rhizobium* population distribution, uneven soil P distribution, and other unknown factors (Parkin, 1993; Androsoff et al., 1995; Stevenson et al., 1995). The local variability of surface roughness, soil texture and SOM may result in variation in run-off and water redistribution, and subsequent soil water status, further influencing the net mineralization of soil N and the dynamics of soil mineral N.

Rupela et al. (1987) observed that chickpea *Rhizobium* populations fluctuated most in the top 5 cm, being reduced during periods of high soil temperature in summer and recovering after rains. The distribution of *Rhizobium* likely was affected by soil microclimate. One might speculate that variable populations of indigenous *Rhizobia* and unequal nodule numbers formed by the indigenous and the introduced *Rhizobia* may

have caused the micro-site variability in nodulation and symbiotic N_2 fixation. Chickpea, however, has not been grown extensively in Saskatchewan prior to 1996 and has never been grown in the study field. Likely, no indigenous chickpea *Rhizobia* were present in the study field. Thus, it is unlikely that indigenous chickpea *Rhizobia* affected the estimates of symbiotic N_2 fixation in the current study.

It is crucial to understand the spatial variability of soil variables, so data can be properly interpreted. Knowledge of the factors controlling variability at the micro-scale level will lead to a mechanistic understanding of how soil microbes interact in and with their environment. The process level information at the micro-scale level is important to improve our predictive capabilities at the larger scale. As we move from the smaller scale to the larger scale, soil variables become more integrated in nature (Parkin, 1993). Soil moisture, mineral N, total N content, pH and total C content were significantly correlated to each other ($P < 0.01$) at the 14-m sampling interval, indicating that these soil variables were intimately linked and would regulate symbiotic N_2 fixation and other biochemical processes in concert. It becomes, therefore, increasingly more difficult to identify the key factors driving biological processes in soil, primarily because the driving factors are more integrated at the larger scales. In the current study, symbiotic N_2 fixation and independent soil variables, with the exception of mineral N, were poorly correlated at the 14-m sampling interval, i.e., the correlations between %Ndfa and pH ($r = 0.26$, $P = 0.09$), EC ($r = 0.13$, $P = 0.43$), and total C content ($r = -0.07$, $P = 0.65$) were not statistically significant at the 5% level of probability. A poor correlation between symbiotic N_2 fixation and soil variables also was observed by Androsoff et al. (1995) in their landscape study in Saskatchewan.

The micro-scale variability of soil variables and symbiotic N_2 fixation probably masked any landform effects on symbiotic N_2 fixation. The analysis of semivariance showed that spatial correlation contributed little to the total variance of %Ndfa, whereas the majority of the variance of %Ndfa was random. The range of %Ndfa was approximately 1.6 m, which was much less than the 14-m sampling interval used for estimation of %Ndfa at the landscape scale. If this spatial pattern of %Ndfa existed across the landscape, it probably masked any landform effect on %Ndfa.

Studies were conducted to compare different approaches for estimating symbiotic N_2 fixation at the landscape scale in Saskatchewan (Androsoff et al., 1995; Stevenson et al., 1995). Neither Androsoff et al. (1995) nor Stevenson et al. (1995) detected a strong correlation among estimates of symbiotic N_2 fixation when two different approaches were used to estimate symbiotic N_2 fixation. These authors suggested, that although the lack of correlation may reflect inherent differences between symbiotic N_2 fixation estimation approaches, it was more likely that micro-scale variability in symbiotic N_2 fixation accounted for the lack of good correlation. This hypothesis is in agreement with the results from the micro-scale study which suggest that the ranges of mineral N and %Ndfa were approximately 2.8 m and 1.6 m, respectively, i.e., mineral N measured 2.8 m apart and %Ndfa measured 1.6 m apart were spatially independent.

A conceptual model was developed to help elucidate the factors controlling symbiotic N_2 fixation at different scales (Fig. 4.4). According to Parkin (1993), the impact of soil and environmental variables on the soil biochemical processes can occur at the regional, field or landscape, plot and micro scale. The major control factors at the regional scale are climate and land use pattern. The main component of climate is precipitation.

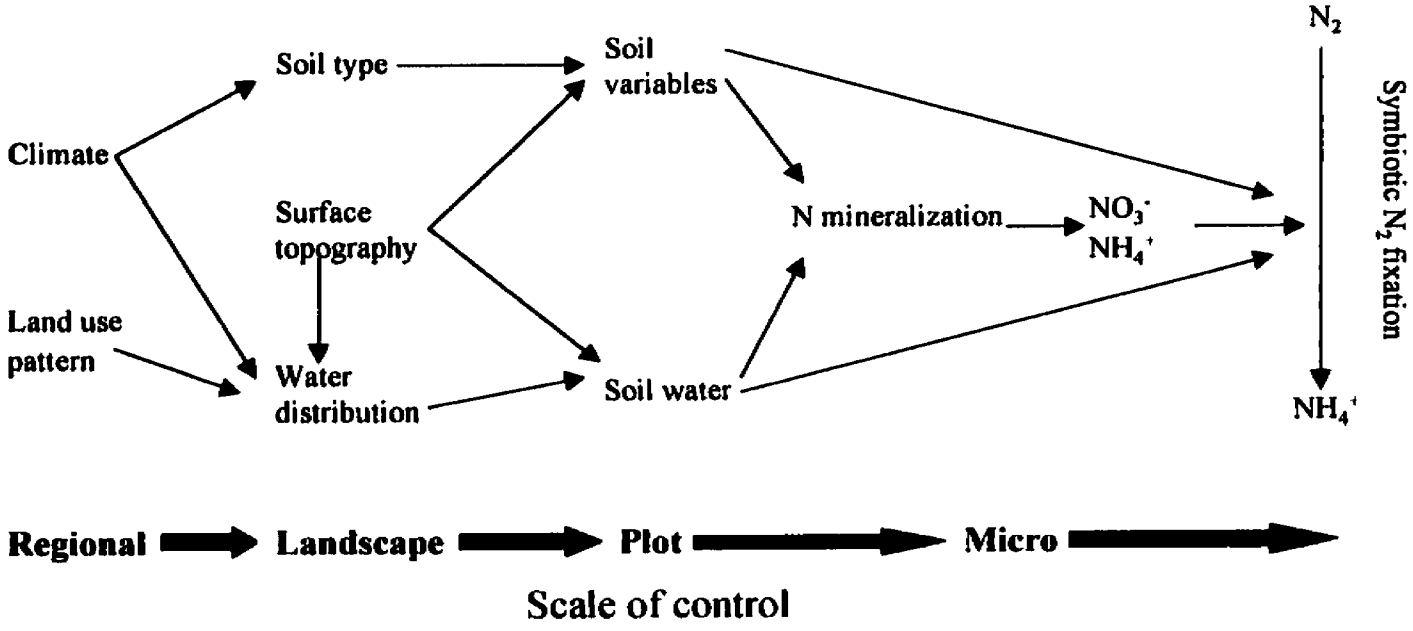

Figure 4.4. A conceptual model for the different scales of regulation on symbiotic N_2 fixation (modified from Stevenson et al., 1995).

Precipitation and its temporal pattern largely control the regional variability of soil and regulate the pattern of water redistribution in the field. Land use patterns may include different tillage systems and crop rotations, which largely influence the status of the soil surface and soil structure, thus, further influencing water redistribution on the soil surface and in the soil.

The major control factors at the landscape scale are soil type, surface topography and water redistribution (Parkin, 1993). Soil types determine soil properties, such as organic matter, N, texture, pH, and EC. The influence of surface topography and water redistribution on soil variables at the landscape-scale level were discussed in detail in Section 2.5.

The plot-scale level can be regarded as the scale at the level of the treatment plot or sampling unit (i.e., grid cell) in the landscape study. The soil variables and soil water status at the plot-scale level are largely dependent on the soil type and its position in the landscape. Heterogeneity of soil variables and soil water status may cause local variation in N mineralization, further influencing the dynamics of soil mineral N at the micro scale. Soil mineral N, soil water and other factors, such as microbial activity and legume growth, ultimately determine the magnitude of symbiotic N_2 fixation at the local or micro scale.

The relative importance of factors that influence symbiotic N_2 fixation at each scale can be site specific, which is largely dependent on which factors are the most limiting factors for legume growth and symbiotic N_2 fixation. Moreover, these factors are intimately correlated and may have an additive effect on symbiotic N_2 fixation. For example, soil water is the most important factor controlling crop growth, N

mineralization and the dynamics of soil mineral N. When soil water is favorable for crop growth, mineralized N and mobile nitrate may be at levels sufficient to inhibit symbiotic N_2-fixing activity. When soil water is a limiting factor, crop growth will be inhibited, but decreased N mineralization and decreased mobility of nitrate may result in an increase in symbiotic N_2-fixing activity.

4.1.3 Chickpea yield and nitrogen contribution to soil via symbiotic dinitrogen fixation

Redistribution of water toward convergent areas exerts the most important landscape-scale control on crop productivity in a natural landscape (Pennock et al., 1987), because many soil properties and biological processes are primarily controlled by soil water status. The yield of chickpea residue was higher in the footslopes as compared to the shoulders, but no landform effect occurred for grain yield of chickpea (Table 4.7). Apparently the relatively high moisture and mineral N content in the footslopes (Table 4.2) increased the residue yield, but not the grain yield of chickpea. Similarly, total N accumulation in the chickpea residue was higher in the footslopes than in the shoulders due to the higher residue yield of chickpea in the footslopes, whereas no landform effect occurred for the amount of N accumulated in the chickpea grain.

Doughton et al. (1993) similarly observed that increased N fertilization rate had no effect on chickpea grain yields in Australia. Their data also suggest that grain yields of chickpea were not limited by N nutrition, and the source of N, whether soil-derived or symbiotically-fixed, was of no consequence to grain yield. Bonfil and Pinthus (1995) observed that increasing the supply of N during chickpea grain development by a pre-

Table 4.7. Median values and results of the Mann-Whitney U test for chickpea yield and N accumulation measured at the landscape scale at the experimental site at harvest in 1996.

Landform element complex	n	Yield (kg ha^{-1})		N accumulation (kg ha^{-1})	
		Residue	Grain	Residue	Grain
Shoulder	15	1856(849)†	1726(552)	11.7 (9.4)	60.9(18.7)
Footslope	27	2376(1094)	1826(560)	15.3(13.3)	58.3(21.2)
Shoulder vs. footslope‡		0.12	0.84	0.01	0.95

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulders and the footslopes.

Table 4.8. Median values and results of the Mann-Whitney U test for N contribution to soil via symbiotic N_2 fixation in chickpea measured at the landscape scale at the experimental site at harvest in 1996.

Landform element complex	n	Residue Ndfa	Grain Ndfs	N contribution	N harvest index†
		kg ha^{-1}			
Shoulder	15	7.3(8.1)‡	20.9(10.7)	-12.9 (15.9)	0.83(0.07)
Footslope	27	10.9(5.5)	19.5 (8.1)	-8.7 (9.0)	0.78(0.08)
Shoulder vs. footslope§		0.01	0.87	0.15	0.01

† N harvest index = (grain N)/(residue N + grain N).
‡ The values in the parentheses are the IQR.
§ The *P* value associated with the comparison between the shoulder and the footslope.

sowing application of nitrate or by top-dressing N at the onset of flowering increased the percentage of N in the chickpea straw, but had no significant effect on the grain yield of chickpea. Chickpea was, therefore, capable of providing all the N required for grain yield, even at the lowest soil mineral N level. The practice of providing 'starter' N fertilizer at sowing would, therefore, lead to better crop growth and N accumulation in chickpea. However, it would not improve chickpea grain yields (Doughton et al., 1993).

Legume crops will add N to the soil system via symbiotic N_2 fixation, if the total N fixed by legume crops is greater than the quantity of N removed in the harvested grain (Doughton et al., 1993). The N contribution to soil N via symbiotic N_2 fixation was negative in both the shoulders and the footslopes (Table 4.8). Only one sampling grid cell in the shoulders and three sampling grid cells in the footslopes had a positive N contribution. The negative N contribution to soil N by chickpea, apparently, was due to the lower kg ha^{-1} Ndfa (or N content) in the chickpea residue and higher N harvest index as compared to other legume crops (Evans et al., 1989; Stevenson and van Kessel, 1997). In Australia, Doughton et al. (1993) observed that chickpea provided a positive contribution to soil N at high rates of symbiotic N_2 fixation and a negative contribution to soil N at low rates of symbiotic N_2 fixation.

Soil N accumulation in the grain (i.e., kg ha^{-1} Ndfs) did not differ between the shoulders and the footslopes (Table 4.8). The accumulation of symbiotically-fixed N (i.e., kg ha^{-1} Ndfa) in chickpea residue was higher in the footslopes than in the shoulders because the residue yield was higher in the footslopes. The N contribution to soil was less negative in the footslopes as compared to the shoulders because more N was contributed by the chickpea residue in the footslopes. Evans et al. (1989) and

Armstrong et al. (1994) found that the N contribution by pea to the soil generally was greatest at sites having the highest soil water availability. This finding is in agreement with the observation in the current study that a higher N contribution to the soil by chickpea occurred in the footslopes (i.e., less negative), where relatively high moisture and mineral N levels stimulated crop growth and resulted in a greater accumulation of N in chickpea residue as compared to the shoulders.

4.2 Availability of Nitrogen from Crop Residues and the Interaction between Residue Nitrogen and Native Soil Nitrogen

4.2.1 Characteristics of labeled residues

The yield, and C content and N content of labeled chickpea residue and labeled wheat residue were determined at harvest in 1996. The residue yield of wheat was significantly higher than that of chickpea in both the shoulders and the footslopes (Table 4.9). The residue yields of both chickpea and wheat were significantly higher in the footslopes as compared to the shoulders. The percentage N content of chickpea residue was higher than that of wheat residue in both the shoulders and the footslopes. The percentage N content of the chickpea residue was higher in the footslopes as compared to the shoulders, whereas no landform effect was evident on the N content of the wheat residue.

The quantity of N input from the residues, i.e., N content of residues expressed as kg ha^{-1}, was variable in different landscape positions. The N input from the wheat residue was greater than from the chickpea residue in the shoulders, whereas no difference occurred in N input of the two residues in the footslopes. The footslopes resulted in a higher N input from the chickpea residue than the shoulders, but a landform effect on the quantity of N input from the wheat residue was not detected (Table 4.9).

The C:N ratio of the chickpea residue was lower in the footslopes than in the shoulders, whereas no landform effect was evident on the C:N ratio of the wheat residue (Table 4.9). Although the C:N ratio of the chickpea residue was significantly lower than that of the wheat residue in both the shoulders and the footslopes, it was relatively high as compared to the C:N ratios of other legume residues. For example, Stevenson and

Table 4.9. Yield, N content and C:N ratio of ^{15}N-labeled chickpea residue and wheat residue measured at the experimental site at harvest in 1996: Median values and results of the Mann-Whitney U test related to landform element complexes and residue types.

Landform element complex	Yield (kg ha^{-1})		N content (%)		N content (kg ha^{-1} N)		C:N ratio	
	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue
Shoulder	1880(860)†	2920(692)	0.54(0.14)	0.46(0.19)	11.6 (6.6)	12.7(6.8)	72(20)	99(52)
Footslope	2730(1380)	3510(905)	0.59(0.16)	0.45(0.18)	17.7(10.7)	14.5(9.7)	64(19)	103(49)
Shoulder vs. footslope‡	0.01	0.05	0.03	0.64	0.01	0.35	0.01	0.62
Shoulder, chickpea vs. wheat§	< 0.01		0.08		0.06		< 0.01	
Footslope, chickpea vs. wheat¶	< 0.01		< 0.01		0.35		< 0.01	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between chickpea residue and wheat residue in the shoulder.
¶ The *P* value associated with the comparison between chickpea residue and wheat residue in the footslope.

van Kessel (1997) reported that the C:N ratios of pea residue were 23:1 and 19:1 in the shoulders and the footslopes, respectively, in a landscape study in Saskatchewan.

The chemical composition of plant tissue changes as the plant matures (e.g., Walton, 1983; Hooda et al., 1986). Hooda et al. (1986) observed a C:N ratio of 22:1 for chickpea straw at 35 days after sowing, and the C:N ratio of the chickpea straw continued to increase over the growing season, reaching 59:1 at 135 days after sowing. Manna et al. (1997) reported a C:N ratio of 60:1 for chickpea straw at physiological maturity.

Legume residues should contribute more N to the succeeding crops as compared to cereal residues because legume residues typically are characterized by relatively low C:N ratios and high N contents. This expectation, however, may not always be true. For example, Bremer and van Kessel (1992) observed that of the ^{15}N added in lentil straw and wheat straw, approximately 5.5% was assimilated by the succeeding wheat crop from both the lentil straw (C:N ratio: 31:1) and the wheat straw (C:N ratio: 43:1).

4.2.2 Residue nitrogen recovery in the soil microbial biomass

In the early spring after fall application of labeled residues, a significant portion of the residue N was recovered in the microbial biomass (Table 4.10), suggesting that both the chickpea residue N and the wheat residue N were utilized rapidly by soil microbes, irrespective of the observation that both the chickpea residue and the wheat residue had relatively high C:N ratios. Bremer and van Kessel (1992) suggested that most of the N in plant tissues was microbially available. Crop residues contain easily decomposable fractions, which are readily available to the microbial biomass (van Veen et al., 1984). In addition, physical decomposition associated with freeze-thaw cycles would enhance

Table 4.10. Median values and results of the Mann-Whitney U test for residue N recovery in microbial biomass, expressed as kg ha^{-1} N and percentage of N in the labeled residues at the experimental site in April and September 1997.

Landform element complex	April 1997				Sept. 1997			
	N recovery (kg ha^{-1} N)		N recovery (%)		N recovery (kg ha^{-1} N)		N recovery (%)	
	Chickpea residue	Wheat residue	Chickpea residue	Wheat Residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue
Shoulder	1.51(1.72)†	2.35(2.86)	14.0(16.9)	16.9(13.9)	2.05(5.05)	1.92(3.95)	34.8(52.7)	12.5(28.2)
Footslope	2.38(4.14)	1.97(2.07)	11.1(21.5)	8.1(12.7)	1.70(2.49)	2.59(3.90)	10.5(20.0)	15.0(32.7)
Shoulder vs. footslope‡	0.07	0.33	0.86	0.05	0.46	0.79	0.08	0.86
Shoulder, chickpea vs. wheat§	0.08		0.44		1.00		0.22	
Footslope, chickpea vs. wheat¶	0.53		0.12		0.18		0.19	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between chickpea residue and wheat residue in the shoulder.
¶ The *P* value associated with the comparison between chickpea residue and wheat residue in the footslope.

the release of N from fall-applied residues, regardless of the crop type (Ivarson and Snowden, 1970). This process will allow soil microbes to rapidly assimilate N from the partially decomposed residues as the soil temperature begins to increase in the spring (Stevenson and van Kessel, 1996b).

Residue N recovery in the microbial biomass was determined in April 1997 and expressed as kg ha^{-1} N. More N was recovered from the wheat residue than from the chickpea residue in the shoulders. However, no difference was found between the two residues in terms of the quantity of N recovered in the footslopes (Table 4.10). More N was recovered from the chickpea residue in the footslopes as compared to the shoulders, but no landform effect was found for N recovery from the wheat residue.

The N recovery from these residues (Table 4.10) showed the same spatial pattern as their N contents (kg ha^{-1} N) (Table 4.9). Thus, the observed levels of N recovery were probably related to the N content (kg ha^{-1} N) of the labeled residues. The spatial pattern of N recovery from these residues may also have been attributable to differences in soil conditions in the various landscape positions. Soils in the footslopes inherently have higher organic C contents (Table 4.2), the greater C availability to soil microbes in the footslopes likely would stimulate their activity (Parkin, 1993). The absence of any landform effect on N recovery from the wheat residue was probably related to the absence of any landform effect on the C:N ratio and percentage N content of the labeled wheat residue (Table 4.9).

In September 1997, no difference was evident in the immobilization of N from the chickpea residue and the wheat residue in the shoulders, whereas immobilization of N associated with the wheat residue was higher than that of the chickpea residue in the

footslopes. The patterns of N recovery from the chickpea residue and the wheat residue in the microbial biomass differed, depending on when the samples were collected. For example, a greater quantity of wheat residue N was recovered in the microbial biomass in the shoulders in April than that of chickpea residue N. No difference, however, occurred in the quantity of N recovered from the two residues in the shoulders in September.

Nitrogen recovery of the chickpea residue in the microbial biomass, expressed as a percentage of N in the chickpea residue, was relatively low (Table 4.10) as compared to other reports of N recoveries from various legume residues in the microbial biomass. For example, Stevenson and van Kessel (1997) observed that 51% and 71% of the N from pea residue applied in the previous fall was recovered in the microbial biomass in the spring in the shoulders and footslopes, respectively. The high C:N ratio of the chickpea residue and residue application method (i.e., surface-applied) may have contributed to the relatively low recovery of chickpea residue N in the microbial biomass.

The quantity of residue N recovered in the microbial biomass at harvest in 1997 was three to sixteen times greater than the quantity of residue N recovered in the wheat crop (Tables 4.10 and 4.11), indicating that microbes are an important intermediary in the cycling of crop residue N. Smith and Paul (1990) stated that the soil microbial biomass is both a catalyst and a sink during the decomposition of crop residues. Consequently, temporal fluctuations in the microbial biomass N and N immobilization from residues may have a considerable influence on the quantity of plant-available N from residue decomposition.

4.2.3 Availability of nitrogen from the chickpea residue and the wheat residue to the succeeding crops

Nitrogen derived from the chickpea residue accounted for as little as 1.2 and 2.1% of the N in the wheat crop (i.e., the second phase of rotation) in the shoulders and the footslopes, respectively (Table 4.11). The data suggested that most of N in the wheat crop was derived from the mineralization of SOM. Bremer and van Kessel (1992) and Jensen (1994b) found that approximately 5% of the N accumulated in the first succeeding cereal crop was derived from the residue of the previous lentil or pea crop. Stevenson and van Kessel (1997) similarly observed that approximately 6% of the N accumulated in the succeeding wheat crop was derived from the residue of the previous pea crop. The comparatively low N contribution from the chickpea residue to the wheat crop, as compared to other legume residues, was probably due to limited N mineralization from the chickpea residue, low N content of the chickpea residue, the relatively dry soil, and the residue application approach.

The C:N ratio of residue must be less than approximately 30:1 or the N content must be more than approximately 1.5% in order for net N mineralization to occur (Power et al., 1986). The C:N ratios of the chickpea residue and the wheat residue in the current study were higher than 30:1 and their N contents were lower than 1.5%. Thus, during the early stage of decomposition, soil mineral N would be immobilized and soil N was required for decomposition to proceed. The high C:N ratio of the chickpea residue probably was a limiting factor for its decomposition. Janzen and Kucey (1988) found that the critical value of the C:N ratio required for net mineralization to occur was time-dependent, i.e., a gradual decline occurred in the critical C:N ratio with incubation time.

Table 4.11. Median values and results of the Mann-Whitney U test for N contribution from labeled chickpea residue and wheat residues, and N recovery of labeled residues to the succeeding wheat (straw plus grain) at the experimental site at harvest in 1997.

Landform element complex	Ndfr (%)		Ndfr (kg ha^{-1} N)		Residue N recovery (%)	
	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue
Shoulder	1.2 (1.9)†	1.5 (1.1)	0.20 (0.41)	0.38 (0.21)	2.2 (2.9)	2.1 (3.6)
Footslope	2.1 (1.4)	1.6 (0.6)	0.65 (0.49)	0.31 (0.14)	3.3 (2.7)	1.7 (1.6)
Shoulder vs. footslope‡	0.16	0.71	0.01	0.39	0.11	0.57
Shoulder, chickpea vs. wheat residue§	0.84		0.63		0.76	
Footslope, chickpea vs. wheat residue¶	0.02		< 0.01		< 0.01	

† Values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between chickpea residue and wheat residue in the shoulder.
¶ The *P* value associated with the comparison between chickpea residue and wheat residue in the footslope.

They concluded that over the long term all crop residues would be expected to show significant net mineralization. The time required for net mineralization to occur, however, increases as the initial N concentration decreases.

The proportion of crop N derived from the decomposition of legume residue and mineralization of soil organic N is determined mainly by the relative quantities and mineralization rates of these two N sources. Approximately 12 and 18 kg ha^{-1} N input was available from the labeled chickpea residue in the shoulders and footslopes, respectively (Table 4.9), which is much less than the reported levels of N input from other legume residues. For example, Stevenson and van Kessel (1997) reported that the N contents of labeled pea residue were 49, 70 and 82 kg ha^{-1} in the shoulders, and low-catchment and high-catchment footslope area, respectively. The amount of N in the chickpea residue was very small compared with the soil N pool (Table 4.1). Thus, the relative N contribution from the chickpea residue to the succeeding crop would be low.

Soil moisture influences moisture available to the decomposing organisms, soil aeration status and other soil properties. Soil moisture content for maximum residue decomposition is near field capacity (Paul and Clark, 1996). The relatively droughty conditions experienced during the 1997 growing season (Fig. 3.6) likely decreased the decomposition of residues.

Since a minimal disturbance direct seeding system was practiced in the study field, the labeled residue was applied on the soil surface. Buried straw decomposes faster than the straw placed above or on the soil surface (Christensen, 1986) because soil microbes and residue are in more intimate contact, and conditions are more favorable and stable for microbial activity when residues are buried. Holland and Coleman (1987) in

Colorado found greater N immobilization, slower decomposition and increased fungal abundance in wheat straw on the soil surface than in the buried wheat straw.

The labeled chickpea residue and the labeled wheat residue did not differ in the amount of N recovered (kg ha^{-1} Ndfr) in the wheat crop (i.e., the second phase of the rotation) in the shoulders (Table 4.11). The wheat crop recovered only 0.3 kg ha^{-1} more N from the labeled chickpea residue than from the labeled wheat residue in the footslopes. The data suggested that neither chickpea residue nor wheat residue was an important N source for the succeeding crops. The footslopes had a higher residue N recovery (%) for the chickpea residue than the shoulders, but the landform had no effect on residue N recovery (%) for the wheat residue, probably because the N content and C:N ratio of wheat residue did not differ between the shoulders and the footslopes.

Norman et al. (1990) observed that, generally, the lower the C:N ratio and the higher the amount of N in the residue, the lower was the amount of residue N recovered in the soil organic fraction at harvest and the higher was the amount of residue N mineralized. Although the C:N ratio of the chickpea residue was relatively high as compared to other legume residues, the C:N ratio of the chickpea residue was significantly lower than that of the wheat residue in both the shoulders and the footslopes. Consequently, the N dynamics of chickpea residue and wheat residue would be different. The wheat crop recovered more N from the labeled chickpea residue than from the labeled wheat residue in the footslopes, but not in the shoulders. The data suggested that the expression of the difference in the N dynamics between the chickpea residue and the wheat residue likely was dependent on the soil conditions or landscape positions.

In the third phase of the rotations (i.e., 1998), the labeled chickpea residue applied in 1996 contributed 0.6% and 0.4% of the N to the canola stubble in the shoulders and the footslopes, respectively (Table 4.12). The labeled wheat residue applied in 1996 contributed 1.1% and 0.9% of N to the canola stubble in the shoulders and the footslopes, respectively. For both the labeled chickpea residue and the labeled wheat residue applied in 1996, the N contribution from the labeled residue (i.e., %Ndfr) to the canola stubble in 1998 (Table 4.12) was less than the N contribution from the labeled residues to the wheat crop in 1997 (Table 4.11), indicating that the availability of residue N decreased with time.

Table 4.12. The N contribution (%Ndfr) from labeled chickpea residue and wheat residue (applied in October 1996) to the canola stubble, measured at the experimental site in August 1998: Median values and results of the Mann-Whitney U test.

Landform element complex	Chickpea residue (%Ndfr)	Wheat residue (%Ndfr)
Shoulder	0.6 (0.5)†	1.1 (0.4)
Footslope	0.4 (0.3)	0.9 (0.5)
Shoulder vs. footslope‡	0.30	0.05
Shoulder, chickpea vs. wheat residue§	< 0.01	
Footslope, chickpea vs. wheat residue¶	< 0.01	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the chickpea residue and wheat residue in the shoulder.
¶ The *P* value associated with the comparison between the chickpea residue and wheat residue in the footslope.

The availability of crop residue N to the succeeding crops is largely controlled by the overall outcome of the mineralization-immobilization processes and the stabilization of residue N in SOM pools after incorporation of the residues in the soil (Jansson and Persson, 1982). Even though a considerable amount of crop residue N remained in the soil after the first succeeding year, it might not be readily available to the second succeeding crop. Jensen (1994a) showed that only 1 to 2% of the residual organic ^{15}N was potentially mineralizable after two years of decomposition, indicating that the remaining residue ^{15}N was present in rather recalcitrant SOM fractions. Ladd et al. (1985) found that 31 to 38% of the added legume ^{15}N was still in the organic fraction of the soil eight years after ^{15}N-labeled medic (*Medicago littoralis* cv. Harbinger) material was applied. They concluded that legume residue did not contribute significant portions of N to the first succeeding crop. In contrast, legume residue N increased the pool of soil organic N and contributed to the supply of mineral N by mineralization in the long-term.

4.2.4 Added nitrogen interaction and residue nitrogen recovery

In both the shoulders and the footslopes, the quantity of soil N accumulated (kg ha^{-1} Ndfs) in the wheat crop in 1997 did not differ significantly in grid cells from which labeled chickpea residue had been applied in 1996 as compared to grid cells from which all aboveground residue had been removed in 1996 (Table 4.13). Although the presence or absence of chickpea residue had no effect on the uptake of soil N, landscape position affected uptake of soil N, i.e., more N in the crop was derived from the soil in the footslopes as compared to the shoulders.

Application of labeled wheat residue had little effect on the contribution of soil N to the N accumulated in the wheat crop as compared to grid cells from which wheat residue had been removed in 1996 (Table 4.13). However, the application of wheat residue enhanced the Ndfs in the shoulders. Although wheat crop recovered more soil N in the footslopes as compared to the shoulders in microplots where the residue had been removed, a similar effect was not detected in microplots where wheat residue had been retained.

Table 4.13. Median values for N contribution from soil (Ndfs, kg ha^{-1} N) to the wheat crop (straw + grain) measured at the experimental site at harvest in 1997 and results of the Mann-Whitney U test.

Landform element complex	Ndfs (kg ha^{-1} N)			
	Chickpea residue	No residue	Wheat residue	No residue
Shoulder	18.5(6.5) †	20.2(13.1)	18.5(8.7)	15.6(5.3)
Footslope	32.3(13.0)	28.9(19.7)	21.4(9.9)	23.9(8.6)
Shoulder vs. footslope‡	< 0.01	0.01	0.37	0.01
Shoulder, residue vs. control§	0.58		0.18	
Footslope, residue vs. control¶	0.59		0.45	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the chickpea residue and wheat residue in the shoulder.
¶ The *P* value associated with the comparison between the chickpea residue and wheat residue in the footslope.

The ANI is a comparative measure of the quantity of N contributed from soil where labeled residue was added as compared to a control treatment where residue was

removed. If the ANI is positive, it is assumed that the applied residue increased the availability of soil N. If the ANI is negative, it is assumed that the applied residue decreased the availability of soil N. A negative ANI was detected more frequently in the shoulders and a positive ANI was typically detected in the footslopes where the impact of the chickpea residue was being evaluated. The ANI associated with the chickpea residue in the footslopes was higher than in the shoulders (Table 4.14). For the wheat residue, a positive ANI was detected more frequently in the shoulders and a negative ANI was detected more frequently in the footslopes, and these differences were significant (Table 4.14).

Table 4.14. Median values and results of the Mann-Whitney U test for ANI (kg ha^{-1} N) of labeled residues measured at the experimental site at harvest in 1997.

Landform element complex	Chickpea residue (chickpea-**wheat**)	Wheat residue (wheat-**wheat**)
Shoulder	-1.2 (11.7)†	1.3 (3.8)
Footslope	1.1 (11.3)	-0.8 (8.0)
Shoulder vs. footslope‡	0.17	0.02
Shoulder, chickpea vs. wheat residue§	0.21	
Footslope, chickpea vs. wheat residue¶	0.06	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the chickpea residue and wheat residue in the shoulder.
¶ The *P* value associated with the comparison between the chickpea residue and wheat residue in the footslope.

The difference in ANI between the chickpea residue and the wheat residue in shoulders was not statistically significant, due to the large variability of the data (Table 4.14). The median value of the ANI for chickpea residue were significantly higher than that of the wheat residue in the footslopes.

The magnitude of the ANI (Table 4.14) was small as compared to the N contribution from the soil (Table 4.13), indicating that the influence of the applied chickpea residue and wheat residue was relatively insignificant with respect to subsequent uptake of soil N.

Due to the high C:N ratio of the chickpea residue and the wheat residue (Table 4.9), both residues likely caused net immobilization of soil mineral N during the early stage of decomposition (Power et al., 1986). The availability of immobilized N from residue and soil to the succeeding crops largely depends on the rate of subsequent remineralization and temporal variation of microbial biomass N, which are controlled by the soil and environmental conditions. The spatial variability of soil properties, the size of the soil N pool, and the characteristics of the applied residue may cause diverse degrees of pool substitution and MIT, which are the major processes responsible for ANI occurrence across the landscape. This would have resulted in the large variation in the magnitude of ANI, and the landform effect on the ANI and residue N recovery (Tables 4.11 and 4.14).

Pool substitution is the process by which added labeled N stands proxy for native unlabeled soil N that would otherwise have been removed or immobilized from that pool (Jenkinson et al., 1985). Microbial immobilization of N, driven by the decomposition of SOM or crop residue, can lead to pool substitution and is the

dominant cause of apparent ANI (Jenkinson et al., 1985). Pool substitution, however, can occur only if labeled N and unlabeled N occupy the same pool at the same time (Jenkinson et al., 1985; Hart et al., 1986). The labeled residue was applied on the soil surface simulating the minimal disturbance direct seeding system practiced in the study field. Thus, it is unlikely that strong pool substitution occurred in the field. Since the decomposition of residue applied on the soil surface is slower than the residue incorporated into the soil (Christensen, 1986), only a portion of the N in the applied residues would be available for pool substitution. Consequently, the observed levels of ANI were relatively low (Table 4.14).

The low magnitude of ANI (Table 4.14) can also be attributable to the limited C and N input from applied chickpea residue and wheat residue. Jensen (1994b) observed that the incorporation of ^{15}N-labeled pea residues slightly increased the accumulation of non-labeled soil N in autumn-sown crops by 6% and 2%, when harvested in December and at maturity, respectively, in a field study in Denmark. The ^{15}N-labeled pea residue with an N content of 2.6% and a C:N ratio of 15:1 was applied at the rate of 322 g m^{-2} dry matter and mixed with soil in his study. In contrast, Yaacob and Blair (1980) observed that in a pot experiment after 12 wk, the extractable mineral N in the labeled soybean residue amended pots was three to seven times as much as in the unamended pots. The larger ANI effect in their study likely was due to the high N content and low C:N ratio of the applied residue, the fact that the residue was well mixed with soil, and the more favorable soil moisture and temperature conditions for residue decomposition, as compared to the field experiments.

The ANI and residue N recovery (%) in the 1997 wheat crop were significantly correlated ($r = 0.37$, $P < 0.01$), although the correlation coefficient was not high. The lack of a high correlation suggested that different types and rates of pool substitution and MIT probably occurred across the landscape. Exchange of ^{15}N for ^{14}N during mineralization-immobilization processes can result in low apparent N recovery rates because a portion of the applied ^{15}N was not accessible to the crops (Jansson and Persson, 1982). The ANI can lead to an increase or decrease in the availability of native soil N. Moreover, ANI can conserve applied ^{15}N through MIT, pool substitution and biological exchange reactions (Jenkinson et al., 1985). The quantity of residue ^{15}N recovered in the microbial biomass pool at harvest in 1997 was three to sixteen times greater than the quantity of residue ^{15}N recovered in the 1997 wheat crop, suggesting that a large portion of the released ^{15}N from the decomposition of crop residue was stored in the microbial biomass pool. As a result, the contribution of residue N to the 1997 wheat crop was low (Table 4.11).

The N contribution from labeled residues to the 1998 canola crop may have been from the mineralization of the remaining crop residue and mineralized ^{15}N recovered in the organic N pool at harvest 1997. A portion of the residue ^{15}N recovered in the microbial biomass would be released and become available to the 1998 canola crop due to the subsequent turnover of microbial biomass N. Jensen (1994a) demonstrated that the turnover of microbial biomass ^{15}N was faster than the total residual organic ^{15}N pool. Ladd et al. (1981) also noticed that the microbial biomass ^{15}N pool declined faster than the total residual organic ^{15}N pool.

Although the microbial N pool frequently is considered the labile organic N pool of the SOM (Parton et al., 1987), the N derived from residue in the microbial biomass during the year following the incorporation of residues probably could not be used to predict the availability of N to the succeeding crop because not all of the residue ^{15}N recovered in the microbial biomass pool will be released and become available to the succeeding crops. In fact, most of the N from legume residue is transferred into a slowly mineralizable fraction of SOM (Wagger et al., 1985; Bremer and van Kessel, 1992).

4.2.5 Factors influencing added nitrogen interaction

Soil N is stored in the organic matter fractions. The availability of soil N to plants is controlled by N turnover or exchange of N between organic and inorganic pools, a process mediated by the soil microorganisms. The quantity of soil N mineralized and the temporal pattern of soil N mineralization largely regulate the processes, such as pool substitution and MIT. The magnitude of pool substitution and N availability of residue N are linked and proportional to the quantity of soil N mineralized and added N. The characteristics and quality of SOM are important factors that control the mineralization of soil organic N. Thus, the ANI of incorporated residues will be directly related to the SOM. In an incubation study whereby soybean tops, vetch tops or corn stover were added to Mollisols in Illinois, Azam et al. (1993) reported that both soil type and the nature of the applied residues influenced the occurrence and extent of the ANI. In the current study, the ANI was significantly correlated with the C:N ratio of the soil ($r = 0.38$, $P = 0.02$) and pH ($r = 0.45$, $P < 0.01$).

The ANI was negatively correlated with soil N content ($r = -0.48$, $P < 0.01$) and hot-KCl extractable N ($r = -0.49$, $P < 0.01$). Due to the high C:N ratio of the added chickpea residue and wheat residue, initially high levels of soil mineral N were required to sustain the residue decomposition process. As a result, mineral N, initially present in the soil, was immobilized during the decomposition of freshly added crop residues and was subsequently mineralized to a lesser extent than in unamended soil (i.e., soil in the control treatment). Thus, a negative correlation occurred between the ANI and soil N content. The ANI was not significantly correlated with the C:N ratio of applied residues, probably because residue C:N ratio is not the only indicator of residue quality (Fox et al., 1990).

4.3 Variability of the Light Fraction Organic Matter and the Transfer of Residue Nitrogen into Soil Organic Matter Fractions

4.3.1 Spatial variability of the light fraction related to landform

Landform had no effect on the C content and C:N ratio of the LF (Table 4.15). Although the N content of the LF was higher in the footslopes as compared to the shoulders ($P = 0.09$), the difference in the median value of the N content of the LF was only 0.1%. The C content, N content and the C:N ratio of the bulk soil was higher in the footslopes than in the shoulders. The N content and C:N ratio of the HF was higher in the footslopes as compared to the shoulders, but landform had no effect on the C content of the HF.

The C content of the LF (Table 4.15) was lower than that of crop residues (Table 4.9). Moreover, the C:N ratio of the LF was closer to that of the bulk soil as compared to that of the residues. These results indicated that plant debris in the LF had undergone considerable decomposition and fractionation. The higher N content of the LF as compared to that of the HF or the bulk soil was attributable to the fact that the LF pool includes N-rich components, such as rhizodeposits, microbial products and microbial cell walls (Spycher et al., 1983).

The ranking of the C content, N content and C:N ratio among the LF, HF and bulk soil was LF > bulk soil > HF, suggesting that the LF and HF are two distinct pools of SOM with different characteristics and in different stages of humification. The data indicate that density fractionation can physically separate SOM into fractions differing in composition. Christensen (1992) observed that the density fractionation method

Table 4.15. Median values and results of the Mann-Whitney U test for C content, N content and C:N ratio of LF, HF and bulk soil (0- to 5-cm depth) measured at the experimental site in October 1996.

Landform element complex	n	C content (%)			N content (%)			C:N ratio		
		LF	HF	Soil	LF	HF	Soil	LF	HF	Soil
Shoulder	20	24.8(4.7)†	2.0(0.5)	2.5(0.5)	1.47(0.28)	0.15(0.04)	0.19(0.05)	15.9(3.1)	12.8(3.6)	12.9(3.1)
Footslope	20	24.7(5.3)	2.2(0.7)	2.9(1.4)	1.57(0.21)	0.21(0.08)	0.28(0.13)	15.1(1.2)	10.5(1.3)	10.8(0.8)
Shoulder vs. footslope‡		0.39	0.30	0.02	0.09	0.04	< 0.01	0.48	< 0.01	< 0.01

† The values in the parentheses are IQR.

‡ The *P* value associated with the comparison between the shoulder and the footslope.

isolated specific SOM constituents with high turnover rates and physically divided SOM into pools differing in composition and biological functions.

Variability in the LF content, expressed as a percentage of soil weight, was higher (i.e., wider interquartile range and higher index of variability) in the footslopes as compared to the shoulders (Table 4.16). The distribution of the LF content (%) in the shoulders was skewed. The LF content (%) was significantly higher in the footslopes than in the shoulders (P = 0.17). When the quantities of C and N in the LF were expressed as kg ha^{-1}, the values, however, were not significantly higher in the footslopes as compared to the shoulders. One possible explanation is that soil bulk density in the footslopes was lower as compared to the shoulders.

Data on LF content (%), LF N (kg ha^{-1}) and LF C (kg ha^{-1}), especially for the footslopes, were highly variable (Table 4.16). The high variability for the LF content (%) was due to the spatial variability of the soil across the landscape. The magnitude of the variability in the LF N content and LF C content reflects the spatial variability of various soil properties. Overall, LF N and LF C were useful measures of N and C in the LF.

Janzen et al. (1992) observed that much of the LF in agricultural soils is derived from plant residues. Boone (1994) discovered that aboveground litter is a major source of the LF in mull forest soils. Crop residue input was higher in the footslopes than in the shoulders as a consequence of higher residue yields of crop (Table 4.9). According to Parkin (1993), the higher soil moisture and higher C and N in the footslopes, as observed in the current study (Table 4.2), are likely to enhance microbial growth, further facilitating the decomposition of crop residue and increasing the input from the

Table 4.16. Descriptive statistics for LF content, and the N and C content of the LF (0- to 5-cm depth) measured at the experimental site in October 1996, and results of the Mann-Whitney U test related to landform element complexes.

Landform element complex	n	Median	Mean	Min.	Max.	IQR	Index of variability (%)†	Skewness
			LF content (%)					
Shoulder	20	0.95	1.07	0.80	1.64	0.42	44	1.09
Footslope	20	1.20	1.28	0.48	2.37	0.78	65	0.35
Shoulder vs. footslope‡		0.17						
			LF N content (kg ha^{-1})					
Shoulder	20	70.7	74.6	49.2	125.1	22.1	31	1.28
Footslope	20	78.7	83.5	22.3	146.0	67.1	85	0.14
Shoulder vs. footslope		0.43						
			LF C content (kg ha^{-1})					
Shoulder	20	1081	1219	766	2225	448	41	1.15
Footslope	20	1207	1324	310	2519	824	68	0.45
Shoulder vs. footslope		0.74						

† Index of variability = (IQR/ median) × 100.
‡ The *P* value associated with the comparison of the median between the shoulder and the footslope.

crop residue to the LF pool. As a consequence, the LF content (%) was higher in the footslopes as compared to the shoulders. The size of the LF pool, however, is a balance between residue input and decomposition, both of which are controlled by the soil and environmental conditions (Biederbeck et al., 1994). The conditions favorable for residue decomposition should also be favorable for the decomposition of the LF and subsequent transfer of LF organic matter into other SOM fractions. The LF is not physically protected by soil particles (Spycher et al., 1983; Strickland and Sollins, 1987) and is readily available to soil microbes. This may be the reason why landform had no effect on the quantity of C and N in the LF when they were expressed as kg ha^{-1}. Alternatively, variability in the data, including bulk density estimation, may have obscured real differences.

4.3.2 Temporal variability of the light fraction

In the chickpea-wheat rotation, the LF content (%) was highest in June as compared to other sampling dates in both the shoulders and the footslopes (Table 4.17). The high LF content in June may be attributable to the temporal variation of precipitation (or soil moisture content) and temperature in the field. Bremer and van Kessel (1992) observed that net mineralization of labeled lentil residue and labeled wheat residue increased rapidly from May to June and remained quite constant for the remainder of the growing season. They related the rapid mineralization of crop residues in June to the increase in soil temperature from April to June and adequate precipitation in June, which apparently activated the soil microorganisms. The enhanced mineralization of crop residues likely increased the input from residue to the LF pool in the current study.

Table 4.17. Temporal variation for LF content (%) (0- to 5-cm depth) measured at the experimental site at different sampling dates: Median values and results of the Kruskal-Wallis H test related to the sampling dates.

Rotation	Landform element complex	Oct. 96	April 97	June 97	Sept. 97	Sensitivity index†
Chickpea-wheat	Shoulder	0.92b‡ (0.21)§	1.05ab (0.40)	1.34a (0.73)	0.92b (0.35)	0.45
	Footslope	1.27ab (1.05)	1.20ab (0.53)	1.52a (1.11)	1.20b (0.39)	0.27
Wheat-wheat	Shoulder	1.16a (0.46)	0.89b (0.25)	1.15a (0.38)	1.06a (0.65)	0.30
	Footslope	1.18a (0.87)	1.16a (0.67)	1.23a (0.43)	1.16a (0.54)	0.06

† Sensitivity index = (highest – lowest) / lowest.

‡ Median values in the same row followed by the same letter are not significantly different and no comparison is available within columns.

§ The values in the parentheses are IQR.

Spycher et al. (1983) also observed that LF concentrations were highest in June in the uppermost two layers (i.e., 0 to 5 cm and 5 to 10 cm) and declined during late summer in a forested site.

In the wheat-wheat rotation, the LF content in June did not differ from that of other sampling dates in either the shoulders or the footslopes (Table 4.17). The data suggest that the transfer of wheat residue into the LF pool was not as much as the transfer of chickpea residue into the LF pool. The lower quantity of the transfer of wheat residue into the LF pool likely suggested lower decomposition rates of the wheat residue as compared to the chickpea residue. Thus, residue composition likely is an important factor controlling residue decomposition and its transfer into SOM fractions.

Although the C content of the LF was highest in April in both rotations and in both the shoulders and the footslopes, the C content of the bulk soil in April was not significantly different from the C content of the bulk soil at most other sampling dates (Table 4.18). The sensitivity index for LF C content was two to three times higher than that for soil C content. Similarly, the C:N ratio of the LF was highest in April in both rotations, and in both the shoulders and the footslopes, but the C:N ratio of the bulk soil was not affected by sampling date, rotations, or landforms (Table 4.19). Moreover, the C:N ratio of the LF was wider than that of the bulk soil at all four sampling dates. The finding that the C:N ratio of the LF was wider than that of the bulk soil and the sensitivity index of the C:N ratio of the LF was higher than that of the bulk soil suggests that crop residue inputs had their greatest impact on the LF pool and least effect on total SOM. Hassink (1995) also found that the effect of the residue input on the C:N ratio

Table 4.18. Temporal variability for C content of LF and bulk soil (0- to 5-cm depth) measured at the experimental site at different sampling dates: Median values and results of the Kruskal-Wallis H test related to the sampling dates.

Rotation	Landform element complex	LF C content (%)					Soil C content (%)				
		Oct. 96	April 97	June 97	Sept. 97	Sensitivity index†	Oct. 96	April 97	June 97	Sept. 97	Sensitivity index
Chickpea-wheat	Shoulder	22.4b‡ (4.3)§	25.8a (3.7)	23.9a (5.0)	21.8c (2.1)	0.18	2.49a (0.39)	2.49a (0.68)	2.44a (0.49)	2.59a (0.54)	0.06
	Footslope	25.2c (5.1)	28.8a (7.5)	27.8b (5.8)	24.0c (3.8)	0.20	3.07a (1.54)	3.12a (1.59)	3.29a (1.34)	3.07a (1.01)	0.07
Wheat-wheat	Shoulder	25.1a (4.9)	25.3a (2.3)	22.9b (2.2)	19.9b (4.3)	0.27	2.43a (0.73)	2.24b (0.51)	2.26b (0.56)	2.29ab (0.62)	0.08
	Footslope	24.5b (5.2)	25.6a (5.0)	24.2b (5.9)	22.8b (3.7)	0.12	2.83a (0.97)	2.68a (1.07)	2.29a (0.83)	2.68a (0.67)	0.06

† Sensitivity index = (highest – lowest) / lowest.

‡ Median values of LF C content (%) or soil C content (%) in the same row followed by the same letter are not significantly different and no comparison is available within columns.

§ The values in the parentheses are IQR.

Table 4.19. Temporal variability of C:N ratio for the LF and bulk soil (0- to 5-cm depth) measured at the experimental site at different sampling dates: Median values and results of the Kruskal-Wallis H test related to the sampling dates.

Rotation	Landform element complex	C:N ratio (LF)					C:N ratio (Soil)				
		Oct. 96	April 97	June 97	Sept. 97	Sensitivity index†	Oct. 96	April 97	June 97	Sept. 97	Sensitivity index
Chickpea-wheat	Shoulder	15.3b‡ (2.4)§	18.1a (4.3)	17.4a (5.2)	14.8b (1.7)	0.22	14.2a (3.3)	13.8a (3.6)	13.9a (3.3)	13.0a (3.1)	0.09
	Footslope	14.9c (1.8)	18.6a (3.2)	16.5b (2.7)	14.9c (1.1)	0.25	11.0a (0.8)	10.9a (0.8)	10.7a (0.7)	10.4a (1.1)	0.06
Wheat-wheat	Shoulder	16.9a (3.7)	17.3a (5.4)	17.4a (2.9)	15.6b (1.8)	0.12	12.6a (2.5)	11.8a (2.2)	11.9a (1.6)	11.9a (1.9)	0.07
	Footslope	15.3b (1.5)	17.7a (4.6)	16.1ab (3.7)	15.0b (1.9)	0.18	10.7a (0.5)	10.6a (0.9)	10.4a (0.5)	10.3a (1.0)	0.04

† Sensitivity index = (highest – lowest) / lowest.

‡ Median values of C:N ratio of LF or bulk soil in the same row followeded by the same letter are not significantly different and no comparison is available within columns.

§ The values in the parentheses are IQR.

was greater for the LF than for the intermediate and the HF, whereas the C:N ratio of non-macroorganic matter was hardly affected by residue input.

The larger sensitivity index of the LF C content and C:N ratio of the LF, as compared to the C content and C:N ratio of the bulk soil (Tables 4.18 and 4.19), also indicates that the LF was more sensitive to residue input and that the temporal fluctuations in the C of the SOM occur mainly in the labile SOM fractions. Thus, the LF may provide an earlier indication of the magnitude of the effects of soil management and cropping systems on soil quality than total organic matter in the soil, as was suggested by Janzen et al. (1992) and Bremer et al. (1994). Biederbeck et al. (1994) argued that it may be possible to manipulate the timing of residue inputs and moisture through cropping practices and thereby maintain adequate labile SOM concentrations and improve the synchrony of mineralization with crop requirements.

In the chickpea-wheat rotation, the LF C as a percentage of the soil C and the LF N as a percentage of the soil N had the highest value in June in both the shoulders and the footslopes (Table 4.20). The temporal trend of the LF C as a percentage of the soil C and the LF N as a percentage of soil N coincided with the temporal trend of the LF content (Table 4.17), i.e., the LF content was highest in June. The highest value of the LF C as a percentage of the soil C and the LF N as a percentage of the soil-N in June (Table 4.20) was due to the highest LF content in June. This observation likely was related to the enhanced mineralization of chickpea residue at this time due to soil moisture and temperature conditions favorable for residue decomposition. In contrast, in the wheat-wheat rotation, the LF C as a percentage of the soil C and the LF N as a percentage of the soil N did not differ among the various sampling dates in either the

Table 4.20. Median values for LF C or LF N as a percentage of soil C or soil N (0- to 5-cm depth) measured at the experimental site at different sampling dates and results of the Mann-Whitney U test related to landform element complexes and results of the Kruskal-Wallis H test related to the sampling dates.

Rotation	Landform element complex	Oct. 96	April 97	June 97	Sept. 97
		LF C as % of soil C			
Chickpea-wheat	Shoulder	8.5b†	10.7a	11.6a	7.9b
		(1.9)‡	(5.7)	(5.4)	(3.6)
	Footslope	8.9b	11.3a	12.9a	9.2b
		(5.8)	(7.2)	(8.7)	(2.9)
	Shoulder vs. footslope§	0.55	0.33	0.36	0.36
Wheat-wheat	Shoulder	11.8a	10.9a	11.7a	9.5ab
		(5.5)	(2.7)	(4.3)	(3.9)
	Footslope	9.7a	13.1a	11.4a	10.7a
		(4.6)	(6.4)	(3.2)	(3.3)
	Shoulder vs. footslope	0.29	0.19	0.65	0.82
		LF N as % of soil N			
Chickpea-wheat	Shoulder	7.9ab	8.4b	9.9a	7.6b
		(2.3)	(2.6)	(4.3)	(3.5)
	Footslope	6.0ab	6.9ab	8.3a	7.1ab
		(4.4)	(2.9)	(6.2)	(2.5)
	Shoulder vs. footslope	0.19	0.45	0.76	0.49
Wheat-wheat	Shoulder	8.0b	6.9c	8.1ab	8.4ab
		(3.9)	(3.1)	(3.8)	(3.6)
	Footslope	7.6bc	7.4c	7.2bc	8.1ab
		(3.5)	(1.2)	(3.6)	(3.0)
	Shoulder vs. footslope	0.29	0.71	0.29	0.41

† Median values in the same row followed by the same letter are not significantly different and no comparison is available within columns.

‡ The values in the parentheses are IQR.

§ The *P* value associated with the comparison between the shoulder and the footslope (i.e., within columns).

shoulders or the footslopes. This trend was similar with the temporal trend of the LF content (Table 4.17).

Landform had no effect on the LF C as a percentage of the soil C and the LF N as a percentage of the soil N, irrespective of the crop rotations or sampling dates (Table 4.20). The quantity of the C and N in the LF pool and the bulk soil will be at equilibrium if inputs and outputs occur at the same rate. The C and N inputs and outputs to the LF and the SOM are controlled mainly by the soil and environmental conditions. It is likely that because the decomposition of the LF and SOM proceeded under identical soil and environmental conditions in a specific landscape position or sampling unit, i.e., under the conditions favorable for the decomposition of the LF, the conditions also were favorable for the decomposition of SOM. As a consequence, landform had no effect on the LF C as a percentage of the soil C or the LF N as a percentage of the soil N.

4.3.3 Vertical variability of the light fraction

The LF content decreased abruptly below the 5-cm depth in both the shoulders and the footslopes (Table 4.21), then much more gradually over the remainder of the 60-cm soil profile (Table 4.21). The LF content in the 5- to 15-cm soil layer was less than 10% of the LF content in the 0- to 5-cm soil layer. The LF N expressed as a percentage of soil N and the LF C expressed as a percentage of the soil C had a vertical distribution that was similar to that of the LF content. The data suggest that the LF was concentrated in the top soil layer in the minimum tillage field, likely because all the crop residues had been retained on the soil surface and had not been mechanically incorporated. Hassink

Table 4.21. Vertical distribution for LF content (%), LF N as a percentage of soil N and LF C as a percentage of soil C, measured in the chickpea-wheat rotation strips at the experimental site in September 1997.

Depth (cm)	LF content (%)		LF N as % of soil N		LF C as % of soil C	
	Shoulder	Footslope	Shoulder	Footslope	Shoulder	Footslope
0- 5	0.92(0.35)†	1.20(0.39)	7.6(3.5)	7.1(2.5)	7.9(3.6)	9.2(2.9)
5-15	0.08(0.04)	0.07(0.12)	1.6(0.7)	0.9(0.8)	1.0(0.6)	0.8(0.9)
15-30	0.06(0.05)	0.04(0.03)	1.5(1.6)	0.5(0.3)	0.7(0.6)	0.6(0.5)
30-60	0.05(0.06)	0.02(0.02)	1.5(1.5)	0.5(0.7)	0.6(1.1)	0.5(0.8)

† The values in the parentheses are IQR.

(1995) and Janzen et al. (1992) also observed that the percentage of organic C and N in the LF declined with increasing soil depth in agricultural soils in The Netherlands and Canada, respectively.

Soluble organic matter can be adsorbed on the mineral surfaces, immobilized by the microbial biomass or leached (Swift et al., 1979; Tiessen et al., 1984). Residue inputs are less important to the LF pool and the percolation of dissolved organics downward is probably more important to the LF pool below the top soil layer. Meanwhile, turnover of roots and microbial tissues probably represents an important source of organic matter for the LF pool below the top soil layer. Typically, the quantities of crop roots (van Rees et al., 1994) and soil microbial organisms (Alexander, 1977; Stevenson and van Kessel, 1997) decrease with soil depth in the soil profile. Thus, the declining LF content with increasing depth probably reflected the distribution of root and microbial debris within the soil profile. Spycher et al. (1983) found that, although, much of the LF was derived from the crop residue, the LF also contained appreciable amounts of microbial and microfaunal debris, including fungal hyphae and spores. Ladd et al. (1977) observed that fumigation of soils resulted in a significant decrease in the N content of the LF, suggesting that soil microbial biomass contributed significantly to this fraction. Meanwhile, some material adsorbed in the HF pool can be transferred to the LF pool as a result of microbial activity and transformation (Spycher et al., 1983; Janzen et al., 1992).

4.3.4 Transfer of residue nitrogen into soil organic matter fractions

Crop residues incorporated into the soil must pass through the soil microbial biomass which partly mineralizes residues and partly converts the residues into new products (van Veen et al., 1984). The residue C and residue N remaining in the soil is gradually transferred from the labile pools to the more stabilized pools (Hassink and Dalenberg, 1996). In the early spring (i.e., April) following the application of labeled residues, a significant portion of the chickpea residue N and the wheat residue N existed in the LF pools and the HF pools (Tables 4.22 and 4.23). The data suggest that the transfer of residue N into the LF pools and the HF pools occurred rapidly for both the chickpea residues and the wheat residues, irrespective of the fact that both the chickpea residue and the wheat residues had relatively high C:N ratios. In an incubation study of added ^{14}C-labeled rye shoot material to the soil, Hassink and Dalenberg (1996) similarly observed that two days after application, a significant portion of the ^{14}C label was present in the soluble (26 to 28%) and light macro-organic matter fractions (31 to 32%). They also observed that the residue C was transferred from the light macroorganic matter and soluble organic matter fractions and accumulated in microaggregates. It is generally accepted that most soil organic matter is finally protected by its association with clay and silt particles and by its location in microaggregates (Tisdall and Oades, 1982; Skjemstad et al., 1993; Golchin et al., 1994). In addition, Strickland et al. (1992) observed that the binding of organics to mineral particles (leading to the formation of heavy material) can take place rapidly.

The transfer of chickpea residue N and wheat residue N into the LF pool was greatest in June in both rotations, irrespective of the landscape position (Table 4.22). In the chickpea-wheat rotation, the recovery of chickpea residue N in the HF pool (expressed

Table 4.22. Median values for ^{15}N recovery of labeled residues expressed as kg ha^{-1} N and percentage of residue N in the LF pool (0- to 5-cm depth) measured at the experimental site in April, June and September 1997, and results of the Mann-Whitney U test related to landform element complexes and residues.

Landform element complex	April 97				June 97				Sept. 97			
	kg ha^{-1} N		%		kg ha^{-1} N		%		kg ha^{-1} N		%	
	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue
Shoulder	1.41 (1.30)†	2.48 (2.86)	15.8 (25.8)	17.2 (24.4)	2.47 (3.90)	3.65 (3.22)	33.5 (23.3)	29.1 (31.5)	1.29 (1.43)	2.78 (0.98)	13.2 (27.1)	21.7 (16.4)
Footslope	2.83 (6.14)	2.72 (1.59)	23.8 (39.6)	20.2 (13.0)	5.34 (4.11)	4.24 (4.00)	40.6 (33.3)	28.9 (12.5)	3.21 (4.33)	2.98 (2.62)	29.1 (15.6)	24.6 (19.5)
Shoulder vs. footslope‡	0.05	0.65	0.26	0.71	0.29	0.33	0.33	0.88	0.03	0.37	0.22	0.87
Shoulder, chickpea vs. wheat residue§	0.36		1.00		0.86		0.65		< 0.01		0.19	
Footslope, chickpea vs. wheat residue¶	0.59		0.26		0.94		0.09		0.68		0.62	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the chickpea residue and wheat residue in the shoulder.
¶ The *P* value associated with the comparison between the chickpea residue and wheat residue in the footslope.

Table 4.23. Median values for ^{15}N recovery of labeled residues in HF pool (0- to 5-cm depth) expressed as kg ha^{-1} N and percentage of residue N measured at the experimental site in April, June and September 1997, and results of the Mann-Whitney U test related to landform element complexes and residues.

Landform element complex	April 97				June 97				Sept. 97			
	kg ha^{-1} N		%		kg ha^{-1} N		%		kg ha^{-1} N		%	
	Chickpea residue	Wheat Residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue	Chickpea residue	Wheat residue
Shoulder	2.56 (1.21)†	2.25 (2.14)	28.8 (14.7)	17.3 (19.8)	3.33 (3.68)	4.36 (4.08)	30.3 (37.5)	30.3 (39.7)	2.51 (2.67)	5.71 (1.64)	29.9 (51.9)	37.7 (33.5)
Footslope	5.59 (3.19)	2.74 (2.43)	47.6 (29.8)	19.3 (14.8)	4.29 (4.79)	4.31 (2.37)	46.9 (25.2)	26.6 (12.9)	5.21 (3.65)	5.95 (4.39)	43.9 (28.5)	32.8 (27.8)
Shoulder vs. footslope‡	< 0.01	0.17	0.02	0.59	0.11	0.89	0.39	0.49	0.02	0.92	0.37	0.29
Shoulder, chickpea vs. wheat residue§	0.59		0.04		0.59		0.73		< 0.01		0.33	
Footslope, chickpea vs. wheat residue¶	< 0.01		< 0.01		0.37		0.06		0.77		0.39	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the chickpea residue and wheat residue in the shoulder.
¶ The *P* value associated with the comparison between the chickpea residue and wheat residue in the footslope.

as kg ha^{-1} N) was higher in the footslopes than in the shoulders (Table 4.23). The temporal trend of the transfer of chickpea residue N into the HF pool was similar to the transfer into the LF pool. However, the temporal trend of the transfer of wheat residue N into the HF pool differed from that of chickpea residue N (Table 4.23), i.e., the N recovery of wheat residue in the HF pool had the highest value in September. Moreover, the quantity of wheat residue N recovered in the HF pool (kg ha^{-1} N) was less than that of chickpea residue N in April. These data suggest that the decomposition and N dynamics of chickpea residue and wheat residue were variable, likely due to the difference in the composition between the two residues (Table 4.9). Norman et al. (1990) found that the lower the C:N ratio and the higher the quantity of N in the residue, the lower was the quantity of residue N recovered in the soil organic fractions at harvest and the higher was the quantity of residue N mineralized. Ladd et al. (1977) also observed that the nature of the ^{15}N-labeled C amendment influenced both the extent to which N of soil fractions became labeled and the amount of ^{15}N recovered in the SOM fractions with time.

The temporal trend of residue N transfer into the LF pool was similar to that of the LF content (%) (Table 4.17), especially in the chickpea-wheat rotation. Thus, it is likely that the highest LF pool in June was attributable to the highest rate of transfer of chickpea residue N into the LF pool at that time. The temporal variation of the LF content (%) and residue N transfer into the LF pool indicates that the LF pool is a short-term reservoir of plant nutrients, due to its high turnover rate. Paul and Juma (1981) observed that the LF content (%) in the soil at any time is small due to its rapid turnover.

In September, residue ^{15}N recovered in the LF was lower than that in June (Table 4.22), likely due to the high turnover rates of the LF after June during which high temperature facilitated decomposition of the LF. The decline in the LF N content and transfer of the residue ^{15}N into the LF from June to September also can be explained by the transfer of N from the LF to other fractions such as the HF, microbial biomass N and mineral N pool. Changes in the quantity of ^{15}N in a specific fraction are the result of decay of ^{15}N in this fraction and the supply of ^{15}N to this fraction from other fractions. Consequently, the gross transfer of residue N into the LF and the transfer of ^{15}N between SOM fractions may be larger than the measured net change of ^{15}N in a specific fraction. The cycling of residue ^{15}N among SOM fractions could occur more than once during the study period.

Due to the relatively high C:N ratio of the chickpea residue and the wheat residue (Table 4.9), net immobilization likely occurred during the early stages of residue decomposition as discussed in Section 4.2.3. A significant portion of the residue N was transferred into the LF (Table 4.22), HF (Table 4.23) and microbial biomass pools (Table 4.10) in the early spring after the fall application of labeled residues, suggesting that residue decomposition and transfer of residue N into SOM fractions still proceeded during the net immobilization phase. McGill et al. (1975) demonstrated that the transfer of N through different particle size fractions occurred during net N immobilization phases of residue decomposition. Ladd et al. (1977) similarly observed that added ^{15}N became distributed among all soil fractions during a net immobilization phase.

In the chickpea-wheat rotation, the quantity of chickpea residue N (kg ha^{-1}) recovered in the LF pool and the HF pool was higher in the footslopes than in the shoulders at

different sampling dates (Tables 4.22 and 4.23). However, landform had no effect on the N recovery of chickpea residue in the LF pool and the HF pool, when the recovery was expressed as a percentage of N in the applied residues (Tables 4.22 and 4.23). Thus, the greater quantity of chickpea residue N recovered in the LF pool and the HF pool (expressed as kg ha^{-1}) in the footslopes than in the shoulders in the chickpea-wheat rotation was largely attributable to the higher residue N input in the footslopes (Table 4.9). In the wheat-wheat rotation, landform had no effect on the transfer of wheat residue N into the LF pool and the HF pool, probably because landform had no effect on the C:N ratio of wheat residue or the N input from the wheat residue (Table 4.9).

4.3.5 Availability of nitrogen from soil organic matter fractions

The quantity of NH_4^+ extracted by the hot KCl from the LF, HF and bulk soil was used as an index of potentially mineralizable N. Anaerobic and aerobic incubation approaches have been used to estimate the potentially mineralizable N from SOM fractions (e.g., Sollins et al., 1984; Boone, 1994; Motavalli et al., 1995; Barrios et al., 1996). Differences in conditions and duration of incubation used in these studies, however, preclude comparisons among studies. In addition, in many of these studies, the LF was separated from the bulk soil using a NaI solution and it is likely that the residual NaI in the fractions adversely affected the community of soil microbes, thus influencing the incubation results (Sollins et al., 1984). In contrast, the hot-KCl extraction is a relatively simple and reproducible approach to estimate the mineralization potential of the soils (Jalil et al., 1996). This method, however, has not been used to estimate N availability of SOM fractions.

At all four sampling dates, hot-KCl extractable N per kg of LF mass was approximately 10 or 12 times higher than the hot-KCl extractable N per kg of bulk soil or per kg of HF mass, respectively (Table 4.24). The ranking of hot-KCl extractable N among the LF, HF and bulk soil was similar to the ranking of C, N content and C:N ratio among the LF, HF and bulk soil (Tables 4.15 and 4.24), i.e., LF > bulk soil > HF. The LF generally is free of mineral particles and, therefore, lacks the protection from decomposition that such particles impart. Thus, the LF decomposes more rapidly as compared to organic matter in the bulk soil or associated with mineral particle fractions, despite having a wider C:N ratio (Sollins et al., 1984; Bonde et al., 1992). The physical protection of soil organic matter and microbial biomass may occur via (1) adsorption of organics to the surfaces of clays or coating of organics by clay particles (Tisdall and Oades, 1982); and (2) entrapment of organics in small pores in microaggregates inaccessible to microorganisms (Elliott and Coleman, 1988).

The quantity of hot-KCl extractable N from the LF, HF and bulk soil was negatively correlated with the C:N ratio of the LF ($r = -0.45$, $P < 0.01$), HF ($r = -0.34$, $P = 0.03$) and soil ($r = -0.57$, $P < 0.01$), as determined in October 1996. Similar correlation between the quantity of hot-KCl extractable N and the C:N ratio also existed at other sampling dates. Since the fresh plant detritus accumulates mainly on separate and non-mineral particles, the turnover rate is regulated mainly by its chemical composition (Waid, 1974; Staaf, 1980).

Not surprisingly, the hot-KCl extractable N from the soil was positively correlated with LF content at all four sampling dates, i.e., $r = 0.56$ ($P < 0.01$) in October 1996, $r = 0.59$ ($P < 0.01$) in April 1997, $r = 0.87$ ($P < 0.01$) in June 1997, and $r = 0.62$ ($P < 0.01$)

Table 4.24. Median values for hot-KCl extractable N in the LF, HF and bulk soil (0- to 5-cm depth) expressed as mg per kg N in the LF, HF and soil measured at the experimental site at different sampling dates, and results of the Mann-Whitney U test related to landform element complexes and crop rotations.

Sampling date	Landform element complex	mg kg^{-1} N (LF)		mg kg^{-1} N (HF)		mg kg^{-1} N (soil)	
		Chickpea -wheat	Wheat -wheat	Chickpea -wheat	Wheat -wheat	Chickpea -wheat	Wheat -wheat
Oct. 96	Shoulder	111	119	7.7	8.1	9.3	9.5
	Footslope	116	109	12.8	9.3	14.4	11.2
	Shoulder vs.footslope†	0.29	0.76	<0.01	0.02	<0.01	0.13
	Shoulder, chickpea vs. wheat‡	0.33		0.76		0.24	
	Footslope, chickpea vs. wheat§	0.45		0.15		0.13	
April 97	Shoulder	98	103	7.8	8.1	9.2	9.6
	Footslope	114	94	12.4	11.6	14.2	13.6
	Shoulder vs.footslope	0.29	0.94	0.01	0.01	0.01	0.02
	Shoulder, chickpea vs. wheat	0.71		0.49		0.65	
	Footslope,chickpea vs.wheat	0.09		0.23		0.55	
June 97	Shoulder	102	95	9.6	8.7	11.7	10.4
	Footslope	117	99	12.9	11.7	15.1	14.5
	Shoulder vs.footslope	0.19	0.71	<0.01	0.03	0.02	0.09
	Shoulder, chickpea vs. wheat	0.59		0.49		0.41	
	Footslope, chickpea vs. wheat	0.17		0.88		0.26	
Sept. 97	Shoulder	109	96	6.3	8.9	7.6	10.5
	Footslope	106	102	10.7	9.8	12.2	11.9
	Shoulder vs.footslope	0.41	0.33	<0.01	0.01	<0.01	0.19
	Shoulder, chickpea vs. wheat	0.01		0.23		0.02	
	Footslope, chickpea vs. wheat	0.33		0.04		0.57	

† The *P* value associated with comparison between the shoulder and the footslope.
‡ The *P* value associated with comparison between the two rotations in the shoulder.
§ The *P* value associated with comparison between the two rotations in the footslope.

in September 1999, respectively. These data suggest that the LF is an important contributor to total soil N mineralization and an important pool for plant available nutrients. Janzen et al. (1992) similarly observed that the LF content of soils was strongly correlated to soil respiration rates, suggesting that the LF may be an important C and energy source for soil microorganisms, but that the correlation between the LF and N mineralization was not strong nor as consistent as that with respiration.

The hot-KCl extractable N (kg ha^{-1}) derived from the LF was lower than that from the HF or the bulk soil (Table 4.25). When the hot-KCl extractable N from the LF was expressed as a percentage of hot-KCl extractable N from the bulk soil, the percentage was approximately 10% (Table 4.26). Boone (1994) similarly found that the LF represented 11% of the N mineralization potential (via anaerobic incubation) in a corn field, 13% in a pine field, and 2% in a maple field for the whole mineral soil. These data suggest that, although the LF is relatively labile, it was not the primary N source for plant available N because it represents only a small proportion of the SOM (Tables 4.20 and 4.21). A substantial part of the N extracted from the soil must have originated from dying microbial biomass and more stabilized organic matter fractions, e.g., the HF. The hot-KCl extractable N from the HF (kg ha^{-1}) was greater than that from the LF (Table 4.25), suggesting that the HF is not just a reservoir of older and recalcitrant SOM. Like SOM, the organic matter in the HF may not be homogenous. Elliott and Cambardella (1991) noticed that organic matter associated with clay and silt particles and micro aggregates can be a heterogeneous pool of SOM. Organics can form different layers around clay and silt particles and aggregates, and organics on external layers are less protected against decomposition than organics on internal layers (Skjemstad et al.,

Table 4.25. Median values for hot-KCl extractable N in the LF, HF and bulk soil (0- to 5-cm depth) expressed as kg ha^{-1} N measured at the experimental site at different sampling dates, and results of the Mann-Whitney U test related to landform element complexes and crop rotations.

Sampling date	Landform element complex	kg ha^{-1} N (LF)		kg ha^{-1} N (HF)		kg ha^{-1} N (soil)	
		Chickpea -wheat	Wheat -wheat	Chickpea -wheat	Wheat -wheat	Chickpea -wheat	Wheat -wheat
Oct. 96	Shoulder	0.49	0.59	3.69	3.29	4.39	4.41
	Footslope	0.56	0.57	4.46	4.14	5.83	5.65
	Shoulder vs.footslope†	0.29	0.88	0.05	0.01	0.03	0.08
	Shoulder, chickpea vs. wheat‡	0.07		0.08		0.71	
	Footslope, chickpea vs. wheat§	0.94		0.82		0.76	
April 97	Shoulder	0.49	0.36	3.74	3.08	4.57	4.11
	Footslope	0.51	0.54	4.19	4.78	5.59	6.55
	Shoulder vs. footslope	0.41	0.01	0.05	< 0.01	0.17	0.01
	Shoulder, chickpea vs. wheat	0.06		0.13		0.41	
	Footslope,chickpea vs. wheat	0.88		1.00		0.36	
June 97	Shoulder	0.55	0.48	4.73	3.31	5.70	4.53
	Footslope	0.62	0.55	4.57	4.31	6.09	6.03
	Shoulder vs. footslope	0.26	0.36	0.09	0.03	0.59	0.03
	Shoulder, chickpea vs. wheat	0.82		0.76		0.23	
	Footslope, chickpea vs. wheat	0.29		0.29		0.65	
Sept. 97	Shoulder	0.48	0.39	3.23	3.28	3.83	4.32
	Footslope	0.51	0.46	3.51	4.14	4.65	5.73
	Shoulder vs. footslope	0.76	0.41	0.03	0.02	0.07	0.15
	Shoulder, chickpea vs. wheat	0.49		0.23		0.11	
	Footslope, chickpea vs. wheat	0.94		0.29		0.26	

† The *P* value associated with comparison between the shoulder and the footslope.
‡ The *P* value associated with the comparison between the two rotations in the shoulder.
§ The *P* value associated with the comparison between the two rotations in the footslope.

Table 4.26. Median values for hot-KCl extractable N in the LF expressed as a percentage of hot-KCl extractable N in the soil, and results of the Mann-Whitney U test related to landform element complexes and crop rotations.

Landform element complex	Oct. 96		April 97		June 97		Sept. 97	
	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat	Chickpea -wheat	Wheat -wheat
Shoulder	11.2(2.5)†	13.3(3.6)	10.1(2.5)	9.2(3.0)	10.6(3.7)	11.7(3.4)	12.9(3.9)	9.9(3.8)
Footslope	9.4(5.8)	10.8(4.6)	9.5(3.1)	7.7(3.4)	11.0(4.3)	9.4(1.7)	10.0(4.7)	8.5(3.9)
Shoulder vs. footslope‡	0.17	0.08	0.45	0.36	0.36	0.17	0.05	0.62
Shoulder, chickpea-wheat vs. wheat-wheat§	0.04		0.23		0.55		0.01	
Footslope, chickpea-wheat vs. wheat-wheat¶	0.55		0.09		0.08		0.74	

† The values in the parentheses are IQR.
‡ The *P* associated with the comparison between the shoulder and the footslope.
§ The *P* associated with the comparison between the two rotations in the shoulder.
¶ The *P* associated with the comparison between the two rotations in the footslope.

1993). Christensen (1992) observed that a decomposable pool of SOM was associated with clay-size particles in the soil. Spycher et al. (1983) indicated that LF organic matter, together with organic matter adsorbed on accessible aggregate surfaces, provided an important reservoir of rapidly cycling C and nutrients in forest ecosystems.

Landform had little effect on the hot-KCl extractable N (kg ha^{-1}, or mg kg^{-1} per kg of LF mass) from the LF at different sampling dates (Tables 4.24 and 4.25). The quantity of hot-KCl extractable N from the LF was related to the C:N ratio of the LF, as mentioned previously. The C:N ratio of the LF was similar in the shoulders and the footslopes (Table 4.15). Much of the LF was derived from the plant residues in agricultural soils (Janzen et al., 1992). Thus, the C:N ratio of the LF is directly related to the quality of plant residues. The difference in residue C:N ratio between the shoulders and the footslopes is relatively small (Table 4.9), i.e., landform had no effect on the C:N ratio of the wheat residue. The C:N ratio of chickpea residue was significantly lower in the footslopes than in the shoulders. However, the difference of median value was only 8 units of the C:N ratio (Table 4.9). This probably explained why landform had little effect on the quantity of hot-KCl extractable N from the LF.

In contrast, a distinct landform effect occurred with hot-KCl extractable N (kg ha^{-1} N or mg kg^{-1} N per kg of HF or soil mass) from the HF and the bulk soil (Tables 4.24 and 4.25), indicating that soils in the footslopes can supply more plant available N than soils in the shoulders. This observation is in agreement with that of Qian and Schoenau (1995) who used an anion exchange membrane technique in landscape studies in Saskatchewan and found that soils in the footslope positions released more mineral N than soils in the shoulder positions. The difference in the N supplying power of soil

between the shoulders and the footslopes is related to the spatial variability of crop growth and N-cycling processes, such as ANI and mineralization of plant residues.

When the hot-KCl extractable N from the LF was expressed as a percentage of the hot-KCl extractable N from the soils, the percentage in the shoulders generally was higher than that in the footslopes, irrespective of crop rotations and sampling dates (Table 4.26). The results suggested that a smaller proportion of the N in the labile SOM fraction, as compared to the bulk soil N, was mineralized (or extractable) in more fertile soils (e.g., greater C and N content) because such soils usually are more highly aggregated which may prevent or delay access by roots and microorganisms (Rovira and Greacen, 1957). Campbell and Souster (1982) observed that the Nm (N mineralization):Nt (total N) ratio usually decreased with increasing C content in Borolls and Boralfs.

In April 1997, the hot-KCl extractable N from the LF was higher in the chickpea-wheat rotation than in the wheat-wheat rotation in the shoulders, whereas no difference existed between the two rotations in the footslopes (Table 4.25). In June and September 1997, the hot-KCl extractable N from the LF was not significantly different between the two rotations. This result was likely due to the fact that the C:N ratio of the chickpea residue was relatively high, even though the C:N ratio of the chickpea residue was statistically lower than that of the wheat residue (Table 4.9). Biederbeck et al. (1994) found that application of N fertilizer and substitution of winter wheat (with chemical fallow) for spring wheat (with tilled bare fallow) usually increased labile SOM, while substitution of flax or lentil for spring wheat had little effect or even reduced labile SOM. The hot-KCl extractable N in the bulk soil did not differ in the two rotations

(Table 4.25), indicating that the one-year inclusion of chickpea in the crop rotation did not significantly improve the quality and N availability of SOM. The reason probably was due to the fact that the difference in the composition between the chickpea residue and the wheat residue was relatively small and the C input and N input from the chickpea residue were small compared with the soil C pools and soil N pools (Tables 4.3 and 4.9).

In the chickpea-wheat rotation, the amount of hot KCl extractable N (kg ha^{-1}) from the LF had the largest value in June in both the shoulders and the footslopes, whereas no peak occurred in June in the wheat-wheat rotation (Table 4.25). The temporal trend of hot-KCl extractable N from the LF was similar with the temporal trend of the LF content (Table 4.17). This observation suggested that the amount of available N in the LF was controlled largely by the absolute quantity of the LF. Meanwhile, the amount of hot-KCl extractable N from the HF and the bulk soil had the highest value in June in both rotations (Table 4.25). The peak value of hot-KCl extractable N in the bulk soil in June likely was due to the higher soil temperature, which was favorable for microbial activity and mineralization.

4.4 The Rotation Benefit of Chickpea: Nitrogen Effect and Non-Nitrogen Effect

4.4.1 Nitrogen effect

Soil moisture content and mineral N content in spring 1997 (i.e., the spring of the second phase of the rotation) were significantly higher in the chickpea-wheat rotation as compared to the wheat-wheat rotation in the footslopes, whereas moisture and mineral N content did not differ between the two rotations in the shoulders (Table 4.27). Similarly, the *A* value measured at harvest 1997, which represented a measure of soil N availability, was higher in the chickpea-wheat rotation than in the wheat-wheat rotation in the footslopes. No difference in the *A* value, however, existed between the two rotations in the shoulders.

It has been suggested that a legume can increase the yield of a succeeding cereal crop by increasing the availability of soil N (i.e., N effect) (Pierce and Rice, 1988; Stevenson and van Kessel, 1996a). High concentrations of soil mineral N can result from the release of mineral N from legume residues incorporated into the soil (Doughton and McKenzie, 1984). Legume residue can contribute more mineral N to the soil through mineralization, as compared to the cereal residue, because legume residue generally had a higher N content and a lower C:N ratio. For example, Stevenson and van Kessel (1996b) observed that pea residue with percentage N content of 2.42% and a C:N ratio of 18:1 contributed 6 to 14 kg ha^{-1} N more than the wheat residue with N content of 0.39% and a C:N ratio of 120:1 in a rotation study at Star City, Saskatchewan.

Table 4.27. Median values and results of the Mann-Whitney U test for gravimetric soil moisture content and mineral N content (0- to 15-cm depth) measured at the experimental site in spring 1997, and *A* value of wheat residue measured at the experimental site at harvest in 1997.

Landform element complex	Moisture (%)		Mineral N (kg ha^{-1})		*A* value (kg ha^{-1})	
	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat
Shoulder	17.9(4.7)†	18.8(5.3)	30.9(14.1)	32.6(10.9)	117(100)	96(29)
Footslope	25.4(8.9)	20.9(5.1)	40.1(23.9)	34.9 (8.9)	186(104)	129(66)
Shoulder vs. footslope‡	< 0.01	0.06	0.01	0.15	0.01	0.19
Shoulder, chickpea-wheat vs. wheat-wheat§	0.49		0.98		0.38	
Footslope, chickpea-wheat vs. wheat-wheat¶	< 0.01		0.09		< 0.01	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the two rotations in the shoulder.
¶ The *P* value associated with the comparison between the two rotations in the footslope.

In the current study, the direct N contribution from chickpea residue to the succeeding wheat generally was low (Table 4.11). The relatively low contribution may have been due, in part, to the low N content (approximately 12 to 18 kg ha^{-1} N) of the chickpea residue (Table 4.9). Moreover, most of the chickpea residue N was immobilized in the microbial biomass (Table 4.10) or was transferred into the SOM fractions (Tables 4.22 and 4.23). Consequently, the majority of the chickpea residue N was not available to the succeeding wheat crop.

A minimal difference occurred in the N contribution to the succeeding wheat by the labeled chickpea residue and the labeled wheat residue. No difference existed in the N contribution between the labeled chickpea residue and the labeled wheat residue in the shoulders, and the labeled chickpea residue contributed only 0.34 kg ha^{-1} N more than the labeled wheat residue in the footslopes (Table 4.11). Bremer and van Kessel (1992) also observed that N availability between lentil straw (C:N ratio: 31:1) and wheat straw (C:N ratio: 43:1) was similar, i.e., of the ^{15}N added in the lentil straw and the wheat straw, 5.5% was assimilated by the succeeding wheat crop.

The N effect of legumes estimated from the N availability of ^{15}N-labeled legume residue to the succeeding crop includes only the N contribution from the aboveground legume residue. Thus, the N effect of a legume in a crop rotation might be underestimated. Legumes can provide an N effect to the succeeding cereal crops by several other mechanisms including (i) increased availability of native soil N due to the application of legume residues (Yaacob and Blair, 1980), and (ii) root decomposition and rhizodeposition (Janzen and Bruinsma, 1993; Jensen, 1996).

The influence of the chickpea residue on the availability of soil N was discussed in detail in Section 4.2.4. The ANI effect associated with the labeled chickpea residue and the labeled wheat residue generally was low (Table 4.14). The labeled chickpea residue and the labeled wheat residue did not differ in ANI in the shoulders. The magnitude of ANI associated with the labeled chickpea residue, however, was significantly higher than that associated with the labeled wheat residue in the footslopes. The results indicated that the chickpea residue increased the availability of soil N as compared to the wheat residue in the footslopes. The increased availability of soil N would contribute to the overall N effect of chickpea in the rotation.

Legume root decomposition and rhizodeposition might contribute a significant portion of the N to the succeeding crop. Decomposition patterns of crop roots are related to their N concentration, diameter, lignin concentration, and presence of non-structured carbohydrates (Berg, 1984; Larsson and Steen, 1988). A considerable debate exists concerning whether mineralization of N in the decomposing root is a rapid (Eason and Newman, 1990) or a relatively slow process (Jenkinson, 1965; Wardle and Greenfield, 1991). The debate is due, in part, to the differences in the growth conditions and crop genotypes in these studies. Jawson and Elliott (1986) observed that in an incubation study more CO_2-C evolved from the wheat straw than from the wheat roots and the difference was nearly equal to the difference in their respective water-soluble C fractions.

Decomposition of root nodules of legume plants may be an important potential source of mineral N released from nodulated plants (Whitney and Kanehiro, 1967). Decomposition of dead roots and nodules may be the dominant pathway for release of

symbiotically-fixed N from living legume plants (McNeill and Wood, 1990; Lory et al., 1992). In the current study, significant differences in root biomass were not detected between the chickpea crop and the wheat crop (Table 4.28), as determined at harvest 1996 (i.e., the first phase of the rotation). Relatively low estimates of root biomass likely reflect the fact that some older roots had begun to decompose (Sauerbeck and Johnen, 1977) and, therefore, were not recovered intact from the soil core samples. Generally, N accumulation in the wheat root was higher than that in the chickpea root, although differences were not significant in the shoulders (Table 4.28). The reduced N accumulation in the chickpea root, as compared to the wheat root, was due to a lower production of chickpea root biomass. A portion of N in the chickpea root and the wheat root would be available to the succeeding wheat crop due to decomposition. Jensen (1996) observed that the rhizodeposition of N amounted to 19 mg plant^{-1} (7% of total plant N) for pea and 17 mg plant^{-1} (20% of total plant N) for barley at maturity. This author also observed that the pea rhizodeposits were more labile than those of barley. Janzen and Bruinsma (1993) also found that the amount of N deposited in the rhizosphere of wheat may constitute up to 20% of total plant N. Thus, the N from root, nodules and rhizodeposits is another significant source of plant available N, which will contribute to the overall N effect of legumes.

The *A* value can account for all possible sources of N that may differ between the chickpea-wheat rotation and the wheat-wheat rotation and is not limited to that associated with the aboveground residues. The *A* value was determined from the %Ndff of the plant at harvest. Thus, the *A* value is a time-integrated assessment of the N supplying power of the soil expressed in kg ha^{-1} equivalents of fertilizer N applied

Table 4.28. The yields of chickpea root and wheat roots, and N accumulation in the root (0- to 30-cm depth) measured at the experimental site at harvest in 1996: Median values and results of the Mann-Whitney U test related to the chickpea and wheat and landform element complexes.

Landform element complex	Root yield (kg ha^{-1})		N accumulation (kg ha^{-1} N)	
	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat
Shoulder	637(646)†	758(771)	3.9(4.3)	4.3(4.2)
Footslope	366(362)	647(659)	2.5(3.4)	4.8(3.9)
Shoulder vs. footslope‡	0.49	0.81	0.36	0.81
Shoulder, chickpea-wheat vs. wheat-wheat§	0.79		0.89	
Footslope, chickpea-wheat vs. wheat-wheat¶	0.43		0.13	

† The values in the parentheses are IQR
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the chickpea and the wheat in the shoulder.
¶ The *P* value associated with the comparison between the chickpea and the wheat in the footslope.

(Fried and Broeshart, 1975). It is a yield-independent index that can be used to detect relative differences in N availability from all possible sources between the two rotations. The *A* value in the chickpea-wheat rotation was 22% higher than the *A* value in the wheat-wheat rotation in the shoulders and 44% higher in the footslopes (Table 4.27). The coefficient of determination showed that the *A* value explained 52% and 39% of the total variation in grain yield and N accumulation, respectively (Table 4.29). The *A* value was also significantly correlated with N accumulation ($r = 0.54$, $P < 0.01$) and wheat grain yield ($r = 0.85$, $P < 0.01$). Stevenson and van Kessel (1996a) argued that the *A* value can be used to estimate the N effect of legume in a crop rotation.

Table 4.29. The coefficient of determination for the effects of the spring moisture and mineral N, *A* value measured at harvest, and root rot and leaf disease severity measured in July 1997 on wheat grain yield and total N accumulation in aboveground biomass.

Variables entered	Wheat grain yield	N accumulation
A value	0.52	0.39
A value, water, and mineral N	0.58	0.49
Root rot and leaf disease severity	0.13	0.08
A value, water, mineral N, root rot and leaf disease severity	0.61	0.50

The yields of wheat straw and grain, and N accumulation measured at harvest 1997 followed the same trend as that of the *A* value (Table 4.30), i.e., the straw yield, grain yield and N accumulation was significantly higher in the chickpea-wheat rotation as compared to the wheat-wheat rotation in the footslopes, whereas no difference occurred in straw yield, grain yield or N accumulation between the two rotations in the shoulders.

Table 4.30. The yield of wheat straw and grain, N accumulation in the aboveground biomass and harvest index measured at the experimental site at harvest in 1997: Median values and results of the Mann-Whitney U test related to crop rotations and landform element complexes.

Landform element complex	Straw yield (kg ha^{-1})		Grain yield (kg ha^{-1})		N accumulation (kg ha^{-1} N)		Harvest index	
	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat
Shoulder	1033 (283)†	985 (571)	749 (147)	695 (329)	18.8 (6.4)	18.9 (8.8)	0.43 (0.05)	0.41 (0.03)
Footslope	1640 (760)	1225 (403)	1113 (432)	781 (332)	32.6 (14.4)	21.7 (10.4)	0.41 (0.05)	0.39 (0.04)
Shoulder vs. footslope‡	< 0.01	0.07	< 0.01	0.34	< 0.01	0.42	0.39	0.16
Shoulder, chickpea-wheat vs. wheat-wheat§	0.68		0.38		0.72		0.10	
Footslope, chickpea-wheat vs. wheat-wheat¶	< 0.01		< 0.01		< 0.01		0.05	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the two rotations in the shoulder.
¶ The *P* value associated with the comparison between the two rotations in the footslope.

The harvest index in the chickpea-wheat rotation was higher than that in the wheat-wheat rotation in both the shoulders and the footslopes (Table 4.30).

Where crop residues were removed after harvest 1996 (i.e., the control treatment of the ANI study), the yields of wheat straw and grain in 1997 were higher in the chickpea-wheat rotation as compared to the yields in the wheat-wheat rotation in the footslopes. However, no difference in the yield of wheat straw or wheat grain existed between the two rotations in the shoulders (Table 4.31). A similar trend for wheat straw and grain yield was observed when crop residues were retained in the field after harvest 1996 (Table 4.30). These observations suggest that the incorporation of crop residues was not necessary in order to demonstrate the rotation benefit of chickpea in a crop rotation. Townley-Smith (1988) also observed that pulse residues did not differ from wheat residues in their effect upon wheat grain yield in Saskatchewan.

No large differences occurred in the N contribution to the succeeding wheat crop between the labeled chickpea residue and the labeled wheat residue (Table 4.11). This probably explains why the aboveground chickpea residue itself was not important for the expression of the yield advantage in the chickpea-wheat rotation, as compared to the wheat-wheat rotation. The data suggest that available N from sources other than from the decomposition of chickpea residue contributed significantly to the total N effect of chickpea in the rotation.

4.4.2 Non-nitrogen effect

The portion of the rotation benefit not associated with the increased availability of N is referred to as the non-N effect. The root rot severity in the second phase of the

Table 4.31. The yield of wheat straw and grain measured in the control treatment for the ANI study at harvest in 1997, i.e., the crop residue was removed after harvest in 1996: Median values and results of the Mann-Whitney U test related to crop rotations and landform element complexes.

Landform element complex	Straw yield (kg ha^{-1})		Grain yield (kg ha^{-1})	
	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat
Shoulder	915(653)†	957(254)	728(426)	676(131)
Footslope	1381(502)	1208(480)	1024(482)	817(379)
Shoulder vs. footslope‡	< 0.01	< 0.01	< 0.01	< 0.01
Shoulder, chickpea-wheat vs. wheat-wheat§	1.00		0.69	
Footslope, chickpea-wheat vs. wheat-wheat¶	0.05		0.04	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the two rotations in the shoulder.
¶ The *P* value associated with the comparison between the two rotations in the footslope.

rotation generally was low in both rotations. No difference occurred in the wheat root rot severity between the two rotations in the shoulders. However, wheat root rot severity in the wheat-wheat rotation was 0.15 units greater than that in the chickpea-wheat rotation in the footslopes (Table 4.32). The wheat leaf disease severity in the second phase of the wheat-wheat rotation was 2 and 3 units greater than in the chickpea-wheat rotation in the shoulders and the footslopes (Table 4.32). These results suggest that the inclusion of chickpea in the crop rotation facilitated a break in the disease cycle, probably because chickpea was not a suitable host plant for these diseases. Martens et al. (1984) also observed that the inclusion of pea in crop rotations decreased the occurrence of wheat pathogens because pea was not a suitable host plant for wheat pathogens.

In a critical review regarding crop rotation, Bullock (1992) stated that "even when pest pressure is minimal, the rotation effect still exists", suggesting that pest control contributed to the benefits of crop rotation, but was not solely responsible for the rotation benefit. Since not all pests detrimentally influencing crops are recognized, it may be hypothesized that much of the rotation benefit is probably due to the alleviation of unrecognized pests. In the current study, the root rot and leaf disease severity explained only 13% and 8% of the total variation in the wheat grain yield and N accumulation, respectively (Table 4.29). When the *A* value, spring soil moisture and mineral N, root rot and leaf disease severity were all considered as independent variables, they explained 61% and 50% of total variation in the grain yield and N accumulation, respectively. Apparently, unmeasured or unknown factors accounted for a considerable portion of the variation in the grain yield and the N accumulation.

Table 4.32. Median values and results of the Mann-Whitney U test for wheat root rot and leaf disease severity measured at the experimental site in the Bear Hills near Biggar, SK, in July 1997, related to the rotations and landform element complexes.

Landform element complex	Root rot (0-4 scale)		Leaf disease (0-11 scale)	
	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat
Shoulder	0.28(0.21)†	0.22(0.73)	4.0(1.0)	6.0(1.0)
Footslope	0.21(1.05)	0.36(0.42)	4.0(0.0)	7.0(1.0)
Shoulder vs. footslope‡	0.92	0.36	0.13	0.02
Shoulder, chickpea-wheat vs. wheat-wheat§	0.92		< 0.01	
Footslope, chickpea-wheat vs. wheat-wheat¶	0.09		< 0.01	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the two rotations in the shoulder.
¶ The *P* value associated with the comparison between the two rotations in the footslope.

It also is possible that other non-N nutrients such as P and K (Bullock, 1992) and the release of growth-promoting substances (e.g., triacontanol) from legume residue (Ries et al., 1977; Fyson and Oaks, 1990) were responsible for a portion of the non-N effect that the legume crops provided to the succeeding cereal crops. Vivekanandan and Fixen (1991) found that maize, at the six-leaf stage, had a larger P concentration when following soybean than when following maize. Hargrove (1986) demonstrated that the inclusion of legume cover crops, such as common vetch (*Vicia sativa* L.), into a crop rotation in the south-eastern U.S. resulted in a beneficial redistribution of K to the soil surface from deeper in the soil profile. Ries et al. (1977) suggested that growth-promoting substances, such as triacontanol, in the legume residues were responsible for the rotation benefit.

In a study in Saskatchewan, Stevenson and van Kessel (1996b) examined the benefits of pea in a pea-wheat rotation as compared to a wheat-wheat rotation and observed that the non-N effect was related to 15 to 23 of the 27 extra kg ha^{-1} N (the additional N content not related to that derived from ^{15}N-labeled pea residue) accumulated by the wheat following pea rather than following wheat. A diminishing response of wheat to N fertilizer rates occurred in the wheat-wheat rotation. Thus, they suggested that at least part of the rotation benefit of pea to wheat was the result of increased demand for N, due to the difference in the wheat crop health between the pea-wheat and wheat-wheat rotation. Rovira (1976) demonstrated that reduced disease severity can increase the N content of wheat tissues. Cook (1992) suggested that no single factor would do more for N-use efficiency in wheat production than having a healthy root system that would take advantage of the N applied to the crop. In the current study, the *A* value was negatively

correlated with leaf disease severity ($r = -0.26$, $P = 0.02$), although a high correlation was not observed. The lack of a high correlation between the *A* value and leaf disease severity was probably due to the fact that leaf disease severity was evaluated using discrete 'scale' values rather than using 'continuous measurement' that could provide more precise estimates, i.e., a high correlation may have been masked by the characteristics of the data.

Plant roots play an important role in N uptake and redistribution. Cook (1992) and Rovira (1976) found that the rotation benefit of pea on wheat root disease damage enhanced wheat root exploration of the soil. At the harvest of the second phase of the rotations, the root biomass of wheat grown on chickpea stubble was higher as compared to the root biomass of wheat grown on wheat stubble in the footslopes, whereas no difference occurred in the wheat root biomass between the two rotations in the shoulders (Table 4.33). No difference occurred in the N accumulation in wheat roots between the two rotations in the second phase of the rotation. The difference in wheat root growth between the two rotations could contribute to the rotation benefits.

Stevenson and van Kessel (1996a) reported that reduced severity of leaf disease and grassy weed infestation was related to 91% of the yield advantage associated with the pea (i.e., non-N effect), whereas the increase in the *A* value was related to only 9% of the yield advantage (i.e., N effect) in the pea-wheat rotation. This study was conducted in a hummocky terrain in the Black soil zone in Saskatchewan during years with above average rainfall. In the current study, the *A* value explained 52% of the total variation in the wheat grain yield, indicating that the N effect at least was as important as the non-N effect. The total rainfall from May to September 1997 was 40 mm less than the average

Table 4.33. Median values and results of the Mann-Whitney U test for wheat root yield (0- to 60-cm depth) and N accumulation in the root measured at the experimental site at harvest in 1997, related to crop rotations and landform element complexes.

Landform element complex	Root yield (kg ha^{-1})		N accumulation (kg ha^{-1} N)	
	Chickpea-wheat	Wheat-wheat	Chickpea-wheat	Wheat-wheat
Shoulder	789(459)†	821(853)	8.7(6.5)	11.6(8.3)
Footslope	920(239)	714(303)	9.0(3.5)	7.8(6.6)
Shoulder vs. footslope‡	0.55	1.00	0.59	0.82
Shoulder, chickpea-wheat vs. wheat-wheat§	0.88		0.71	
Footslope, chickpea-wheat vs. wheat-wheat¶	0.15		0.59	

† The values in the parentheses are IQR.
‡ The *P* value associated with the comparison between the shoulder and the footslope.
§ The *P* value associated with the comparison between the two rotations in the shoulder.
¶ The *P* value associated with the comparison between the two rotations in the footslope.

of previous years. Under the conditions of dry climate, the development of disease and pest was likely suppressed. As a result, the importance of N availability dominated as compared to the impact of crop diseases. Russelle et al. (1987) indicated that as N becomes less limiting to yield, non-N effect benefits become more important. In the Dark Brown soil zone the soil fertility is not as high as in the Black soil zone, and an increase in N availability could be more important to the rotation benefit under the dry conditions. Mooleki et al. (1995) also argued that the N effect of lentil may be higher than the non-N effect in the Dark Brown soil zone.

4.4.3 Landscape-scale variability

Soil and environmental conditions probably are the main factors affecting the expression of the rotation benefit of legume crops grown in a legume-cereal rotation. Crop rotation often does not result in a yield advantage when moisture is limiting (Wright, 1990; Campbell et al., 1992). Wright (1990) observed that the smallest rotation benefit generally was obtained at the driest sites and that in some instances the rotation benefit of pulse crops could even be negative.

Campbell et al. (1992) argued that available soil moisture, not N, is the major limitation to cereal production in the semi-arid climate. Soil moisture is not only necessary for plant and soil microbial growth, but also is the most important agent for the biochemical processes in the soil. De Jong and Rennie (1969) observed that inadequate growing season rainfall induced moisture stress in plants earlier on upper slopes than lower slopes. In the second phase of the rotation in my study, the differences in the yield of wheat straw and grain (Table 4.30), *A* value (Table 4.27), root

rot severity (Table 4.32), residue N recovery (Table 4.11) and ANI (Table 4.14) were not statistically significant between the chickpea-wheat and wheat-wheat rotation in the shoulders, whereas these variables were significantly different between the two rotations in the footslopes.

In a field with undulating or hummocky features, water redistribution is a fundamental control on N-cycling and crop productivity (Pennock et al., 1987; Fiez et al., 1994). Nordbo et al. (1994) and Stevenson and van Kessel (1996a) found that plant disease and weeds exhibited spatial patterns within the field. Thus, the benefit of chickpea in the rotation is also expected to show spatial patterns. In my study, a landform effect was present for yield of wheat straw and grain within the chickpea-wheat rotation (Table 4.30). The relatively high soil C content in the footslopes (Table 4.2) likely enhanced the decomposition of crop residue and mineralization of soil N, thus facilitating possible N-cycling processes in the soil. As a result, the *A* value and residue N recovery were higher in the footslopes as compared to the shoulders. The difference in the N-cycling processes between the shoulders and the footslopes also likely affected the pest and disease development and root growth in the field. For example, a landform effect existed for the leaf disease severity in the chickpea-wheat rotation. The grain yield of wheat following chickpea increased by 8% as compared to the grain yield of wheat following wheat in the shoulders and by 43% in the footslopes, respectively (Table 4.30). These results suggest that the benefit of chickpea in the chickpea-wheat rotation could be better expressed in the more fertile part of the landscape, i.e., the footslopes.

5. GENERAL DISCUSSION AND SUMMARY

All soils are anisotropic natural bodies in a landscape. Soils in a given landscape position, however, will exhibit similar morphological and chemical characteristics because these soils have been subjected to a distinctive set of hydrological and pedological regimes (Pennock et al., 1987). The spatial variability of soil properties can occur at different scales (Parkin, 1993). For example, when the soil was sampled at two scales (i.e., 2 m and 50 m), Cahn et al. (1994) observed that soil organic C had small-scale spatial variation nested within large-scale spatial variation in central Illinois, i.e., the range of spatial dependence was >50 m at the small scale, whereas the range of spatial dependence was >150 m at the large scale. Miller et al. (1988) found that the range of spatial dependence for soil texture and organic C was approximately 80 m, which was the distance roughly equal to the diameter of the hills located in the study area. They concluded that soil properties vary spatially along hillslopes and soil properties of the upslope locations must in some way influence the lower lying positions.

The local or micro-scale variability of surface roughness, soil C and texture can cause micro-site variability of runoff and soil water storage. As a consequence, micro-scale variability of N mineralization and other N-cycling processes are likely to occur because C and N availability and cycling processes are intimately linked. Information regarding the micro-scale variability of soil properties and N-cycling processes is limited. Results from my study demonstrated a landform effect on all measured soil

properties (i.e., soil mineral N, moisture, hot-KCl extractable N, pH, percentage C and N content), except EC, at the landscape scale (i.e., 14-m sampling interval). When soil was sampled at the micro scale (i.e., 0.3-m sampling interval), soil moisture, percentage C and N content had similar ranges of spatial dependence and similar semivariogram curves, which were different from those of soil mineral N and EC. These observations suggested that the degree of variability and spatial distribution of soil properties were diverse, because each soil variable was controlled and regulated by its own suite of associated controlling factors. For example, soil moisture was regulated mainly by soil texture and weather conditions, whereas SOM was largely determined by soil texture, cultivation and organic input.

Spatial variability inherent in field soils can be a problem for the interpretation of results from field studies of symbiotic N_2 fixation (Reichardt, 1990). Estimates of symbiotic N_2 fixation are highly variable in the field and several scientists have suggested that the landscape controls on symbiotic N_2 fixation are site specific (e.g., Androsoff et al., 1995; Stevenson et al., 1995). My results indicated that 72% of the variation in the estimates of %Ndfa was random and only 28% could be accounted for by spatial correlation when the chickpea plant was sampled at 0.3-m intervals (Fig. 4.3).

The determination of symbiotic N_2 fixation at different scales has both scientific and practical implications. Understanding symbiotic N_2 fixation at the landscape scale is important for site-specific management of legume crops in a hummocky terrain. The micro-scale variability of symbiotic N_2 fixation can improve the precision of estimates of symbiotic N_2 fixation. In addition, knowledge of the factors controlling variability at the micro scale leads to a mechanistic understanding of how symbiotic N_2 fixation is

controlled by soil factors and how the microorganism interacts with its environment. Process information at a micro scale also is crucial to improving our estimation and predictive capabilities at a larger scale.

A portion of the symbiotically-fixed N in a legume crop is available to the succeeding crops through the decomposition and mineralization of legume residues. Most recent estimates of N availability from legume residues to the succeeding cereal crop have been based on ^{15}N uptake by the succeeding cereal crop from previous ^{15}N-labeled legume residues. Using this methodology, the contribution of N from the legume residue to the succeeding cereal crop also has been treated as an estimate of the N effect of a legume in a legume-cereal rotation. This method provides a direct measurement of N availability from legume residue to the succeeding cereal crops. Although the ^{15}N technique has invariably been more precise than other approaches, such as the N yield difference approach (Fried and Broeshart, 1975), it does not necessarily represent the true value of residue ^{15}N recovery in the succeeding cereal crops. The substitution of ^{15}N for ^{14}N (i.e., pool substitution) during microbial immobilization and denitrification will cause the uptake of ^{15}N by the succeeding cereal crops from legume residue to be low because a portion of the ^{15}N in the legume residue was not accessible to the crops (Jansson and Persson, 1982; Jenkinson et al., 1985).

In addition to serving as a direct N source to the succeeding crops, residue incorporated into the soil also can influence the availability of soil N via the ANI (or 'priming effect') process. The 'priming effect' is an acceleration of the rate of the decomposition of SOM brought about by the addition of plant residues to the soil

(Bingeman et al., 1953). The rate of decomposition of the added plant residues increases when they are mixed with SOM (Hallam and Bartholomew, 1953).

Considerable debate has been centered on the occurrence of ANI (e.g., Jansson, 1958; Broadbent, 1965; Jenkinson et al., 1985). Previous studies demonstrated that the increased uptake of N from SOM in fertilized plots could be attributed to: (i) osmotic effects due to the addition of N fertilizer (Broadbent and Nakashimha, 1971); (ii) changes in physiological processes of the crop induced by fertilizer N (Sapozhnikov et al., 1968); (iii) nitrification of NH_4^+, causing acid hydrolysis of soil organic substances (Turchin, 1964); and (iv) increase of microbial activity due to the addition of N fertilizer (Westerman and Kurtz, 1973). According to Jenkinson et al. (1985), microbial N immobilization, whether driven by the decomposition of SOM or plant material, can lead to pool substitution and is the dominant cause of 'apparent' ANI. Thus, the magnitude of the ANI is directly proportional to the rate of immobilization. Anything that increases immobilization, such as addition of crop residue with a high C:N ratio, will increase the magnitude of the ANI. In an incubation study, Azam et al. (1993) added ^{15}N-labeled soybean tops, vetch tops and corn stover to the soil and reported that both soil type and the quality of applied residues regulated the occurrence and magnitude of the ANI. They proposed that the negative ANI in their study was likely due to the fact that the mineral N present initially in the soil was immobilized during the decomposition of freshly added organic matter and was subsequently mineralized to a lesser extent than in unamended soil.

Few reports are available regarding the ANI of crop residues under field conditions. My results demonstrated a landform effect on the ANI of both the chickpea residue and

the wheat residue with a wide range in the magnitude of the ANI. The data indicated that the spatial variability of the soil regulated the ANI processes of the residue. The ANI process can conserve applied ^{15}N through pool substitution, MIT or biological exchange reactions. Thus, residue N recovery will be regulated by the ANI process. In my study, the magnitude of the ANI and residue N recovery in the succeeding wheat crop were significantly correlated ($r = 0.37$, $P < 0.01$). The lack of a high correlation between the ANI and residue N recovery suggested that diverse degrees of pool substitution and MIT occurred across the landscape. Future studies must investigate the contribution of the ANI of legume residue to the N effect of legumes in the rotation, and investigate the biochemical principles controlling the occurrence and magnitude of the ANI.

The SOM is heterogeneous and consists of fractions differing in turnover rates. The labile fractions account for only a small proportion of the total SOM. However, they are very dynamic and account for much of the SOM fluctuations over time. It has been suggested that the density methods used for the physical separation of the labile fractions of the SOM are straightforward, reliable, and reproducible (Gregorich and Ellert, 1993). My results showed that the ranking of C content, N content, and C:N ratio among the LF, HF and bulk soil was LF > bulk soil > HF, indicating that the density fractionation can physically separate SOM into fractions differing in composition. The temporal variation of the LF was larger than the SOM, suggesting that the LF was more sensitive to the added crop residues and soil conditions. The quantity of hot-KCl extractable N from the LF was approximately 10% of that of the hot-KCl extractable N

from the bulk soil, meaning that at least part of the SOM associated with the HF is also labile.

Crop residues incorporated into the soil are colonized by soil microorganisms and adsorbed by mineral particles (Swift et al., 1979). The influence of residue input on the quality and quantity of SOM fractions decreased in the order of light, intermediate, heavy macroorganic matter and non-macroorganic matter (Hassink, 1995). The fate of crop residues added to the soil has been investigated extensively. However, only the effects of soil and environmental conditions on the decomposition of crop residues have been emphasized (e.g., Jenny et al., 1949; Amato and Ladd, 1992). The transfer of residue C and residue N between active and passive SOM fractions has received less attention (Amato and Ladd, 1980). In an incubation study, Hassink and Dalenberg (1996) added ^{14}C-labeled rye residue to the soil and found that 26 to 28% of the label was present in the soluble fractions and 31 to 32% of the label in the light fractions two days after application. They also observed that the residue C was transferred from the soluble and light fractions and finally accumulated in the microaggregates. My results showed that in the early spring following fall application of labeled residue, approximately 15 to 24% of the chickpea residue N and 17 to 20% of the wheat residue N were present in the LF (Table 4.22), and approximately 28 to 48% of the chickpea residue N and 17 to 19% of the wheat residue N were recovered in the HF (Table 4.23). These results indicate that the transfer of residue N into the SOM fractions can occur very quickly and most of the residue N remained in the soil and was gradually transferred from labile pools to more stable pools, i.e., added ^{15}N from residues would be stabilized in the different fractions of SOM. This result suggests that the crop

residues incorporated into the soil will have a long-term effect on soil fertility and soil N. The data regarding the distinction of the LF and HF and the transfer and sequestration of residue N into these fractions can help us understand the mechanisms of residue decomposition and the transfer of residue N among SOM fractions. Future studies should investigate the transfer rates of ^{15}N from the legume residue between SOM fractions and the factors controlling the transfer.

The fate of the N in the chickpea residue and the wheat residue incorporated to the soil after the harvest 1996 is summarized in Fig. 5.1. Approximately 70% of the N in the chickpea residue was derived from symbiotic N_2 fixation. At harvest in the second phase of the rotation, the wheat crop recovered 3.2% and 4.4% of the chickpea residue N in the shoulders and the footslopes, respectively. Most of the chickpea residue N was transferred to the soil microbial biomass and SOM fractions (Fig. 5.1a). A portion of residue N may have been lost from the soil system via N-cycling processes, such as denitrification and leaching. A small portion of chickpea residue N that remained in the soil at the harvest of the second phase of the rotation was available to the canola crop in the third phase of the rotation due to the subsequent turnover of N. Approximately 0.6% and 0.4% of the N in canola stubble grown in 1998 was derived from the chickpea residue applied in fall 1996 in the shoulders and the footslopes, respectively. The data suggested that only a small portion of N or the symbiotically-fixed N in the chickpea residue was available to the succeeding crops, since the majority of chickpea residue N was transferred to the soil microbial biomass and SOM fractions. These data suggest that the beneficial effects of chickpea residue in a crop rotation may be due mainly to the fact that chickpea residue can increase the long-term fertility of the soil. Ladd et al.

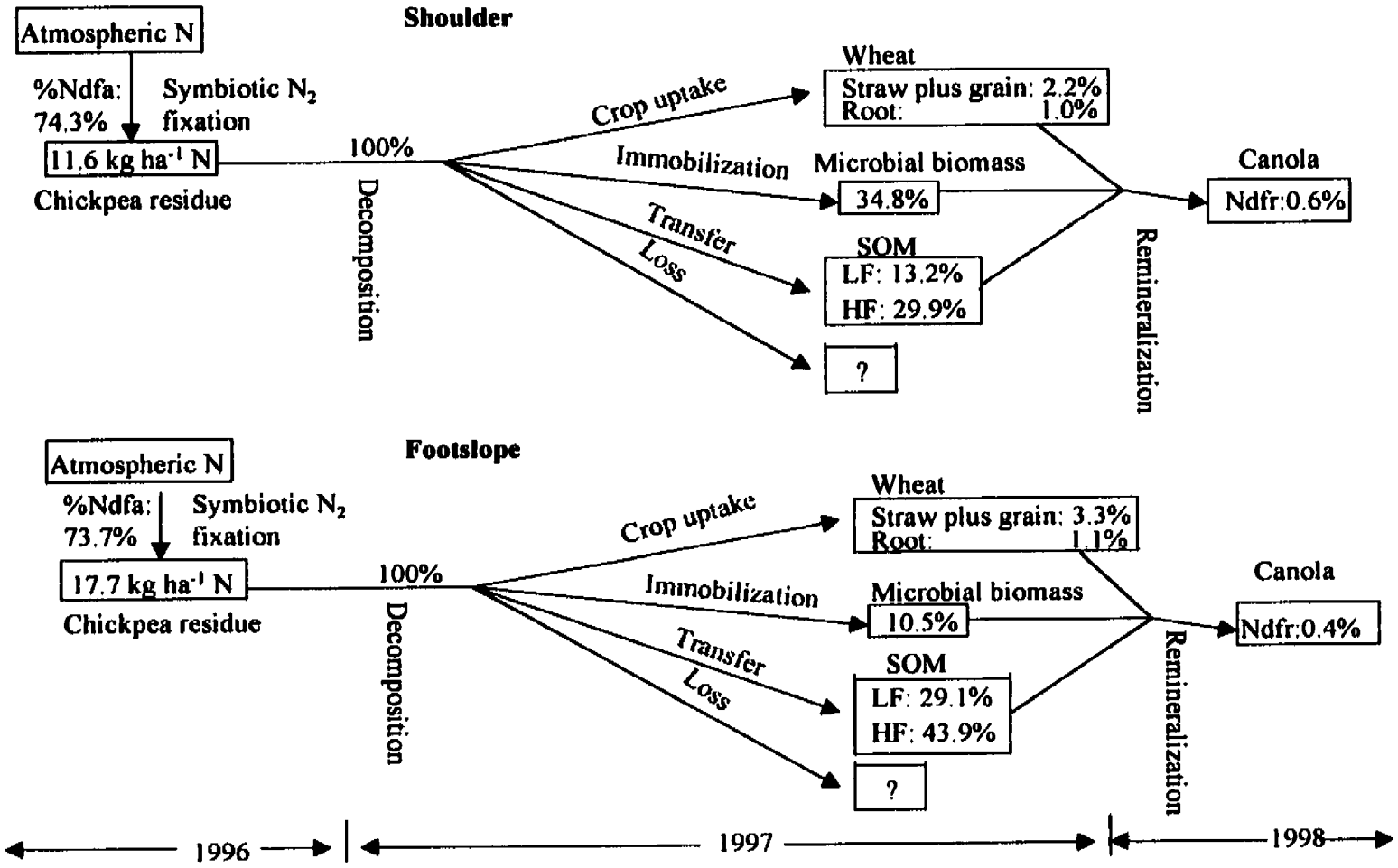

Figure 5.1a. A simplified flowchart of N dynamics for the labeled chickpea residue applied in October 1996 at the study site.

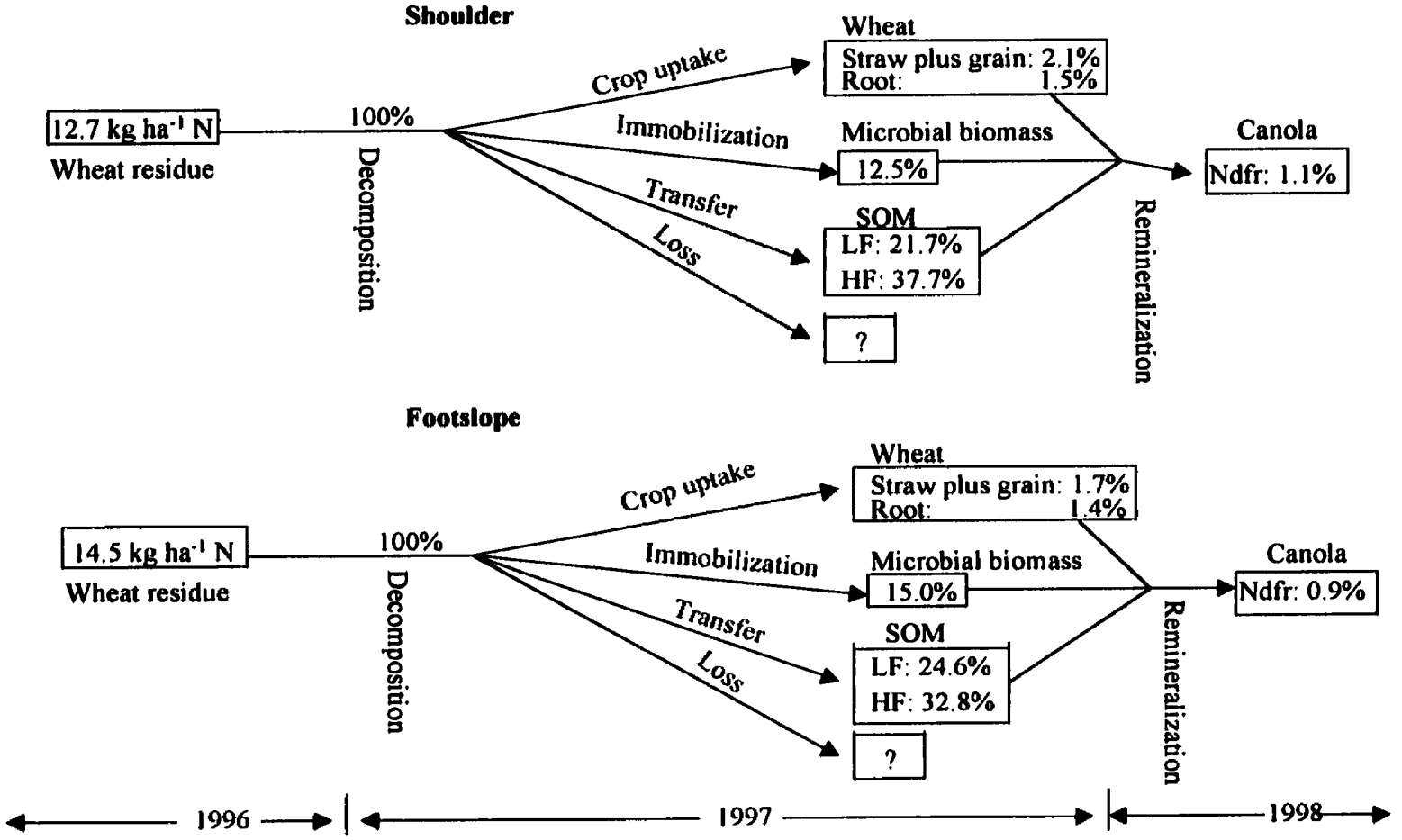

Figure 5.1b. A simplified flowchart of N dynamics for the labeled wheat residue applied in October 1996 at the study site.

(1983) also argued that the increase in the long-term soil fertility is likely due to the conversion of a portion of the symbiotically fixed N in legume residues into stable humus which directly and indirectly improves soil fertility.

The wheat crop in the second phase of the rotation recovered 3.6% and 3.1% of N from previous wheat residue in the shoulder and the footslope, respectively (Fig. 5.1b). As was observed with the chickpea residue N, most of the wheat residue N was transferred to the soil microbial biomass and SOM fractions after one year. Due to the relatively small difference in the N content and C:N ratio between the chickpea residue and the wheat residue, both chickpea residue and wheat residue contributed similar quantities of N to the succeeding crops. Therefore, no difference was detected in the quantity of N contributed from the chickpea residue and the wheat residue to the succeeding wheat crop in the shoulders and the chickpea residue contributed only 0.3 kg ha^{-1} more N to the succeeding wheat crop as compared to the wheat residue in the footslopes (Table 4.11).

The basic assumption of the *A* value concept is that a plant having two sources of a nutrient will access this nutrient from these two sources in direct proportion to the amounts available (Fried and Broeshart, 1975). Since identical quantities and enrichment of ^{15}N-labeled N fertilizer were applied at each grid cell in both the chickpea-wheat and the wheat-wheat rotation, the *A* value can be used to compare the difference in the quantity of non-labeled N from all sources between the two rotations. If the soil supplies more non-labeled N, the applied ^{15}N will be more diluted and a higher *A* value will be achieved. Thus, a higher *A* value indicates that the soil can supply more mineral N to the plants. The *A* value measured at the harvest in the second

phase of the rotation was 22% and 44% greater in the chickpea-wheat rotation as compared to the wheat-wheat rotation in the shoulders and footslopes, respectively. It has been suggested that the *A* value will reflect the N effect of legume more accurately and meaningful as compared to the quantity of N available to the succeeding cereal crop from the decomposition of ^{15}N-labeled legume residue (Stevenson et al., 1998).

The mechanisms of rotation benefits of legume in a legume-cereal rotation are not completely understood. Not all pests detrimentally affecting crops are recognized (Bullock, 1992). Thus, much of the rotational benefit is due to alleviation of unknown pests. Crookston (1984) noticed that yield increases from rotation persist even beyond optimum levels of fertility, soil tilth, soil moisture, and pest control, suggesting that some unknown factors result in the rotation benefit. Future studies are required to examine soil microbiological factors, which might account for the rotation benefit, and to investigate the possible existence of crop antibodies which likely produce a healthy soil in response to the proliferation of the roots of a crop into that soil (Crookston, 1984).

Stevenson and van Kessel (1996a) were not able to detect a rotation benefit of pea beyond the first year following pea in a pea-wheat-canola rotation as compared to a wheat-wheat-canola rotation in a landscape study in Saskatchewan. Thus, they considered the rotational benefit of pea to be short-term. Long-term rotation benefits, however, also may exist. For example, Ladd et al. (1985) found that after 8 years of application, 31 to 38% of added legume ^{15}N was still in the organic fractions of the soil tested, suggesting that building up SOM likely was the main benefit of legume residues. Campbell et al. (1992) observed that after five cycles of a lentil-wheat rotation, the C:N

ratio of the SOM was narrowed. They proposed that the narrowed C:N ratio of the SOM might increase soil N availability in the long-term. In my study, chickpea residue incorporated in the fall 1996 contributed 0.6% and 0.4% of the N in the canola stubble grown two years after chickpea in the shoulders and the footslopes, respectively. It can be expected that the contribution of N from chickpea residue to the succeeding crops will continue for a number of years. Thus, the inclusion of legume in the crop rotation likely will have a long-term effect on the soil fertility and soil N status.

One of the major benefits expected from a legume crop is that it will add N to the soil via symbiotic N_2 fixation. A very consistent effect of the legume crops is to increase mineral N in the soil. The higher concentration of soil mineral N results from conservative use of N by the preceding legume crop (i.e., 'N sparing'), coupled with the release of mineral N from the legume residues (Doughton and McKenzie, 1984). Herridge (1987) found 30 kg ha^{-1} more post-harvest nitrate in the root zone in the legume-cereal rotation as compared to the cereal-cereal rotation. This amount of nitrate was considered as 'spared' N. In Australian, Evans et al. (1989) found that spared N contributed more to the average N effect of 40 and 33 kg ha^{-1} for lupin and pea, respectively, than did N released from the legume residues.

Few reports are available regarding N sparing by chickpea. Herridge et al. (1995) reported that soil nitrate spared by chickpea in a chickpea-wheat rotation ranged from 6 to 31 kg ha^{-1} as compared to the wheat-wheat rotation. In my study, the N derived from soil (Ndfs) in the chickpea straw and grain measured at harvest 1996 was 25.8 and 23.9 kg ha^{-1} in the shoulders and the footslopes, respectively. The Ndfs (kg ha^{-1}) in the wheat straw and grain measured at harvest in 1996 was 47.6 and 62.4 kg ha^{-1} in the shoulders

and the footslopes, respectively. The inclusion of legume crops in cereal cropping can theoretically increase soil N concentrations and at the least, arrest the decline of soil N fertility associated with intensive cereal cropping. The 'spared' N by legume crops in the legume-cereal rotation will contribute to the overall N effect of the legume crops, regulate the ratio of the N effect:non-N effect, and influence other N-cycling processes, such as mineralization of crop residue and denitrification, as compared to the cereal-cereal rotation. This speculation should be investigated in detail in future studies.

My results showed that the wheat grain yield when grown on chickpea stubble increased by 8% as compared to wheat grown on wheat stubble in the shoulders and by 43% in the footslopes. The results suggested that landscape position is an important factor controlling the expression of the rotation benefits of the chickpea. The *A* value, residue N recovery in the second phase of rotation and the ANI effect were significantly higher in the chickpea-wheat rotation, as compared to the wheat-wheat rotation, in the footslopes, whereas no difference in these variables existed between the two rotations in the shoulders. Consequently, it was expected that the crop yield and N effect were higher in the footslopes as compared to the shoulders. Wheat diseases and weeds, however, also were higher in the footslopes due to relatively higher moisture and soil mineral N. As a consequence, the resulting yield benefit was a combination of these two opposing mechanisms.

The landscape-scale approach encompasses a larger field area than the small-plot approach and covers all of the landform elements in a landscape. Thus, it can be used to examine and explain the spatial variability of investigated processes and the landscape controls on these processes. Water redistribution in a hummocky terrain, and its effects

on the soil properties, crop growth and N-cycling processes is not expected to influence results when the experiment is conducted in a small-plot experimental approach.

Although the advantages of the landscape-scale approach are compelling, several practical and statistical concerns must be dealt with for the landscape-scale approach to be useful. Milne (1936) suggested that soils along a landscape are related in much the same manner as links in a chain. Soils at one location influence the surrounding soils by affecting drainage conditions, erosion, deposition, leaching, translocation and redeposition of chemical constituents. Thus, the size of the experimental unit or the distance between sampling points is of concern, if one is attempting to establish independent treatments or measurements. The definition of the experimental unit and the randomization of treatments to the experimental units are not as straightforward in landscape-scale studies as compared to the traditional small-plot studies. The statistical analysis for the small-plot studies is relatively straightforward and based on well-established procedures. However, statistical treatment of data from landscape-scale studies remains unclear.

Overall, my study investigated the N-cycling processes at the landscape-scale, including symbiotic N_2 fixation, decomposition of the chickpea residue, the effect of chickpea residue on the availability of soil N, and the transfer of chickpea residue N to soil organic matter fractions. The results provide a good picture regarding the N dynamics of the chickpea residue and improve our understanding in the mechanisms of the N effect of chickpea in the rotation. Spatial variability of these processes suggests that landscape position and environmental conditions regulate these processes. Knowledge regarding the spatial variability of these processes is very important to

improve our understanding of the N dynamics of legume residues, the N effect and the non-N effect of legume crops in the crop rotation, and site-specific management of legumes in the crop rotation.

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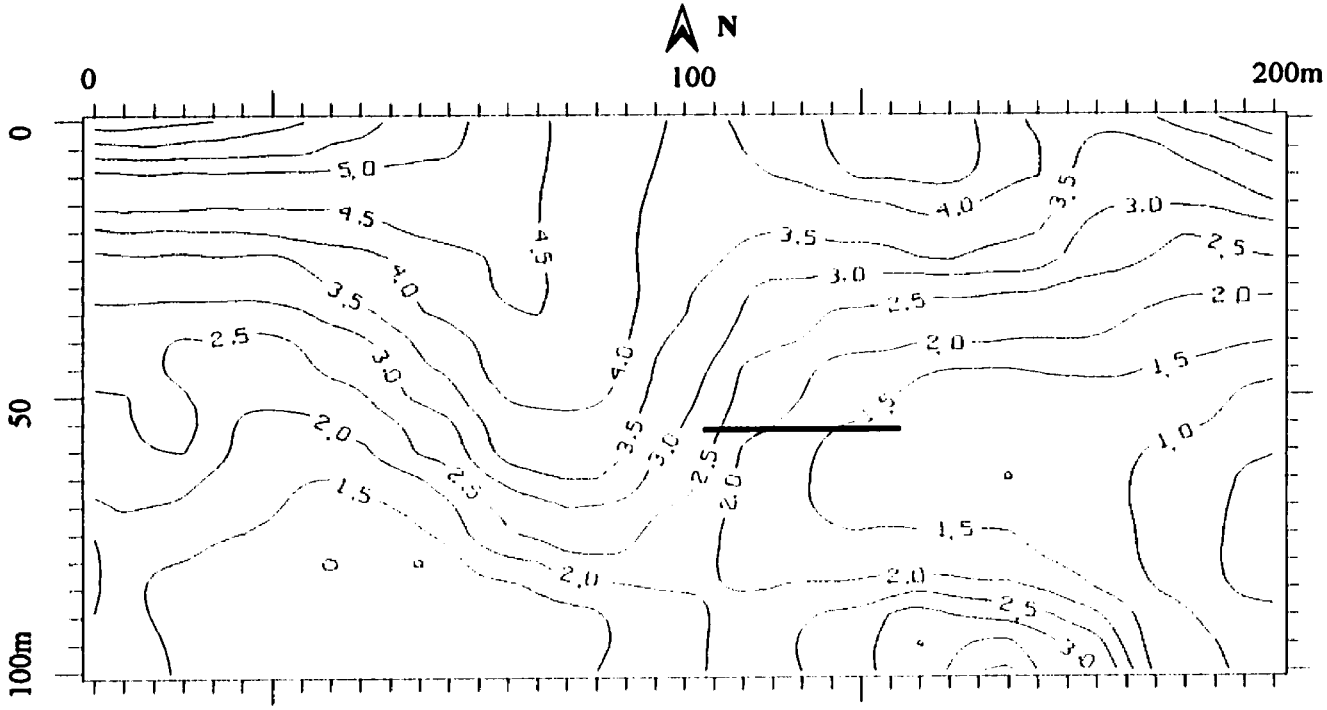
APPENDIX A

A contour map of the study field in the Bear Hills near Biggar, SK. The black line in the figure indicates the position of the transect used for estimating symbiotic N_2 fixation at the micro scale.



APPENDIX B

SPATIAL DEPENDENCE: ANALYSIS OF SEMIVARIANCE

According to Milne (1936), soils at one location affect their surrounding soils by influencing drainage conditions, erosion, deposition, leaching, translocation and redeposition of chemical constituents. Thus, soil properties in the field, especially in the non-level field, vary continuously in space. Soils along a catena or a hummocky landscape are related much the same as links in a chain. As a consequence, the values at sites that are close together in the field will be more similar as compared to those further apart. Consequently, the observations cannot be regarded as independent. They depend upon one another in a statistical sense, and a more advanced statistical analysis is required.

In practice, the variation of soil properties is very irregular. However, the variation of soil properties is not wholly erratic. Some structure occurs in the variation in the sense that the values at positions near one another usually are more similar as compared to others. The only practicable approach is to regard such a property as a random variable and to treat its variation in space statistically.

Spatial variability of soils has been investigated by soil scientists during the past decades. One approach has been to use geostatistical analyses to study spatial variability of soil properties (Yost et al., 1982). The geostatistical analyses was originally used in the mining industry. According to Matheron (1963), the geostatistical approach in mining differs from classical approaches in agronomic studies in the following aspects:

(1). Classical methods do not consider the spatial aspects of data;
(2). Neighboring samples of ore reserves may not be independent of each other. Samples taken close together tend to be more similar than those that are far apart; and
(3). Ore deposits differ from most field experiments because they cannot be replicated. A specific deposit occurs only once and it is usually unique.

Geostatistics has proven useful in soil science for characterizing and mapping spatial variation of soil properties. In addition, geostatistical analysis of within-field variation of soil nutrients and plant growth parameters can help identify cause-effect relationships among these parameters (Tabor et al., 1984). The following text will explain how to use analysis of semivariance to examine the spatial dependence of soil properties.

In order to use the analysis of semivariance to investigate the spatial dependence of measured soil properties and biochemical processes, typically samples are taken at a regular space (or distance) along a linear transect (Fig. A1). Let us start with the simplest situation. Consider two places some distance (h) apart at which a property Z has the values z(x) and z(x+h), the relation between the two values can be defined by their variance,

$$s^2 = [z(x) - \bar{z}]^2 + [z(x+h) - \bar{z}]^2 = \tfrac{1}{2}[z(x) - z(x+h)]^2 \qquad \text{(E1)}$$

where $\bar{z}$ is the mean of z(x) and z(x+h), and the quantity s^2 is therefore called the semivariance.

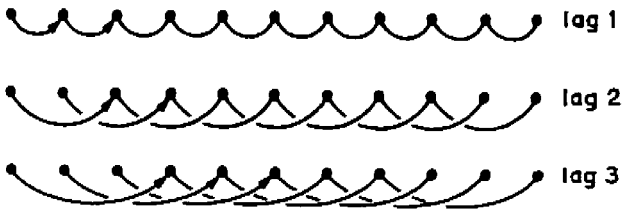


Figure A1. Lagged comparisons for estimating semivariances on a linear transect.

Suppose that Z has been measured many times along the linear transect and that m pairs are separated by the vector h or the same lag distance (Fig. A1). The average semivariance at this lag (i.e., lag1, h) can be calculated from

$$\gamma(h) = \left(\frac{1}{2m}\right) \sum_{i=1}^{m} [z(x_i) - z(x_i+h)]^2 \qquad \text{(E2)}$$

where $z(x_i)$ is the value of the variable Z at sampled location x_i and $z(x_i+h)$ is the value of the variable Z at sampled location x_i+h, a distance h away from x_i. Thus, m pairs of sample locations are a distance h apart.

The calculation can then be repeated for any integral multiple of the sampling interval h along the transect to obtain the semivariances for increasing long lags, i.e., h = 2, 3, 4, ...(Fig. A1). Equation (E2) can then be generalized to:

$$\gamma(h) = \frac{1}{2(n-h)} \sum_{i=1}^{n-h} [z(i) - z(i+h)]^2 \qquad \text{(E3)}$$

We then obtain the ordered set of semivariance values $\gamma(1)$, $\gamma(2)$, $\gamma(3)$, ..., the sample semivariance subsequently is plotted with their associated lag distance on the abcissa and $\gamma(h)$ (the semivariance) on the ordinate to obtain the sample semivariogram (Fig. A2). Characteristics of the semivariogram, such as nugget, sill and range can be useful in explaining the structure of spatial dependence in the field.

If the semivariogram curve passes through the origin, it fully describes the spatial dependency of the soil property, with spatial dependency accounting for all of the semivariance within the range. Usually, however, the curve does not pass through the origin and this discontinuity is called the nugget or nugget effect. The nugget (i.e., *y*-intercept) is the residual and random variation not removed by close sampling due to the fine-scale variability or measurement error. Typically, the magnitude of semivariance increases as the lag distance increases and will reach a maximum at which it levels off. The maximum semivariance is known as the sill. The lag distance where the variance approaches an asymptotic maximum (i.e., sill) is the range across which data are spatially correlated (Clark, 1979). The range is of considerable importance. The range, expressed as

sampling distance, can be interpreted as the diameter of the influence zone which represents the average maximum distance over which a soil property of two samples is related. At distances less than the range, measured properties of two samples become more alike with decreasing distance between them.

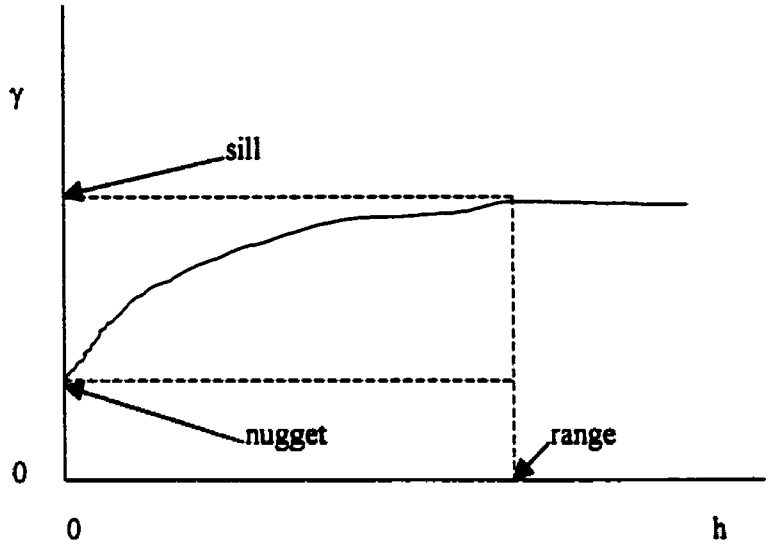


Figure A2. Elements of idealized semivariogram with range, sill and nugget.

Small nugget variances suggest that little variation was present at distances shorter than the first lag of the semivariogram. Percentage of sill, or relative nugget effect, i.e., nugget semivariance expressed as percentage of sill, indicates the relative quantity of this random variance in the maximum variance (i.e., sill). If the value of percentage of sill is small, it indicates that the majority of variance in the sill is derived from spatial correlation among samples (Trangmar et al., 1987; Gonzalez and Zak, 1994).

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