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RESIDUE DECOMPOSITION OF CO-I-I-ON, PEANUT AND SORGHUM

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To my family

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ABSTRACT

Diack, Mateugue. MS., Purdue University, December 1994. Residue decomposition of cotton, peanut and sorghum. Major Professor: D.E. Stott.

Developing effective management strategies that protect soil against erosion requires an understanding of residue decomposition. While the impact of environmental factors such as temperature and water content has been studied, little has been done to understand how the characteristics of the residue itself impact the decomposition rate. Traditionally, the C:N ratio has been used as a predictor of decomposition rates for agronomic crops, but has recently been shown to be poorly correlated. This study relates the chemical composition of residue components (aboveground biomass and roots) to the decomposition rates for three cultivars each of three crops: cotton (*Gossypium hirsutum*), sorghum (*Sorghum bicolor*) and peanut (*Arachis hypogaea*). The rates were determined by mass loss and CO_2 evolution. Change in the specific surface area of the residue as related to mass loss was also measured. The three crops were from slowest to the most rapid loss: sorghum > cotton > peanut. From the initial chemical and physical residue characteristics, the following equation was developed to predict decay in the first stage:

$P_D = (N * Sugars * Hemicellulose * K_{in.})$ / Lignin, where P_D is the predictive decay rate, $K_{in.}$ is the initial specific surface area-to-mass ratio. For mass loss, $r^2 = 0.96$, and for CO_2 evolution, $r^2 = 0.95$. Since varietal differences within crops have led to significant variation in decomposition rates, cultivars with slower decaying residues might be recommended for highly erodible lands.

CHAPTER 1

LITERATURE REVIEW

Soil erosion is a major problem facing land managers, conservation planners, environmental scientists and those concemed with construction sites. At the farming level, erosion destroys the inherent fertility of the soil, and that means higher farm and food costs. Maintaining crop residue on the soil surface is an effective and cost-effective practical method for controlling wind and water erosion. Douglas et al. (1992) noted that if residues are burned, removed, buried or decomposed before a critical erosion period, there may be insufficient cover to protect the soil.

Critical time periods for wind erosion, when the potential for erosion is the greatest, occur from the time of the last tillage before seeding until the crop has grown enough to provide adequate ground cover (Siddoway and Fenster, 1983). This is when soil clods have dispersed due to freezing and thawing or wetting and drying, and when residue is usually positioned flat on the soit surface.

Residue protects the soil surface from water erosion by absorbing the impact energy of raindrops, thus reducing soil particle detachment. Residue, also reduces surface crusting and sealing thereby enhancing infiltration and crop seediing emergence. Surface residue slows the velocity of runoff water by creating small obstructions along the flow path. This action reduces both the amount of soil transported and the amount of additional soil particles detached by flowing water.

Managing crop residues on soil surface is a primary method for controlling soil erosion. One of the main goals of conservation tiliage is to keep enough crop residues on the soil surface to control or minimize erosion. Generally, a conservation tillage system that leaves 30% of surface covered by residue, can reduce soil loss by 60-70%. On steep slopes, greater cover is required to achieve 60-70% soil loss reduction. Quantities of residue biomass left after harvest depend on climatic conditions and soil nutrient availability during the growing season. Surface residues in the standing position are twice as effective in controlling wind erosion as the same quantity of residues lying flat on the soil surface (Tanaka, 1986). However, flat residues are the most effective for controlling erosion by water.

Understanding how rapidly surface managed crop residues are decomposed and lost from a field site, is a prerequisite to the design of erosion prediction and control that will ensure sustainable and profitable agriculture. The major factors controlling crop residue decomposition are residue physical and chemical characteristics, soil physical, chemical, and biological composition, and finally climatic conditions (Stott et al., 1989).

1.1. Factors Influencing Crop Residue Decomposition

1.1.1. Residue C haraderistics

1.1.1.1. Residue Type, Positioning and Placement

Crop residue types are generally separated into two main entities which are the above-ground biomass of the plant (sheath, stem and leaves), and the roots.

The residue position within a field is important in determining the type of soil erosion that can be best controlled. For protection from water erosion, flat residues contribute more cover than standing, residues and this protects the soil surface from raindrop impact. However, standing residues persist longer because of slower decomposition rates. For wind erosion, standing residues reduce the wind velocity near the soil surface (Steiner et al., 1993). Flat residue cover increases surface roughness, acts as non-erodible material, and prevents soil particle detachment. Tanaka (1986) studied the effect of chemical and stubble-mulch fallow on residue orientation and decomposition, and to compare residue biomass of standing vvinter wheat residue on chemical fallow plots to that of spring wheat. From the chemical fallow plot, standing residue with an angle < 45^0 from vertical, and flat residue with an angle > 45^0 from vertical were collected separately. He found that quantities of chemical fallow standing residue decreased, while flat residue increased at constant rates during the summer fallow period. Tanaka hypothesized that the loss and gain of residue were due to repositioning of the standing residue.

Surface placement of crop residues can be an effective practical method for erosion control. Microorganisms involved in the decomposition of crop residues are sensitive to residue placement. Puig-Gimenez and Chase (1984) showed that under identical incubation conditions in the laboratory, straw kepf near the surface of the soil and resiclue mixed uniformily through the 7 cm deep soil sample were not significantly different in decomposition rate. In contrast to these results, field studies have shown significantly greater decomposition of buried residues than of surface-applied residues (Greb et al., 1974). The decrease in decomposition parallels, but likely not due to the drop in the soil organic carbon level. Parr and Papendick (1978) stated that buried residues are likely to decompose faster than surface residues because buried residues are exposed to more uniforrn temperature and moisture conditions within the soil profile. Furthermore, in a study of wheat straw residue loss under simulated field conditions, Brown and Dickey (1970) observed that buried wheat residue had a greater mass loss than residue on the soil surface.

1.1.1.2. Residue Particle Size

Few data are available on the effects of particle size on residue decomposition. In some faboratory decomposition studies, crop residues were chopped into 4 to 5 cm sections, in others, ground residues were used. Large particles generally decompose slower than small particulate materials (Allison, 1973). Jensen (1994) related decomposition rate with residue particle size and C:N ratio, noting that the decomposition of plant residues was slower with small, than with coarse residues in the early decomposition stage of materials of low C:N ratio. He concluded that it was probably due to a better protection of the smaller residues and biomass by clay minerals. For residues with high C:N ratio, the decomposition of larger sized residues may be N-limited, resulting in a slower rate of decomposition compared to smaller residues.

Residue type, particle size, position and placement in the field are all important factors contributing to the regulation of the decomposition process.

1.1.1.3. Chemical Composition of Plant Residues

Chemical quality of the crop residues is one of the most important factors controlling the rate of breakdown of the residues by microbes. Although microbes do not have absolute control on nutrient availability, they are strong competitors for available nutrients. The overall rate of decomposition is infiuenced by the types of organic molecules and the nutrient content of the plant residues (as well as by other factors being discussed). Nitrogen is a key nutrient for microbial growth and hence for organic material breakdown. Residue with high nitrogen contents favor rapid initial decomposition. Also, the comportent most frequently limiting microbial adivity is the availability of utilizable C substrate (Alexander, 1977).

In plants, about 75% of the dry weight is polysaccharide, with cellulose, the most abundant of all naturally occurring organic compounds, constituting at least 10% of all vegetable matter (Cheshire, 1979). The cellulose has a structural role; in the plant cell wall, linear chains of cellulose molecules occur in cross-link bundles embedded in a highly branched polysaccharide matrix consisting of hemicellulose. Hemicelluloses have been defined as the alkali-soluble polysaccharides in plant and are a mixture of homo- and heteropolysaccharides with xylans predominating. Plants also contain small amounts of water-soluble polysaccharides.

Lignin is the second most abundant polymer synthesized by plants (Stott et al., 1989). According to Lewis et al. (1990), lignins are plant polymers derived from the hydroxycinnamyl alcohols or monolignols *p*-coumaryl, coniferyl, and sinapyl. They also noted that the aromatic portions of these phenylpropanoids are described as *p*-hydroxyphenyl (h), guaiacyl (g), and syringyl (s) moieties, respectively, and that lignins are classified according to this distinction.!

Polysaccharide and lignin contents are important factors in the plant residoe decomposition. Their initial concentrations play a major role in predicting the kinetics of residue decomposition.

1 .1.1.5. Biodegradation and Stabilization of Plant Residues in Soil Humus

Young succulent tissues are metabolized more readily than residues of mature plants. As the plant ages, its chemical composition changes; the content of nitrogen, proteins, and water-soluble substances fall, and the proportion of cellulose, hernicallulose and lignin rises. Soluble C compounds degrade first, followed by structural polysaccharides (hemicellulose and cellulose), with lignin decomposing later at much slower rate (Wessen and Berg, 1985; Summerell and Burgess, 1986; Reber and Schara, 1971). Residues having relatively high lignin contents, low N content or high C:N ratio degrade at a slower rate (Ladd et ai., 1981; Parr and Papendick, 1978). However, more recent work has shown that C:N ratio was closely related to the nature of the plant residue (grain vs iegume), residue placement (Smith et al., 1986), and residue particle size (Jensen, 1994). Lignin is a very complex, slowly degrading compound, and high lignin content retards decomposition.

Lignin is thought to be the major source of polyphenols. The role of lignin as a regulator in the decomposition process has been elucidated in several studies (Meentemeyer, 1978; Berendse et al., 1985). Increasing lignin concentration reduces the decomposition rate of plant residues. High lignin content of plant residues could also enhance nutrient immobilization, especially of nitrogen (Melillo et al., 1982). Simple phenolic substances and other aromatic compounds may *be* present in plant and microbial residues, and are released during biodegradation of aromatic polymers such as lignins (Flaig et al., 1975; Kassim et al., 1982; Linhares and Martin, 1979).

Labeling of plant and model lignins has greatly facilitated our knowledge of the biodegradation and transformations of lignin during humification in soil (Kirk et al., 1977). Within the soil humus, lignin biodegradation studies indicate that lignin is an important substrate for humus formation (Stott et al., 1989).

The use of ^{14}C-labeled substrates has made it possible to more precisely follow the degradation and stabilization in humus of specific carbons (Stott et al., 1989). After one year, (Martin et al., 1980), in a 2-year biodegradation and stabilization of specific crop, lignin, and polysaccharide carbons in soils study, about 10 to 20% of the residual C will be present in the soil biomass, and 80 to 90% of the residual C will be in new humus (Stott et al., 1983a, b). With time, the proportion of residual substrate carbon in biomass will decline and that in humus will increase (Kassim et al., 1982; Stott et al., 1989). In most soils, the biomass constitutes about 2 to 4% of the organic carbon (Anderson and Domsch, 1978; Jenkinson and Powlson, 1976). About 20% of the residual C from readily biodegradable substrates will be associated with the humic acid fraction of soil humus, with some of it being present in aromatic molecules. Martin et al. (1974a) found ^{14}C activity in over 16 phenolic compounds upon Na-amalgam degradation of soil humic following incubation of soil amended with ^{14}C-labeled glucose or wheat straw. Still, the greater part of the residual C is present in peptides and polysaccharides and is released as sugar or amino acid units upon acids hydrolysis (Jenkinson, 1971; Martin et al., 1980; Oades and Wagner, 1971; Stott et al., 1983a; Wagner and Mutatkar, 1968). As Stott et al. (1983a) reported, this would be expected as the majority of metabolized C not released as CO_2 would be transformed into microbial protoplasm, cell wall material, and polysaccharides. Sixty percent or more of most organic residues consist of cellulose and other polysaccharides. Some residues, such as legumes and microbial tissues, contain from 6 to as much as 65% protein (Stott et al., 4989). Most of these materials are very biodegradable, but they will decompose at slower rates than simple sugars and amino acids, especially during the early stages of decomposition. Still, after 6-12 months, Sauerbeck and Gonzalez (1977) reported that about 70 to 85% of the C will evolve as CO_2 in a field decomposition of ^{14}C-labeled plant residues in the various soils study. About 6 to 16% of the residual C will be present in soil biomass (Stott et al., 1983a).

A vast number of residue decomposition studies have found that plant residue disappearance rates generally follow an exponential decay curve. The absolute mass loss is relatively rapid in early stages, but slows with time. This has been expressed by Stott et al., (1994) by the equation:

$$M_t = M_y * e^{-(R_{opt}} \tag{1.1}$$

where M_t is the residue mass per unit area remaining on the surface today and M_y is the mass per unit area remaining on the ground the previous day, R_{opt} is a decomposition constant specific to a residue type and EF, measured as the lower limit of moisture and temperature factors, is the environmental factor determining the fraction of a decomposition day that has occurred during day t. This curve will fit the decomposition pattern of most types of plant residues within the same environment. The key variable is the R_{opt} value. In general, the pattern of decomposition is explained by the chemistry of the organic molecules present in the crop residues. Molecules that are readily degraded, such as sugars, disappear quickly, whereas, recalcitrant lignin and phenolic molecules are degraded very slowly. Usually, a ranking order of decomposition of the organics present in plant litter is as follows: sugars > hemicellulose > cellulose > lignin > waxes > phenols. Varietal differences have been shown to have an impact on decomposition rates of cereal and legume residues (Smith and Peckenpaugh, 1986; Stott, 1992). These differences are likely to be due to the proportions of these compounds.

Residue decomposition rate depends on the amount of residue as well as the chemical and physical quality of the residue. Three pools of compounds are generally identified as one readily decomposable pool including simple sugars, starches, and other proteins, an intermediate pool with non structural carbohydrates, and a more recalcitrant pool including lignin and other structural compounds. These pools along with the environmental factors determine the kinetics of residue decomposition.

Ghidey et al. (1985) established a residue decay equation based on change in residue surface area with time. However, they made an assumption that crop residue consists of solid stems of uniform length and diameter, and that decomposition starts from the outside surface of the material and proceeds linearly inward. Based on what we know, microorganisms attack preferably the most readily degradable part of a plant material first which is the inside part of the stem in this case. In general, stems have more pronounced lignification on the outside surface than in the internal part. Stott et al. (1992) have found that corn and soybean stem surface areas changed insignificantly over time, while leaf area changes were very significant. Steiner et al.(1993) mentioned that decomposition may occur in the stem's interior, leaving the stem exterior (and cover) relatively intact.

1.12. Soil Physical, Chemical and Biological Properties

1.1.2.1. Soil Type

It has been shown that the presence of clay will increase microbial numbers and activity in soil and pure cultures, especially during the early stages of degradation of readily available organic substrates (Filip, 1975). Gregorich et al. (1991) also reported that the rate of decomposition of substrate C was greater in soils with more clay, in a study of the influence of soil texture on the turnover of C through the microbial biomass, For organisms, association with clay may offer a favorable ecological niche because the clay surface may have concentrated substrate for the organisms. Bacteria adhere to both charged and noncharged surfaces, and it has been suggested that surface charges are not important (Oades et al., 1989). However, the interaction of clay particles and cells is dependent on the size and the charge of exchangeable cations and on electrolyte concentration, just as for other negatively charged colloidal particles. The interaction of microorganisms with clays is an area of expanding interest, as clays may prevent the potential spread of a disease-causing organism-e.g., Fusarium-or may protect bacteria and viruses against extremes in the environment and against sferilants (Strozky, 1980). Clays may also increase O_2 uptake by microbial cultures (Filip et al., 1972; Haider et al., 1970; Strozky, 1967). The presence of clay, however, may reduce total C loss as CO_2 through increasing the efficiency of C conversion to biomass and through forming complexes with decomposition products and new humus colloids (Greaves and Wilson, 1973; Greenland, '1971; Martin et al., 1976). In a 10-year study by

Jenkinson (1977), soils with higher clay contents retained greater amounts of the C of added ^{14}C-labeled residues. Guekert et al. (1977) observed that intimate association of glucose, microbial polysaccharide, and bacterial cells with clay reduced the evolution of C as CO_2 during incubation in soil.

Soil texture and soil organic matter have a great effect on residue decomposition. Microbial population and activity are expected to be high with a soil high in organic matter and clay content.

1.1.2.2. Soil Acidity

Hydrogen ion concentration is another factor influencing carbon turnover rates. Each microbial species has an optimum pH for growth and a range outside of which no cell proliferation takes place. Loss of C from organic substrates may be slower in acid soils especially during the early stages of decomposition (Jenkinson, 1971). Consequently, the treatment of acid soils with lime accelerates the decay of plant tissues, simple carbonaceous compounds, or native soil organic matter (Afexander, 1977).

Measurements of pH are important criteria for predicting the capability of soils to support microbiat activity.

1.1.2.3. Soil Fertility

Crop residues play an important role in maintaining soil fertility and productivity by providing a source of nutrients and inputs to organic matter. Soil organic matter is the major source of N, S, P, and many micronutrients in soils. Organic matter is critical to efficient crop production because of its cation exchange and water holding capacities. Crop residues , including roots, are the primary source of organic material added to soil in many cropping systems. They represent a major contribution to nutrient cycling. C and N availability within crop residues along with lignin content greatly influence decomposition rates and N availability to plants. Decomposition of residues with low N contents such as wheat and grain sorghum may result in microbial immobilization of soil and fertilizer N, and effectively reducing N availability to plants (Reinertsen et al.,1 984; Vigil et al., 1991).

1.1.2.4. Soil Microbial Population, Tillage and Management Practices

Soil microbial population in relation with management practices influences crop residue decomposition in the field.

In a 2-year decomposition study conducted on corn, wheat, and soybean residue, Brader (1988) found that bacterial and actinomycete populations were consistently higher on soybean residue in comparison with corn and wheat residue. However, fungal populations were consistently highest on corn residue and lowest on wheat residue. Stott et al. (1989) reported that in arid zone soils, which are predominantly alkaline, the bacteria and streptomycetes would be more active in organic resiclue decomposition. The fungi however, have a much greater biomass (Anderson and Domsch 1973); they are able to grow at lower moisture contents, and are no doubt important contribution to residue biodegradation in desert soils. Soil microbial populations have been found to differ between conventional tillage and no-till systems. Plowing and cultivation accelerate the microbial processes involved in oxidizing organic matter. Doran (1980) reported that no-till had more total biomass than did conventional tillage soils in the surface O-7.5 crn, which was related to an increase in soil water content, percent organic carbon, and nitrogen levels. Doran (1980) also found that these results were reversed at the 7.5-l 5 cm depth. He concluded that this was probably due to the placement of crop residue at depth with plowing, which raised the soil water and organic carbon content.

Changes in soil organic t-natter reactions, as determined by organic car-bon content, have strong implications on the microbial activity. The distribution of organic carbon (OC) in the soil profile is a direct reflection of the management practices in a given soil. The percent of OC tends to be greater in the no-till surface O-7.5 cm than under conventional tillage, although the two systems show similar organic carbon content through the remainder of the soil profile (Dick, 1984; Doran, 1980). The buildup of OC at the surface from no-till management reflects the localized distribution of plants residues on the surface.

1 .1.2.5. Soil Fauna

Soil meso and macro animals are also involved in organic debris degradation in many ecosystems, and interest in their activities is increasing. Soil fauna are known to play a critical role in the biological turnover and nutrient release of plant residues by fragmenting the plant residues, resulting in enhanced microbial activities and grazing of microflora by fauna . Edwards and Heath (1963) reported that when soil animals are excluded from decomposing litter, via small mesh litterbag, fragmentation is insufficient and this leads to reduce consumption by microorganisms. Schaller (1968), pointed out that earthworms and soil insects are ver-y active in the disintegration of organic litter accumulated on soil surfaces. Earthworm activity, greater in no-till systems, has been implicated in increased rates of corn residue breakdown (Zachman et al., 1989). Termite feeding activities were observed in litter decomposition and they accounted for much of the mass loss in a litter decomposition study (Cepada et al., 1990).

Soil macrofaunal activity can have an important effect on residue decomposition in an ecosystem appropriate for their living conditions. Not only do they break down the relatively large particles of residue and trigger the decomposition process, but also feed themselves on the residues, reducing considerably the amount of residues present.

1.1.3. Climatic Conditions

1.1.3.1. Soil Temoerature

Temperature is a major environmental factor for controlling residue decomposition rates in soil. Qrganic residue decomposition rates increase as temperature increases (Stott et al., 1989). Although each species of the soil population has a temperature optimum, the overall optimum range in soils is generally about 20 to 27°C in temperate climatic zones. Below this range, the decomposition rate will decrease and will essentially be stopped when surrounding environs freeze (Stott et al., 1989). In a study on wheat decomposition, Stott et al. (1986) established equations for the relationship between the amount of residue decomposition and temperature. They observed that there was still significant amount of residue decomposition at 0°C, with 12 to 17% $[^{14}C]CO_2$ evolved as CO_2 in 30 days. The decomposition decreased with the temperature.

1.1.3.2. Soi! Moisture and Aeration

Soil moisture status is another important environmental factor regulating residue decomposition (Kowalenko et al., 1978). Favorable moisture /conditions for organic residue decomposition in soils range from about 50 to 90% of the moisture-holding capacity (-50 to -15 kPa) as reported by (Stott et al. 1 989). As the moisture content decreases below 50% of capacity, the activity of the soil organisms decreases, but some biodegradation occurs even at about 2% moisture (-1.5 MPa), which is the permanent wilting point for most pl ants (Focht and Martin, 1979). In a laboratory study on wheat residue decomposition, Stott et al. (1986) found that significant decomposition still occurred at -5 MPa, with 10% of the residue C evolving as CO_2 over one month. Brown (1976), and Griffin (1972) reported that many soil organisms will live and even thrive at water potentials much lower than -1.5 MPa. Wilson and Griffin (1983) estimated that 6 out of 11 basidiomycetes tested grew at water potentials below -10 MPa.

A decreased rate of decomposition of ^{14}C-labeled plant residues in planted soil compared with fallow soil has been attributed to lower microbial activity resulting from restricted aeration (Füer and Sauerbeck, 1968). Linn and Doran (1984) found that aerobic microbial respiration increased with soil water content and reached a maximum at 60% water filled pore space. Above 60% water filled pore space, air became limiting. In well-drained soils, acids and alcohols are formed, but they rarely accumulate in appreciable amounts because they are readily metabolized by aerobic bacteria, actinomycetes, and fungi. The main products of aerobic carbon mineralization are CO_2, water, microbial cells, and soil humus components. In the absence of O_2, organic carbon is incompletely metabolized, intermediary substances accumulate, abundant quantities of CH4 and smaller amounts of H_2 are evoived.

1.1.3.3. Effects of Wettina and Dryinq, Freezinq and Thawinq

Under the low humidity and high temperatures frequently encountered in arid zones, soils are subject to rapid drying following rains and irrigation (Stott et al., 1989). They also reported that in areas where the winter temperatures drop below freezing, soils are subject to freezing and thawing cycles. Shields et al. (1974) noted that the drying and rewetting or the freezing and thawing of soils cause *a* marked flush in CO_2 evolution. A decrease in bacterial numbers upon drying and an increase in soluble amino acids and bacterial numbers following rewetting have been observed by Stevenson (1956). Shields et al. (1974) found that freezing and thawing were more effective than wetting and drying cycles in causing the release of previously stabilized ^{14}C as CO_2 from the soils. The wetting and drying increased the evolution of previously stabilized ^{14}C from 16 to 121% compared to controls kept continuously moist (Stott et al., 1989). Salonius (1983) pointed out that a major factor in the increase CO_2 evolution was related to death of vegetative microbial cells during the freezing or drying process. After conditions become favorable for growth, the surviving organisms quickly decompose the killed cells (Shields et al., 1974).

1.2. Living Roots and Root Decomposition

The value of roots as a source of organic matter is ably demonstrated by the high organic matter content of grassland soils (Cook, 1962). Among the extremely diverse soil microsites, which govern the activity and survival of microorganisms, the soil-root interface plays an important role, particularly in modifying the density, activity and structure of the microbial communities. Plant roots continuously provide the soil with small amounts of a wide variety of easily decomposable materials, thereby creating a rhizosphere effect (Curl and Truelove, 1986). The rhizosphere is a microhabitat for microorganisms, most of thern dependent on soluble exudates from the root (Dormaar, 1990). The microbial and chemical composition of the rhizosphere differs considerably from that in the soil not influenced lby roots (Curl and Truelove, 1986). Billes and Bottner (1981) and Bottner (1982) observed that wheat root litter seemed to disappear faster when living roots were present. The release of all organic material, both soluble and insoluble from roots, occurs during plant growth (Newman, 1985). Cheng and Coleman (1990) reported that living roots had a stimulatory effect on soil organic matter decomposition due to higher microbial activity induced by the roots.

There have been few studies of decomposition of roots in any ecosystem (Berget ai., 1984), and there are numerous difficulties in following the decomposition of roots in the soil under natural conditions (Jenkinson, 1965). However, as Berg et al. (1984) pointed out, not only is quantification of root decomposition necessary, but also it is important to understanding the factors regulating the decomposition process. In a study of in *situ* decomposition of root-derived carbon from wheat, Martin (1989) observed that the decomposition of root-derived organic material, present in the wheat rhizosphere, was more complete in undisturbed soil than when air-dried roots were mixed with moist or air-dry soil. His explanation was based on the assumption that the airdrying and mechanical disturbance killed a large part of the rhizosphere biota present around roots in undisturbed soils. Berg et al. (1987) found that organic matter mass loss, from red clover root decomposition, was fast during the first 13 days (44%) and almost ceased after 30 days when about 29% of the organic material remained. They also noticed that there was no notable difference in mass or nitrogen loss from roots of different diameters. The C:N ratio of the root remains decreased from initially 2527 to 11:13 at the end of the incubation. Root decomposition occurs continuously and peaks in early summer, then declines to low levels during winter, and is in phase with soil temperature (Santantonio et al., 1987). Joslin et al. (1987) also reported that root decomposition rate (% weight loss) was highest during the August-September inter-val, showing a positive correlation with soil temperature when studying the association of organic matter and nutrients with fine root turnover in a white oak stand. Rates of mass losses of roots in a desert soil were equal to or higher than those reported from mesic ecosystems by Whitford et al. (1988).

The hypotheses to test were that there is difference in decomposition rate between cultivars of a given plant species based on their initial chemical and physical composition, and that these characteristics can be used to predict decomposition rate.

The objectives of this study were to: (i) determine decomposition rades for cotton, peanut and sorghum aboveground residues and roots by carbon loss and mass loss; (iii) determine the impact of initial chemical and physical characteristics of the residues on decomposition; (iii) determine if plant species affects decomposition rate observed; (iv) determine changes in the mass-to-specific surface area during decomposition, and (v) develop predictive decay equations for plant residues based on mass loss or CO_2 loss and the chemical and physical characteristics off the residues.

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CHAPTER 2

SURFACE RESIDUE AND ROOT DECOMPOSITION OF COTTON, PEANUT AND SORGHUM FOR USE IN EROSION PREDICTION MODELS

2.1. Abstract

Developing effective management strategies that protect soil against erosion requires an understanding of residue decomposition. While the impact of environmental factors such as temperature and water content has been studied, little has been done to understand how the characteristics of the residue itself impact the decomposition rate. Traditionally, the C:N ratio has been used as a predictor of decomposition rates for agronomic crops, but has recently been shown to be poorly correlated. This study relates the chemical composition of residue components (aboveground biomass and roots) to the decompbsition rates for three cultivars each of three crops: cotton (*Gossypium hirsutum*), sorghum (*Sorghum bicolor*) and peanut (*Arachis hypogaea).* The rat& were determined by mass loss and CO_2 evolution. Clhange in the specific surface area of the residue as related to mass loss was also measured. The three crops were from slowest to the most rapid loss: sorghum > cotton > peanut. From the initial chemical and physical residue characteristics, the following equation was developed to predict decay in the first stage:

P_D = (N*Sugars*Hemicellulose*$K_{in.}$) / Lignin, where P_D is the predictive decay rate, $K_{in.}$ is the initial specific surface area-to-mass ratio. For mass loss, r^2 = 0.96, and for CO_2 evolution, r^2 = 0.95. Since varietal differences within crops

have led to significant variation in decomposition rates, cultivars with slower decaying residues might be recommended for highly erodible lands.

2.2. Introduction

Soil erosion is a problem with many consequences. It can limit soil productivity, denude the landscape, transport sediments, organic matter and pollutants from one place to another. Surface-managing crop residues is a primary method of controlling soil erosion by water or wind. In many areas of the world, insufficient amounts of residue are produced to provide adequate erosion protection. In ‘other areas, the accumulation of crop residues is frequently viewed as a nuisance to crop establishment and growth, and a disposal problem (Elliott et ai., 1987).

The root system of a crop is as important as surface residue in preventing water erosion by limiting lateral runoff. In some areas there is not enough surface residue due to low productivity, burning for management purposes, or utilization as animal feed or even as fuel. In these areas, roots may be the only type of residues left in the field. Consequently, while residue cover may not be sufficient to protect surface soil, roots systems can play a major role in reducing sediment loss from water erosion.

The rate of residue decomposition will determine the amount of soil surface covered during critical erosion periods throughout the year, as well as the amount of residues in top portion of the soil profile. Therefore, understanding the mechanisms of residue decomposition is necessary for developing a viable crop residue management system for erosion control.

Plant residues consist of two parts: the aboveground portion, mainly composed of stems and leaves, and the roots. The aboveground biomass may be standing, flat on the soil surface, or become buried through tillage and other management operations. The physical nature and the initial chemical composition of the plant residues largely determine the ability of microbrganisms to assimilate them. In the traditional agronomic literature, the C:N ratio has been assumed to be a controlling factor, while in the traditionai forestry literature, the lignin-to-N has been considered most important. However, the C:N ratio is apparently not the deterrnining factor, nor is the lignin-to-N ratio solely responsible (Stott, 1992). Decomposition rate for plant residue varies between plant species and between cultivars within a species (Stott, 1993).

Most knowledge about crop residue decomposition is based on above-ground residue, mostly winter wheat (Brown and Dickey, 1989; Knapp et al., 1983; Tanaka, 1985; Stott et: al., 1988 and 1990; Broder et al., 1988; Stroo et al., 1989; Collins et al., 1990; Douglas et al., 1992; Steiner et al., 1993), whereas there have been few studies of decomposition of roots in any ecosystem (Berg et al., 1987; Bottner et al., 1988; Cheng et al., 1990). There may be some difficulties iin following the decomposition of roots in the soil under natural conditions. An *in situ* study of decomposition of rootderived carbon from wheat revealed that the degradation of rootderived organic material present in the wheat. rhizosphere was more complete in undisturbed soil than when air-dried roots were mixed with moist or air-dry soil (Martin, 1989).

The specific-surface-area-to-mass ratio (k) represents a fraction of an area (ha) of soil covered by one kg of residue and is specific for a crop type. The k value is a conversion constant (ha kg^{-1}) used in an equation for converting residue mass to cover (Gregory, 1982):

$$C = 1-e^{(-km)} \tag{2.1}$$

where:

C = fraction of the surface cover remaining

m = mass (kg ha^{-1}) of residue present on the surface

The Gregory equation is currently used in all the USDA erosion models: WEPP (Water Erosion Prediction Project), WEPS (Wind Erosion Prediction

System), RUSLE (Revised Universai Soil Loss Equation), and RWEQ (Revised Wind Erosion Equation).

The residue mass-surface cover relationship is closely related to the levels of residues, and considerable decomposition of mass may occur before a large decrease in cover is measured (Steiner et al., 1993). For residues having high proportion of leaf material following harvest, there may be tremendous loss in mass with little loss in cover, because leaf material decomposes rapidly and is light compared to stem material (Stott, 1992). Stern will lose mass, not surface area.

The objectives of this study were to: (i) determine decomposition rates for cotton, peanut and sorghum above-ground residues and roots by two methods: CO_2 evolution and mass loss; (ii) determine how the initial physical and chemical properties of the roots and residues impact the decomposition rates; (iii) determine if differences in decomposition exist between plant varieties within a species; (iv) determine changes in the mass-to-specific surface area during decomposition; and (v) develop predictive decay equations for plant residues based on mass loss or CO_2 loss and the chemical and physical characteristics of the residues.

2.3. Materials and Methods

2.3.1. Soil

A Russell silt-loam (fine-silty, mixed, mesic Typic Hapludalf) soil was used in this study. It was obtained from the Ap horizon at the Purdue Agronomy Research Center in West Lafayette, IN. The soil was airdried (to minimize microbial action before use), crushed to pass a 2-mm mesh screen, then stored until use. The soil had a pH of 5.3, a total C content of 7.8 g kg^{-1}, and a total N content of 1.2 g kg-‘.

2.3.2. Plant Materials

Plant materials from three crops: cotton (*Gossypium hirsutum*), peanut (*Arachis hypogaea*) and sor'ghum (*Sorghum bicolor*) were used for this experiment. Each crop was represented by three genetically different cultivars. For each cultivar, the residue was split into two residue types (above-grbund biomass ancl toots). These components were used to determine the residue decomposition rates.

Table 2.1. Dates and locations of the crop sample collection.

Crops	Cultivars	Sampling Dates	County	State
Cotton	DLP-5690	9/1 oi93	Sumter Co.	Georgia
	DP-5215	8/1 oi93	Duval Co.	Texas
	HS-46	9/13/93	Pike Co.	Alabama
Peanut	Florunner	9/10/93	Sumter Co.	Georgia
	NC-7	9/25/93	Stoney Creek	Virginia
	NC-11	9/25/93	Stoney Creek	Virginia
Sorghum	Triumph-266	7/14/93	Duval Co.	'Texas
	GW-744BR	10/15/93	Payne Co.	Oklahoma
	NorthrupKing-300	11/23/93	Saluda Co.	Si. Carolina

Plant residue samples were collected by USDA-SCS personnel from fields in several states (Table 2.1), within one or two days of harvest in order to be in unweathered condition and maximize their use. Five plant samples, representatiive of the whole field, were taken as follows: one plant was picked from the center of the field, and the other four were collected each between one corner and the center of the field, avoiding the end rows. When removing the whole plant from the ground, care was taken so that the roots within the top 10-20 cm of the soil did not break apart. The residues were shipped overnight to the National Soil Erosion Research Laboratory (NSERL) in West Lafayette, IN. The leaves and stems (above-ground biomass) were separated from the roots. The residues were gently washed with water to remove any remaining soil and airdried before chemical analysis.

23.3. Chemical Analysis of Plant Materials

Each plant residue component was chemically analyzed for total C content, total N content, simple sugar content and the structural and non-structural carbohydrate contents. Total C and N content were measured by dry combustion (Model CHN-600; Leco Corp., St Joseph, MI). Hemicellulose, cellulose, and lignin contents were determined by sequential fiber analysis (Goering et al., 1970). This fiber analysis system was designed to provide estimates of forage fiber composition.

For the sequential fiber analysis, four different solutions, neutral detergent fiber (NDF), acid detergent fiber (ADF), demineralizing solution, and a potassium permanganate solution were used. The neutral detergent solution was made from sodium lauryl sulfate, ethyl diamine tetra acetic, sodium phosphate dibasic, and water; the acid detergent solution was prepared from hexadecyl trimethyl ammonium bromide, sulfuric acid, and water; the demineralizing solution was a solution of oxalic acid, and the saturated potassium permanganate solution was obtained from potassium permanganate plus silver sulfate mixed with water.

Following is a brief description of the steps involved in the sequential fiber analysis. First, a 0.5-g of ground residue was placed into a Berzelius beaker, and 100 ml of neutral detergent solution was added for digestion on a hot plate for 1 hr. A 4.25 cm glass microfiber filter (Whatman GF/A) was placed into a standard sintered glass crucible (Pyrex 50 ml, C porosity). Neutral detergent fiber residues were then filtered under vacuum onto the glass filter-crucible combination, dried at 105°C for 24 hr, cooled for 20 min in a dessicator, and weighed. The glass filter plus NDF residue was removed from the crucible and placed into another Berzelius beaker for the ADF digestion. Remaining residue from the NDF analysis adhering to the crucible wall was removed with a rubber-tipped glass rod and ADF solution and added to the beaker. The glass filter and NDF residues in the beaker were broken up using the rubber-tip glass rod. Acid detergent solution was then poured into the beaker up to 100 ml for the digestion of the residue. The same procedure described above for NDF was followed for ADF determination in the second step. In the third step, the crucible containing ADF residue was placed into a shallow pyrex pan. About 1/3 to 2/3 cm of water was added to the pan. Enough of the permanganate mixture was added to the crucible to wet the sample. The residue was again broken up with the glass rod in the crucible. Then the crucible was allowed to stand for 1.5 hr, while stirring every 15-20 min, and adding more of the mixture if necessary. After filtration, the crucibie is placed in a clean pyrex pan and filled half full with demineralizing solution. After rinsing the residue several times with the dernineralizing solution. the finished fiber should be white. Then, the crucible was washed 3-4 tirnes with ethanol 80%. The white residue was dried at 105°C overnight, cooled for 20 min in a dessicator, and weighed. Afterwards, the crucible was put into a muffle furnace, at 500°C for the ash determination. After 4 hr, the crucible was removed from the muffle furnace, put back in the 1 05°C oven ovemight, cooled in a dessicator and weighed. NDF was calculated as the ratio between the sample weight after digestion with NDF solution and the initial sample weight times the sample dry matter; ADF was the ratio between the sample weight after digestion with ADF solution and the initial sample weight times the sample dry matter; hemicellulose was determined as the difference between NDF and ADF; lignin content was assumed to be known as the remaining of the residue sample after digestion; cellulose was determined as the difference between lignin and ash (Chemey et ai., 1985).

Two plant monosaçcharides, or simple sugars, sucrose and fructose were measured colorimetrically. Sucrose analysis (Handel, 1968), was determined by placing into a small test tube, a 100 µl aliquot extracted from a 1:1 weight-volume ratio of finely ground residue and 50% ethanol solution. 100 µl of 30% KOH was added to destroy the sugars. Then the test tube was placed into a boiling water bath for 10 min, and cooled to room temperature. Ptior to mixing on vortex-type mixer, 3.0 ml of anthrone reagent was added. The samples were Yncubated at 40°C for 15 min before reading the absorbance on a spectrophotometer set at 620 nm.

Fructose analysis (Davis et al., 1967) was determined using 100 µl aliquot from the same extract that was used to determine sucrose. To each sample, 3 ml of conoentrated HCI was added plus 1 ml of 0.05% resorcinol reagent. The sample was well mixed on a vortex-type mixer, and incubated in a water bath set at 77°C for 8 min. Then the samples were allowed to cool to room temperature just prior to measuring absorbance at 420 nm on the spectrophotometer.

2.3.4. Plant Residue Mass loss Experiment

The mass loss experiment consisted of a randomized complete block design with one soil, three crops, three cultivars for each crop, and two residue types (above-ground biomass and roots) for each cultivar. The treatments were done in triplicate.

Each treatment consisted of leaves and stems in the same proportion as was present in the aboveground biomass after harvest. Roots were incubated separately (Table 2.2).

Table 2.2. Plant residue corrponents and loading rates.

Crops	Leaves		Stems		Roots	
	(%)	(g/100g soil)	(%)	(g/100g soil)	(%)	(g/100 g soil)
Cotton	45.0	0.90	55.0	1.10	100	2.00
Peanut	26.5	0.57	71.5	1.43	100	2.00
Sorghum	42.5	0.85	57.5	1.15	100	2.00

Residues were chopped into 4 to 5-cm long and the pieces were spread evenly on the soil surface in a 10 by 7.5 cm^2 polystyrene dish. Optimum moisture conditions were assumed to be the water content at -1/3 bar water potential as equalled to 60% water holding capacity, plus 300% of the re$idue mass (Myrold et al., 1981). Afler the appropriate amount of water was added, the incubation dish was loosely wrapped with a food service film (PYA / Monarch, Inc., Greenville, SC), to allow some aeration. The samples were incubated at $22^oC \pm 1^oC$.

Samples were withdrawn on day 3, 7, 14, 28, 56, and 84 of the incubation for mass measurement. At each destructive sampling, the incubation mixture were oven-dried at 40^oC, for 48 hr. When dry, the residues were carefully separated from the soil, gently washed to remove the soil particles, and put back into the oven at 40^oC for 48 hr. The residues were weighed then placed into crucibles for ashing at 800^oC for 2 hr.

The equations used to calculate the percent mass remaining were:

$$M_T = M_F - M_A \quad (2.2)$$

$$\% M_R = (M_T / M_I) * 100 \quad (2.3)$$

where:

M_T = corrected mass (g) remaining at time T

M_F = mass (g) of the residue after incubation (oven dry basis)

M_A = mass (g) of the ashed residue

M_R = % of initial mass remaining at day T

M, = initial residue mass (g)

T is the incubation time in days

2.3.5. CO, Evolution

A second method for determining decomposition rate is to measure the amount of C evolved as CO_2. To monitor microbial respiration, a known mass of residue, chopped into 4 to 5-cm lengths was spread evenly on 100 g airdried soil in an incubation jar. Addition of an amount of water to achieve the water content at -1/3 bar water potential as equalled to 60% water holding capacity plus 300% residue mass (Myrold et al., 1981) gave optimum moisture conditions of residue decomposition. An alkaline trap, 5 ml of a 30% KOH plus tropaelin 0 as indicator, in a 25-ml beaker was placed in each jar on top of the soil and residue. Tropealin 0 (Sigma Chemical Co, St. Louis, MO) was used to check if the KOH solution has reached a 50% CO_2 saturation (pH 11). Respired CO, was absorbed in the KOH trap.

Each jar was placed into a circulating water bath, set at 22°C ± 1°C, and hooked to an electrolytic respirometer. At the top of the respirometer, there was a 25 or 50-ml burette, a positive electrode for oxygen, and a 4-cm tube for overflow. At the bottom, there was a negative electrode for hydrogen. Both electrodes were platinum. The positive electrode was connected to a 500-ml chamber containing the electrolyte solution 8% $(Na)_2SO_4$. KOH was withdrawn after 3 , 7 ,14 ,28 ,56 and 64 days of incubation. To remove all of the KOH, a 22-gauge needle with a Luer-lock fitting was inserted into the jar stopper and lengthened with a piece of capillary tubing to reach the bottom of the KOH trap. Fresh KOH was injected in the same manner. The amount of CO, trapped in the KOH was measured by a potentiometric method (Golterman, 1970) using an automatic titrator (Model DL 25, Mettler instrument Corp., Hightstown NJ).

The CO_2 evolution experiment used the same statistical design as the mass loss experiment with the addition of a control treatment (no residue). To correct the amounl: of CO_2 evolved from the residues, CO_2 evolution from the bare soil (control treatment) was sustracted from CO_2 evollved from treatments (soil plus residue) at each given time.

The reactions involved in KOH trapping the evolved CO2 are as follows:

$$KOH + CO, \longrightarrow HCO, + K+ \quad (2.4)$$

$$HCO, + K+ + HCl \longrightarrow H_2CO_3 + KCl \quad (2.5)$$

Each milliequivalent of 'KOH used to absorb evolved CO, is equivalent to 12 mg of CO, carbon. The formula used to calculate % C-CO, evolved is:

$$\% \text{ C-CO}_2 = [\, K_1 * (1/M) * V * N * C_i \,] \quad (2.6)$$

where:

K, = 0.135, a calculated constant to convert the raw result into the desired unit

M = the mass (g) of the residue

V = the volume (ml) of HCl titrant

N = the concentration (N) of HCl titrant

C_i = the initial carbon content (%) of the residue

2.3.6. Measurement of Specific Surface Area-to-Mass Ratio

Specific surface areas for the leaves and stems were measured using a digitizer (Summagraphics) arnd AutoCad version 10. As decomposition proceeded, the ratio between the specific surface area and the mass remaining was calculated at each sarnpling time.

The equation used to convert residue mass to cover is from Gregory (1982):

$$C = 1 - e^{(-km)} \tag{2.7}$$

where:

C is the fraction of the surface cover remaining

m is the mass (kg ha-‘) of residue present on the surface

The constant k can be derived from the following equation:

$$k = - \log(l\text{-}C) / m \tag{2.8}$$

2.3.7. Statistical Analysis

Statistical analysis of the data was done to deterrnine differencas among treatmenfs, using the PC-SAS, Version 6.09 (SAS Inc., Cary NC). Comparisons between treatment means were made at the P =0.05 level using the Waller-Duncan's multiple range test procedure.

2.4. Results

2.4.1. Initial Chemical Composition

The mean concentrations of total C and N, simple sugars, hemicellulose and lignin {Tables 2.3 and 2.4) were significantly different between the above-ground residues and roots for cotton cultivars DLP-5690 and DP-5215 For DLP-5690 above-ground residues, the total N content was 288% greater than the roots, whereas total carbon, simple sugar, hemicellulose and lignin contents were 3, 30, 22 and 17% lower, respectively. For DP-521 5 above-ground biomass, total N was 147% higher than the roots, whereas total carbon, simple sugar, hemicellulose and lignin contents were 5, 40, 49 and 51 % lower respectively. Cultivar HS-46 above-ground residues had 232% greater total N concentration than the roots, but total C was 0.3% lower, hemicellulose 8% lower, and lignin

15% lower. Simple sugar concentrations of the above-ground biomass were 177% lower than the roots.

Far peanut, the initiai chemical composition (Tables 2.3 and 2.4) of the aboveground residues were significantly different from the roots, except for total C. Cultivar Florunner above-ground biomass had 88% higher simple sugar concentrations than the roots, but total N was 44% lower , hemicellulose 26% lower, and lignin 32% lower. For cultivar NC-7 above-ground residues, simple sugar contents were 31 % greater than the roots, but total N was 44% lower, hemicellulose 65% lower, and lignin 32% lower as well. Cultivar NC-11 above-ground biomass had 27% higher simple sugar concentrations than the roots, but hemicellulose and lignin were lower by 56% and 35% respectively.

Table 2.3. Initial chemical composition of the above-ground residues.

Crop	Cultivar	Total C	Total N	Sugars	Hemicellulose	Lignin
		g kg" residue				
Cotton	OLP-5690	448.9 a'	31.4 a	18.1 c	252.4 b	112.1 a
	DP-521 5	437.1 b	19.3 b	23.1 b	133.1 c	80.7 c
	HS-46	457.3 a	30.9 a	34.0 a	262.5 a	103.3 b
Peanut	Florunner	450.4 a	13.4 b	89.9 a	176.6 a	64.8 a
	NC-7	455.2 a	20.0 a	87.7 a	140.0 b	42.3 c
	NC-11	450.4 a	18.8 a	66.8 a	108.2 c	50.4 b
Sorghum	Triumph-266	438.2 c	11.9 b	41.1 b	208.3 c	47.6 a
	GW7-44BR	452.5 a	17.8 a	32.5 c	327.1 a	32.5 b
	NKing-300	447.9 b	6.9 c	48.7 a	273.7 b	48.2 a

'Values followed by the same letter, within speciies, are not significantly different by the Waller-Duncan's multiple range test at P = 0.05.

Sorghum above-ground residues and roots (Tables 2.3 and 2.4) were significantly different in initial total C and total N, simple sugar, hemicellulose , and lignin concentrations. For cultivar Triumph-266 above-ground residues, total N content was 86% greater than the roots, hemicellulose was 22% greater, but simple sugar and lignin contents were 37% and 41 % lower than the roots respectively. Cultivar GW-744BR above-ground biomass had total N and hemicellulose concentrations of 76 and 9% greater than the roots respectively, but simple sugar and lignin contents were 76 and 41% lower respectively. For cultivar Nking-300 above-ground residues, total C content was 15% higher than the roots but total N, simple sugar, hemicellulose, and lignin concentrations were 22, 67, 14 and 44% lower respectively.

Tables 2.3 and 2.4 indicated significant differences in initial chemical composition between cultivars within species.

Table 2.4. Initial chemical composition of the plant roots.

Crop	Cultivar	Total C	Total N	Sugars	Hemicellulose	Lignin
				g kg^{-1} residue		
Cotton	DLP-5690	463.2 a	8.1 ab'	26.0 c	322.6 a	135.7 b
	DP-521 5	458.9 a	7.8 b	38.5 b	261.2 c	163.1 a
	HS-46	458.9 a	9.3 a	94.3 a	283.5 b	121.5 c
Peanut	Florunner	452.4 a	24.0 b	47.7 b	238.8 c	95.3 a
	NC-7	436.8 b	26.8 b	66.7 a	398.9 a	61.9 c
	NC-11	456.3 a	31.3 a	68.5 a	247.5 b	77.3 b
Sorghum	Triumph-266	404.5 a	6.4 b	65.6 c	266.8 c	80.7 b
	GW-744BR	346.0 c	10.1 a	132.7 b	360.2 a	55.1 c
	NKing-300	388.0 b	8.9 a	148.8 a	317.6 b	86.5 a

'Values followed by the same letter, within species, are not significantly different by the Waller-Duncan's multiple range test at P = 0.05.

2.4.2. Initial Specific Surface Area

For cotton, the specific: surface area (Table 2.5) of the leaves and stems before the incubation did rot significantly differ between the cultivars. The specifk surface area of DL.P-5690, DP-5215, and HS-46 leaves was 101, 73 and 85% greater than the stems respectively.

No peanut cultivar was significantiy different from one another for the above-ground specific surface area (Table 2.5). The specific surface area of the leaves was significantly greater than the stems by 95% for Florunner, 235% for NC-7, and 1113% for NC-l 1.

The initial specific surface area of the sorghum leaves and stems (fable 2.5) showed significant differences between cultivars except for GW-744BR. Triumph-266 leaf specifk surface area was greater by 45% than that of Xhe stems. GW-744BR leaf specific surface area was not significantly different from that of the stems. Nking-300 leaf specific surface area was 87% higher than that of the stems. The leaf specific surface area for Triumph-266 was also 18% greater than that of GW-744BR, but 9% lower lower than that of Nking-300. GW-744BR leaf specific surface area was 23% lower than that of NKing-300.

2.4.3. Initial Residue Mass

For all species, the stem mass was much greater than the leaves (Table 2.5). Within cotton species, cultivar HS-46 above-ground residue mass was higher than those of cultivars DP-5215 and DP-5690. No difference was noted between the initial mass of the roots of these three cultivars.

For peanut, there was no significant difference in either the above/round residue or tlhe root mass between cultivars Florunner, NC-7 and NC-1 1.

Sorghum cultivar GW-744BR presented greatet above-ground residue mass than Triumph-266 and Nking-300. However, GW-744BR root mass was lower than those of Triumph-266 and Nking300 which were not different.

Table 2.5. Relative initial mass and specific surface area of the residue components.

Crops	Cultivars	Relative Initial mass (%)			Relative initial specific surface area (%)	
		Leaves	Stems	Roots	Leaves	Stems
Cotton	DLP-5690	38.5 a[1]	43.9 ab	17.6 a	66.8 a	33.2 a
	DP-5215	34.4 b	49.1 a	16.5 a	63.4 a	36.6 a
	HS-46	40.8 a	45.9 ab	13.3 b	64.9 a	35.1 a
Peanut	Florunner	24.3 b	69.5 a	6.2 a	65.4 b	33.6 a
	NC-7	27.8 ab	67.5 ab	4.7 a	77.0 a	23.0 b
	NC-I 1	29.4 a	65.1 b	5.5 a	68.0 b	32.0 a
Sorghum	Triumph-266	36.9 a	44.5 b	18.6 a	59.2 b	40.8 b
	GW-744BR	33.2 b	52.5 a	14.3 b	50.2 c	49.8 a
	NKing-300	36.2 a	46.9 b	16.9 ab	65.1 a	34.9 c

[1]Values followed by the same letter, within species, are not significantly different by the Waller-Duncan's multiple range test at P = 0.05.

2.4.4. C lost as CO_2

One method of determining residue decomposition rates is to measure the amount of C evolved as CO_2 after correction for the amount evolved from bare soil. For cotton residue, C evolved as CO_2 increased rapidly during the first fourteen days of incubation then started leveling off from day 15, and then showed no significant change after 28 days until the end of the expetiment.

Cultivar OLP-5690 above-ground biomass (Figure 2.1) showed cumulative C lost as CO_2, after 14 days, 35% which was significantly greater than the 22% evolved from the roots. For cultivar DP-521 5 above-ground residues (Figure 2.2), C lost, 30%, was greater than that of the roots, 10%. Cultivar HS-46 (Figure 2.3), showed no significant difference in cumulative CO_2 evolved between the above-ground biomass, 30%, and the roots, 27%.

The decomposition rates differed among the cotton cultivars. DLP-5690 (Figure 2.4) above-ground residues were degraded faster than DP-5215 and HS-46 above-ground biomass. The latter two cultivars did not degrade at significantly different rates. Cumulative CO_2 evolution of the roots for DLP-5690, DP-5215 and HS-46 (Figure 2.5) induced a different scenario with cultivar HS-46 root decay rate (Table 2.6) being fastest followed by OLP-5690 roots, and DP-521 5 presented the slowest decomposition rate.

The total carbon evolved from the peanut cultivars Florunner, NC-7 and NC-11 above-ground residues (Fiigures 2.6, 2.7, and 2.8) was rapid during the first 14 days, 57, 53 and 50% respectively. The C losses were significantly higher than the roots, 15, 10, and 7% lost, respectively. Florunner above-ground residues were significantly greater in C loss than that of NC-7 and NC-1 1 (Figure 2.9). Also, Florunner roots (Figure 2.10) were significantly different than that of cultivars NC-7 and NC-l 1 in C evolved as CO_2.

As a result, the decomposition rate of Florunner above-ground residues (Figure 2.9) was significantly higher than NC-7 and NC-1 1 above-grouncl residue decay rates, and Florunner root degradability (Figure 2.10) were significantly greater than that of cuftivars NC-7 and NC-l 1 roots.

Sorghunn cultivars Triumph-266 and GW-7448R showed significant difference in % C evolved as CO_2 in the first 14 days (Figures 2.11, and 2.12) between the above-ground, 23 and 45% CO_2-C, respectively, and the roots, 18 and 34% CO_2-C, respectively.. For cultivar Nking-300 above-ground residues, (Figure 2.13), the cumulative % C lost as CO_2 was lower, 33% than that of the roots, 40%. Consequently, GW-744BR above-ground residues (Figure 2.14) and Nking300 roots (Figure 2.15) had fastest decomposition rate whereas Triumph-266 and GW-744BR roots were decomposed very slowly (Table 2.6).

Peanut above-ground residues decay rate (Figure 2.16) decomposed significantly faster than cotton and sorghum. Cotton and sorghum above-ground biomass decomposition rates were not significantly different from one another. Sorghum roots have a faster decay rate than either cotton or peanut roots (Figure 2.17).

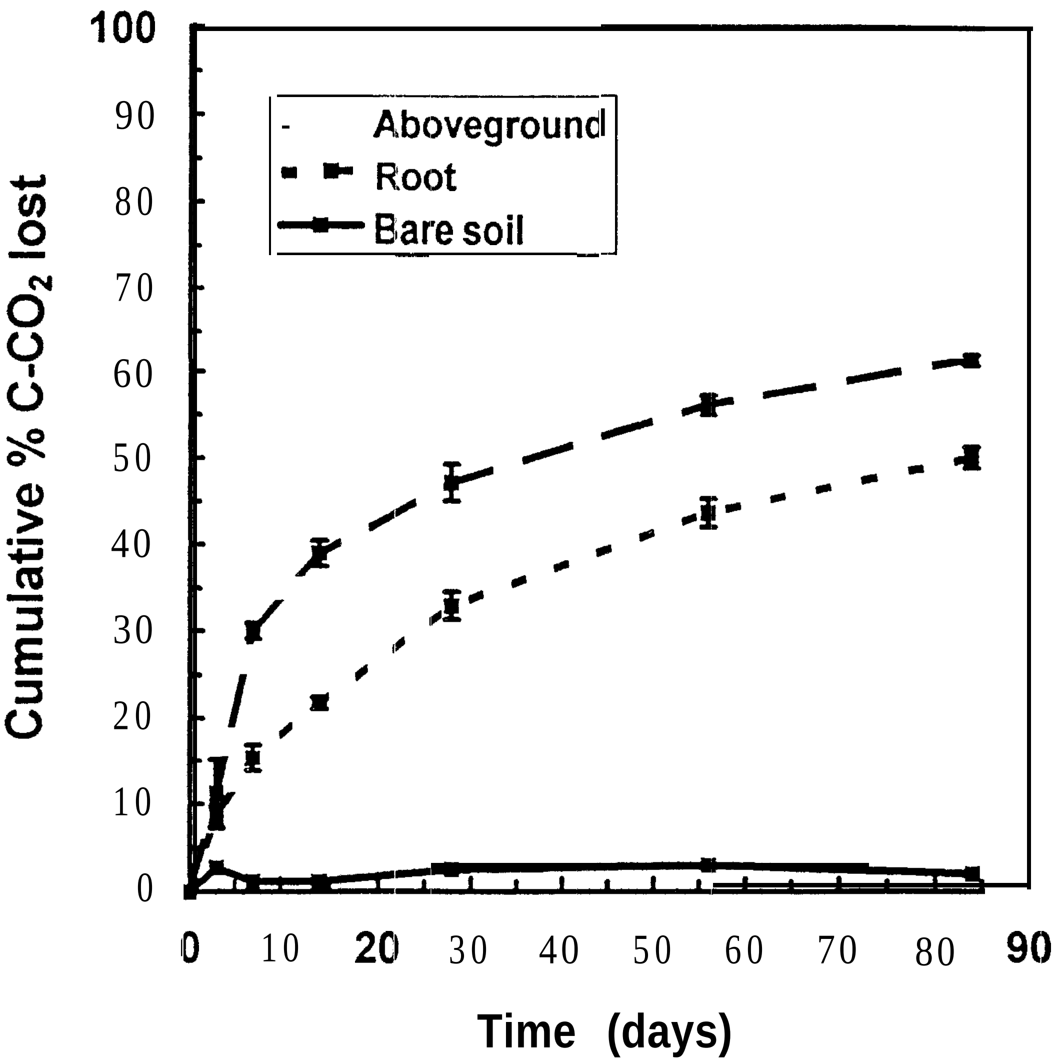


Figure 2.1 Decomposition of cotton DLP-5690 as rmeasured by CO_2 evolution over time. Bars represent standard deviations at given time. CO_2 evolved from the bare soil was used to correct the CO_2 evolution from treatments with residues.

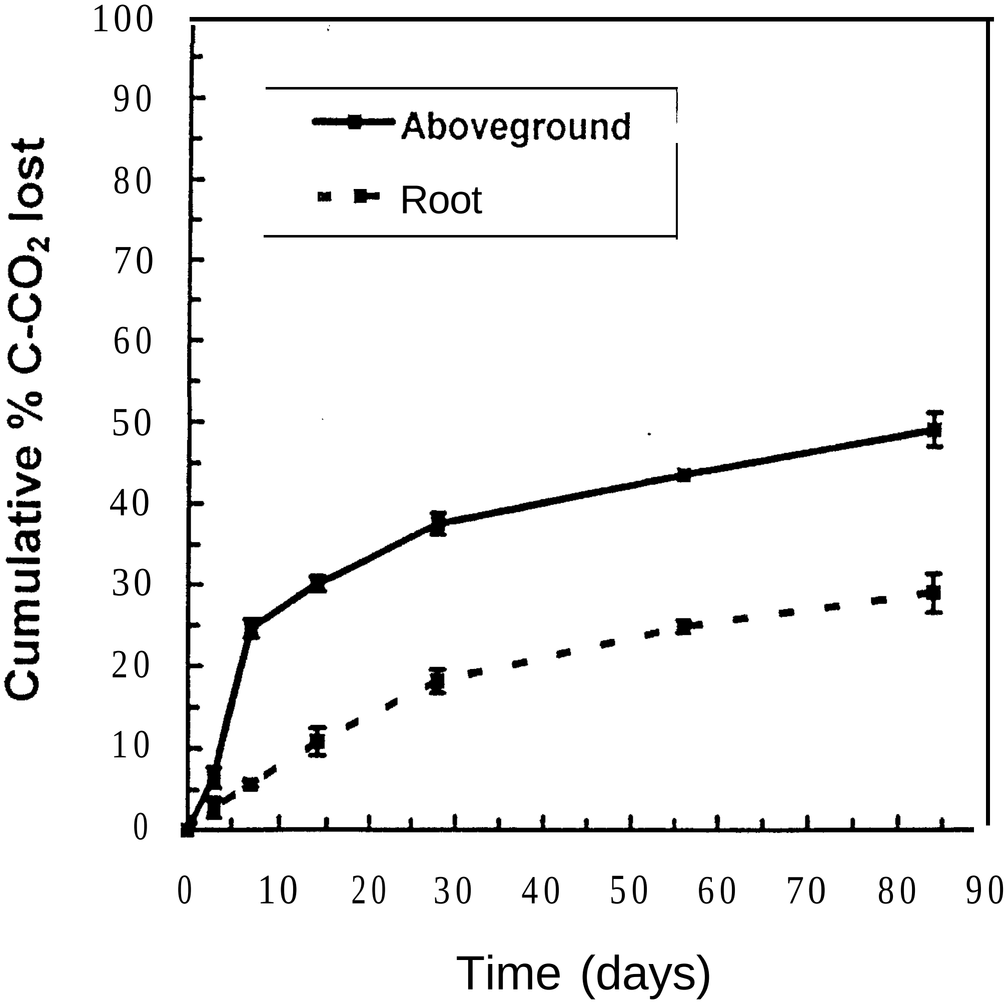


Figure 2.2. Decomposition of cotton DP-521 5 as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

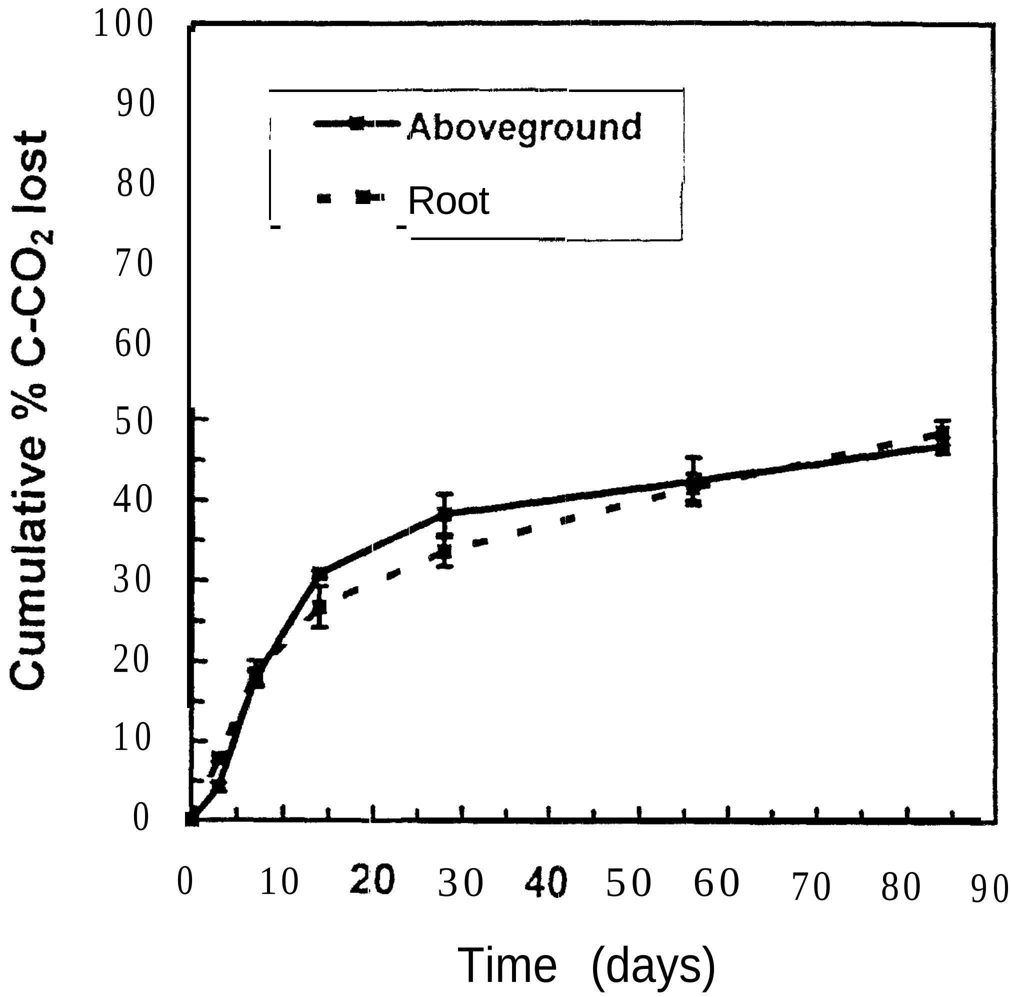


Figure 2.3. Decomposition of cotton HS-46 as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

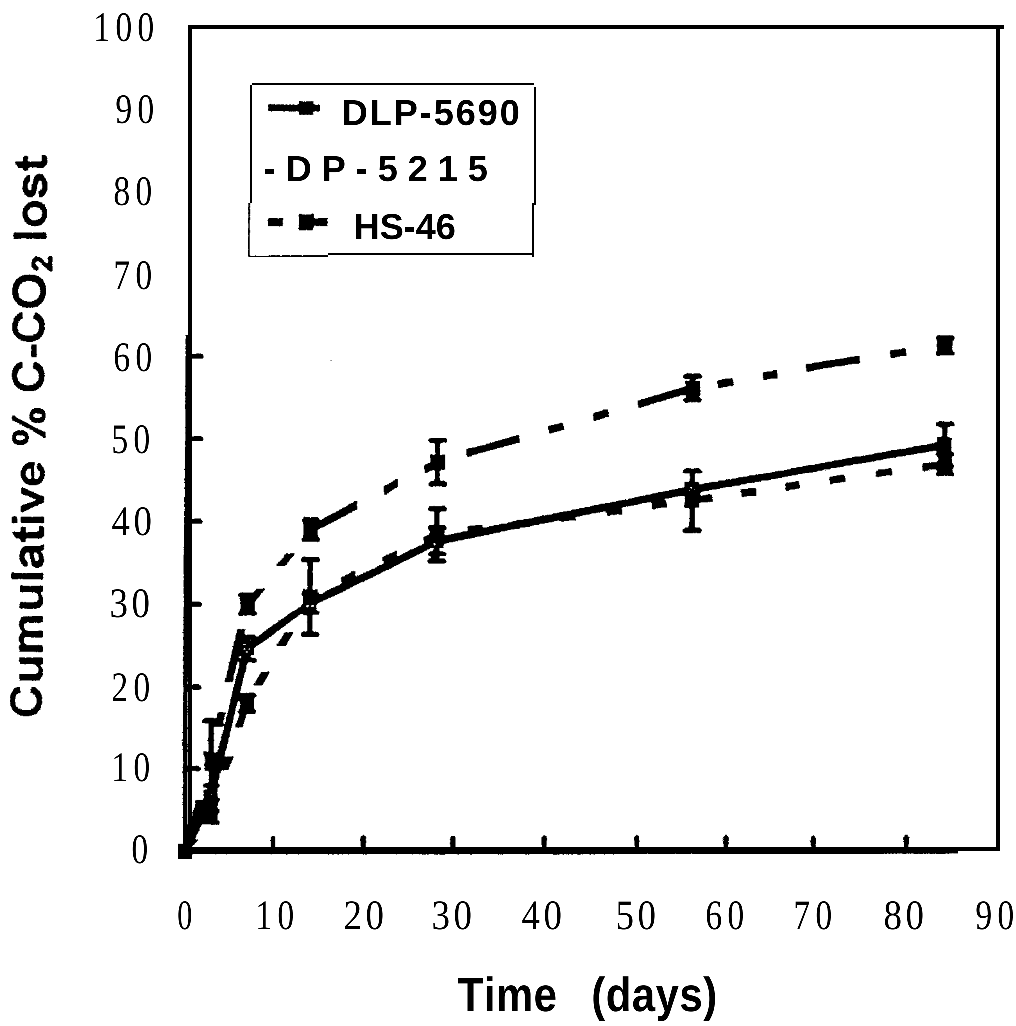


Figure 2.4. Decomposition of cotton above-ground biomass as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

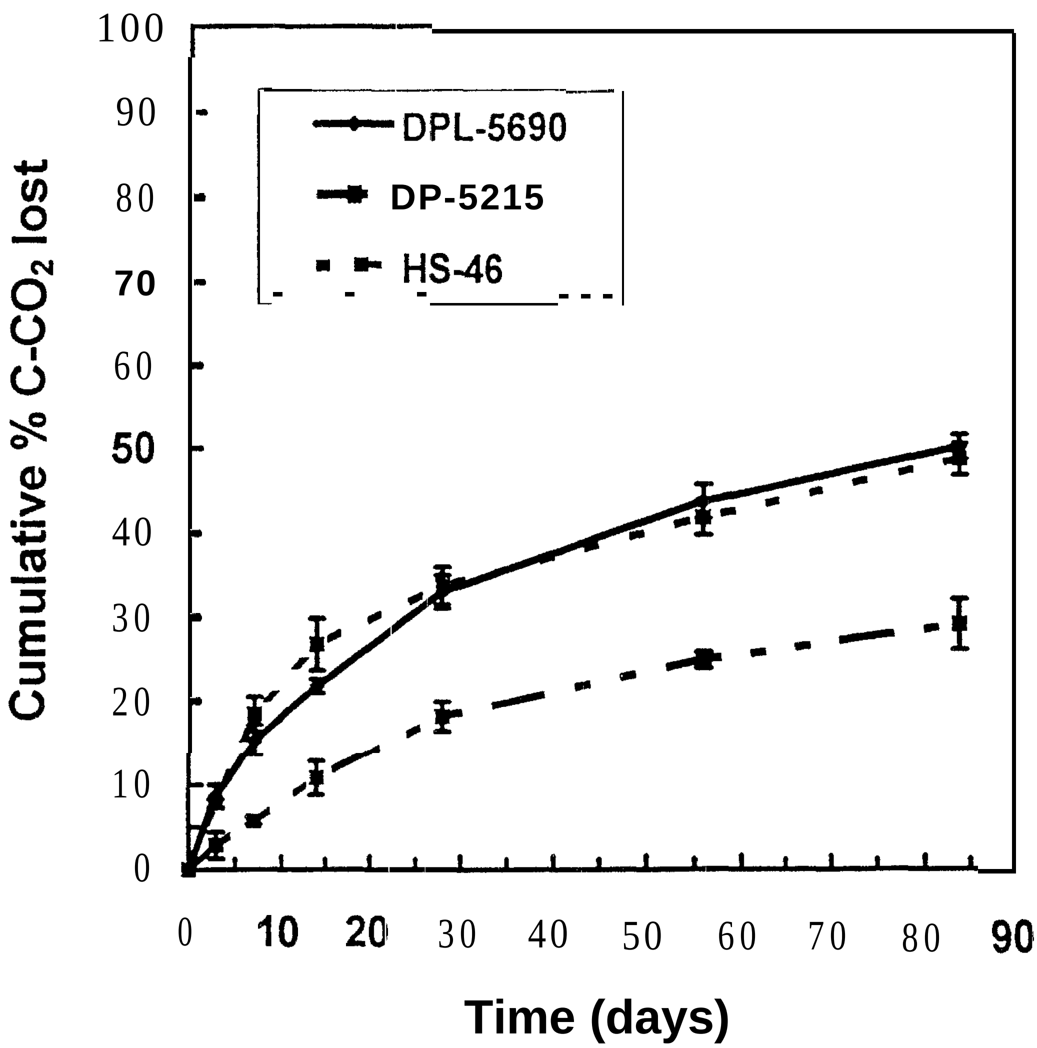


Figure 2.5. Decomposition of cotton roots as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

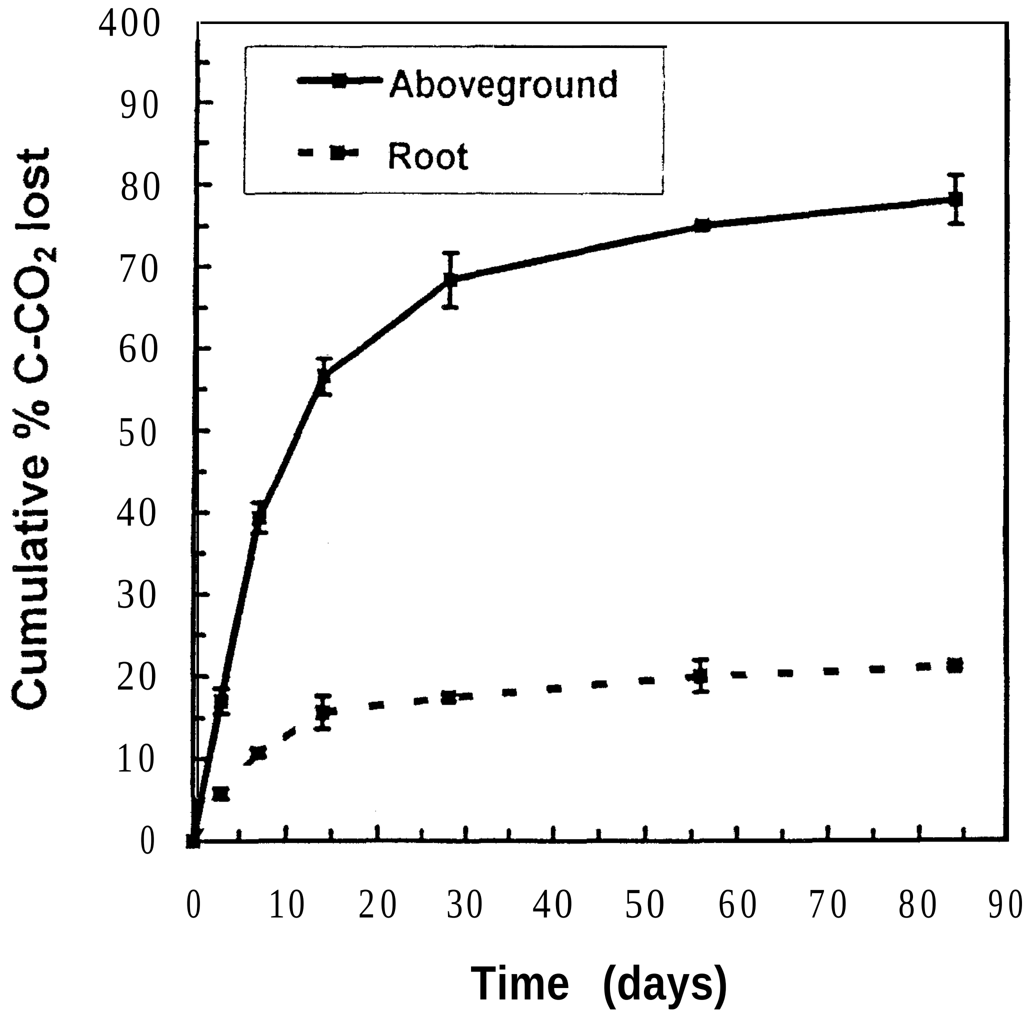


Figure 2.6. Decomposition of peanut Florunner as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

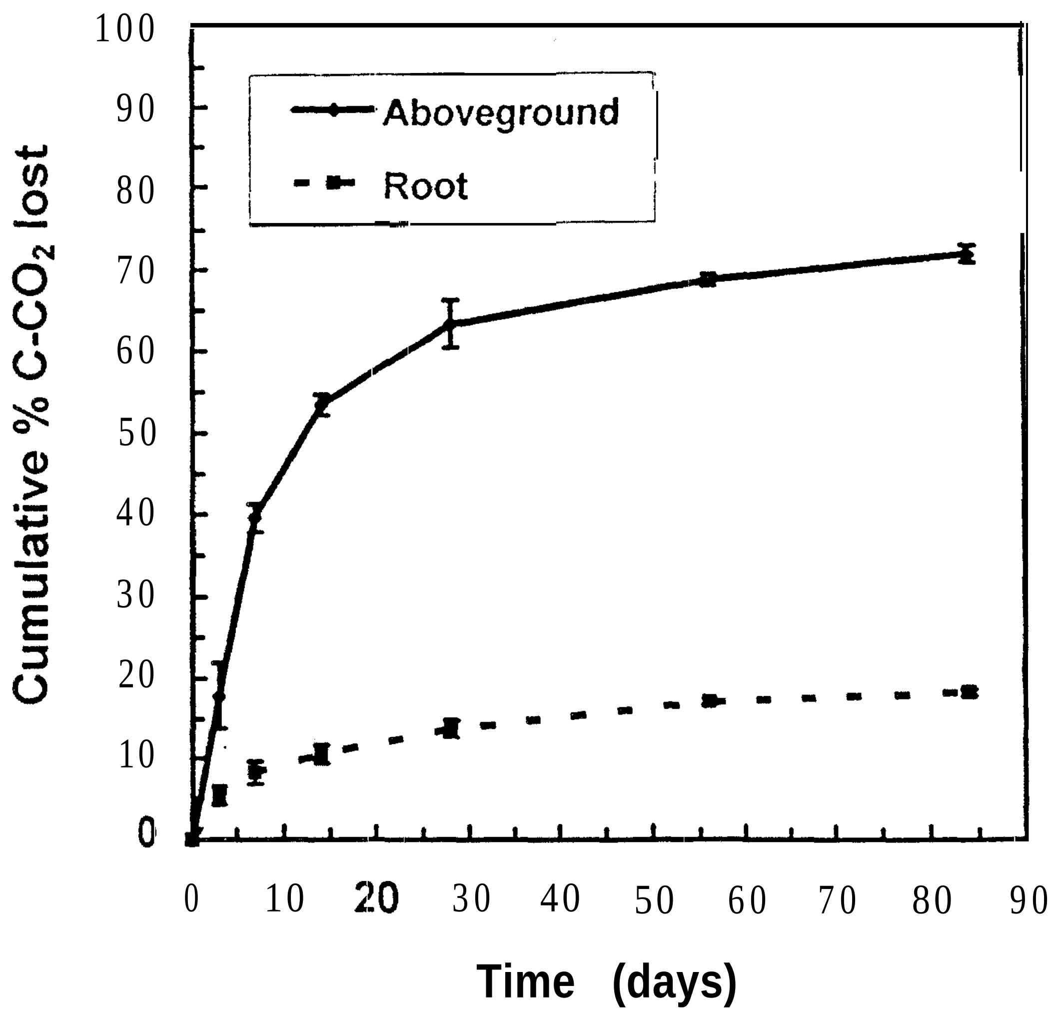


Figure 2.7. Decomposition of peanut NC-7 as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

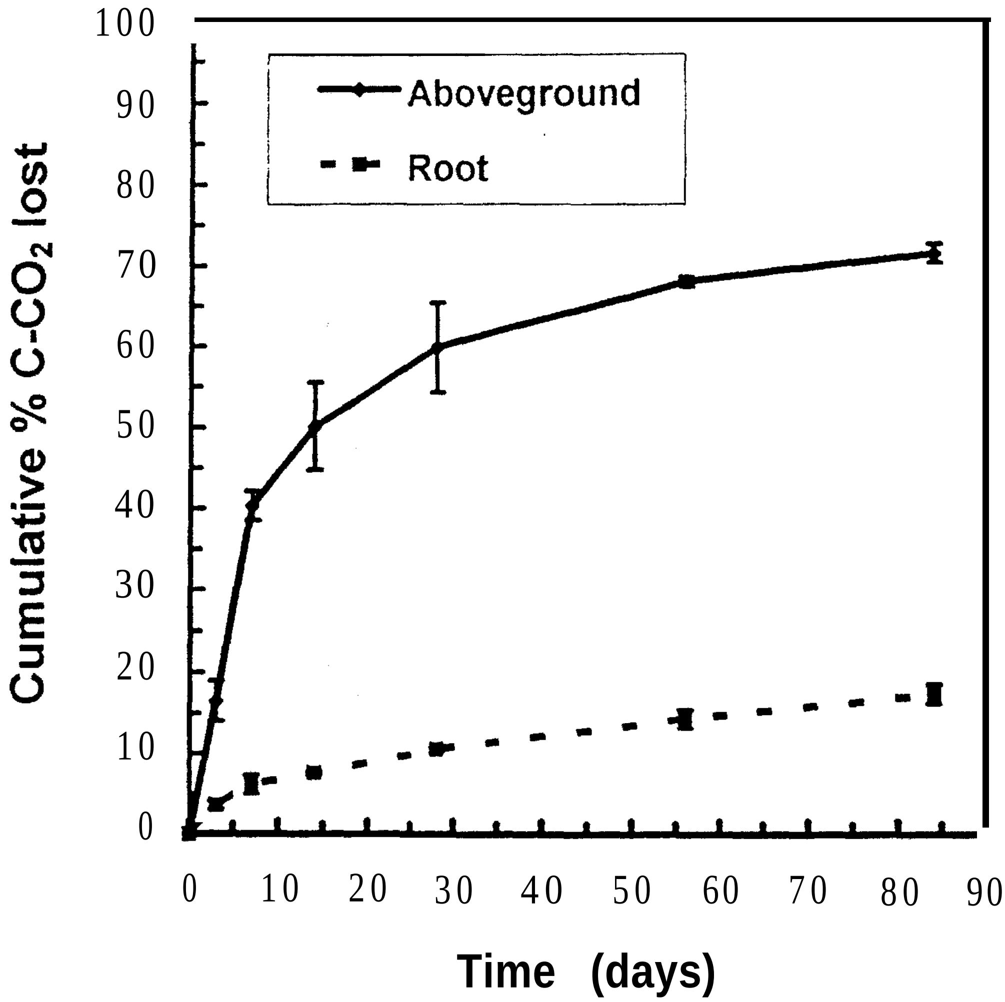


Figure 2.8. Decomposition of peanut NC-l 1 as determined by CO_2 evolution over time. Bars represent standard deviations at given time.

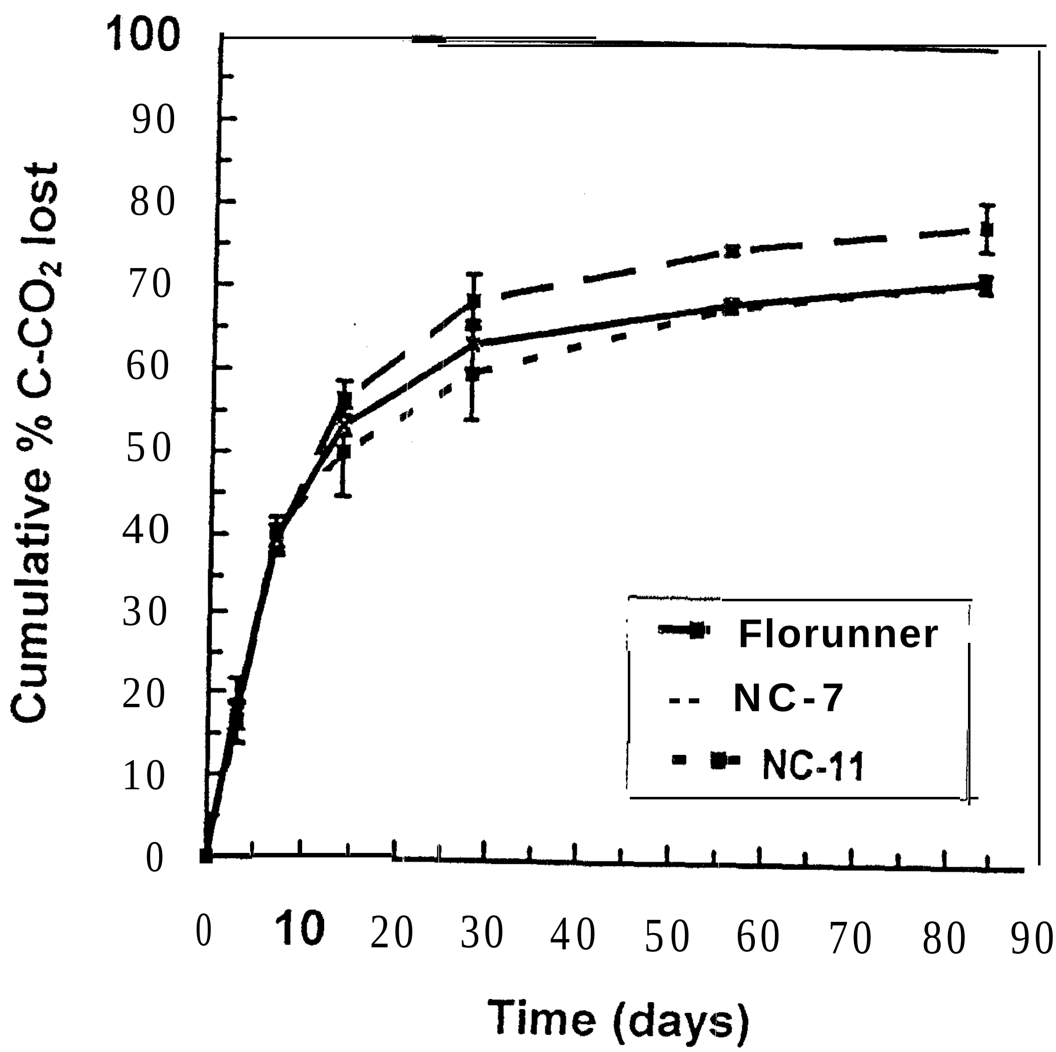


Figure 2.9. Decompositior: of peanut above-ground biomass as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

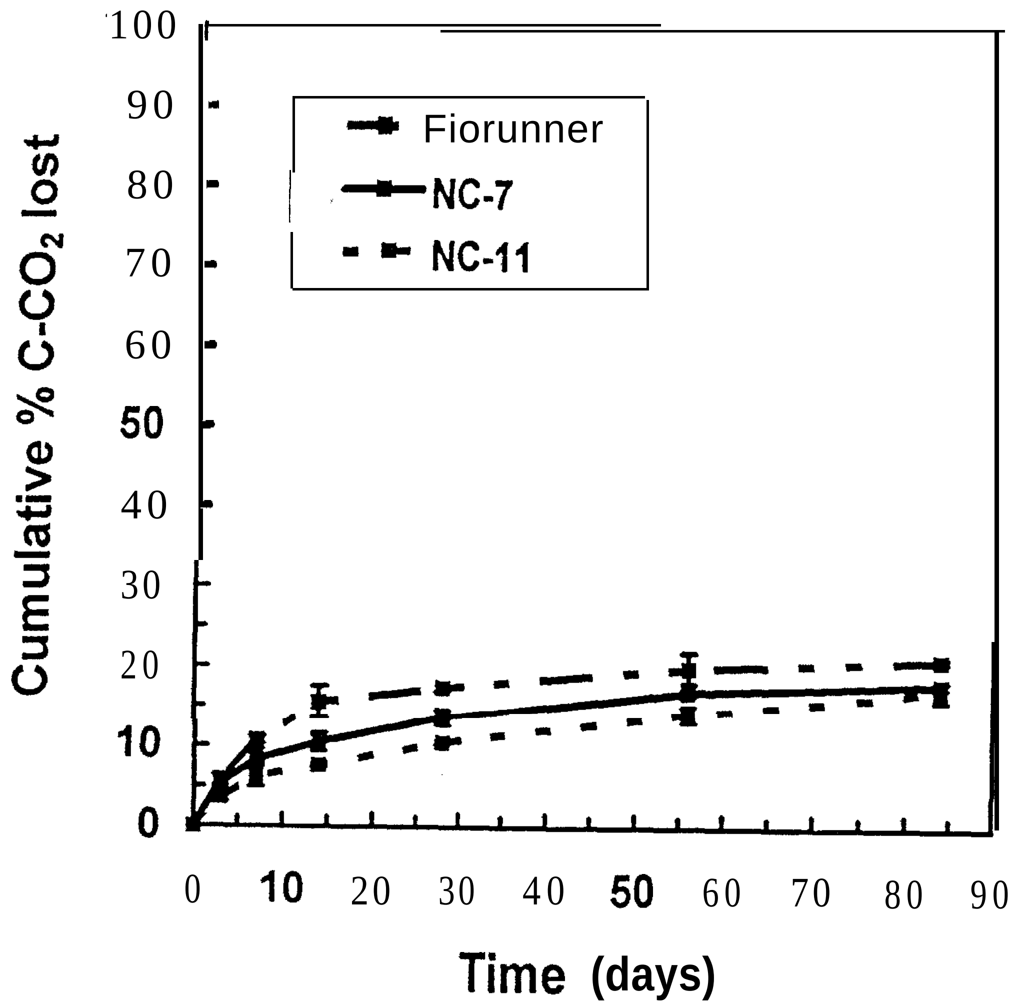


Figure 2.10. Decomposition of peanut roots as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

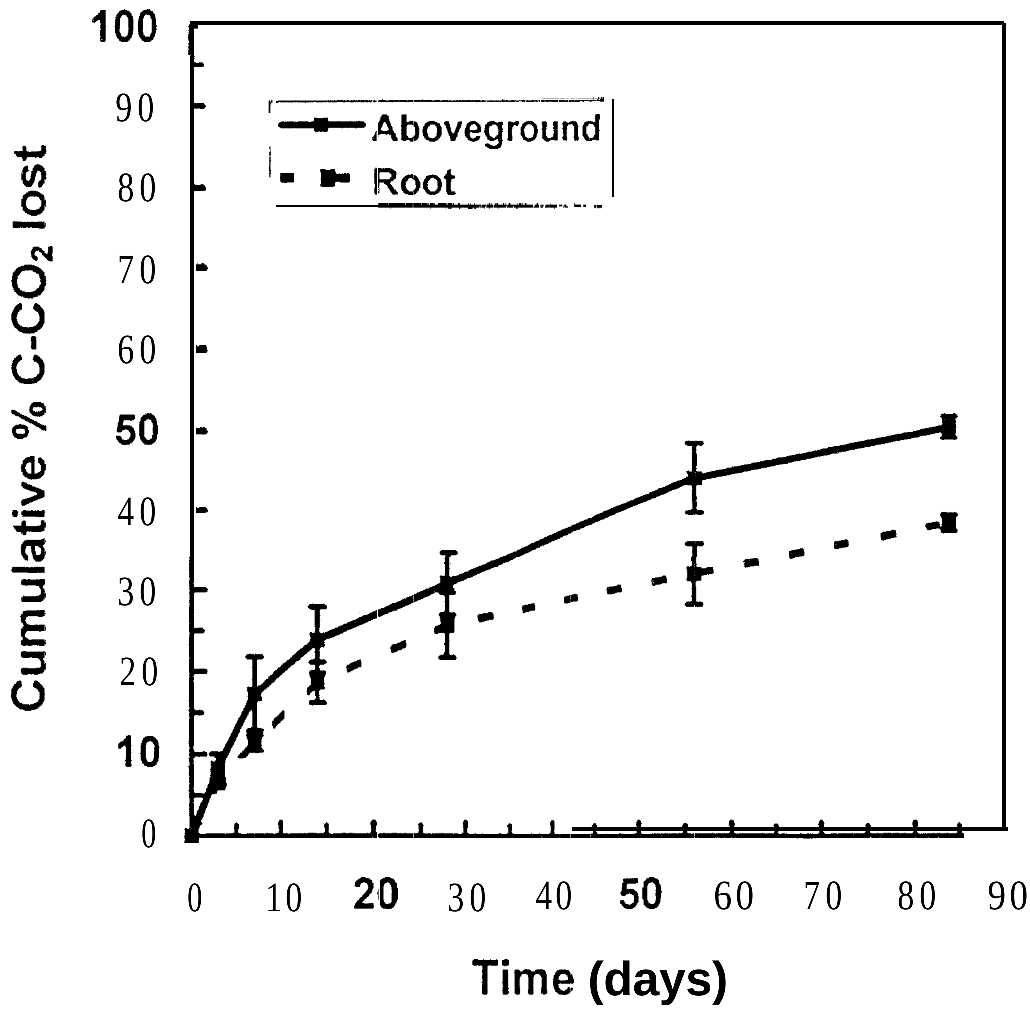


Figure 2.11. Decomposition of sorghum Triumph-266 as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

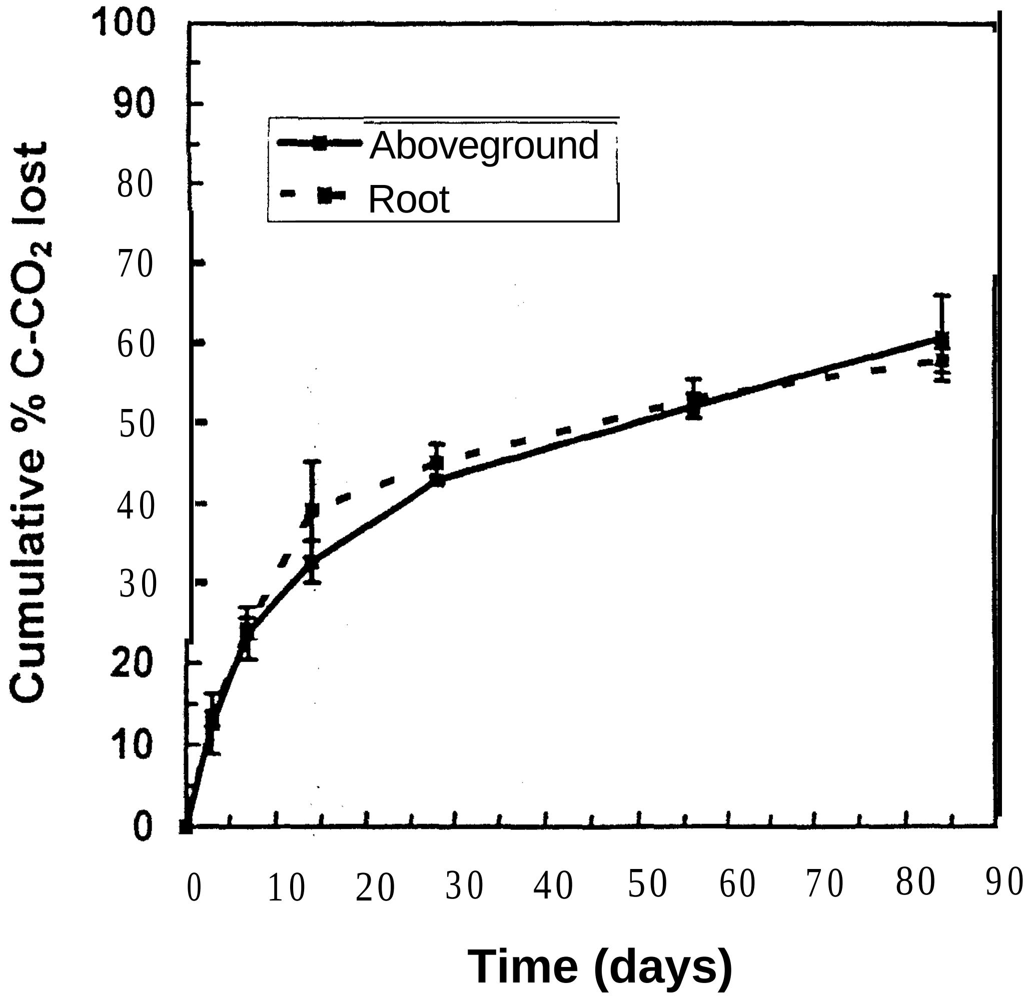


Figure 2.13. Decompos'ition of sorghum NKing-300 as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

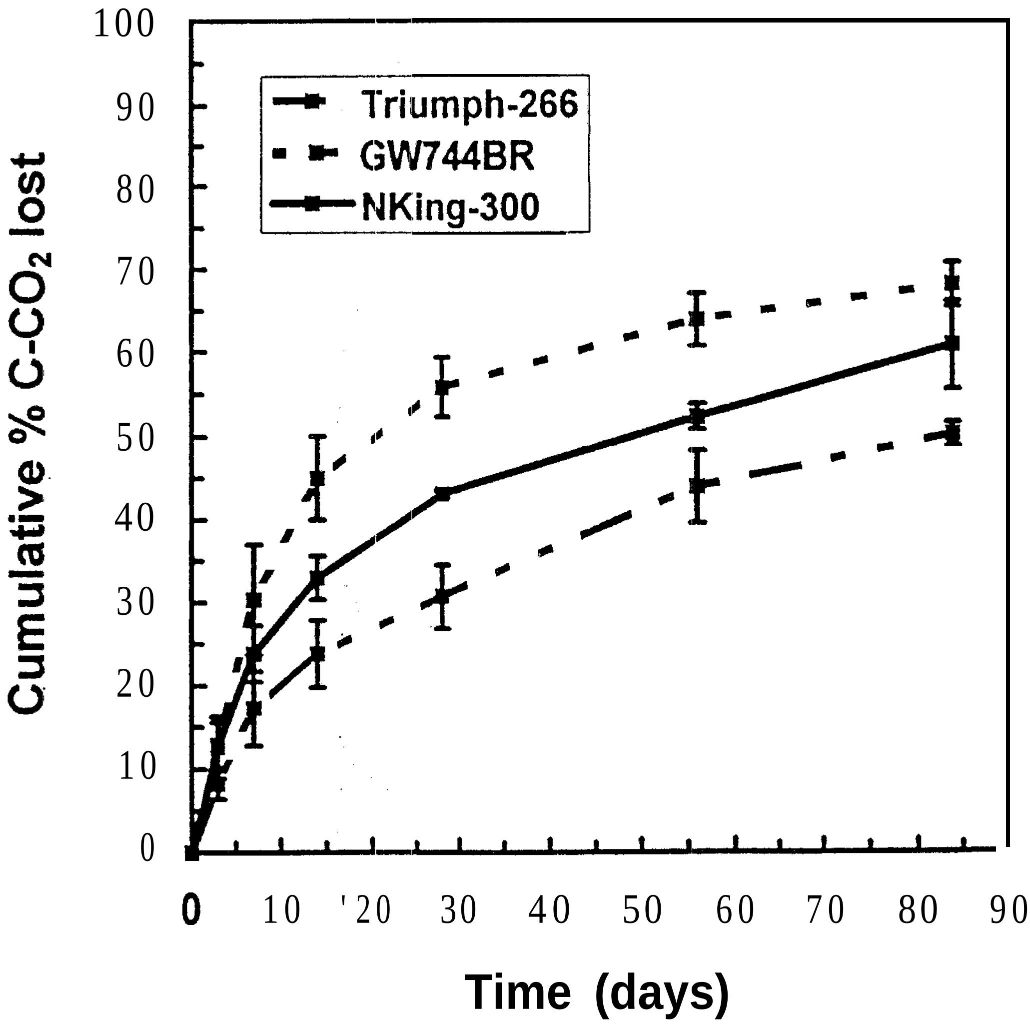


Figure 2.14 Decomposition of sorghum above-ground biomass as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

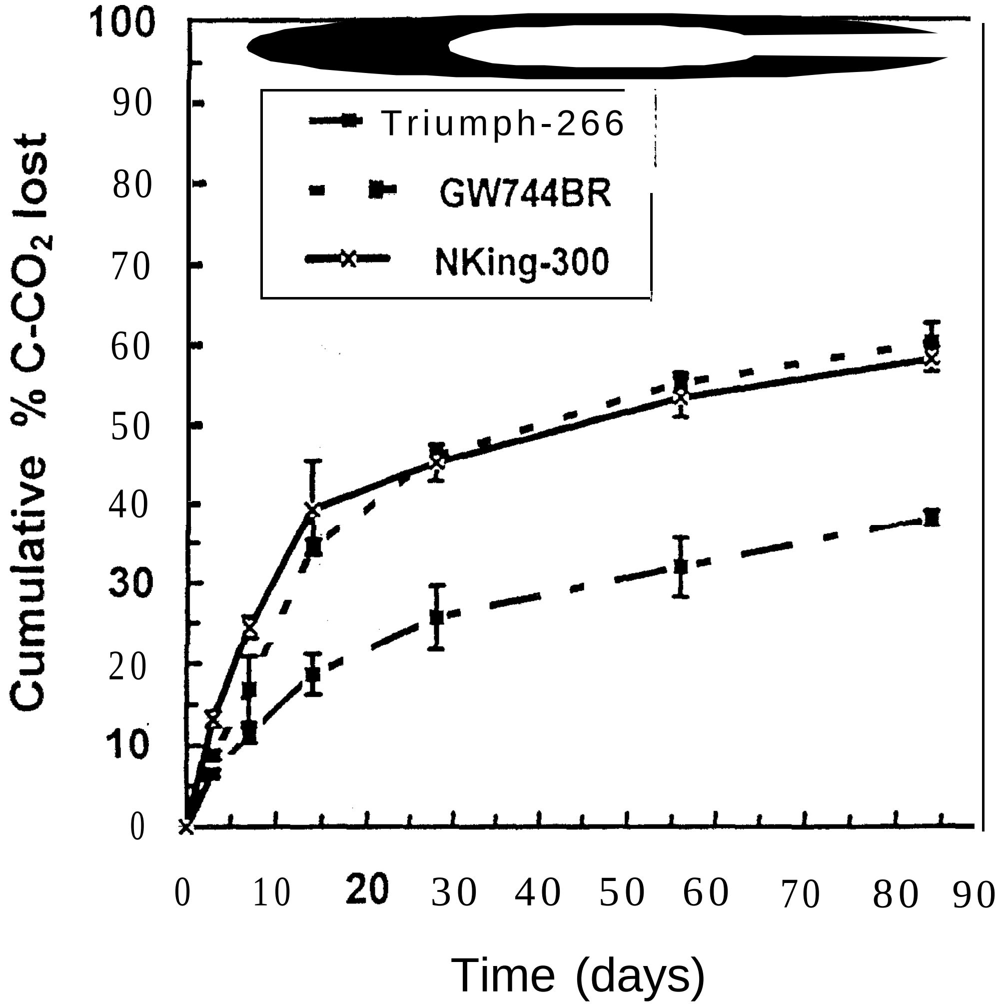


Figure 2.15. Decomposition of sorghum roots as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

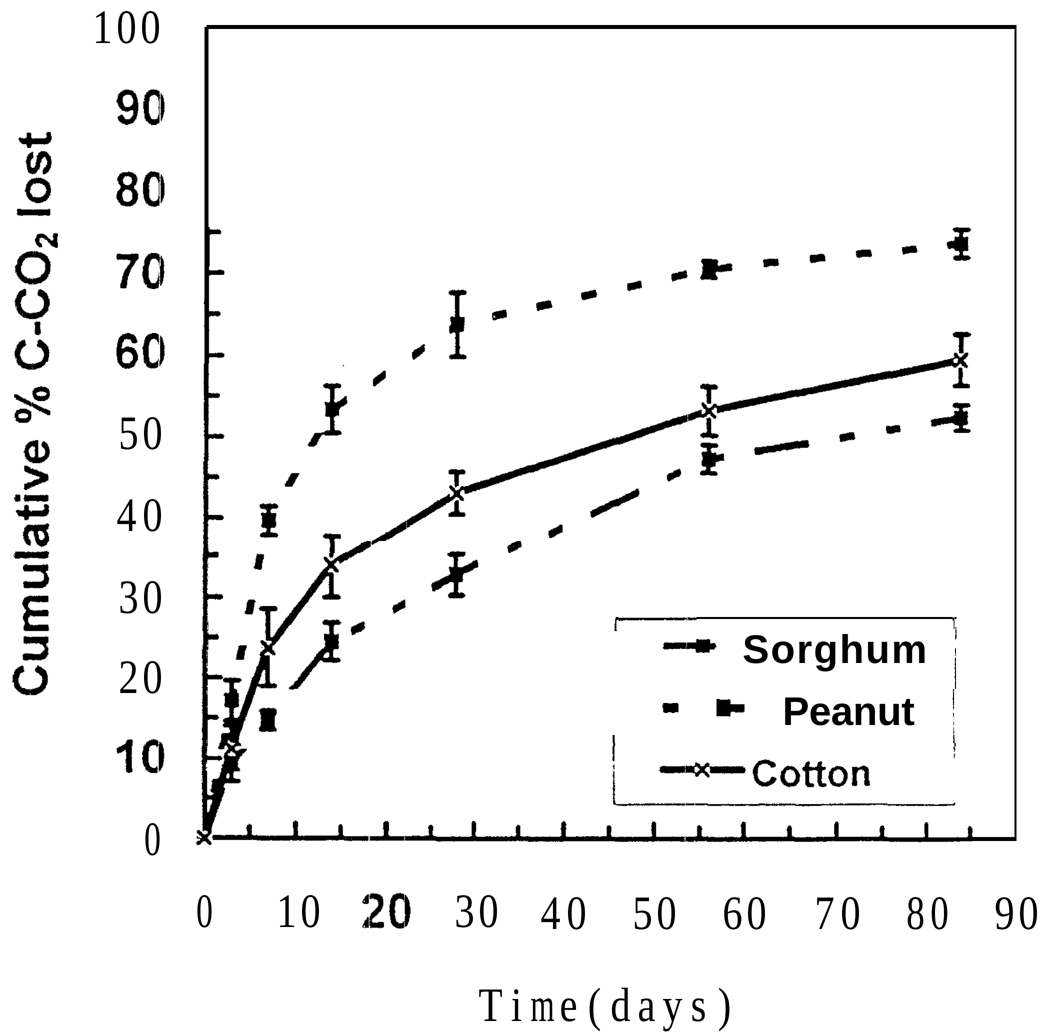


Figure 2.16. Mean decomposition rate of the above-ground biomass for each of the three crops as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

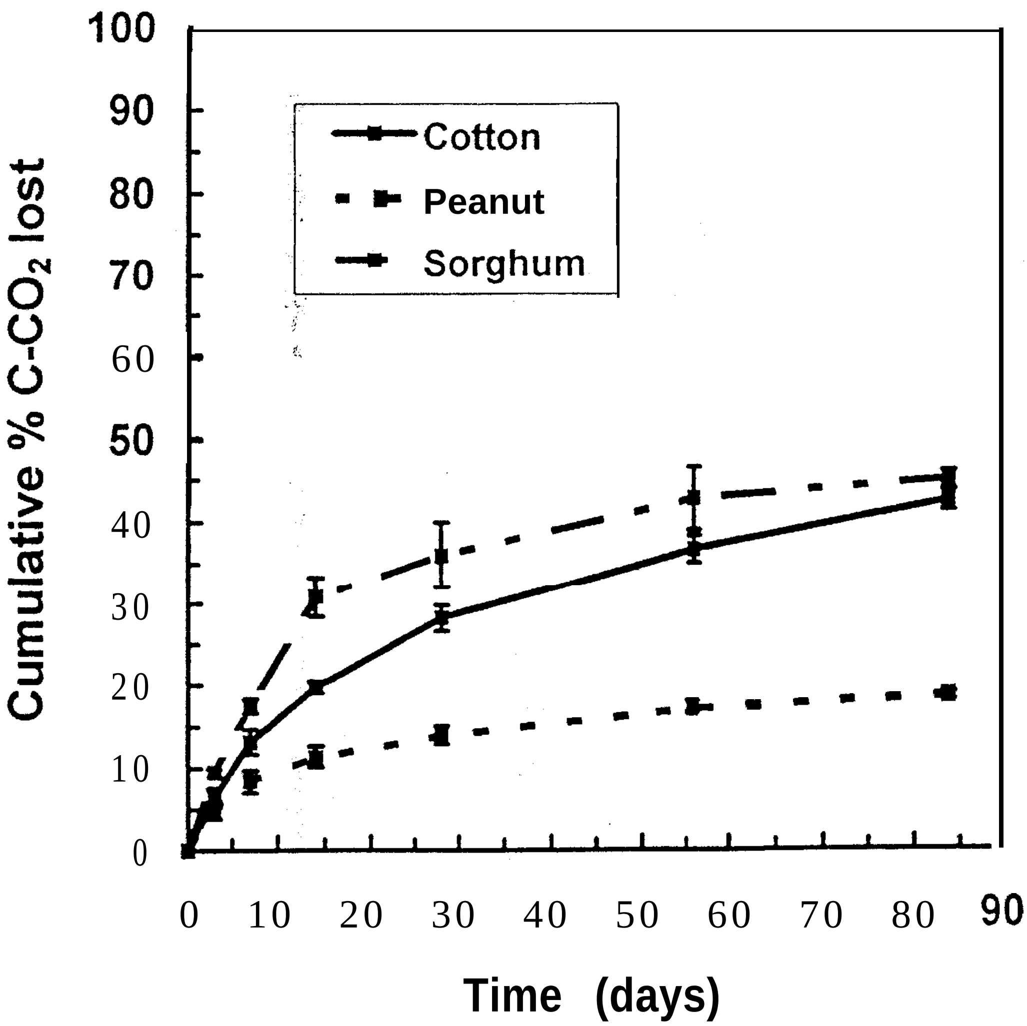


Figure 2.17. Mean decomposition rate of the roots for each of the three crops as measured by CO_2 evolution over time. Bars represent standard deviations at given time.

2.4.5. Change in Mass loss

In determining mass loss, the above-ground residues were split in leaves and stems and each of these components was measured separately. For cotton cultivars (Figures 2.18, 2.19 and 2.20), the rate of mass loss of the leaves was significantly higher than the stems and roots. However, no significant difference was found between stems and the above-ground biomass in any of the three cultivars. DLP-5690 (Figure 2.21) had a faster above-ground residue breakdown rate, 38%, followed by that of DP-5215, 30% and HS-46, 26%. HS-46 root mass loss (Figure 2.22) was higher, 29%, than that of DLP-5690 and DP-5215, 24 and 17% respeciively.

Peanut leaf mass loss was significantly faster than that of the stems which were much faster than roots (Figures 2.23, 2.24 and 2.25) for all cultivars. Cultivars Florunner and NC-'7 showed no significant difference between stems and the total above-ground in the percent mass remaining during the first 14 days. Only NC-1 1 presented higher mass loss for the leaves, 43%, than stems and roots, 26 and 9% respectively. There was no difference in rate of breakdown of the above-ground residues between the three cultivars (Figures 2.26), but Florunner root had a faster mass loss rate than the roots from the other two cultivars (Figure 2.27).

Sorghum cultivars showed significant differences between the above-ground residues and the root breakdown (Figures 2.28, 2.29, and 2.30) in the early decomposition. However, only cultivar Triumph-266 presented a significant difference between leaves and stems. There was no difference in decay rates between the above-ground residues for the three cultivars (Figure 2.31).

Significant differences in mass remaining were observed between the mean mass loss of the cultivars of cotton, peanut, and sorghum above-ground biomass (Figure 2.33) in the early decomposition phase. Peanut mass loss was greater, 45%, than cotton and sorghum, 33 and 25%, respectively. However, sorghum root breakdown (Figure 2.34) was faster, 12%, than that for cotton and peanut roots, 7 and 5%, respectively.

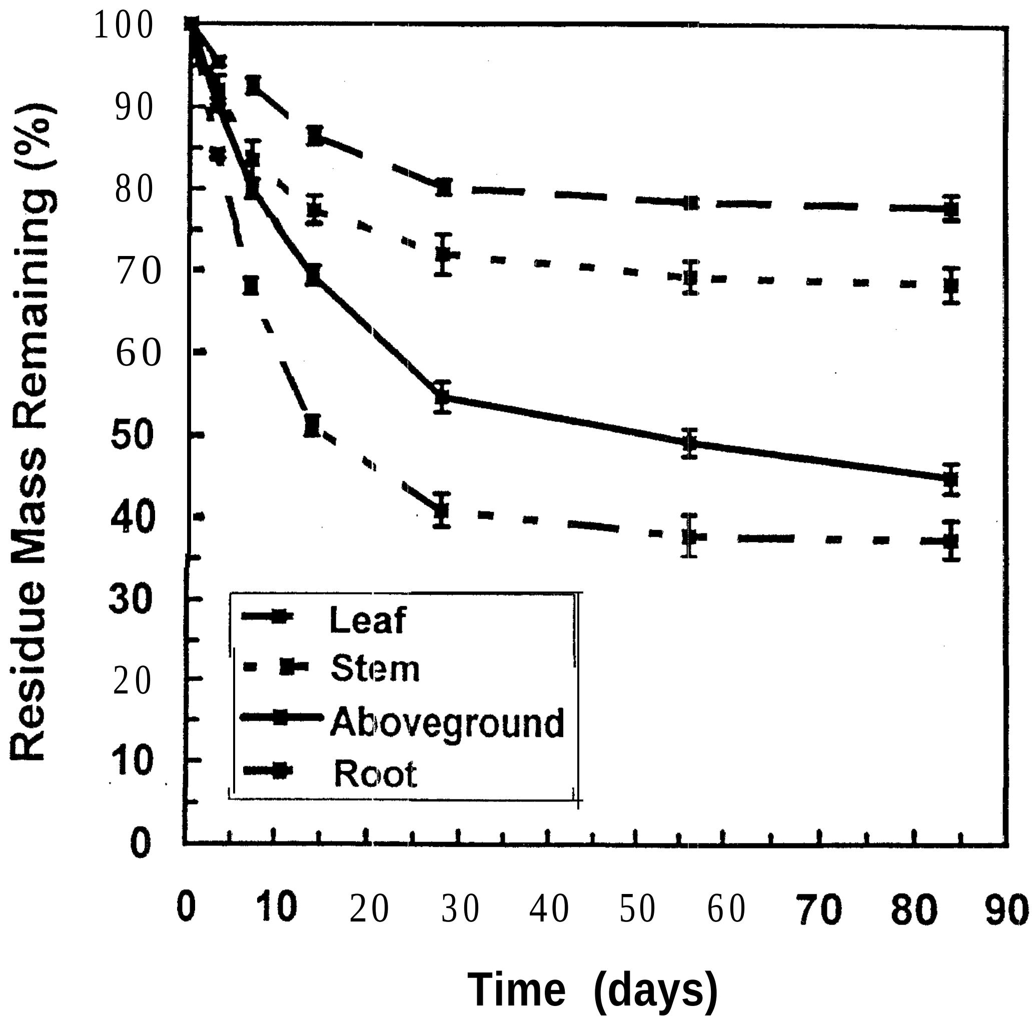


Figure 2.18. Decomposition of cotton DLP-5690 as measured by mass loss over time. Bars represent standard deviations at given time.

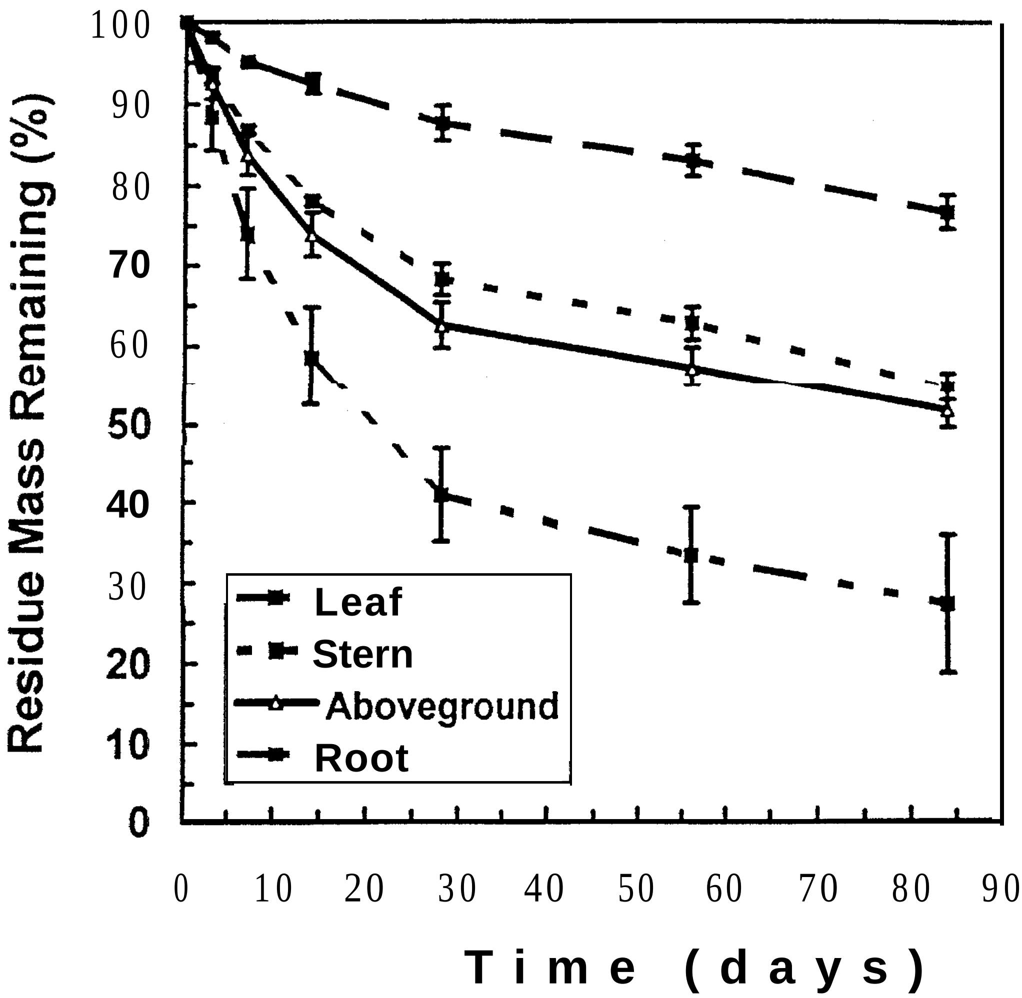


Figure 2.19. Decomposition of cotton DP-521 5 as measured by mass loss over time. Bars represent standard deviations at given time.

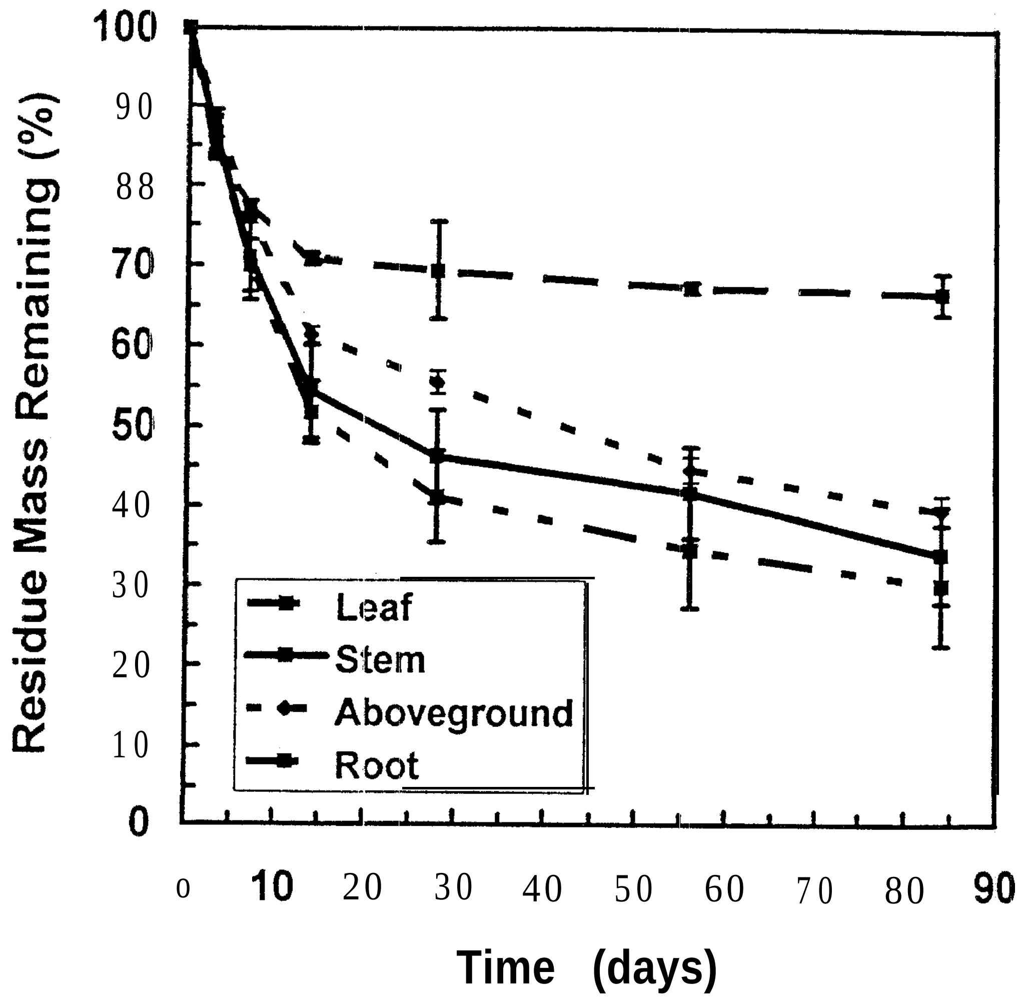


Figure 2.20. Decomposition of cotton HS-46 as measured by mass loss over time. Bars represent standard deviations at given time.

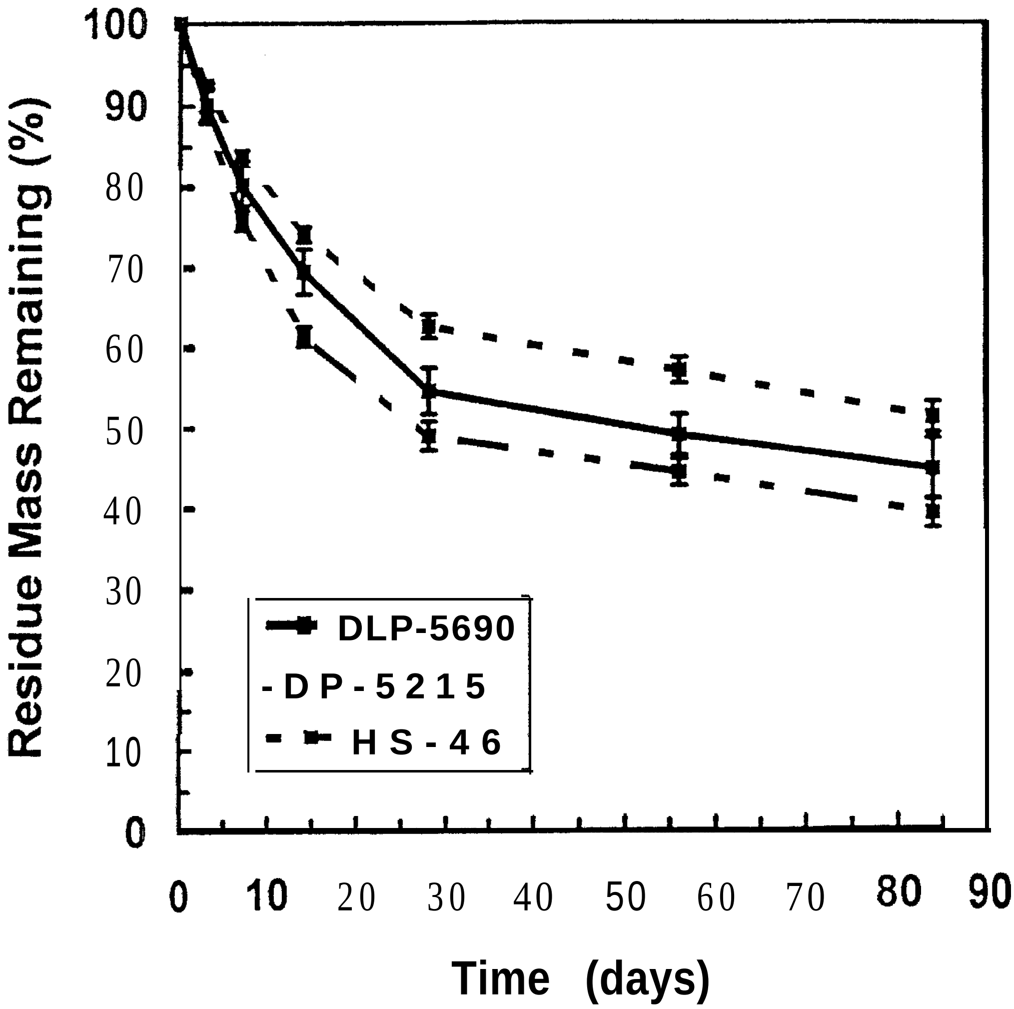


Figure 2.21. Decomposition of cotton above-ground biomass as measured by mass loss over time. Bars represent standard deviations at given time.

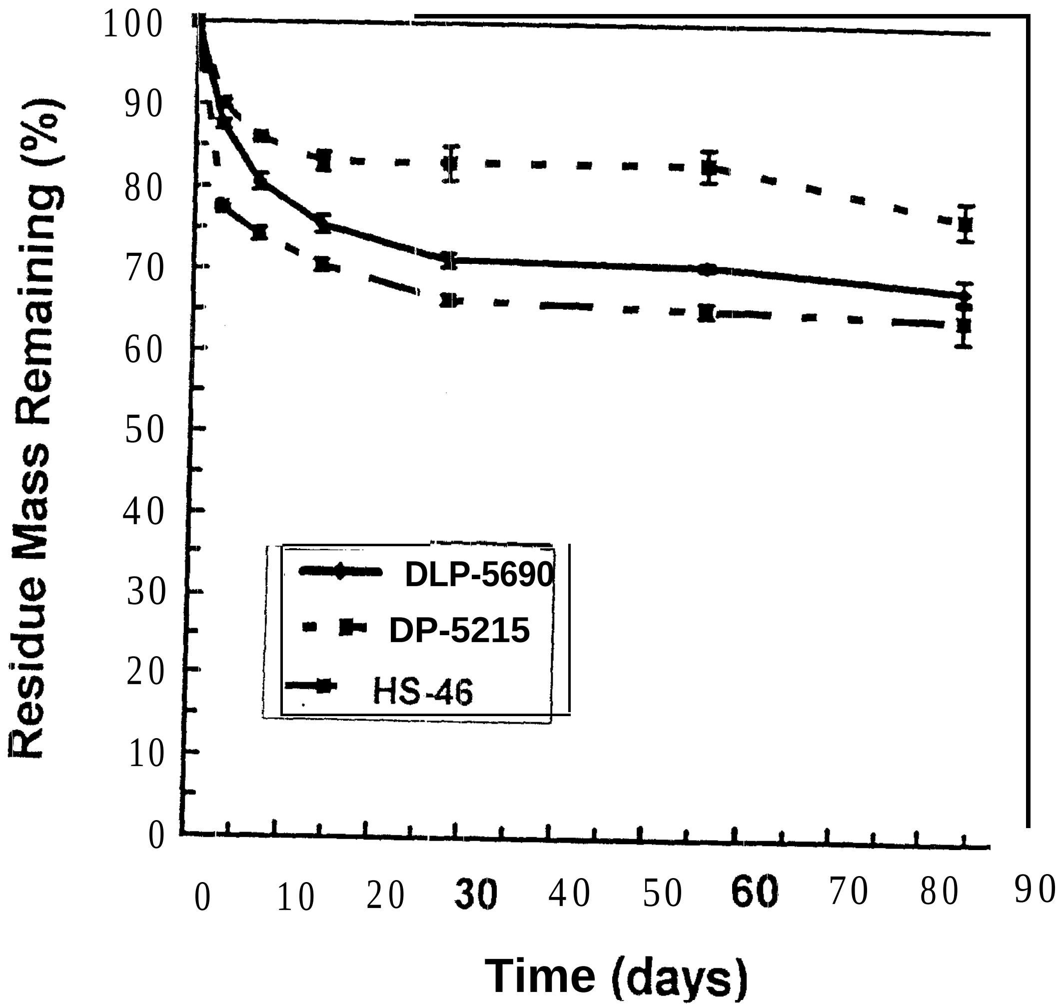
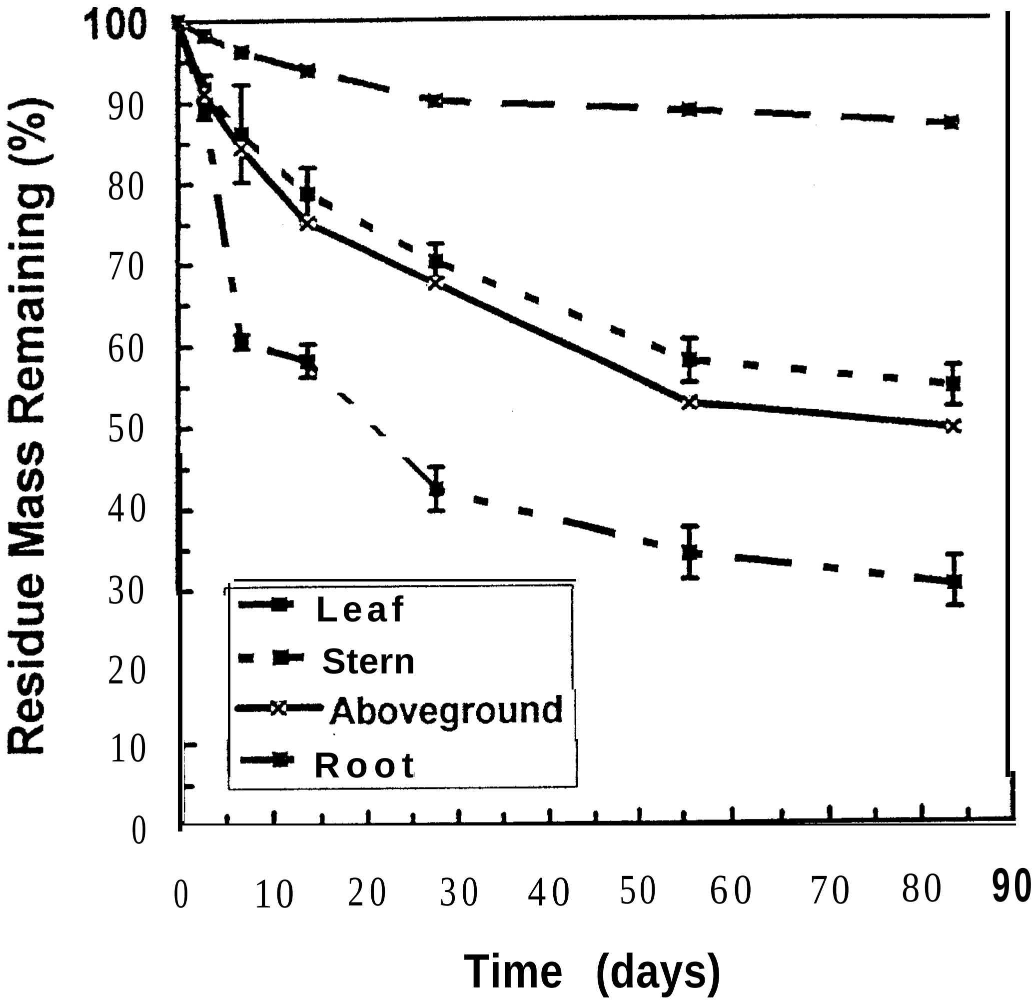


Figure 2.22. Decomposition of cotton roots as measured by mass loss over time. Bars represent standard deviations at given time.



'Figure 2.23. Decomposition of peanut Florunner as measured by mass loss over time. Bars represent standard deviations at given time.

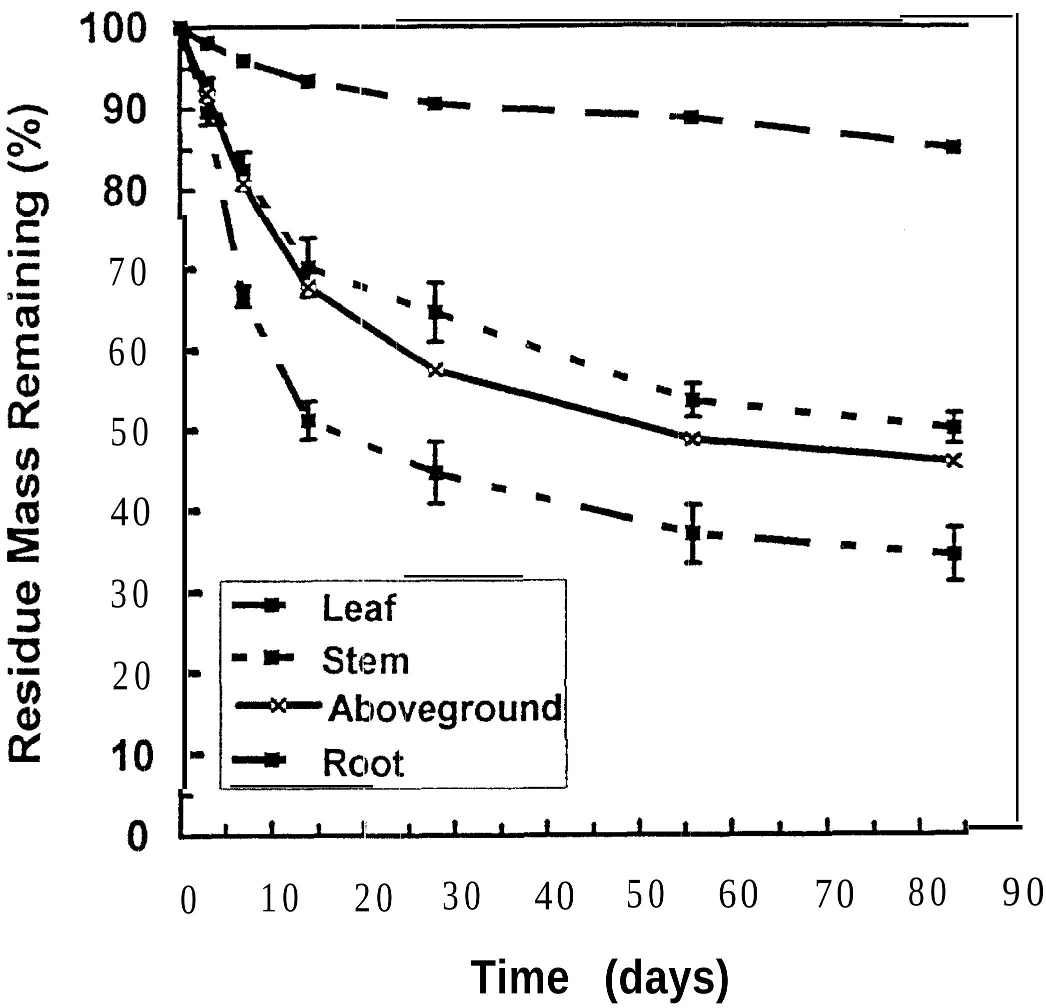


Figure 2.24. Decomposition of peanut NC-7 as measured by mass loss over time. Bars represent standard deviations at given time.

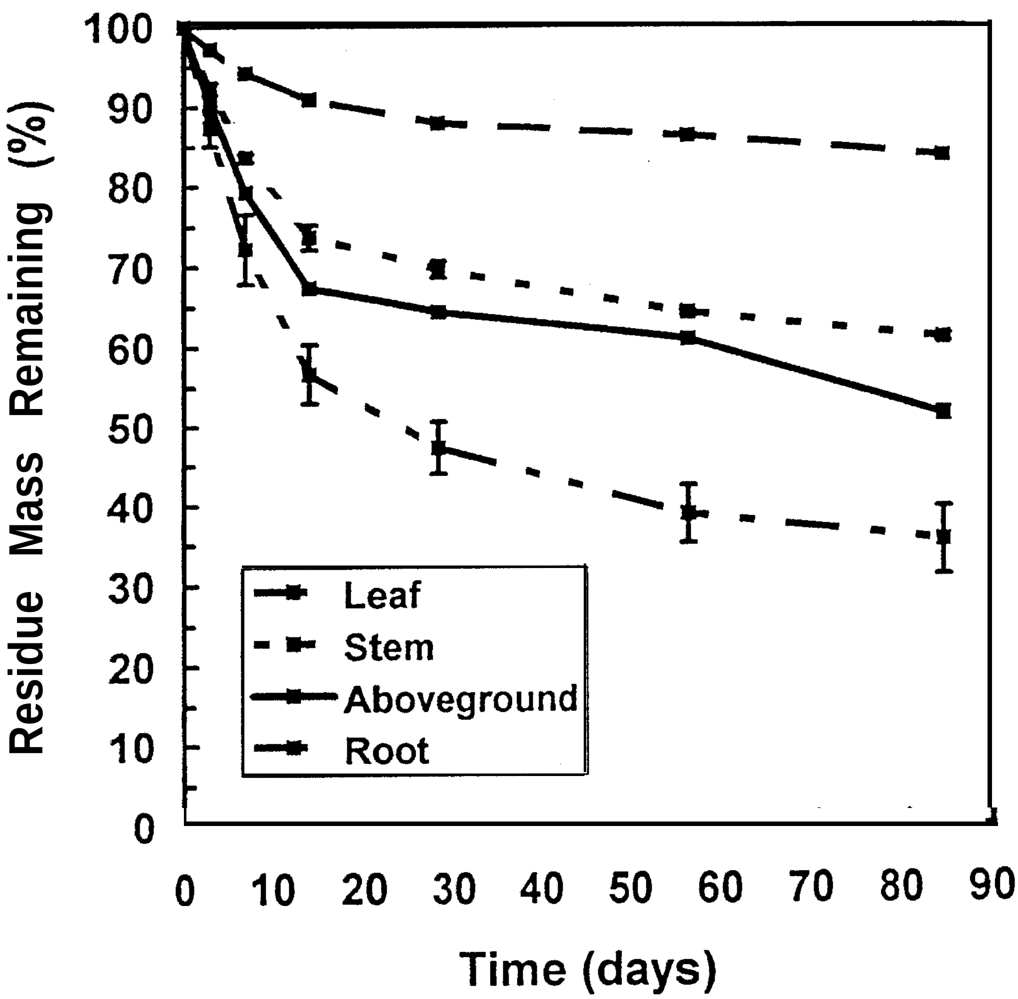


Figure 2.25. Decomposition of peanut NC-11 as measured by mass loss over time. Bars represent standard deviations at given time.

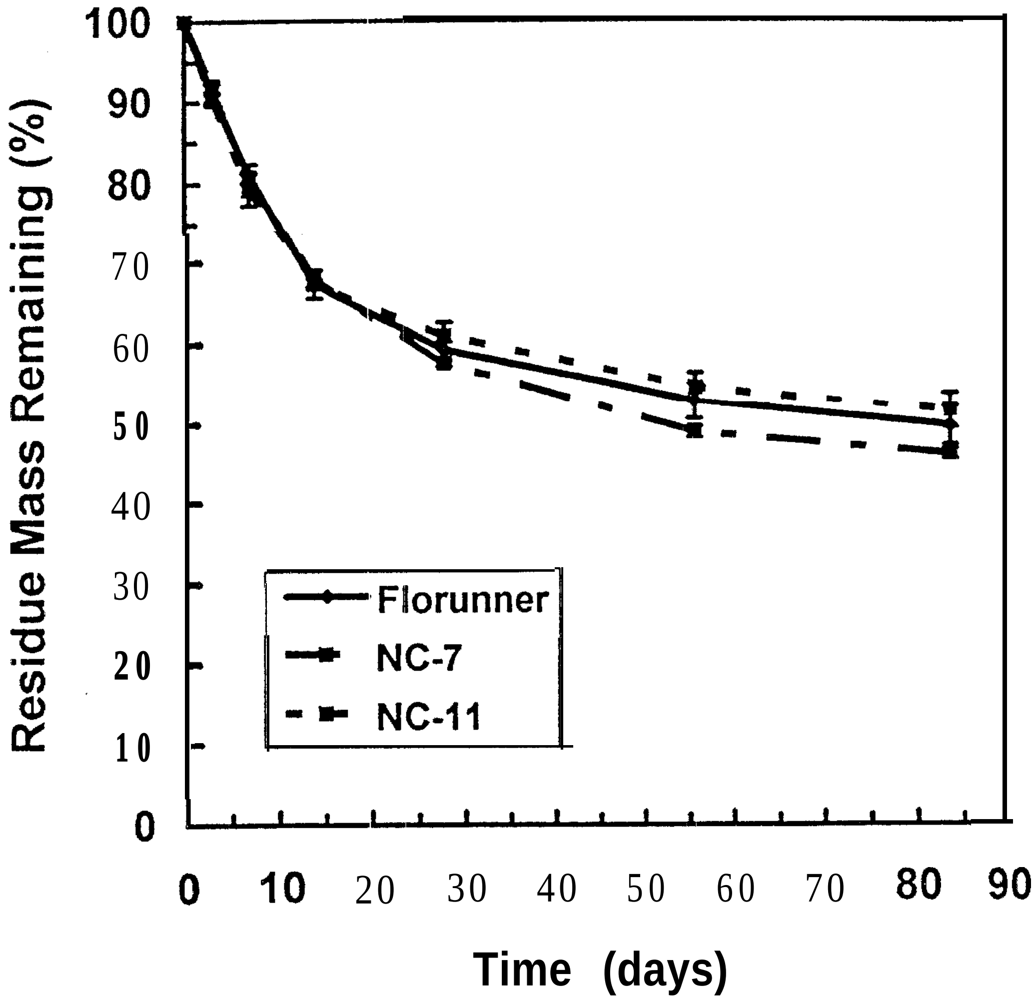


Figure 2.26. Decomposition of peanut above-ground biomass as measured by mass loss over ltime. Bars represent standard deviations at given time.

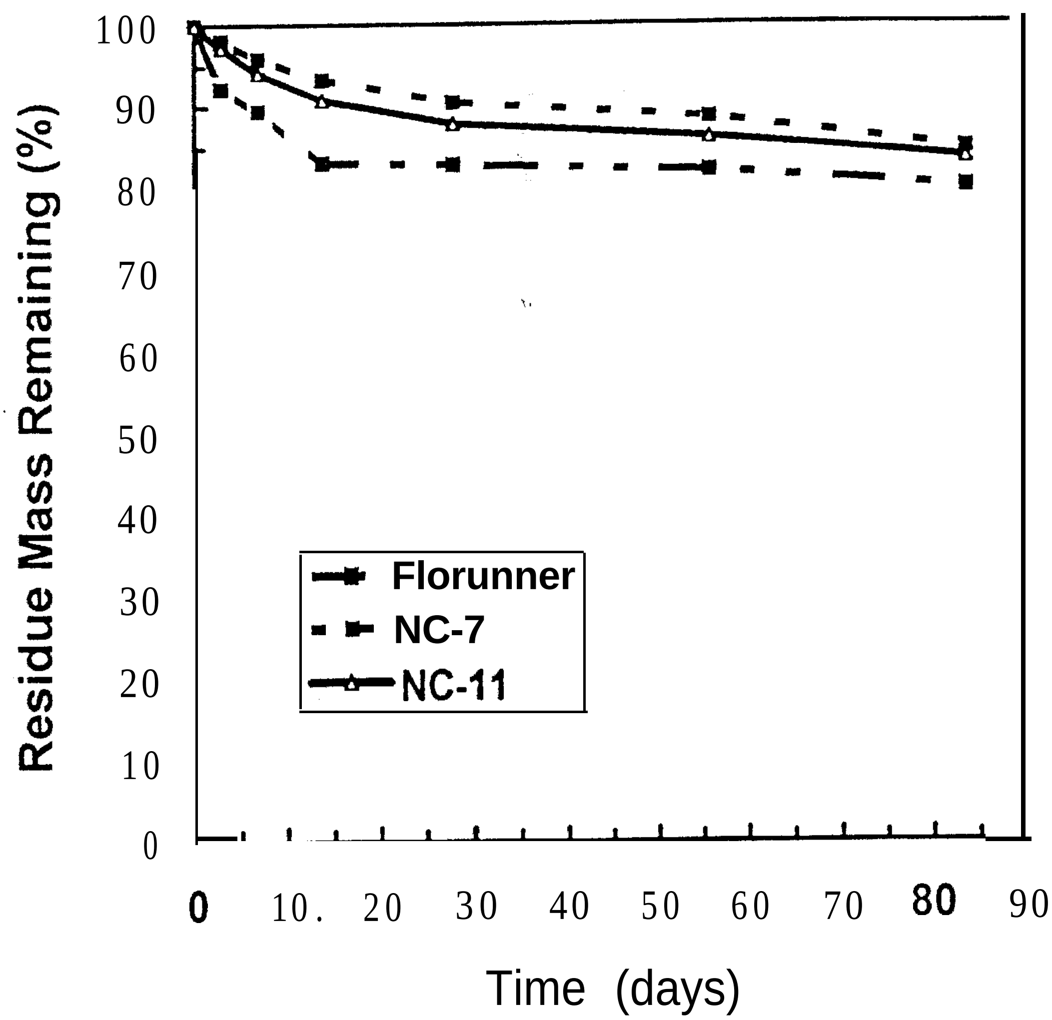


Figure 2.27 Decomposition of peanut roots as measured by mass loss over time. Bars reoresent standard deviations at given time.

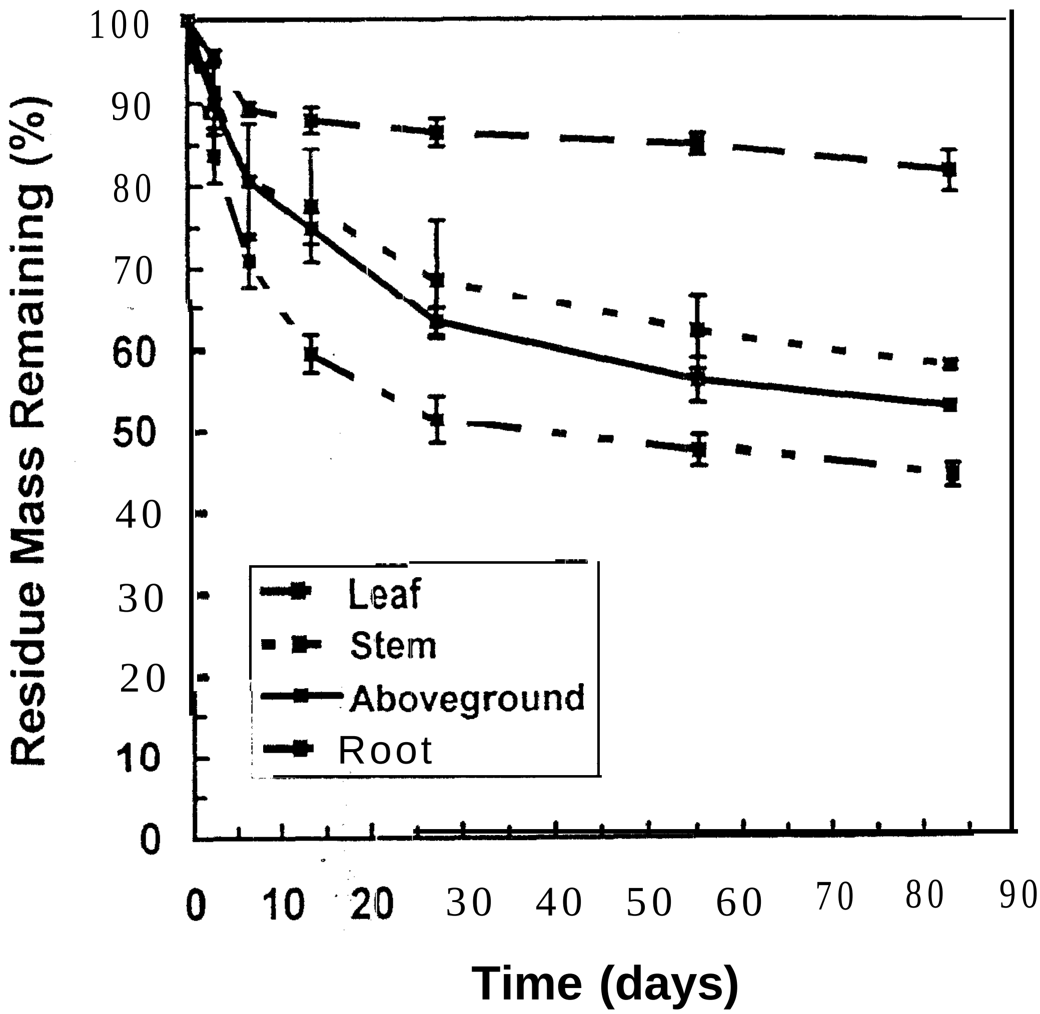


Figure 2.23. Decomposition of sorghum Triumph-266 as measured by mass loss over time. Bars represent standard deviations at given time.

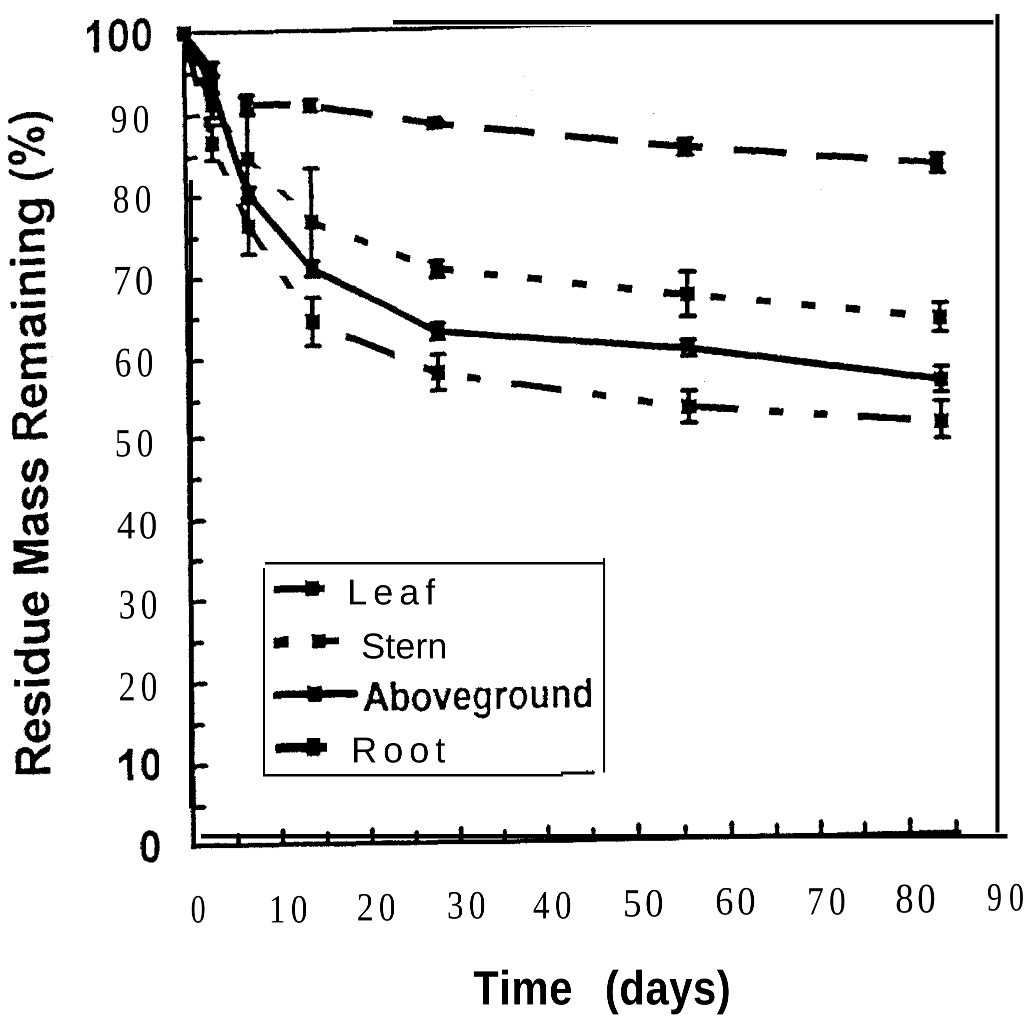


Figure 2.29. Decomposition of sorghum GW-744BR as measured by mass loss over time. Bars represent standard deviations at given time.

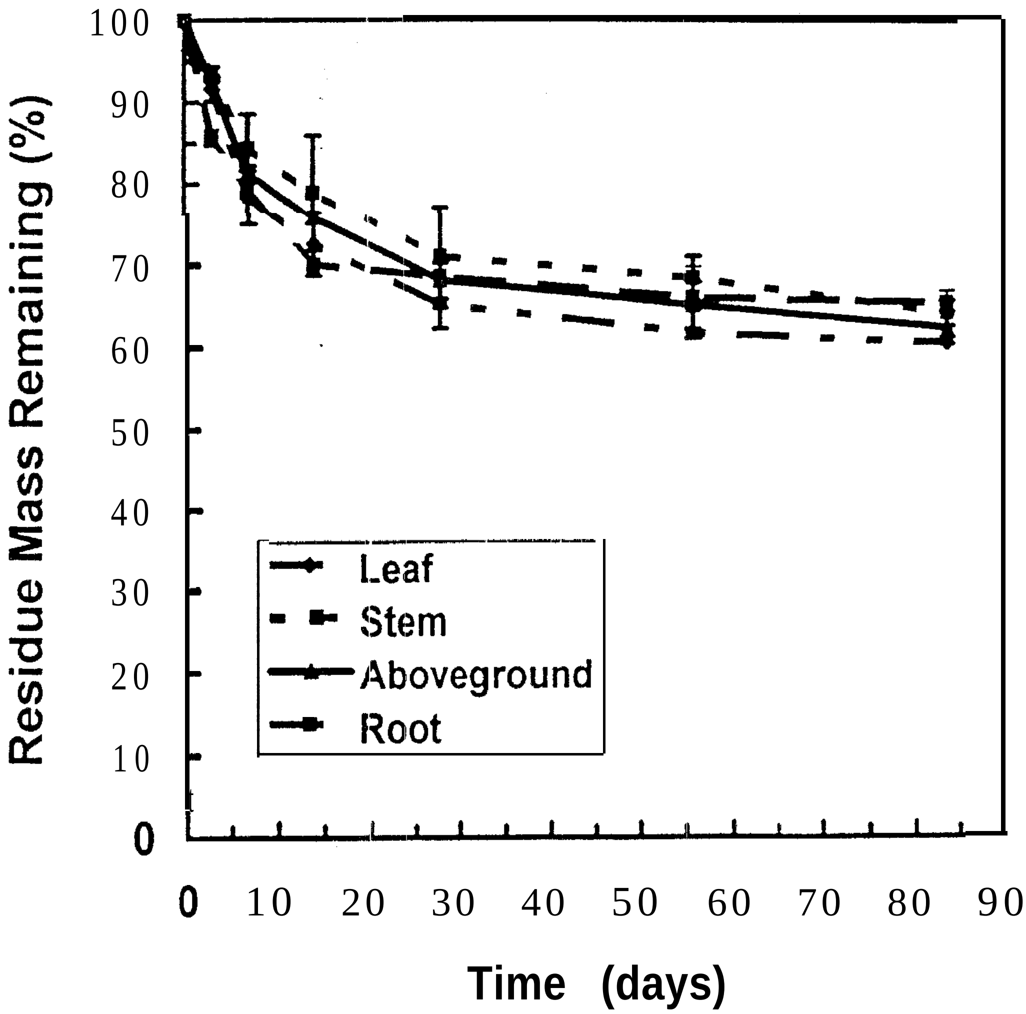


Figure 2.30. Decomposition of sorghum Nking-300 as measured by mass loss over time. Bars represent standard deviations at given time.

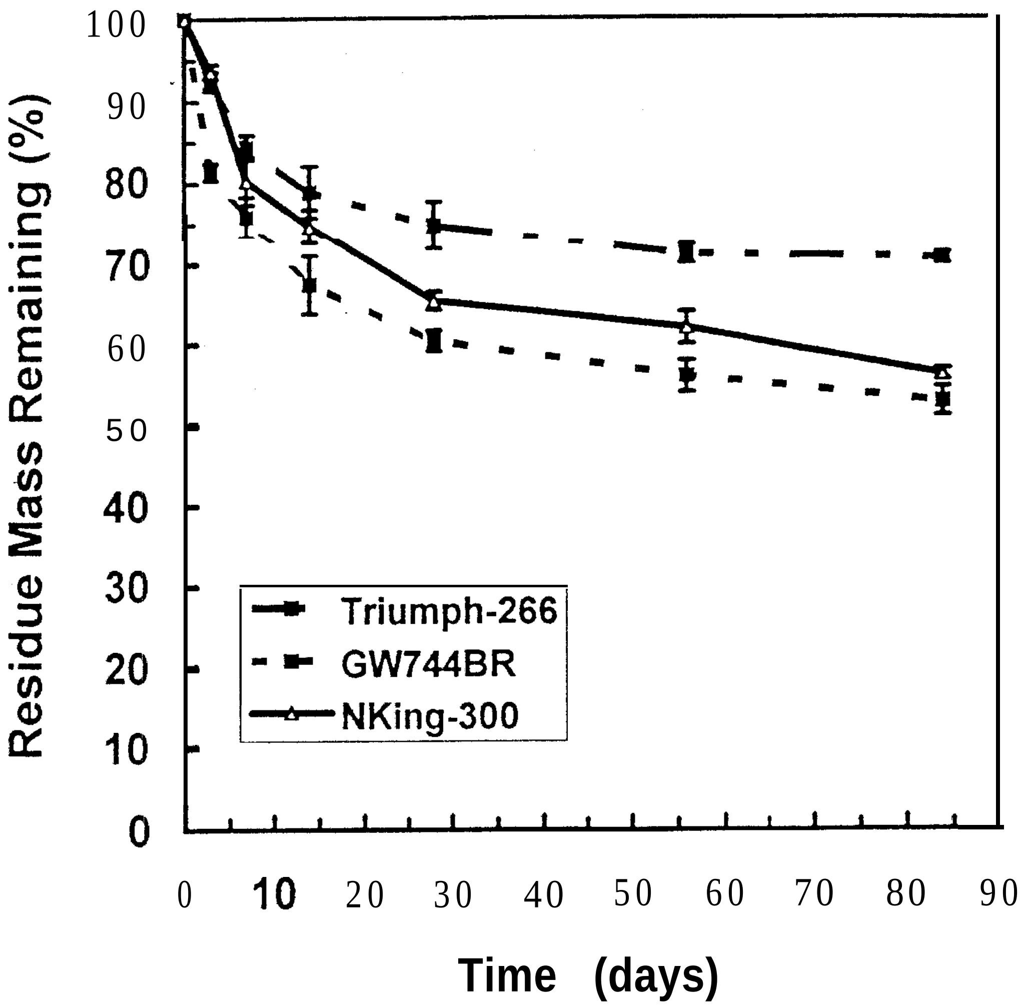


Figure 2.31. Decomposition of sorghum above-ground biomass as measured by mass loss over time. Bars represent standard deviations at given time.

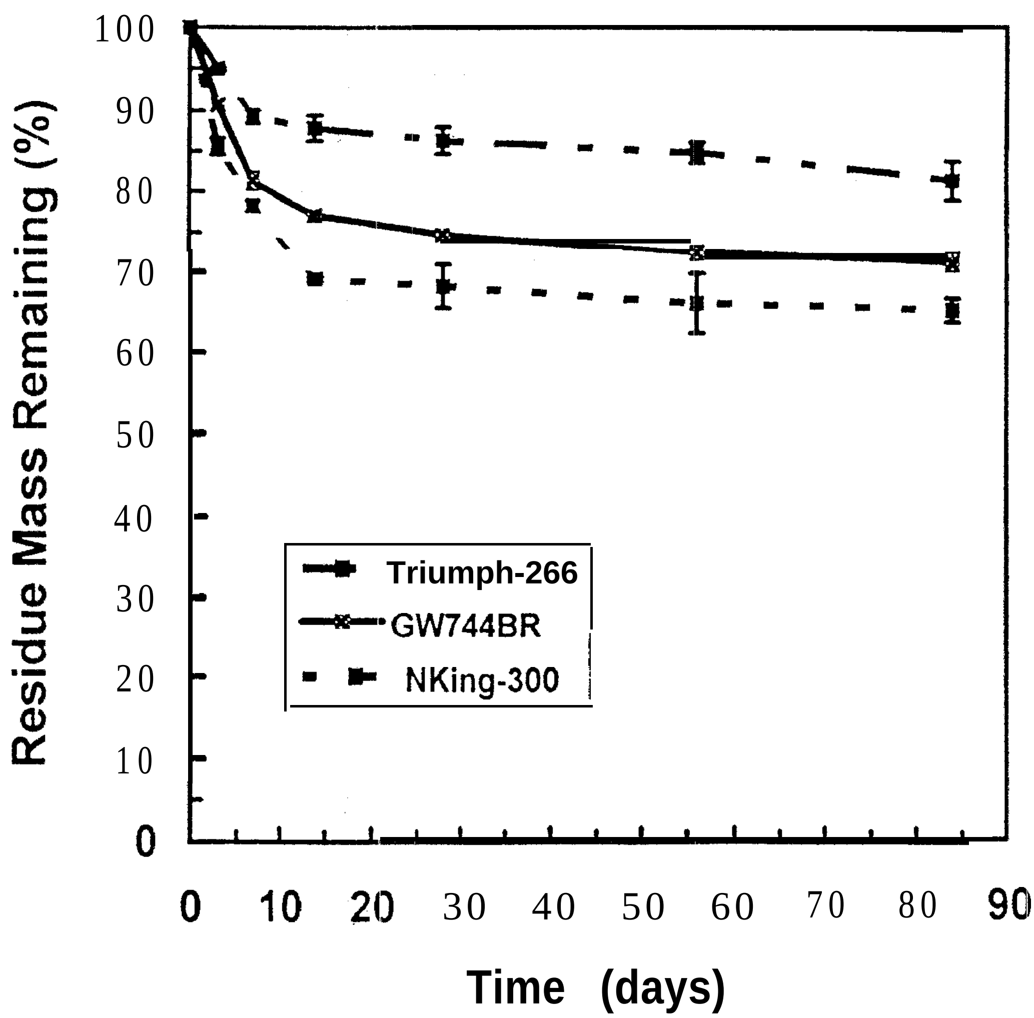


Figure 2.32. Decomposition of sorghum roots as measured by mass loss over time. Bars represent standard deviations at given time.

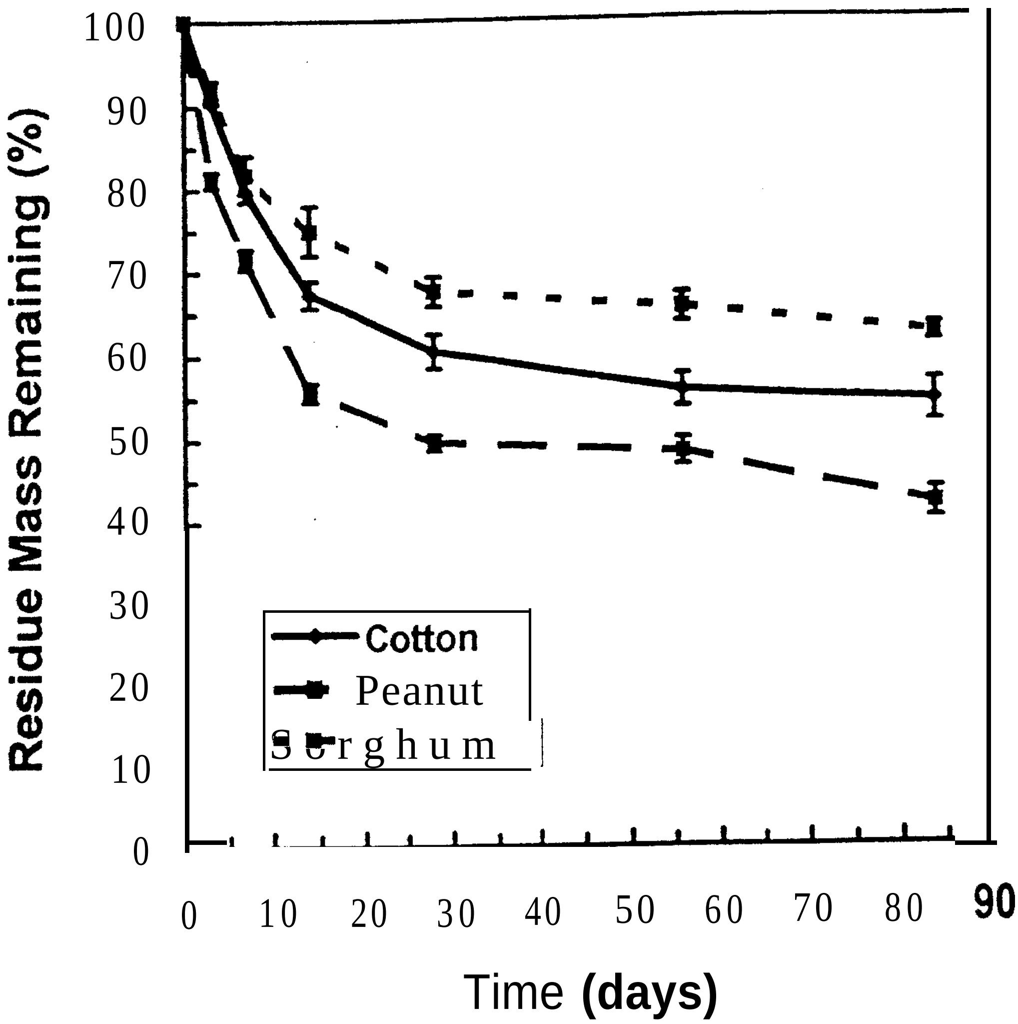


Figure 2.33. Mean decomposition rate of the above-ground biomass for each of the three crops as measured by mass loss over time. Bars represent standard deviations at given time.

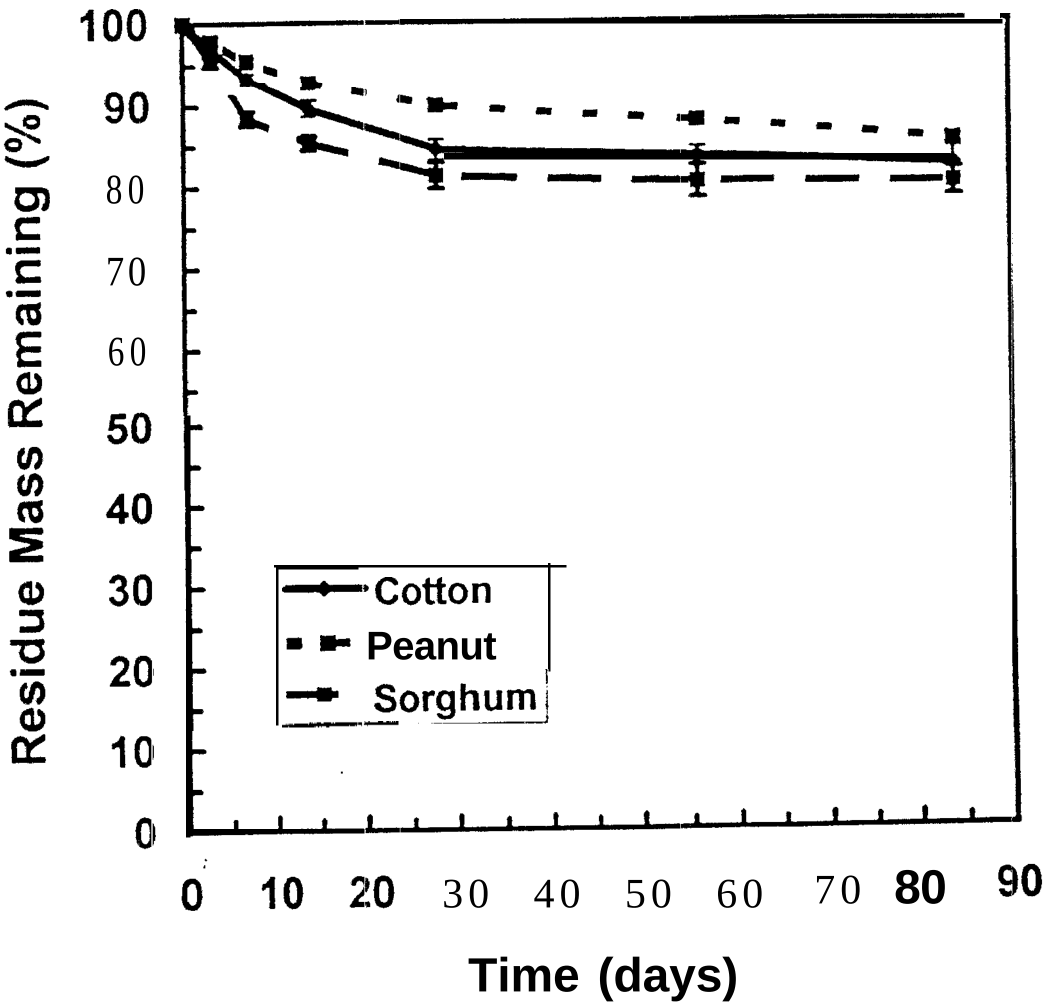


Figure 2.34,. Mean decomposition rate of the roots for each of the three crops as measured by mass loss over time. Bars represent standard deviations at given time.

2.5. Discussion

The decomposition rates for ail cotton (Figures 2.1, 2.2, 2.3, 2.4, and 2.5), peanut (Figures 2.6, 2.7, 2.8, 2.9, and 2.10), and sorghum (2.11, 2.12, 2.13, 2.14, and 2.15) cultivars followed the pattern for Michaelis-Menten first-order kinetics. The rapid increase in CO_2 evolution during the first 14 days was probably due to the high total N content, the high level of readily available C in the form of extractable sugars or a combination of the two (Tables 2.3 and 2.4). Kinetically, the CO_2 evolution from the residues studied exhibited a lineaf dependence on the chemical composition of the residue. The rapid disappearance of these soluble compounds were probably related to a quick buiid up of the microbial activity which would increase the CO_2 respiration. Also, the readily available C and N components in the crop residues might provide the initial energy and nutrients necessary to activate the microorganisms that are responsible for the degradation of the less readily available components of the residue.

The leveling off phase of the CO_2 evolution, between days 15 and 28, would be the period where hemicelllulose was the main fraction available to the microofganisms. As the decomposition process proceeds, CO_2 evolution slows down, following an exponential curve, probably due to change in chemical composition of the remaining residue available to the microorganisms. I think that in this phase of decomposition, the hemicellulose fraction probably disappears initially at a rapid rate, but the subsequent degradation appeafs to be slower. The degradation of hemicellulose is more marked when the environment is aerobic, and when there is availability of inorganic nutrients, especially nitrogen, (Alexander, 1977). At this stage of the decomposition process, I think that there is probably not enough N or readily available C to keep the microbial activity at high level. As a result, there is a decrease in decomposition rate and respiration, resulting in a slower rate of CO_2 evolution.

All residue types show the same trend and similar slopes in this portion of the curve, suggesting that that the second phase of the decomposition is probably not a good element of comparison of CO_2 evolution.

After 28 days of decornposition process, the remaining residues entered the third phase of the decomposition process. At this point, the slowly available residue components dominated the residue substrate. Lignin, known to be resistant to degradation, was probably the major remaining component. The rate and extent of lignin decomposition are affected by temperature, availability of nitrogen, and by constituents of the residues undergoing decay (Sarkanen et al., 1971). At this stage of degradation, all the readily available nutrients are expected to vanish. Lignin is probably being decomposed by relatively slowly growing microorganisms (Witkamp et al., 1963). Consequently, microbial respiration is very low. As a result, CO_2 evolution follows a quasi steady-state for the rest of the decomposition. Lignin continues to disappear however.

Cotton cultivars DLP-5690 and DP-521 5 above-ground biomass (Figures 2.1 and 2.2) showed greater cumulative CO_2 evolution than the roots due to higher total N, lower hemicellulose and lignin concentration of the above-ground residue. In addition, lower lignin content plus high specific surface area-to-mass ratios for the above-ground residue provide microorganisms better access ta available C sources (Collins et al., 1990; Jensen 1994). Cultivar HS-46 above-ground residues and roots (Figure 2.3) were not different in cumulative CO_2 evolved probably due to higher level of total concentration of N, but lower sugar, hemicellulose and lignin contents for the above-ground biomass than the roots. The specific surface area-to-mass was probably too low in above-ground to provide microorganisms good access to available C sources.

For all peanut cultivars (Figures 2.6, 2.7, and 2.8), above-ground residues showed much higher cumulative CO_2 evolved than the roots due to the higher simple sugar contents available to the microorganisms, combined with lower lignin concentration of the above-ground biomass. The insignificant difference in sugar concentrations between Florunner, NC-7, and NC-1 1 above-ground residues (Table 2.3) certainly excludes any difference in their cumulative CO_2 evolved (Figure 2.9). Peanut is a legume, and the highest N level is concentrated not in the above-ground biomass but in the root system where the nuts are produced (Table 2.4).

fhe only sorghum cultivar, GW744BR, showing significant difference in CO_2 evolution between the above-ground biomass and roots (Figure 2.12), had the highest total N, and the lowest simple sugar and lignin concentrations in the above-ground than the roots. For the other cultivars, Triumph-266 and Nking-300 (Figures 2.11, and 2.13), higher available C in the form of simple sugar concentrations in the roots probably contributed to their higher CO_2 evolution level, matching that of the above-ground residues. Sorghum roots are fibrous and high in sugar content (Table 2.4). These results were consistent with Leonard et al. (1963) who observed that high levels of sugars in sorghum roots furnished the energy for the multiplication of soil microorganisms which compete with plants for the available soil nitrogen. The data (Tables 2.3, 2.4, and 2.5) support the differences in cumulative CO_2 evolution among residues. These results agreed with Collins et al. (19903 data in their study of decomposition of winter wheat residues. They found that cumulative CO_2 evolution among residue components increased as the concentration of soluble C increased, and CO_2 production from chaff was initially more rapid than that from stems, but after 15 days, decomposition of the chaffs and stems produced CO_2 at the same rate.

Residue decomposition is a process in which the rate of transformation is proportional to the qualitative amount of residue available to the microorganisms. This qualitative amount of residue is reflected by the concentration of the different chemical compounds and the physical nature of the residue. The chemical composition of the residue constitutes probably the most important regulator of the decomposition (Knapp et al., 1983a). In this study, three pools were sorted out as they represented three different phases of the

CO_2 evolution kinetics: 1) nitrogen and readily available carbon in the form of simple sugars, 2) hemicellulose, and 3) lignin. My data show that this compares well with what Stroo et al. (1989) have observed in predicting rate of wheat decomposition. Nitrogen is required by the microorganisms for the synthesis of amino acids, nucleotides, and other compounds. These microorganisms also requit-e carbon source to construct all their carbon-containing biomolecules, Hemicellulose, a non-structural carbohydrate, second only to cellulose in quantity, represents a significant source of energy and nutrients to the microorganisms. Lignin is the third most abundant constituent of the plant residues and is slow to degrade.

Residue decomposition, as measured by cumulative CO_2 evolution, cannot be related to a single pool, but a set of all defined pools, each of them playing a particular role. However, for legume species, the pool of N and available C in the form of simple sugars seems to play the determinant role. Cheshire et al. (1988) reported that using a single pool tends to underestimate changes in the residue decomposition with time.

In this study, the common trend in the CO_2 evolution rates from the roots did not present any real break between the second phase with decreasing of hemicellulose availability and the steady phase with lignin availability. This was probably due to the high <concentrations of hemicellulose and lignin present in the roots. Most root systems store a relatively appreciable level of readily available C in the form of sugars, but when matched with higher contents of structural carbohydrate and lignin available to the microorganisms, the decomposition process remains slow. The decomposition rate of roots could be an important. information in the management strategies to prevent soil erosion by water. Even though it has been found that root. degradation was more complete in undisturbed soil (Martin, 1989) compared to tilled soil, the results obtained from this study, with air-cfried roots, would still be useful to quantify root decomposition.

The differences in residue decomposition between the above-ground biomass and the roots of these cultivars used in this study is due to differences in initial chemical and physical characteristics of the two residue components of each given cultivar, and also in morphologic variation between cultivars (Stott, 1992). Jensen (1994) related decomposition of plant residues at different total C:N ratios with different particle sizes. But, in the early decomposition process, microorganisms are more likely to utilize the readily available fraction (soluble C in the form of sugars) of the plant residues than the total C pool which includes the more recalcitrant fraction (Stott, 1992).

In the first fourteen days, the residue mass remaining decreased quite rapidly. At day 15, the mass remaining started leveling off and then showed no significant change from day 28 until the end of the experiment. The rapidity at which the breakdown of the residues occurred in the early phase was mainly dependent on the initial chemical and physical nature of the residues. For most cases, high levels of total N and readily available C in the form of sugars were essential to a rapid decomposition. The degradation of the leaves usually was so fast that even if the stems were breaking down slowly, the weight loss of the overall above-ground still remained relatively high. Table 2.3 showed that the peanut aboveground residues (legume) that had the fastest weight loss rate in the early decomposition had highest concentrations of simple sugars, relatively high N content, relatively low hemicellulose and lignin levels compared to cotton and sorghum. Also, peanut residues have the second highest specific surface area-to-mass ratio after cotton (Table 2.6) which provides microorganisms much better access to available C sources. Cotton above-ground residues had the second highest level of N, relatively high concentrations of sugar, hemicellulose, and lignin. For sorghum above-ground residues, a combination of low N content, high hemicellulose level and a relatively low lignin content versus relatively high concentrations of sugars but a lower specific surface area-to-mass ratio of the residues made the rate of breakdown the slowest among the crop species. These results were consistent with previous work of Collins (1988) and Stroo et al. (1989).

The same pattern of CO_2 evolution was observed in mass loss as well in this study.

2.51. Change in the Specific Surface Area-to-Mass Relationship

Specific surface area-to-mass relationship, represented by a k value, is a specific surface area-to-mass ratio with dimension of ha kg" of residue. In Gregory's (1982) equation, (eq. 2.7), k is specific for a given crop and considered to be constant over time. Specific surface area-to-mass relationship (Figures 2.35, 2.36, 2.37) for cotton was significantly different. Cultivars DLP-5690 and DP-5125 above-ground biomass k values were significanly greater than that of cultivar HS-46 The first two cultivars were not significantly different in k value. Figures 2.38, 2.39 and 2.40 did not showed significant difference in k values between peanut cultivars Fiorunner, NC-7 and NC-l 1. Sorghum cultivars Triumph-266, GW744BR and Nking-300, were not significantly different in specific surface area-to-mass ratio (Fgures 2.41, 2.42 and 2.43). However, there was a significant difference between the mean k value of each species. The initial k value (Figure 2.44) for cotton was greater, 0.00048 ha kg-‘, than peanut and sorghum, 0.06029 and 0.00019 ha kg^{-1} respectively. In the first 10-14 days, change in specific surface area-to-mass ratio was relatively rapid for cotton and peanut , residues, but change in sorghum was quite slow.

Stott et al. (1994) found a k value of 0.00023 ha kg^{-1} for com from field data. This was consistent with the range of values from this study as the three crop species used, sorghum is the crop that is physiologically and morphologically closest to com, and both are monocotyledons. Compared to com, sorghum has a lower osmotic concentration of the leaf juices, but the stalks, crown, and root juices are higher in sorghum (Leonard et al., 1963). In addition to its juicy stem, sorghum leaf area is smaller than com. Therefore, sorghum residue decomposition may be somewbat faster than com. Consequently, a k value for sorghum should be smaller, but close to that of com residue.

K was found to be a value specific to each crop species. It changes within a certain range over time during the decomposition process because it is a ratio of specific surface area over mass of the decomposing residue (Eq. 2.8). In this study, significant differences were observed between cultivars of cotton, but not from peanut and sorghum. However, the significant difference in mean k values between cotton, peanut and sorghum species was consistent with its specificity to each crop (Stott, 1994).

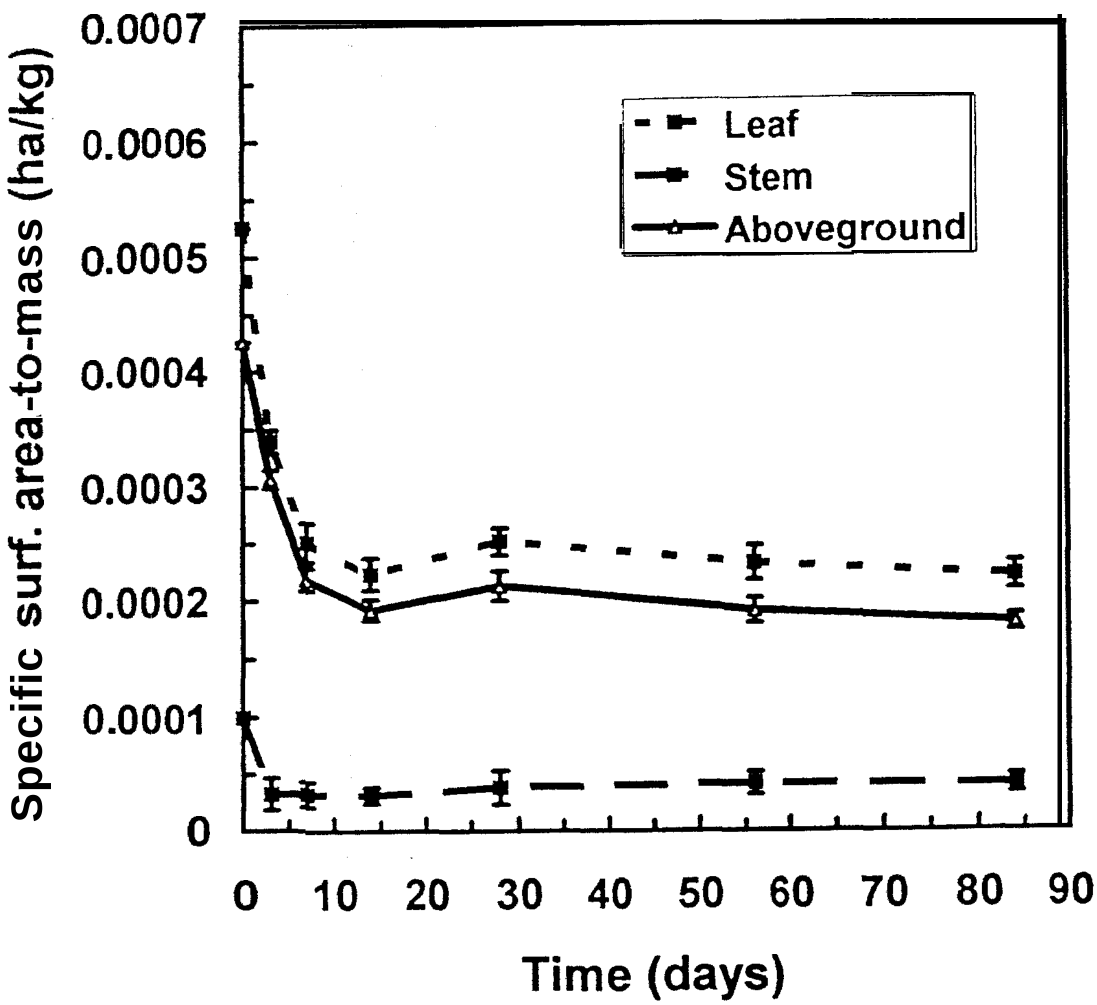


Figure 2.35. Change in specific surface area-to-mass for cotton DLP-5690 over time. Bars represent standard deviations at given time.

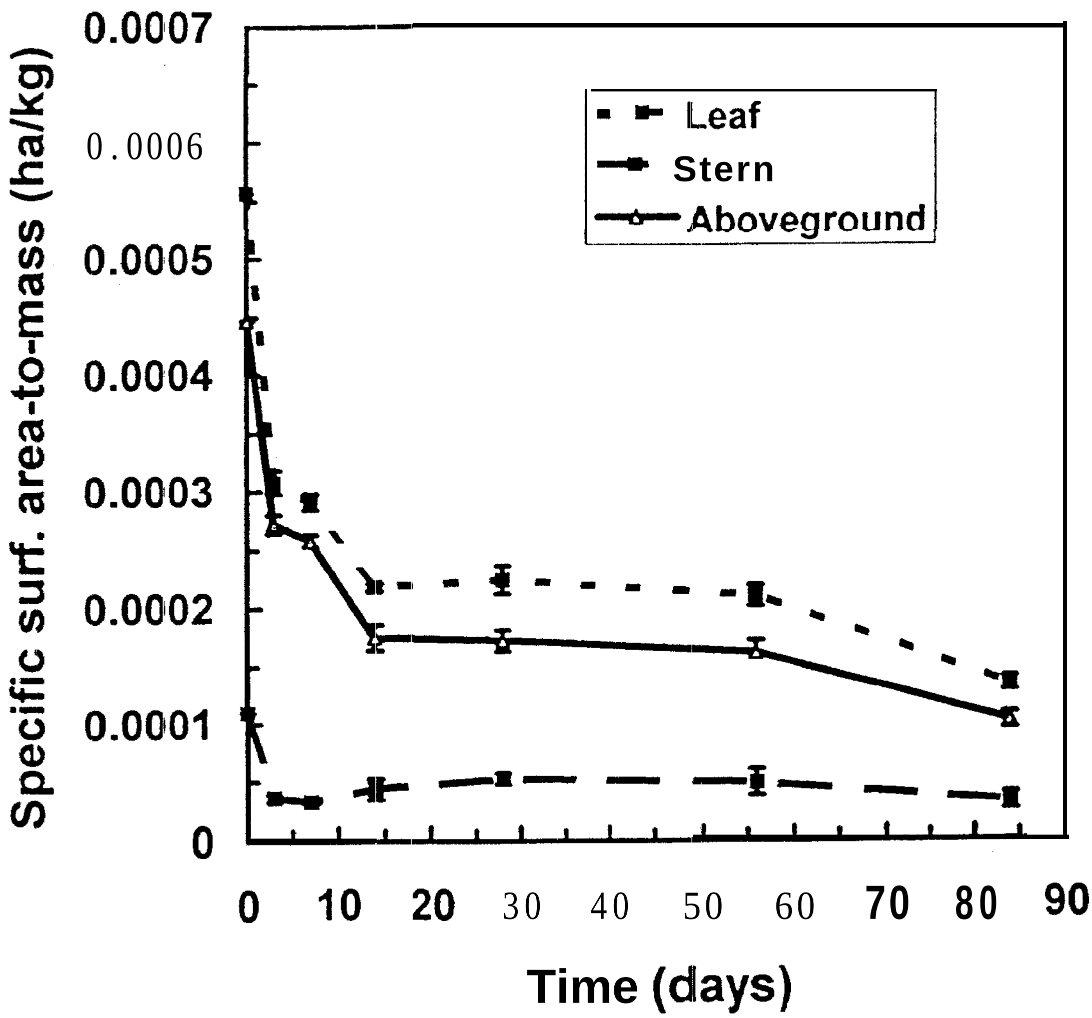


Figure 2.36. Change in specific surface area-to-mass for cotton DP-5215 over time. Bars represent standard deviations at given time.

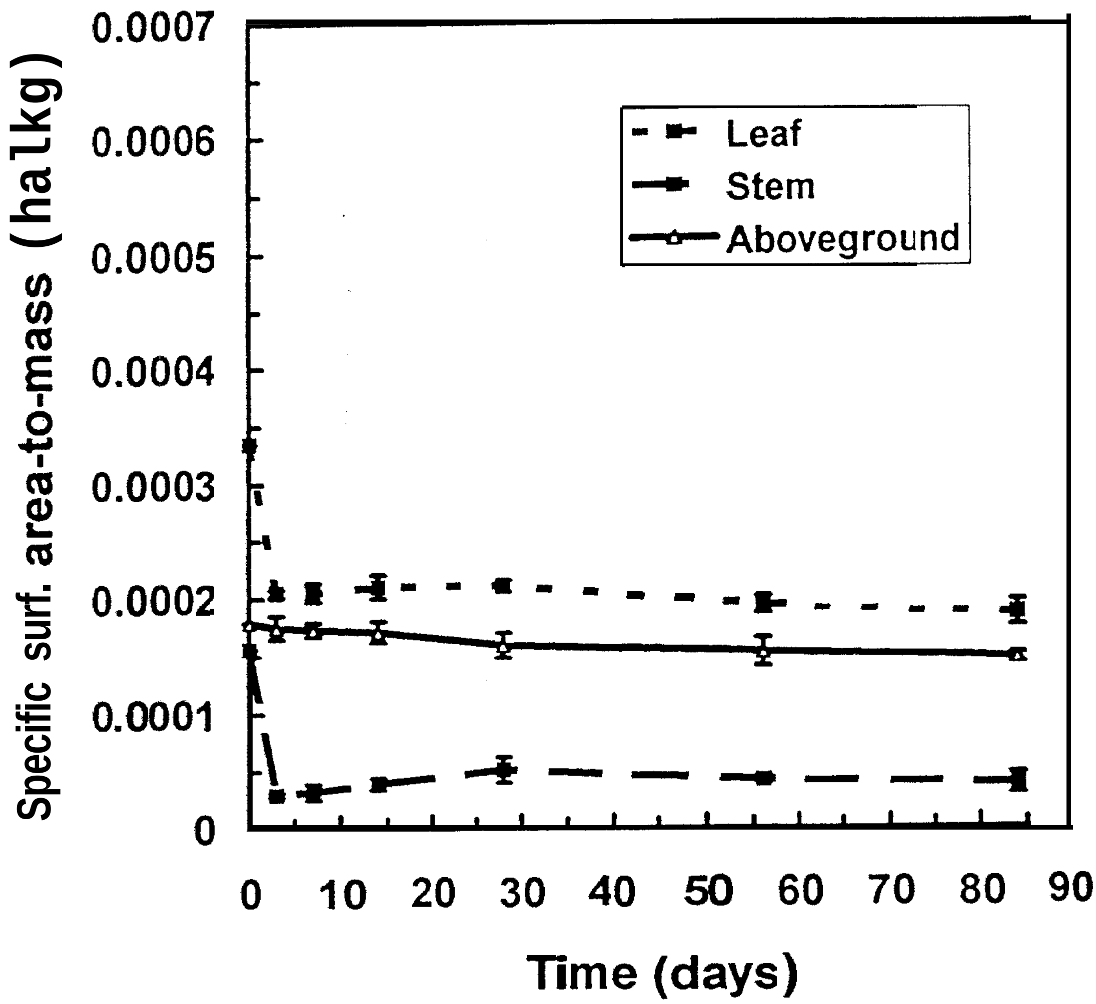


Figure 2.37. Change in specific surface area-to-mass for cotton HS-46 over time. Bars represent standard deviations at given time.

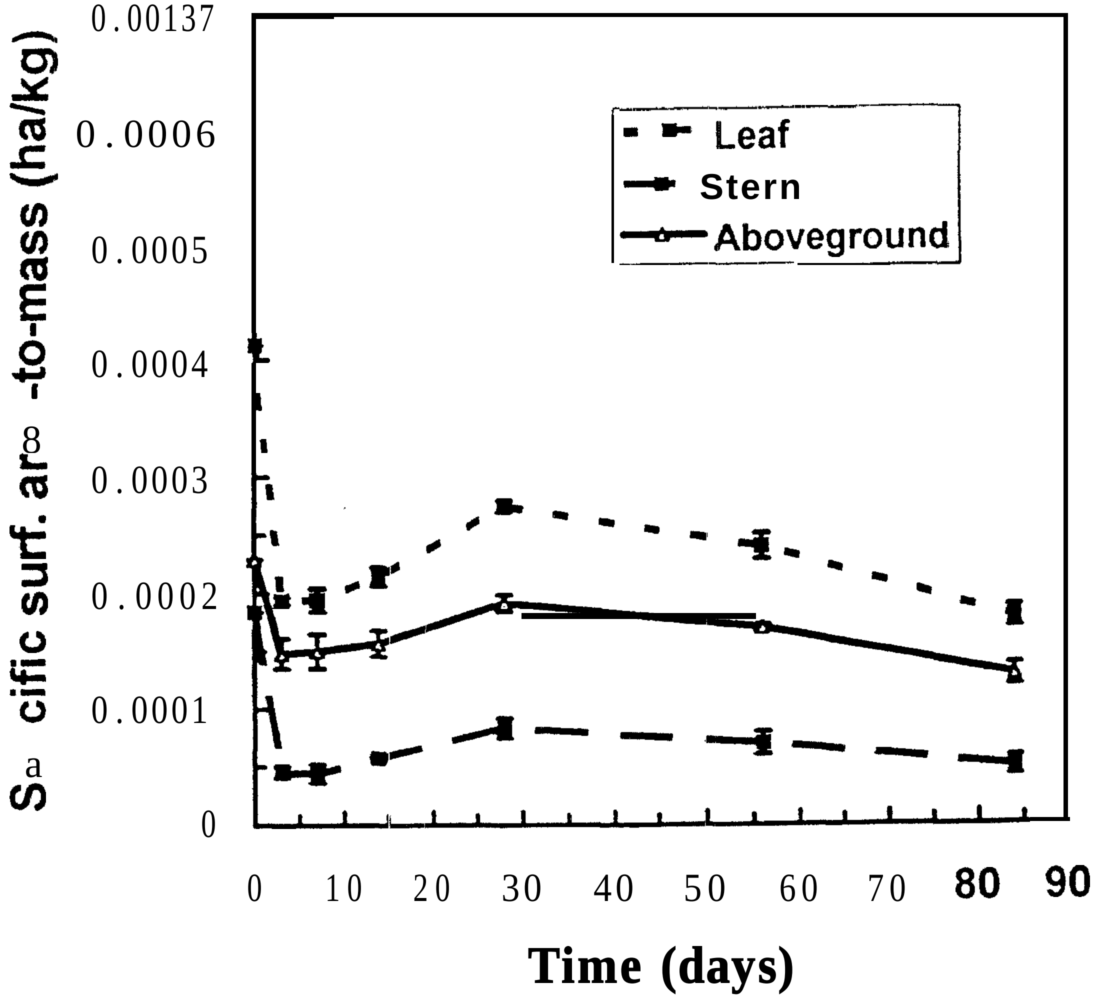


Figure 2.38. Change in specific surface area-to-mass for peanut Florunner over time. Bars represent standard deviations at given time.

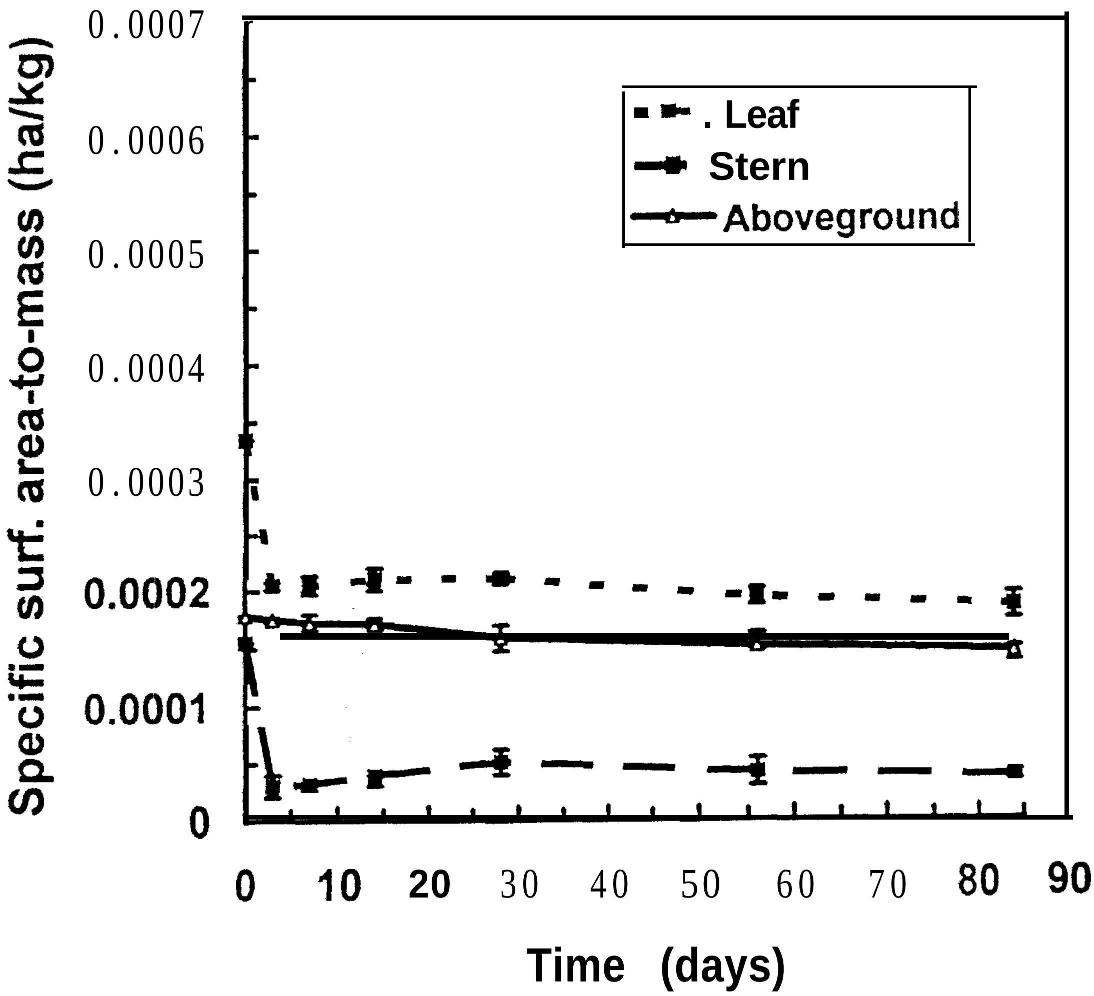


Figure 2.39. Change in specific surface area-to-mass for peanut NC-7 over time. Bars represent standard deviations at given time.

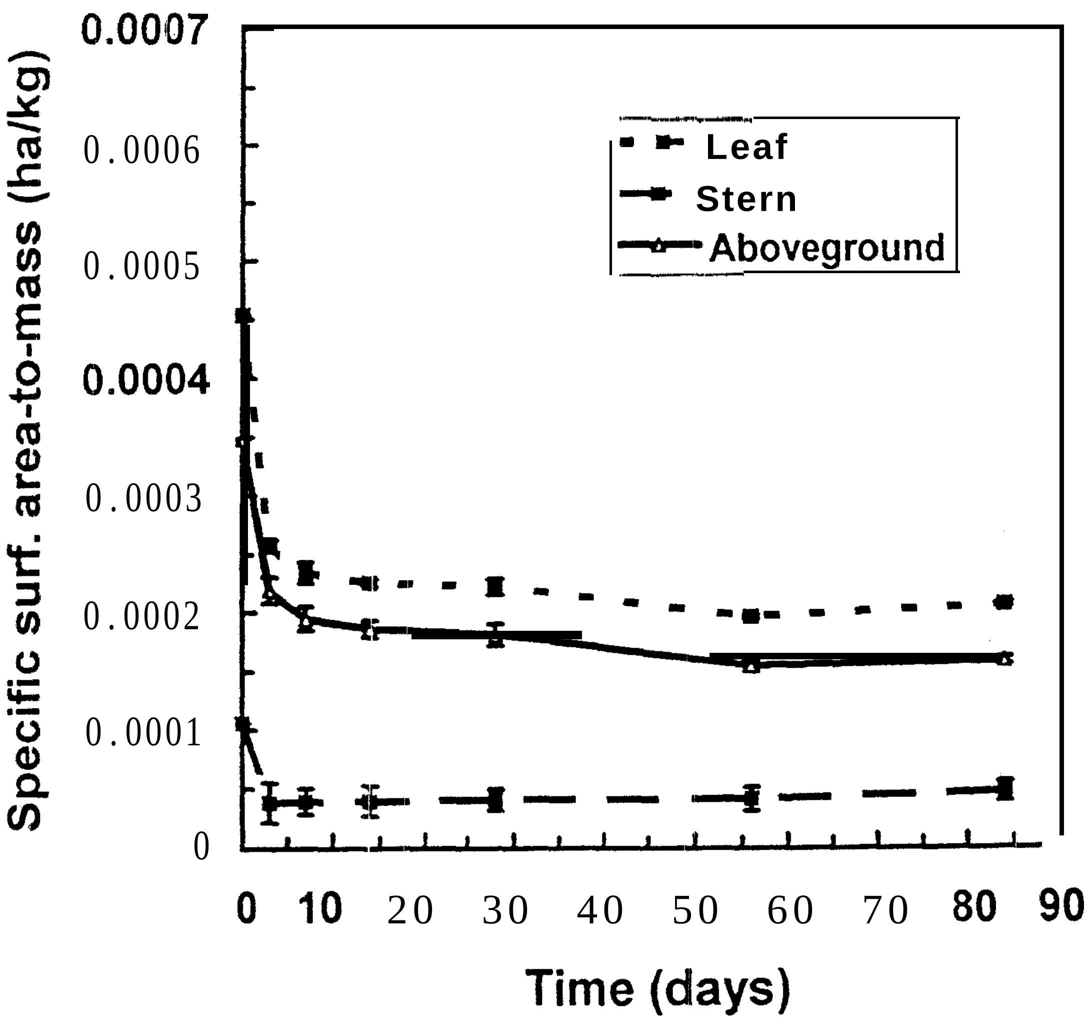


Figure 2.40. Change in specific surface area-to-mass for peanut NC-l Y over time. Bars represent standard deviations at given time.

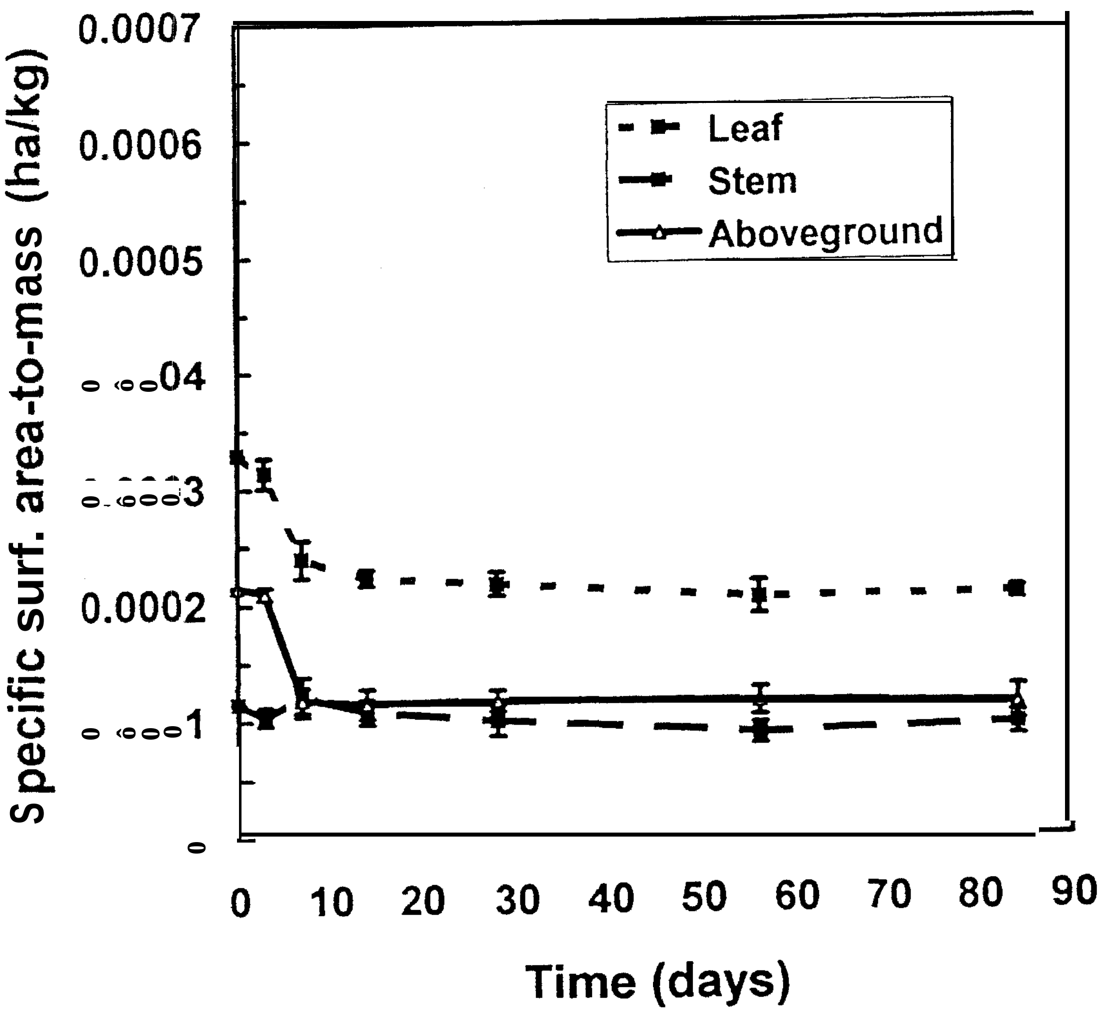


Figure 2.41. Change in specific surface area-to-mass for sorghum Triumph-266 over time. Bars represent standard deviations at given time.

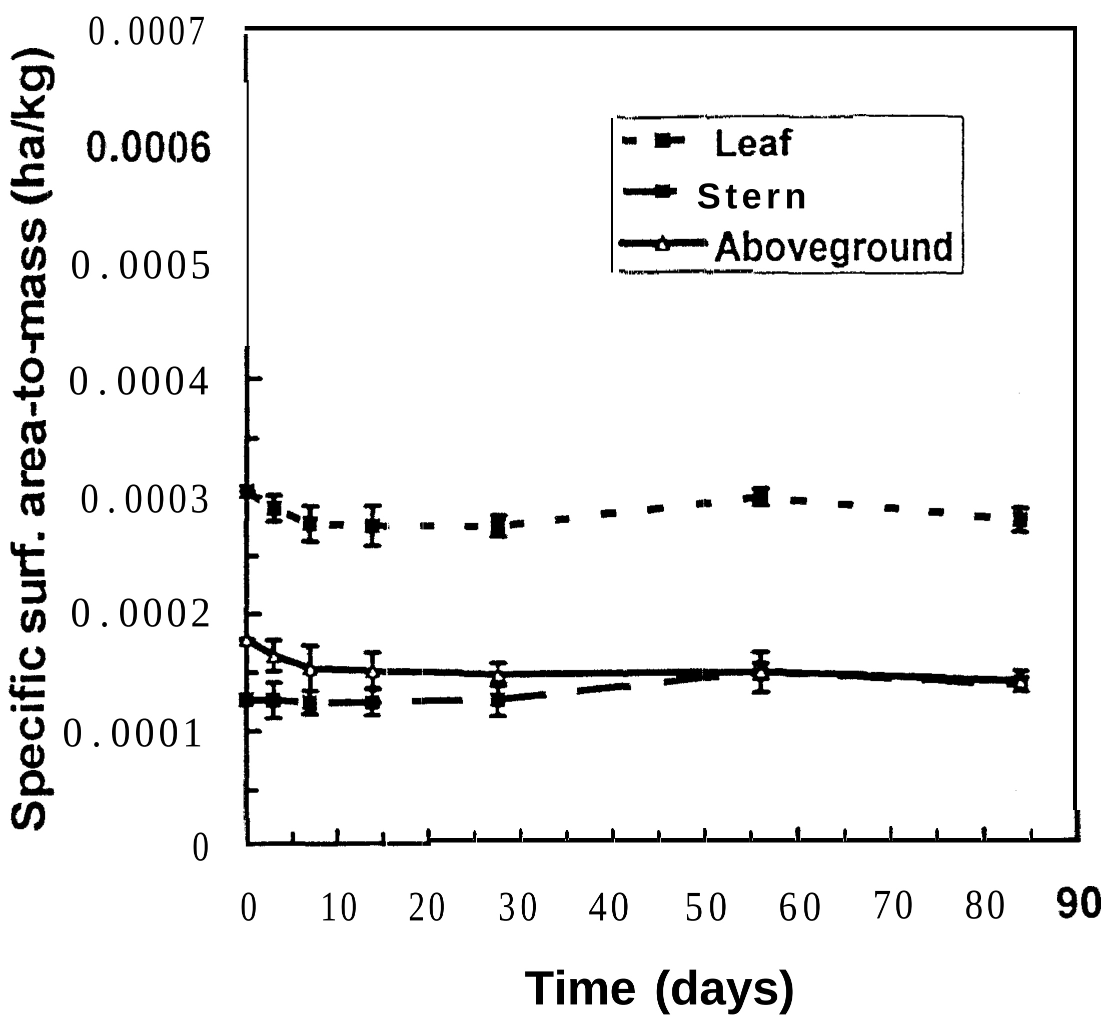


Figure 2.412. Change in specific surface area-to-mass for sorghum GW-744BR over time. Bars represent standard deviations at given time.

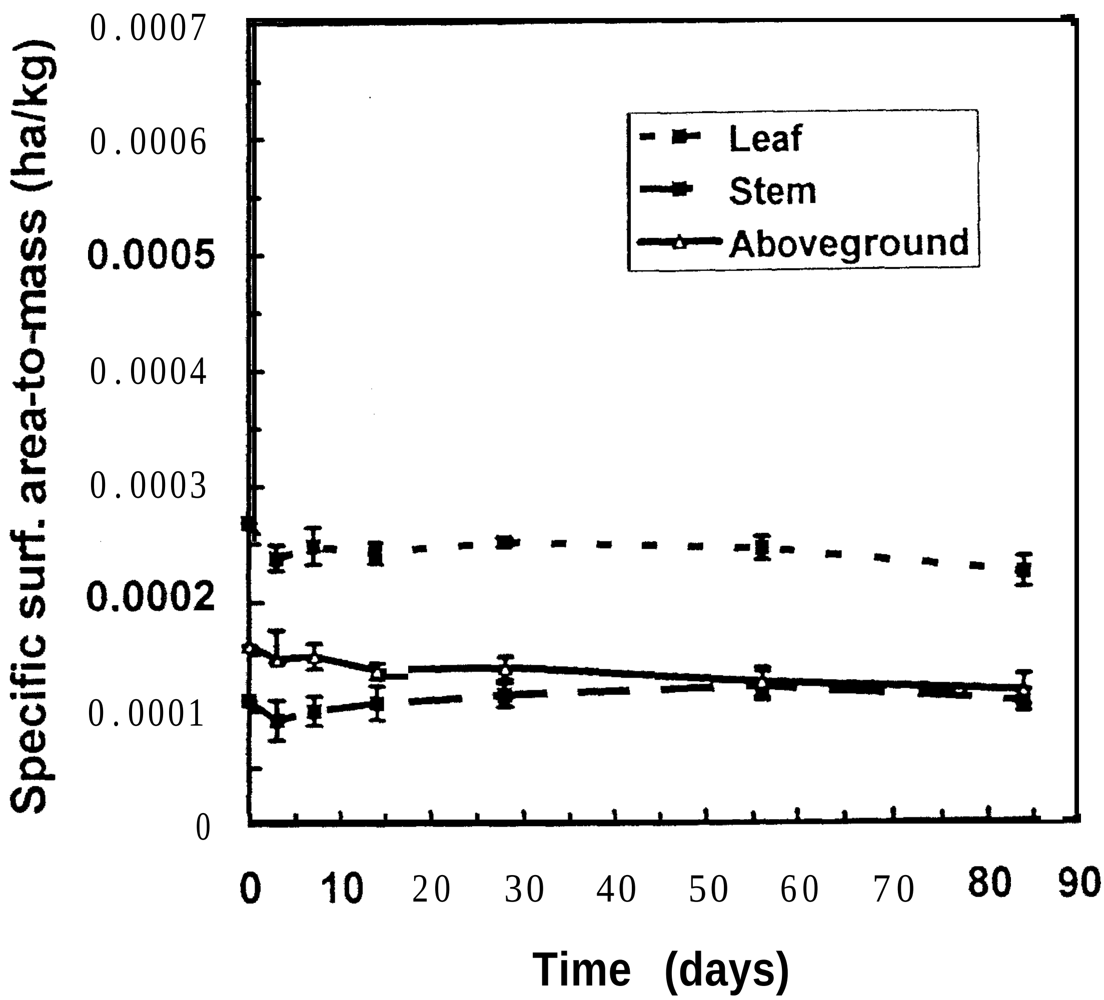


Figure 2.43. Change in specific surface area-to-mass for sorghum Nking-300 over time. Bars represent standard deviations at given time.

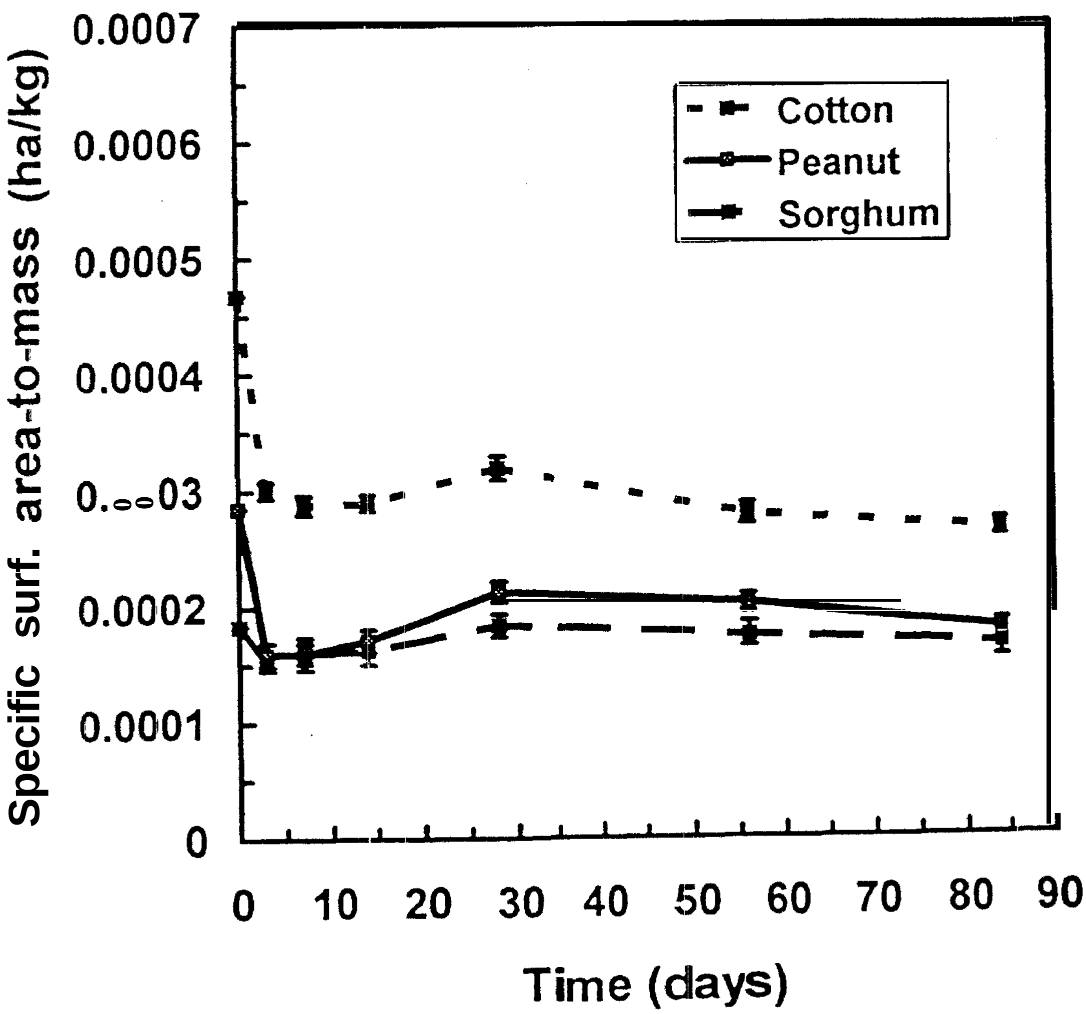


Figure 2.44. Change in specific surface area-to-mass for the three crops over time. Bars represent standard deviations at given time.

2.5.2. Relationship between Mass loss and Car-bon loss

Residue decomposition can be measured by car-bon loss or mass loss. Carbon loss as estimated by CO_2 evolution, is the most used method (Knapp et al., 1983; Stott et al. 1986; Stroo et al., 1989; Collins et al., 1990). Measuring decomposition via mass loss simulates changes in the field and is more important. in natural resource models that need to predict the amount of soil surface covered by residues at any given time. To relate field measurements of residue mass loss to laboratory experiments, in which CO_2 evolution is the variable, a relationship between mass loss and CO_2 evolution was determined using linear regression. The mass loss-carbon loss relation was determined for the above-ground residues and roots of three cultivars each of three species, cotton, peanut, and sorghum (Figure 2.45). The equation of best fit was linear:

$$\text{Mass loss} = 0.16 + 0.58\ CO_2 \text{ evolution} \qquad (2.9)$$

where mass loss (% d^{-1}) and CO_2 evolution (% d^{-1}) rates were calculated based on the first 14 days of incubation.

The residue decomposition measured by CO_2 evolution was higher than the mass loss measurement because the simulation of field measurements of residue mass loss involved uncontrolled field conditions which, with time, did not provide optimal conditions to the microorganisms. Stroo et al. (1989) found that residue rnass loss was greater than the proportion of C lost as CO_2-C, and hypothesized that some physical fragmentation occurred during decomposition preventing full residue recovery. For Collins (1988), the C concentration in the wheat straw decreased slightly as decomposition progressed and some C might be lost as gases other than CO_2, resulting in greater mass loss than carbon loss.

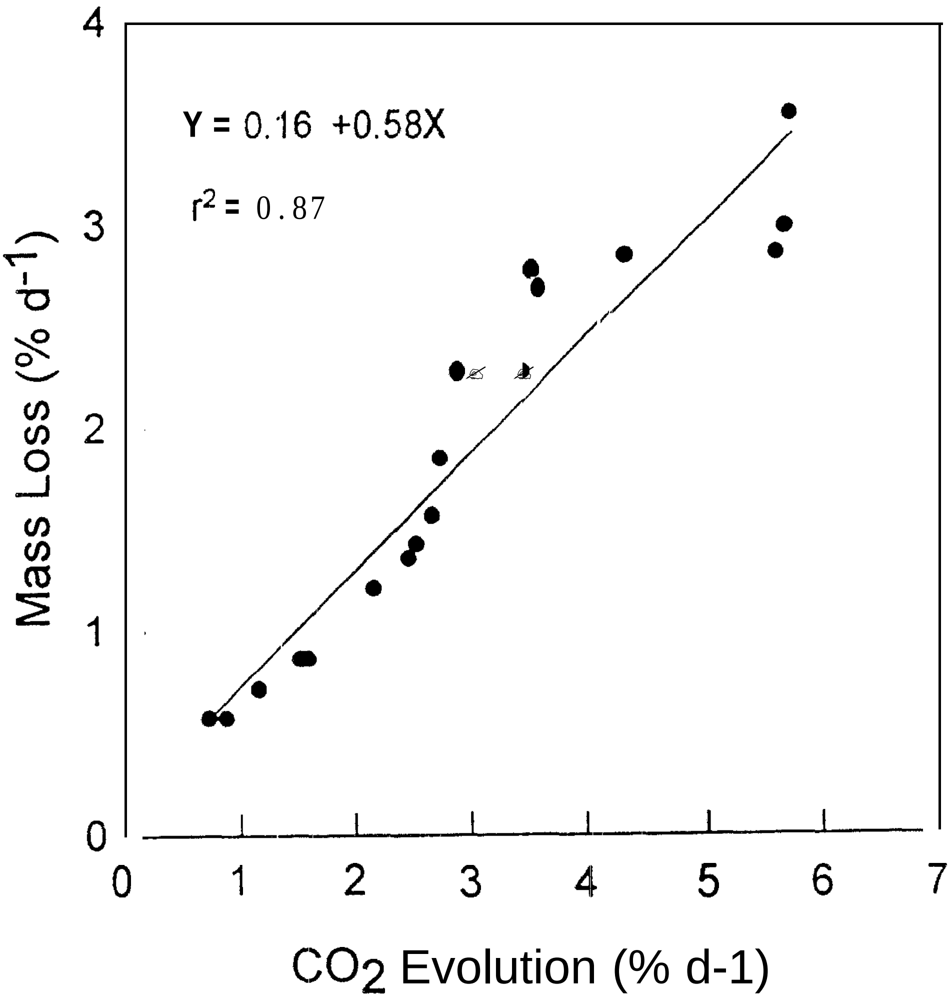


Figure 2.45. Relationship between mass loss and CO_2 evolution for above-ground Ibiomass and roots of three cultivars of cotton, peanut and sorghum in the early stage of decomposition.

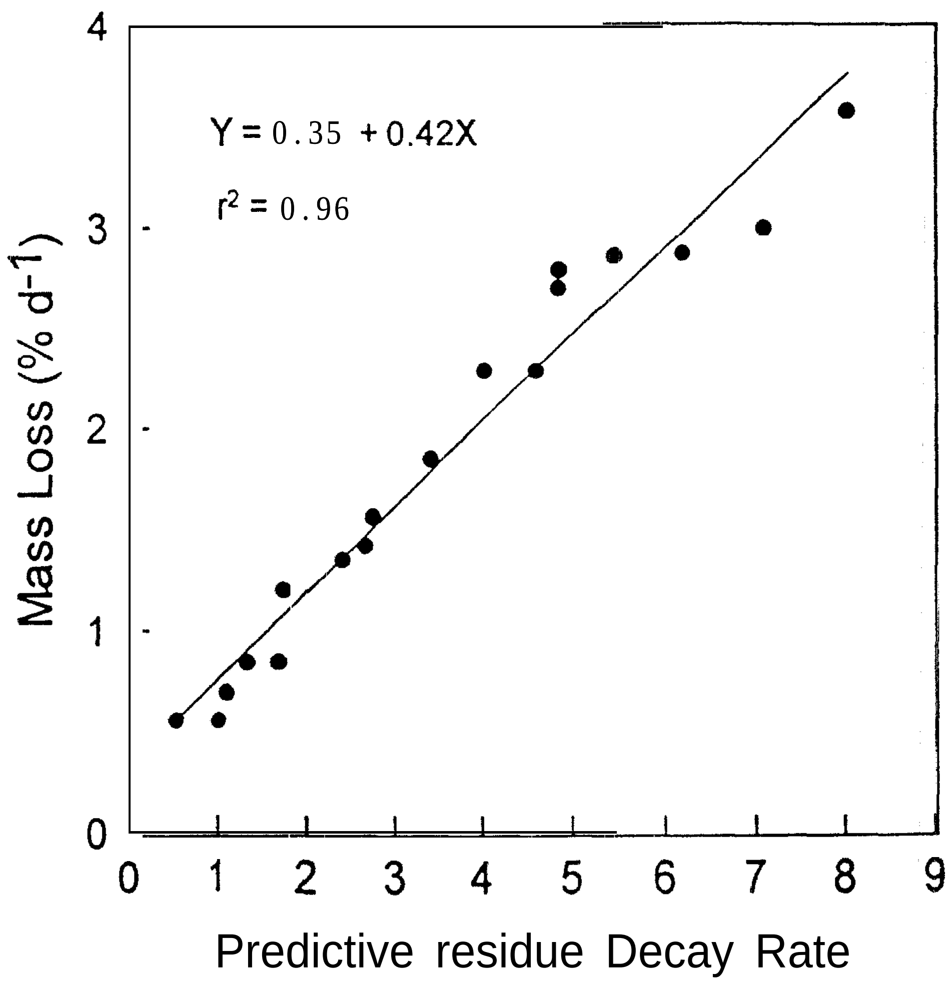


Figure 2.46. Relationship between mass loss and predictive decay rate using above-ground biomass and roots of three cultivars of cotton, peanut and sorghum.

2.5.3. Prediction of Residue Decay

This prediction of residue decay is an attempt to describe in a certain way the contribution of different parameters to the rate of plant residue decomposition. The C:N ratio has been used for long time as a predictor of decomposition, but it has been shown recently that it correlated pooriy with decomposition rate (Stott, 1992). After it has been found that C:N ratio solely could not sufficiently describe the rate of decomposition (Hernan et al., 1977), lignin and lignin-to-nitrogen were also tested for a better prediction of decay rate (Hargrove et al., 1986). Collins et al. (1990) used a relationship with total carbohydrate, C, N and lignin and concluded that the relationship did not seem to hold when the components were mixed before decomposition. The relationship used to predict the plant residue decay rate included total N, simple sugars readily available as fraction soluble C, hemicellulose considered as somewhat available after the soluble fraction, and then lignin which mark the boundary between fractions available and recalcitrant.

The predictive decay rate P_D is expressed in the following equation:

$$P_D = (N \cdot Sugars \cdot Hemicellulose \cdot K) / Lignin \qquad (2.10)$$

where N, (nitrogen), sugars, hemicellulose, and lignin are expressed in g kg-‘, and k is the specific surface area-to-mass ratio (ha kg-‘).

For mass loss (Figures 2.46), the equation of best fit was linear in the form:

$$\text{Mass loss} = 0.35 + 0.42\, P_D \qquad (2.11)$$

For CO_2 evolution (Figure 2.47), a linear regression fitted the equation in the form:

$$CO_2 \text{ evolution} = 0.47 + 0.70\, P_D \qquad (2.12)$$

where mass loss (% d^{-1}) and CO_2 evolution (% d^{-1}) rates were based on the first fourteen days of incubation.

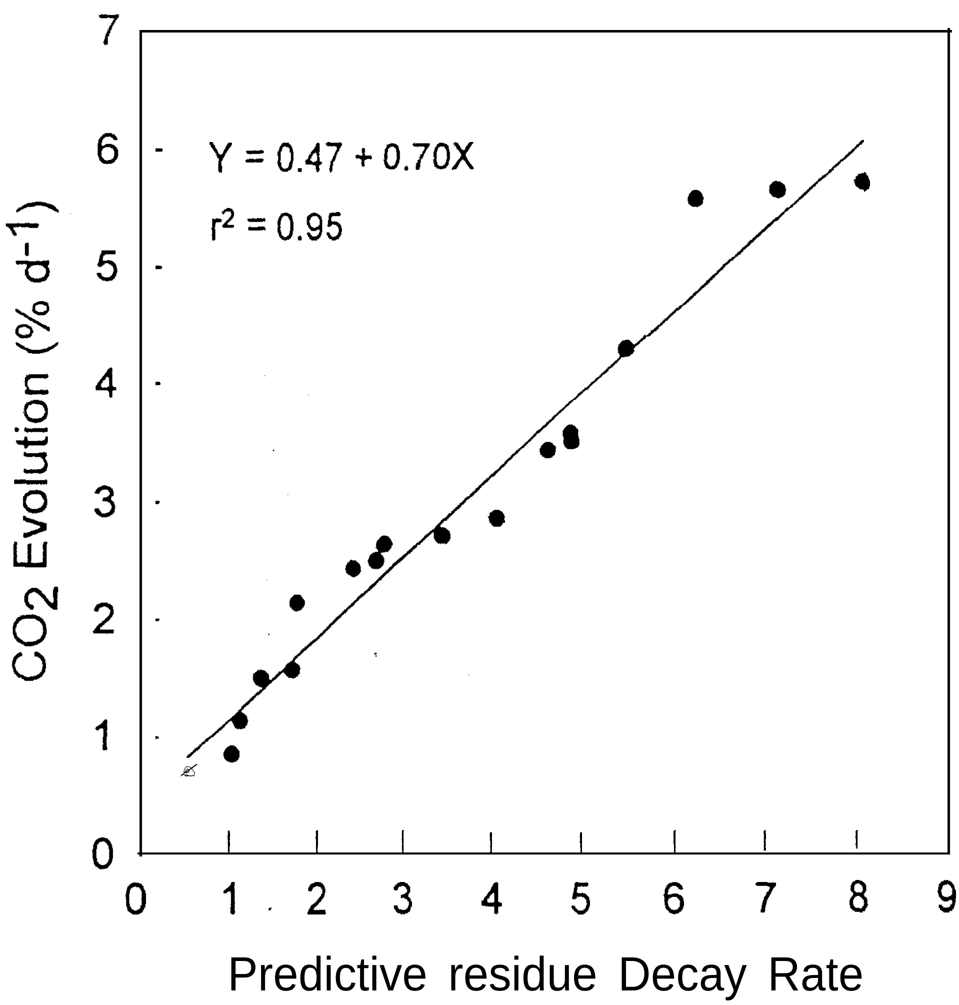


Figure 2.47. Relationship between CO_2 evolution and predictive decay rate using above-ground biomass and roots of three cultivars of cotton, peanut, sorghum.

Table 2.6. Predictive ratio and Rate constants of CO_2 Loss and Mass Loss.

Crop	Cultivar	Residue Type	Component Ratio	Predictive Rate	Rate constant (% d^{-1})*	
					CO_2 Loss	Mass Loss
Cotton	DLP-5690	leaf / stem	46.7:53.3	5.45	4.2	2.9
		root	100	1.13	2.1	1.2
	DP-5215	leaf / stem	41.2:58.8	4.86	3.2	2.8
		root	100	0.53	0.7	0.6
	HS-46	leaf / stem	47.1:52.9	2.66	2.5	1.4
		root	100	2.75	2.6	1.6
Peanut	Florunner	leaf / stem	25.9:74.1	6.20	5.6	2.9
		root	100	1.34	1.5	0.9
	NC-7	leaf / stem	29.2:70.8	7.10	5.6	3.0
		root	100	1.10	1.1	0.7
	NC-11	leaf / stem	31.1:68.9	8.04	5.7	3.2
		root	100	1.01	0.9	0.6
Sorghum	Triumph-266	leaf / stem	45.3:54.7	4.60	2.7	1.9
		root	100	1.70	1.6	0.9
	GW744BR	leaf / stem	38.7:61.3	4.02	3.4	2.3
		root	100	2.40	2.4	1.4
	NKing-300	leaf / stem	43.6:56.4	3.40	2.9	2.1
		root	100	4.85	3.6	2.7

* The rate constant is calculated as the slope of the curve (%) divided by 7 days,

2.6. Conclusions

The initial chemical and physical characteristics of the plant residues and roots impacted the rates of decomposition . The decomposition rates determined by CO_2 evolution and mass loss showed differences between cultivars, for cotton, peanut and sorghum. Due to their leguminous nature, the three peanut cultivars were decomposed rapidly, and were different in decay rates among them. The degradability of peanut above-ground residue was highest followed by cotton, while the sorghum above-ground decomposition fate was the slowest. The plant roots did not follow the same order in degradability as did the plant above-ground residues. Sorghum roots were decomposed faster than cotton and peanut. There was significant difference between the decomposition rates of the cotton and peanut roots. CO_2 evolution and mass loss methods used to determine rates of decomposition were highly correlated.

Changes in specific surface area-to-mass measurements showed significant differences between cultivars within cotton only, but there were differences between species as if k value was a constant specific for each crop.

It was possible to develop a prediction decay equation from the initial chemical and physical characteristics of the residues for the early stage of decomposition.

This predictive decay equation in the early decomposition process is a partial result that can be used to predict decomposition rate of residue in the early stage. A validation of the predictive equation with decomposition rates measured in the field will certainly help predict the decomposition rate of any plant residue over time. Once validated, this predictive decay equation will be a useful tools for land managers, conservation planners, environmental scientists and even those concerned with construction sites. It also could be used as parameter in a crop breeding program. Predicting residue decomposition, used in a management program, can help solve soil erosion problem, but also can help control accumulation of crop residues when it is viewed as a nuisance to crop establishment and growth, or as a disposal problem.

Future work will include using the predictive decay equation to develop residue decay parameters for erosion prediction models such as RUSLE (Revised Universal Soit Loss Equation), RWEQ (Revised Wind Erosion Equation), WEPP (Water Erosion Prediction Project), and WEPS (Wind Erosion Prediction System).

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CHAPTER 3

CROP RESIDUE DECOMPOSITION WITH CHANGE IN SOIL DEPTH

3.1. Abstract

Microorganisms play a major role in the crop residue decomposition psocess, and it has been assumed that microbial activity is uniformed with soil depth in a given tillage system. This study was conducted to determine variation in residue decornposition rates related to the microbial activity with changes in soil depth under established no-till and a moldboard plow tillage system, on a silty clay loam soil at the Purdue Agronomy Research Center, West Lafayette, IN. Soil cores were sampled at O-20 cm and then partitioned into O-l, 1-5, 5-12.5 and 12.520 cm sections constituting the different sampling soil depths. The peanut (*Fastigiata vulgaris)* residue used in the experiment was the Spanish Tampsan 90 cultivar. The decomposition rate was quantified by measuring the amount of CO_2-C evolved from an electrolytic respirometer incubation system, loading 2 g of airdried residues in 100 g air-dried soil for each treatment. Soil depth in no-till soil, signifkantly influenced residue decomposition. After 84 days, cumulative % CO_2 evolution from the surface soil (O-l cm) was high, 50%, whereas, from the lower depth soil (12.5-20 cm), CO_2-C was much lower, 22%. From the intermediate depth soil, (1-5 cm), residue decomposition as measured by CO_2-C evolution was significantly lower, 37%, than from the surface soil, but significantly higher than decomposition from the lower depth soil. From the plowed sites, a reverse situation occurred due to

inverting residues. Residue decomposition rates from lower depth soils (5-12.5 cm and 12.5-20 cm) as measured CO_2-C evolution was 40% and 38% respectiveiy, and not signifkantly different from each other, but were significantly greater than the decomposition rates, 21% and 13% CO_2-C evolved from soil obtained from the shallower depths, l-5 cm and O-1 cm, respectively. Due to lower microbial activity, residue decomposition decreased with soil depth in no-till situation whereas in a moldboard piow tillage system, it increased with soil depth.

3.2. Introduction

The amount of crop residues remaining on the soil surface and within the top 20 cm of the soil profile are critical factors in erosion control. A successful crop residue management system depends upon an understanding of the factors governing crop residue decomposition, and how much residue cover is lost from a field site.

Tillage influences the physical environment near the soil surface, thus affecting biological process in the soif. Soif profile differences between no-till and conventionally tilled soil have been reported and can be detected after a few years of changing from conventional to no-till management practices (Dick, 1983). According to Doran (1980), no-till soils have more total microbial biomass than conventionai tillage soils in the surface O-7.5 cm. In addition, there are increases in soil water content, organic carbon contents, and total nitrogen levels in the no-till soils probably due to higher amounts of residue left on the soil surface in the no-till system. Each tillage event causes a movement of moist soil to the surface, which then dries rapidly.

Surface residues affect soil temperature pattems and soil water content, thus affecting biological activity in the soil (Roper, 1985). Along with soil physical and chemical characteristics, microorganisms play a major role in the crop residue decomposition process. Therefore, knowledge of crop residue decomposition undet a given tillage system, and how decomposing activities of the microbial populations are distributed as soil depth changes would be useful information for predictive rnodels.

The objective of this study was to determine if there is a difference with depth in residue decomposition rates when soil is held under identical environmental conditions.

3.3. Materials and Methods

3.3.1. Soil and Site Description

A Drummer silty clay loam soil (fine-silty, mixed, mesic Typic Haplaquoll) was used in this experiment. The sampling site was a nineteen-year tillage corn/soybean rotation field experiment located at the Purdue Agronomy Research Center in West Lafayette, IN. The site has less than 2% slope, is tiled at a 20-m spacing, and the soil is well structured (Table 3.2).

The plots were established in 1975 and consist of corn/soybean rotation under a variety of tillage managements. The two tillage systems sampled in this study were: (i) fall moldboardl plowing to 20 cm, with one disking and one field cultivation to 10 cm in the spring prior to cultivation and (ii) no-till planting with 2.5-cm-wide fluted coulters to cut through residues and open a slot ahead of standard planter units (Griffith et al., 1988).

The soil samples used for this experiment were taken from the no-till and moldboard plow plots of com following soybean. For each treatment, four replicate plots were sampled, The samples were taken from between rows 2 and 3 within each plot as this row was uncompaded by wheel traffic. In each plot, four soit cores were taken from the O-5 cm layer using rings, and four other soil cores were also sampled from the O-20 cm layer using soil probes. The samples were then partitioned into O-l cm, 4-5 cm, 5-12.5 cm, and 12.5-20 cm soil depths The soil samples taken with the rings were to complete the amount of soil needed for the experiment at depths O-l cm and 1-5 cm. The samples were airdried, ground to pass a Z-mm sieve, and stored until use.

3.32. Plant Materials

Peanut (*Fastigiata vulgaris*), Spanish Tampsan 90 cultivar, was grown in 5-gallon buckets, using a sanitized soil mix. The plants were grown in the greenhouse for 125 days. On a three-week basis, the plants were treated with specific compounds against white flies and spidermites. After harvesting, the aboveground biomass (stems and leaves), the below ground biomass (roots) and the yield biomass (pods) were separated from one another. The residue samples were washed to remove excess soil. After washing, residue samples were dried at 40°C for 48 hr and weighed.

A subsample of plant residue was finely ground (< 0.3 mm) for chemical analysis, using a Straub Grinding Mill (Model 4E, Straub CO, Philadelphia PA). Total C, H, and N contents (Table 3.1) were determined using a dry combustion analyzer (Model CHN-600; Leco Cor-p., St. Joseph, Ml). Lignin, cellulose, and hemicellulose contents were determined by sequential fiber analysis using the Goering et. al. (1970) procedure (see chapter 2 for details). Chemical analysis were done in triplicate.

Table 3.1. Initial chemical composition of the peanut residues

Residue type	Total C	Total N	Cellulose	Hemicellulose	Lignin	Ash
			g kg^{-1} residue			
Aboveground*	397.4	24.4	191.0	241.5	68.7	17.0
Roots	397.0	22.3	286.5	230.7	85.0	22.2

*Aboveground is the non-harvested material, primarily stems and leaves.

3.33 Decomposition Experiment

Residue decomposition rates were determined by the amount of C evolved as CO_2 over time. The experiment consisted of eight treatments. Four treatments were composed of soil from the no-till system at four depths (O-1 cm, l-5 cm, 5-12.5 cm and 12.5-20 cm), the other four were from the moldboard plow system, at the same depths. For each treatment, 100 g soif and 2 g peanut residue (ovendried basis) were placed in an incubation jar. The 2 g residue consisted of 1 g stem, 0.5 g leaf and 0.5 g roots, representing the proportion of each residue component left in the field after harvest. The controls consisted of soil from each treatment with no residue. The incubation jars were connected to electrolytic respirometers (Knapp et al., 1983a). The optimal moisture content for incubation was consiclered to be the water content at -1/3 bar water potential as equalled to 60% water' holding capacity, plus 300% of the residue mass (Myrold et al., 1981). The moistened soil was mixed thoroughiy, the dry residue spread evenly on the soil surface, soil to residue contact insured and then the incubation jar was tightly sealed (Stott et al., 1986). The jars were submerged in a water tank and insulated by putting styrofoam on. The water temperature was maintained at $22^{o}C \pm 1°C$ with a circulating water bath.

The amount of CO_2 respired was captured in an alkaline trap of 5 ml 30% KOH. An indicator, tropaelin 0, (Sigma Chemical CO, St. Louis, MO), was added to the KOH solution to indicate if the solution has reached a 50% CO, saturation (pH 11). To remove the KOH, a 22-gauge needle with a Luer lock fitting through the stopper and lengthened with a sufficient piece of capillary tubing to reach the bottom of the KOH trap will be used. Fresh KOH was injected in the same manner, thus the incubation chamber remained sealed throughout the experiment (Stott et al., 1986). KOH was withdrawn after 3, 7, 14, 28, 56 and 84 days of incubation. The amount of CO, evolved during the decomposiition was measured by titration of the KOH solution using Golterman (1970) potentiometric titration method.

3.3.4. Incubation system

The system used to incubate the soils consisted mainly of a respirometer and an incubation jar held in a circulating water bath to maintain a constant temperature and prevent condensation within the jars. The circulating bath (Model 2095, S/N, Forma Scientific, Marietta OH) was connected to a plexiglas water tank in which the jars were held (Stott et al., 1986). Each incubation jar was connected to an electrolytic respirometer. At the top of each respirometer, there was a 25 or 50-ml burette, a positive electrode for oxygen, and a 4-cm tube for overflow. At the bottom, there is a negative electrode for hydrogen. Both electrodes were platinum. The positive electrode is connected to a 500-ml chamber containing the electrolyte solution 8% $(Na)_2SO_4$.

Within each incubation jar, there was a small glass cup to hold the alkaline trapping solution. Respired CO_2 was absorbed in the KOH trap, thereby reducing the total pressure in the incubation jar. This causes the electrolyte to be drawn up into the capillary tube containing the O_2 electrode. As the electrical circuit is completed, H_2O is hydrolyzed with H_2 being captured in the gas burette.

3.35. Measurement of CO_2 evolution

The reactions involved in the KOH trapping the evolved CO_2 are as follows:

$$KOH + CO_2 \longrightarrow HCO_3^- + K^+ \quad (3.1)$$

$$HCO_3^- + K^+ + HCl \longrightarrow H_2CO_3 + KCl \quad (3.2)$$

Each milliequivalent of KOH used to absorb evolved CO, is equivalent to 12 mg of CO, carbon.

The formula used to calculate cumulative % C-CO, evolved is:

$$\% \text{ C-CO}, = [\, K_1 * (1/M) * V * N * C, \,] \tag{3.3}$$

where:

K_1 = 0.315, a calculated constant to convert the raw result into the desired unit

M = mass of the residue in grams

V = volume of HCI titrant in ml

N = concentration of HCI titrant in normality

C_i = initial carbon content of the residue in percent.

3.3.6. Statistical Design

The experiment consisted in a completely randomized design with treatment soils from two management systems, and four soil depths (eight treatments plus controls). The experiment was done in triplicate.

Statistical analysis of the data was run to determine differences among treatments using the PC-SAS, Version 6.09 (Statistical Analysis System 1985). Analysis System 1985).

3.4. Results and discussion

The mean concentrations of total C (Table 3.2) from the surface 0 to l-5 cm no-till soil were significantly greater than that of plowed soil (P = 0.05). However, below 5 cm, there was no significant difference in total C contents between the two tillage systems. Within the no-till system, total C contents were not significantly different frorn the surface 0 to 1-5 cm, but they were significantly higher than those below 5 cm. No significant differenœ in total C concentration was observed along the profile 0 to 20 cm within the moldboard plowed soii. Total N content (Table 3.2) was significantly greater from the surface O-l cm no-till than plowed soil. Below 12.5 cm, the mean concentrations of totai N of moldboard piowed soil were significantly higher than those of no-till soil. Within no-till system, total N contents were significantly decreasing with depth soils, whereas within the plowed soil, total N contents were increasing.

Table 3.2. Physical and chemical characteristics of the soit samples

Depth (cm)	Tillage	Clay (%)	Silt (%)	Sand (%)	pH	Total C (g kg-‘)	Total N (g kg-‘)
O-1	No-Till	27.9	57.6	14.5	5.84 a	28.4 a’	4.0 a
	M. Plow*	35.9	54.9	9.1	5.98 a	23.7 b	3.0 b
I - 5	No-Till	28.8	59.9	11.2	6.05 a	26.1 a	3.2 b
	M. Plow	39.2	50.8	10.0	5.92 a	23.1 b	3.1 b
5 - 12.5	No-Till	40.3	50.1	9.7	5.01 b	23.9 b	2.9 b
	M. Plow	30.2	58.4	11.3	5.50 ab	23.8 b	3.5 ab
12.5 - 20	No-Till	37.7	51.6	10.7	4.76 b	23.4 b	2.6 c
	M. Plow	28.6	59.3	12.1	5.47 ab	22.4 b	6.3 ab

*M. Plow = Moldboard Plow
‘Values within columns, followed by the same letter are not significantly different by the Waller-Duncan’s multiple range test at P = 0.05.

Soil depth influenced significantly microbial residue decomposition in both tillage systems. After 84 days, high microbial activity resulted in 50% CO& evolved from the surface soil (O-l cm), as compared to 23% C evolved from the lowest depth soil, 12.5-20 cm, (Figure 3.1). The amount of the CO_2-C evolved from the intermediate depth soil, l-5 cm, was significantly (P = 0.05) lower, 38%, than from the surface soil, but significantly higher than the CO_2-C evolved, 25% and 23%, from the lower depth soils, 5-12.5 and 12.5 - 20 cm respectively.

In the moldboard plow system, a reverse situation occurred (Figure 3.2). Residue decomposition rates did not differ from the lower depth soils, 5-12.5 and 12.5-20 cm, 42% and 40% CO_2-C evolved respectively. They were, however, significantly greater (P = 0.05) than the decomposition rates in soils from the shallower depths, 1-5 and O-l cm, measured as 27% and 21% CO_2-C evolved respectively.

In the no-tilled soil, the decomposition rate in the shallow depth soils, O-l cm and 1-5 cm, did not differ significantly from rates in the lower depth moldboard plowed soils, S-12.5 cm and 12.5-20 cm. There was also no significant difference (P = 0.05) in C evolution between the lower depth no-till soils, 5-12.5 un and 12.5-20 cm, and the top layer moldboard plow soils, O-l cm and 1-5 cm.

The amount of CO_2-C evolved measured during the residue decomposition process is an index of the activity of the microorganisms being respiring. Along the top 20-cm of the soil profile, residue decomposition as determined by microbial respiration showed great differences across the no-till and moldboard plow systems. Microbial respiration in surface no-till was significantly greater than that in plowed soit (Figures 3.1 and 3.2). At greater depth, microbial respiration was much higher in moldboard plowed soil than in no-till system. These results were consistent with the observations of Barber et al. (1977) and Doran (1980b) who found that respiration rates from surface no-tilt soils were signifkantly greater than those from plowed soils. However, at a soil depth below 50 and 75 mm, these indexes of microbial activity were often greater in plowed soils, reversing the trend noted in the surface 0- to- 75 mm. In general, the presence of surface crop residues in no-till system results in physical and chemical changes in the soil environment. The organic matter distribution is shifted towards the surface, the pore size distribution induces larger macropores, water is lost more slowly due to iow evaporation, nutrients are translocated by plants from the subsoil to the surface during the plant life cycle. Consequently, optimal conditions for an increase in CO_2 evolution are created through a stratification of the microbial respiration at the top of the soil profile. Most researchers (Campbell et al., 1976; Lal et al., 1976; and Blevins et al., 1977) have concluded that the increased microbial activity observed in the surface layer of reduced or no-tillage soils, is related to their greater organic carbon C and water contents resulting from the maintenance of wop residues on the soil surface. In the moldboard plowed soil, the trend of microbial respiration observed was reversed (Figure 3.2). The increase in CO_2 evolution due to maximal microbial activity extended to a greater soii depth than with no-till. This could be due primarily to the plowing action which inverted the residues into a deeper depth soil. Moreover, soil air diffusion rates resulting from plowing and cultivation accelerate the process by which soil microorganisms oxidize organic matter which becomes considerably reduced at the surface.

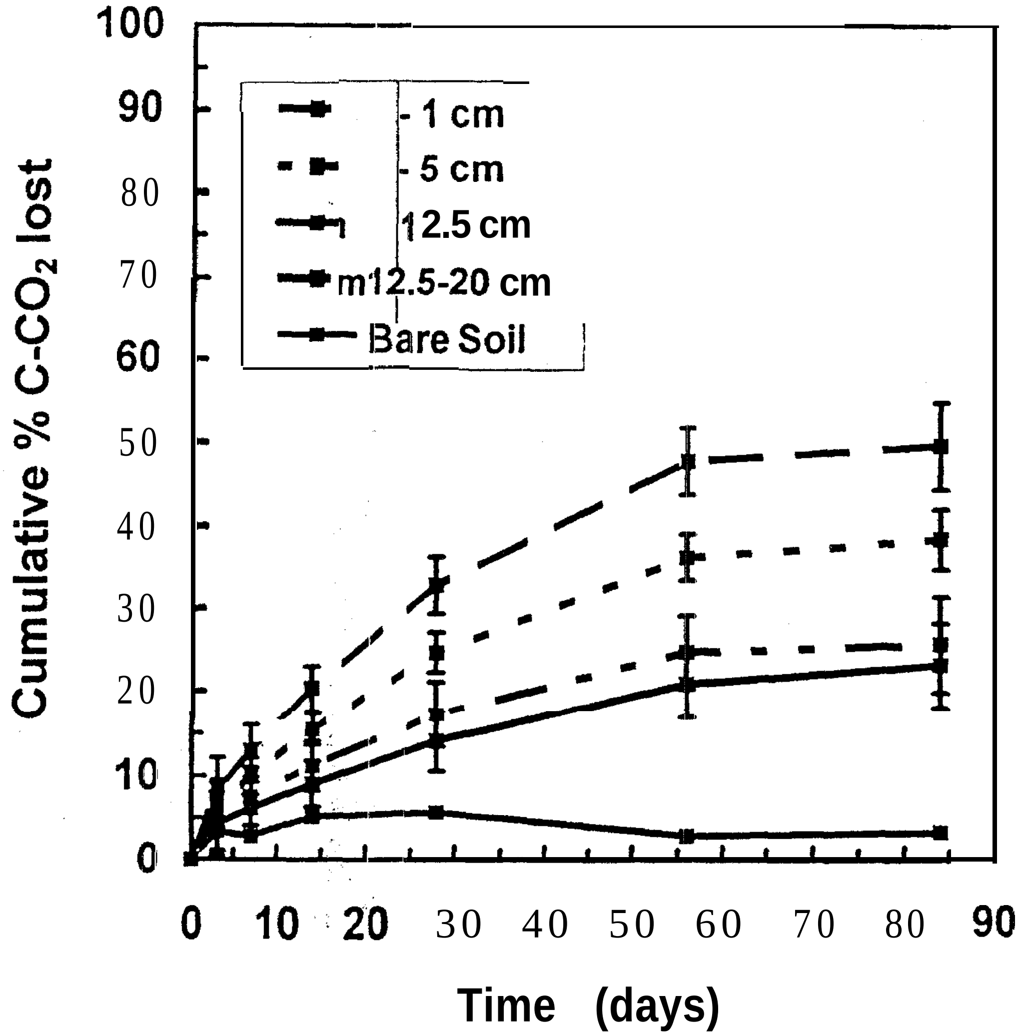

Figure 3.1. Cumulative CO_2-C evolution from different depth no-till soils amended with peanut residue. CO_2 evolved from the bare soil was used to correct the CO_2 evolution from the treatments with residues.

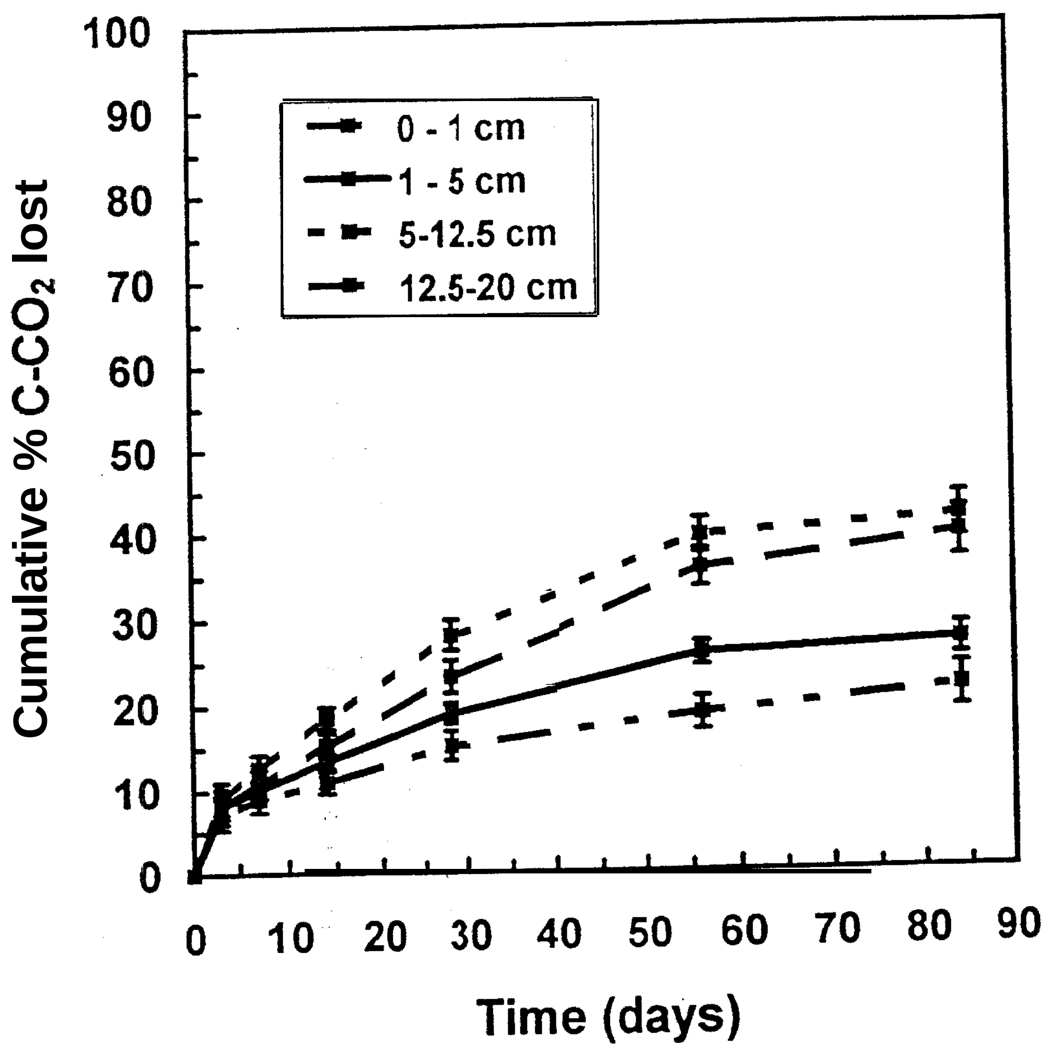

Figure 3.2. Cumulative CO_2-C evolution from different depth moldboard plowed soils amended with peanut residue.

3.5. Conclusion

Residue decomposition fates decreased with soil depth in a no-till management system, whereas in a moldboard plow system, it increased with soil depth, when temperature and moisture are held constant. This might be due to the fact that in no-till soil, crop residues are left at the soil surface whereas in a moldboarcl plow system, surface residue biomass is incorporated into the soil profile. This leads to an enrichment of the microbial population in the lower levels of the plow layer within the moldboard plow system.

Currently, plant residue decomposition models assume a uniformity in the activity of microbial populations with depth and focuses rather on environmental conditions. Since this study has showed that, at least in the top 20 cm of the soil profile, microbial activity is subject to changes depending upon the management practices, the model's assumptions that the extent of potential microbial activity is about the same where the residues are concentrated within the profile seem to be verified.

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APPENDICES

Table A. CO2 evolution from no-till and moldboard plowed soils amended with peanut residue.

Sampling Date	Tillage	Soil depth (cm)	Replicate	Volume HCl (ml)	CO2 evolved (%)
8/28/93	No-Till	0-1 cm	1	25.452	4.607
			2	36.635	8.13
			3	28.71	5.634
			control	10.824	
	No-Till	1-5 cm	1	19.603	3.925
			2	18.963	3.723
			3	14.01	2.163
			control	7.141	
	No-Till	5-12.5 cm	1	26.149	5.185
			2	20.533	3.416
			3	28.919	6.058
			control	9.685	
	No-Till	12.5-20cm	1	21.86	3.965
			2	24.353	4.744
			3	20.845	3.639
			control	9.2909	
	Moldboard Plow	o-l an	1	22.087	5.056
			2	27.51	6.766
			3	2725	6.664
			control	8.028	
	Moldboard Plow	1-5 cm	1	31.179	7.152
			2	37.646	9.189
			3	28.323	6.252
			control	8.473	
	Moldboard Plow	5-12.5 cm	1	22.137	4.067
			2	21.103	3.742
			3	24.361	4.774
			control	9.223	
	Moldboard Plow	12.5-20cm	1	29.74	5.072
			2	30.325	5.256
			3	27.146	4.255
			control	13.638	

Table A. Continued.

Sampling Date	Tillage	Soil depth (cm)	Replicate	Volume HCl (ml)	CO2 evolved (%)
9/01/93	No-Till	0-1 cm	1	111.47	18.437
			2	68.532	18.863
			3	101.6	18.131
			control	9.029	
	No-Till	1-5 cm	1	29.016	6.458
			2	29.383	6.306
			3	32.517	5.169
			control	10252	
	No-Till	5-12.5 un	1	32248	8.368
			2	39.701	7.625
			3	32.639	9.313
			control	8.528	
	No-Till	12.5-20cm	1	29.922	6.697
			2	30.288	7.525
			3	26.12	5.858
			control	9.6649	
	Moldboard Plow	0-1 cm	1	26.631	7.844
			2	16.971	8.248
			3	22.38	8.894
			control	5.993	
	Moldboard Plow	1-5 cm	1	15.539	8.357
			2	23252	11.436
			3	16.974	7.922
			control	6.606	
	Moldboard Plow	5-12.5 cm	1	43.856	8.917
			2	48.578	9228
			3	41.093	9.25
			control	7.935	
	Moldboard Plow	12.5-20cm	1	32.815	8.439
			2	29.983	8.241
			3	26.839	6.816
			control	7.87	

Table A. Continued.

Sampling Date	Tillage	Soildepth (cm)	Replicate	Volume HCI (ml)	CO2 evolved (%)
9/08/93	No-Till	o-1 cm	1	116.3	31.5
			2	130.25	33.81
			3	132.51	33.382
			control	19.534	
	No-Till	1-5 cm	1	36.415	8.743
			2	35.748	8.501
			3	34.35	7.175
			control	19.487	
	No-l-Ill	5-12.5 cm	1	32.303	11.872
			2	34.48	112
			3	26.754	11.848
			control	7.974	
	No-Till	12.5-20cm	1	25.277	8.994
			2	13.442	8.225
			3	28.362	8.572
			control	8.259	
	Moldboard Plow	o-1 cm	1	35.363	11.514
			2	35.983	12.002
			3	37.409	12.841
			control	8.176	
	Moldboard Plow	l-5 cm	1	43.263	11.504
			2	32.929	13.188
			3	38239	10.39
			control	19.956	
	Moldboard Plow	5-12.5 cm	1	101.757	19.464
			2	104.13	20.096
			3	113.12	21.331
			control	23.631	
	Moldboard Plow	12.5-20cm	1	95.9	18.083
			2	98.57	18248
			3	88.987	15.527
			control	24.483	

Table A. Continued.

Sampling Date	Tillage	Soil depth (cm)	Replicate	Volume HCl (ml)	CO2 evolved (%)
9/22/93	No-Till	o-1 cm	1	88.465	36.862
			2	94.14	43.692
			3	92.137	42.994
			control	20.94	
	No-Till	1-5 cm	1	34.915	10.635
			2	39.983	11.077
			3	32.714	8.77
			control	20.902	
	No-Till	S-12.5 cm	1	17.145	12.87
				16.13	12.261
			4	19.003	13.297
			control	a274	
	No-Till	12.5-20cm	1	21.814	10.342
			2	17.51	8.992
			3	22.632	10.057
			control	11.831	
	Moldboard Plow	o-l cm	1	26.005	13.648
			2	30.43	14.733
			3	3429	16.093
			control	10.198	
	Moldboard Plow	1-5 cm	1	17.121	12.674
			2	12.82	13.777
			3	14274	11.175
			control	8.455	
	Moldboard Plow	5-12.5 an	1	87.143	29.692
			2	95.016	31.387
			3	96.121	32.772
			control	11.377	
	Moldboard Plow	12.5-20cm	1	93.507	29.048
			2	86.175	28.218
			3	84.728	25.304
			control	12.303	

Table A. Continued.

Sampling Date	Tillage	Soil depth (cm)	Replicate	Volume HCl (ml)	CO2 evolved (%)
101201'93	No-Till	0-1 cm	1	81.543	48.005
			2	79.698	51.29
			3	85.312	44.854
			control	92705	
	No-Till	1-5 cm	1	67.18	18.621
			2	58.076	17.836
			3	62.668	18.651
			control	8.006	
	No-Till	5-12.5 cm	1	21.183	14.727
			2	14.176	34.871
			3	23.298	15.44
			control	7.42	
	No-Till	12.5-20cm	1	28.31	13.005
			2	22.205	10.831
			3	30.167	11.623
			control	8.584	
	Moldboard Plow	o-l cm	1	13299	9.925
			2	16.321	15.93
			3	20278	17.623
			control	7.456	
	Moldboard Plow	1-5 cm	1	13.626	13.538
			2	16.31	15.002
			3	1525	12.258
			control	7.231	
	Moldboard Plow	5-12.5 cm	1	13.951	30.494
			2	16.449	32.526
			3	18.322	34.104
			control	8.012	
	Moldboard Plow	12.5-20cm	1	59.783	35.891
			2	62.742	35.463
			3	64.044	32.725
			control	9.076	

Table A. Continued.

Sampling Date	Tillage	Soildepth (cm)	Replicate	Volume HCl (ml)	CO2 evolved (%)
11117'193	No-Tilt	o-1 cm	1	118.98	45.678
			2	113.1	51.346
			3	116.41	54.506
			control	10.5261	
	No-Till	1-5 cm	1	136.69	25.361
			2	13329	18.554
			3	124.69	25.385 ,
			control	12.9856	
	No-Till	s-12.5 cfn	1	114.08	21.998
			2	113.74	23.856
			3	118.83	28.489
			control	12.5632	
	No-Till	12.5-20cm	1	10272	24.951
			2	92.911	16.885
			3	105.71	25.304
			control	12.8847	
	Moldboard Plow	o-l cm	1	105.76	1 a.933
			2	127.39	15.884
			3	115.35	17.083
			control	11.4571	
	Moldboard Plow	1-5 cm	1	146.73	18.447
			2	159.03	20.118
			3	14218	24.825
			control	12.0564	
	Moldboard Plow	5-12.5 an	1	155.3	38.4552
			2	14923	40.584
			3	122.83	35.59
			control	10.5238	
	Moldboard Plow	12.5-20cm	1	138.16	34.623
			2	133.10	31.412
			3	145.24	35.959
			control	92351	

Table B. CO2 evolution from soit amended with cotton residues.

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
01/07/94	DLP-5690	Aboveground	1	36.802	8.816
			2	36.147	8.609
			3	39.838	16.072
			control	8.814	
		Root	1	35.465	8.996
			2	37.274	9.585
			3	29.647	7.163
			control	6.906	
	DP-5215	Aboveground	1	42.898	11.081
			2	44.31	11.526
			3	36.44	9.047
			control	7.718	
		Root	1	11.208	1.664
			2	19.712	4.343
			3	13.954	2.529
			control	5.924	
	HS-46	Aboveground	1	30.417	8.04
			2	32.489	8.692
			3	34.084	9.195
			control	4.892	
		Root	1	31.916	7.92
			2	30.111	7.351
			3	32.638	8.147
			control	6.771	
Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
01/11/94	DLP-5690	Aboveground	1	47.774	13.878
			2	42.452	12.199
			3	42.117	12.094
			control	3.722	
		Root	1	22.112	5.649
			2	23.527	6.094
			3	31.598	8.637
			control	4.177	
	DP-5215	Aboveground	1	37213	9.992
			2	33.853	8.934
			3	41.594	11.373
			control	5.489	
		Root	1	14.836	2.75
			2	10.527	3.282
			3	14.19	2.548
			control	6.105	
	HS-46	Aboveground	1	35.986	9.806
			2	41.309	11.483
			3	37.654	10.331
			control	4.854	
		Root	1	43.354	11.657
			2	33.486	8.5487
			3	43.609	11.737
			control	6.347	

Table B. Conlinued

Sampling Date	Cultivar	Residue	Replicale	Volume HCl (ml)	CO2 evolved (%)
01/18/94	OLP-5690	Aboveground	1	45.272	12.382
			2	42.227	II .423
			3	45.224	9.2174
			control	5.862	
		Root	1	28.654	6.411
			2	31275	7.174
			3 .	26.557	5.688
			control	8.499	
	OP-5215	Aboveground	1	36.638	8.675
			2	37.826	9.05
			3	42.816	10.622
			control	9.094	
		Root	1	21.623	3.367
			2	26.762	5.005
			3	32.816	6.912
			control	10.87	
	HS-46	Aboveground	1	34.074	7.961
			2	43.664	11.051
			3	39.345	15.921
			control	6.799	
		Root	1	39.803	8.892
			2	45.515	10.691
			3	38.588	5.359
			control	11.573	
Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
02/01/94	OLP-5690	Aboveground	1	54.596	14.643
			2	39.725	9.959
			3	46.41	12.065
			control	8.106	
		Root	1	50.65	13.097
			2	41.599	10.163
			3	41.159	10.044
			control	927	
	OP-5215	Aboveground	1	28.301	5.88
			2	36.699	8.525
			3	35.295	8.063
			control	9.633	
		Root	1	29.14	5.827
			2	38.976	8.925
			3	33.667	7253
			control	10.641	
	HS-46	Aboveground	1	36.741	4.981
			2	35.502	7.091
			3	46.061	10.417
			control	12.99	
		Root	1	38.138	7.821
			2	39.013	8.098
			3	27.758	4.551
			control	13.309	

Table B. Continued

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
03/01/94	DLP-5690	Aboveground	1	42.405	10.299
			2	37.942	8.893
			3	34.43	7.787
			control	9.709	
		Root	1	43.19	10.173
			2	51.081	12.659
			3	40.299	9.262
			control	10.893	
	DP-5215	Aboveground	1	30.515	6.298
			2	29.354	5.933
			3	30.208	6227
			control	10.519	
		Root	1	30.137	5.897
			2	32.763	6.724
			3	35.492	7.584
			control	11.414	
	HS-46	Aboveground	1	20	1.597
			2	24.378	2.976
			3	29.349	7.691
			control	14.93	
		Root	1	42.395	9.233
			2	32.148	6.006
			3	41.523	8.959
			control	13.081	

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
03/29/94	'DLP-5690	Aboveground	1	20.388	4.455
			2	22.722	5.19
			3	25.546	6.079
			control	6.244	
		Root	1	2828	6.004
			2	34.041	7.819
			3	26.153	5.33
			control	9.218	
	DP-5215	Aboveground	1	21.312	4.023
			2	22.553	4.414
			3	25.714	8.099
			control	8.54	
		Root	1	26.377	4.106
			2	34.433	6.724
			3	28.074	1.571
			control	13.085	
	HS-46	Aboveground	1	23.304	3.802
			2	23.618	3.901
			3	29.596	5.784
			control	11233	
		Root	1	29.388	5.185
			2	35.092	6.981
			3	39.702	8.434
			control	12.927	

Table C. CO2 evofufion from amended with peanut residues.

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
01/07/94	Florunner	Aboveground	1	57.688	16.744
			2	54.927	15.874
			3	63.257	1a.496
			control	4.531	
		Root	1	23.105	6.331
			2	21.229	5.74
			3	19.802	5.291
			control	3.004	
	NC-7	Abovegrd	1	55.344	21.769
			2	53.194	14.791
			3	59.837	16.884
			control	6236	
		Root	1	20.321	4.413
			2	26.458	6.348
			3	23.389	5.379
			control	6.31	
	NC-11	Abovegrd	1	64.357	18.303
			2	60.58	17.113
			3	50.838	14.045
			control	6.25	
		Root	1	20.765	4.0484
			2	18.087	3204
			3	19.426	3.626
			control	7.912	
Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
01/11/94	Florunner	Abovegrd	1	72.47	21.416
			2	72.078	21.292
			3	81.329	24.206
			control	4.482	
		Root	1	19.536	5.283
			2	18.76	5.039
			3	17.021	4.491
			control	2.762	
	NC-7	Abovegrd	1	77.439	22.458
			2	78.409	22.763
			3	69.781	20.046
			control	6.142	
		Root	1	19.818	4.078
			2	12.05	1.631
			3	15.934	2.854
			control	6.871	
	NC-11	Abovegrd	1	78.608	23.048
			2	77.964	22.846
			3	82.152	25.74
			control	5.438	
		Root	1	16.526	3.676
			2	11.794	1.519
			3	13.103	2.597
			control	4.855	

Table C. Continued

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
01/18/94	Florunner	Abovegrd	1	65.556	17.659
			2	69.144	18.789
			3	67.386	15.085
			control	9.494	
		Root	1	23.601	3.521
			2	34.305	6.893
			3	26.134	4.319
			control	12.422	
	NC-7	Abovegrd	1	68.505	14.199
			2	63.581	12.648
			3	70.333	14.775
			control	23.428	
		Root	1	19.943	3.282
			2	13.561	1.272
			3	16.752	2.277
			control	9.522	
	NC-I 1	Abovegrd	1	3529	14.854
			2	37.517	8.94
			3	26.597	5.501
			control	9.133	
		Root	1	13.661	1.602
			2	12.61	1.208
			3	13.551	1.504
			control	8.774	
Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
02/01/94	Florunner	Abovegrd	1	48.081	11.851
			2	57.352	14.771
			3	58.875	8.951
			control	10.457	
		Root	1	18.8	1.497
			2	20.1	1.907
			3	M.867	2.1489
			control	14.045	
	NC-7	Abovegrd	1	36.688	9.285
			2	48.692	12.442
			3	30.816	7.441
			control	7.191	
		Root	1	22.742	4.16
			2	16.893	2.317
			3	19.816	3.239
			control	9.535	
	NC-11	Abovegrd	1	31.055	6.589
			2	36.909	7.122
			3	31.026	15.336
			control	10.137	
		Root	1	14.297	3.072
			2	12.939	2.645
			3	13.618	2.859
			control	4.541	

Table C. Continued

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
03/01/94	Florunner	Abovegtd	1	32.579	6.422
			2	32.468	6.387
			3	35.011	7.188
			control	12.189	
		Root	1	20.668	0.67
			2	29.078	3.319
			3	30.363	3.723
			control	18.541	
	NC-7	Abovegrd	1	34.804	5.342
			2	32.648	4.725
			3	36.702	5.94
			control	17.643	
		Root	1	22.385	3.696
			2	20.284	3.035
			3	21.298	3.354
			control	10.649	
	NC-1 1	Abovegrd	1	38.067	8.797
			2	34.764	7.757
			3	36.099	8.171
			control	10.157	
		Root	1	21.375	2.778
			2	27.712	4.774
			3	24.562	3.782
			control	12.554	

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
03/29/94	Florunner	Abovegrd	1	18263	2.159
			2	10.178	1.495
			3	31.409	6.293
			control	11.429	
		Root	1	17.492	1.343
			2	17.023	1.1954
			3	15.984	0.668
			control	13.228	
	NC-7	Abovegrd	1	23.112	3.853
			2	21.312	3.286
			3	17.329	2.031
			control	10.879	
		Root	1	14.688	1.486
			2	12.55	0.81
			3	13.108	0.989
			control	9.978	
	NC-11	Abovegrd	1	17.325	2.583
			2	23.369	4.487
			3	18.674	3.008
			control	9.123	
		Root	1	22.221	4.162
			2	15.659	2.095
			3	18.948	3.131
			control	9.005	

Table D. CO2 evolution from soil amended with sorghum residues.

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
01/07/94	Triumph-266	Aboveground	1	30.163	7.194
			2	30.702	7.364
			3	39.189	10.037
			control	7.322	
		Root	1	26.159	6.229
			2	28.422	6.942
			3	26.517	6.342
			control	6.362.	
	GW-744BR	Aboveground	1	43.466	11.011
			2	46.74	12.042
			3	47.942	15.571
			control	8.509	
		Root	1	45.661	9.231
			2	44.567	8.817
			3	42.852	a.277
			control	18.575	
	NKing-300	Aboveground	1	37.762	9.962
			2	42.892	11.578
			3	37.676	16.235
			control	6.135	
		Root	1	46.61	12.973
			2	47.412	12.596
			3	52.314	14.1407
			control	7.422	
Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
01/11/94	Triumph-266	Aboveground	1	42.291	11.089
			2	41.432	4.518
			3	43.849	11.58
			control	7.086	
		Root	1	18.632	4.228
			2	19.263	4.427
			3	23.385	5.725
			control	5.208	
	GW-744BR	Aboveground	1	71.661	21.075
			2	70.148	20.536
			3	68.967	10.714
			control	4.953	
		Root	1	23.372	5.703
			2	23.845	12.152
			3	25.322	6.317
			control	5.266	
	NK-300	Aboveground	1	32.136	a.705
			2	37.58	10.42
			3	4Q.52	14.496
			control	4.499	
		Root	1	37.572	9.97
			2	45.316	12.41
			3	41.444	11.19
			control	5.919	

Table D. Continued

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
01/18/94	Triumph-268	Aboveground	1	22.29	4.137
			2	24.511	4.836
			3	22.933	10.63
			control	9.155	
		Root	1	27.526	4.901
			2	30.892	9.112
			3	37.551	8.059
			control	11.965	
	GW-744BR	Aboveground	1	56.014	13.856
			2	45.871	10.661
			3	53.869	19.48
			control	12.025	
		Root	1	59.883	17.142
			2	60.284	17.268
			3	84.588	18.624
			control	5.461	
	NK-300	Aboveground	1	45.046	10.839
			2	42.693	10.097
			3	41.149	a.461
			control	10.637	
		Root	1	78.258	15.478
			2	84.137	23.63
			3	78.099	5.978
			control	9.118	

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
02/01/94	Triumph-M	Aboveground	1	32.78	8.067
			2	36.843	3.046
			3	37.233	9.469
			control	7.1703	
		Root	1	34.93	420Q
			2	46.195	10.907
			3	30.047	5.821
			control	11.567	
	GW-744BR	Aboveground	1	42.528	9.598
			2	39.038	a.499
			3	37.642	14.359
			control	12.0561	
		Root	1	51.488	12.94
			2	45.546	11.076
			3	44.188	10.647
			control	10.386	
	NK-300	Aboveground	1	45.148	10.563
			2	43.64	10.088
			3	42.536	9.741
			control	11.614	
		Root	1	31.526	8.726
			2	32.775	7.119
			3	31.061	3.429
			control	10.173	

Table 0. Conlinued

Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
03/01/94	Triumph-266	Aboveground	1	81.446	10.811
			2	51.582	13.704
			3	57.166	9.169
			control	8.0766	
		Root	1	24.567	2.979
			2	34.39	6.074
			3	35.362	9.5303
			control	15.107	
	GW-744BR	Aboveground	1	31.492	4.947
			2	38.695	10.366
			3	43.645	8.775
			control	15.766	
		Root	1	40.399	10.011
			2	37.0	9.159
			3	33.779	7.955
			control	8.5226	
	NK-300	Aboveground	1	45.086	9.9
			2	45.699	10.1
			3	38.03	7.69
			control	13.612	
		Root	1	30.529	5.694
			2	40.847	8.944
			3	42.851	9.575
			control	12.452	
Sampling Date	Cultivar	Residue	Replicate	Volume HCl (ml)	CO2 evolved (%)
03/29/94	Triumph-265	Aboveground	1	23.184	4.921
			2	29.321	6.854
			3	30.275	7.155
			control	7.5591	
		Root	1	28.514	5.856
			2	33.359	7.382
			3	28.454	5.837
			control	9.9225	
	GW-744BR	Aboveground	1	22.885	1.562
			2	37.475	8.178
			3	32.749	4.69
			control	17.86	
		Root	1	29.653	8.521
			2	26.256	5.451
			3	16.657	2.427957
			control	8.9492	
	NK-300	Aboveground	1	40.508	8.451
			2	25.596	3.754
			3	38.26	13.098
			control	1 3.678	
		Root	1	25.317	3.836
			2	25.528	3.902
			3	32.813	6.197
			control	13.138	

Table E. Mass loss of cotton residue.

Sampling Date	Cultivar	Residue	Replicale	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
01/07/94	OLP-5690	Leaves	1	0.9	0.62	0.17	16.191
		Leaves	2	0.9	0.64	0.17	15.471
		Leaves	3	0.9	0.63	0.17	15.831
		Stems	1	1.1	0.95	0.06	7.947
		Stems	2	1.1	0.99	0.06	6.433
		Stems	3	1.1	0.91	0.06	9.461
		Roots	1	2	1.8	0.44	4.616
		Roots	2	2	1.88	0.44	4.039
		Roots	3	2	1.75	0.44	4.977
	DP-5215	Leaves	1	0.9	0.83	0.16	4.464
		Leaves	2	0.9	0.63	0.16	13.954
		Leaves	3	0.9	0.65	0.16	13.305
		Stems	1	1.1	0.98	0.02	6.137
		Stems	2	1.1	0.95	0.02	7.452
		Stems	3	1.1	0.98	0.02	6.137
		Roots	1	2	1.82	0.1	22
		Roots	2	2	1.91	0.1	1.492
		Roots	3	2	1.85	0.1	1.964
	t-E-46	Leaves	1	0.9	0.74	0.11	10.806
		Leaves	2	0.9	0.65	0.11	14.542
		Leaves	3	0.9	0.6	0.11	16.582
		Stems	1	1.1	0.8	0.16	16.757
		Stems	2	1.1	0.92	0.16	12385
		Stems	3	1.1	0.94	0.16	11.857
		Roots	1	2	1.72	0.1	2.406
		Roots	2	2	1.71	0.1	2.47
		Roots	3	2	1.66	0.1	2.786
Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
01/11/94	OLP-5690	Leaves	1	0.9	0.66	0.17	14.752
		Leaves	2	0.9	0.6	0.17	16.911
		Leaves	3	0.9	0.61	0.17	16.551
		Stems	1	1.1	0.06	0.06	7.568
		Stems	2	1.1	0.93	0.06	8.704
		Slems	3	1.1	0.91	0.06	9.481
		Roots	1	2	1.7	0.44	5.337
		Roots	2	2	1.83	0.44	4.4
		Roots	3	2	1.75	0.44	4.977
	DP-5215	Leaves	1	0.9	0.66	0.16	12.981
		Leaves	2	0.9	0.56	0.16	16.228
		Leaves	3	0.9	0.63	0.16	13.954
		Stems	1	1.1	0.97	0.02	6.575
		Stems	2	1.1	0.98	0.02	6.137
		Stems	3	1.1	0.96	0.02	7.014
		Roots	1	2	1.71	0.1	3.064
		Roots	2	2	1.68	0.1	3.3
		Roots	3	2	1.74	0.1	2.628

Table E. Continued

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Finalweight (g)	Ash (g)	mass loss (%)
	HS-46	Leaves	1	0.9	0.63	0.11	15.35
		Leaves	2	0.9	0.61	0.11	16.158
		Leaves	3	0.9	0.6	0.11	16.582
		Sterns	1	1.1	0.78	0.16	17.485
		Stems	2	1.1	0.84	0.16	15.3
		Stems	3	1.1	0.89	0.16	13.478
		Roots	1	2	1.7	0.1	2.533
		Roots	2	2	1.56	0.1	3.42
		Roots	3	2	1.56	0.1	3.42
Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Finalweight (g)	Ash (g)	mass loss (%)
01/18/94	DLP-5690	Leaves	1	0.9	0.59	0.17	17271
		Leaves	2	0.9	0.63	0.17	15.631
		Leaves	3	0.9	0.58	0.17	1'7.63
		Stems	1	1.1	0.93	0.06	8.704
		Stems	2	1.1	0.89	0.06	10.218
		Stems	3	1.1	0.94	0.08	8.325
		Roots	1	2	1.79	0.44	4.666
		Roots	2	2	1.73	0.44	5.121
		Roots	3	2	1.68	0.44	5.481
	DP-5215	Leaves	1	0.9	0.61	0.16	14.603
		Leaves	2	0.9	0.62	0.16	14279
		Leaves	3	0.9	0.52	0.18	17.524
		Stems	1	1.1	0.92	0.02	8.767
		Stems	2	1.1	0.93	0.02	8.329
		Stems	3	1.1	0.91	0.02	9.206
		Roots	1	2	1.7	0.1	3.142
		Roots	2	2	1.89	0.1	1.85
		Roots	3	2	1.7	0.1	3.142
	HS-46	Leaves	1	0.9	0.58	0.11	17.37
		Leaves	2	0.9	0.6	0.11	16.582
		Laaves	3	0.9	0.5	0.11	20.601
		Stems	1	1.1	0.92	0.16	12.385
		Stems	2	1.1	0.94	0.16	11.657
		Stems	3	1.1	0.95	0.16	11.292
		Roots	1	2	1.53	0.1	3.61
		Roots	2	2	1.47	0.1	3.99
		Roots	3	2	1.6	0.1	3.166
Sampling Date	Cultivar	Residue	Replicate	Initialweight (g)	Finalweight (g)	Ash (g)	mass bss (%)
02/01/94	OLP-5690	Leaves	1	0.9	0.47	0.17	21.588
		Leaves	2	0.9	0.55	0.17	16.71
		Laaves	3	0.9	0.51	0.17	20.149
		Stems	1	1.1	0.67	0.06	18.543
		Stems	2	1.1	0.81	0.08	13.245
		Stems	3	1.1	0.78	0.06	14.381
		Roots	1	2	1.59	0.44	6.131
		Roots	2	2	1.56	0.44	8.347
		Roots	3	2	1.6	0.44	6.059

Table E. Continued

Sampling Date	Cultivar	Residue	Replicate	Initiai weight (g)	Final weight (g)	Ash (g)	mass loss (%)
	DP-5215	Leaves	1	0.9	0.5	0.10	18.173
		Leaves	2	0.9	0.53	0.16	17.2
		Leaves	3	0.9	0.52	0.16	17.524
		Stems	1	1.1	0.91	0.02	9.206
		Stems	2	1.1	0.85	0.02	11.836
		Stems	3	1.1	0.93	0.02	8.329
		Roots	1	2	1.37	0.1	5.735
		Roots	2	2	1.59	0.1	4.007
		Roots	3	2	1.53	0.1	4.478
	HS-46	Leaves	1	0.9	0.51	0.11	20.196
		Leaves	2	0.9	0.53	0.11	19.391
		Leaves	3	0.9	0.46	0.11	22.217
		Stems	1	1.1	0.89	0.10	13.478
		Stems	2	1.1	0.88	0.16	13.642
		Slems	3	1.1	0.89	0.18	13.478
		Roots	1	2	1.39	0.1	4.496
		Roots	2	2	1.43	0.1	4.243
		Roots	3	2	1.37	0.1	4.623
Sampling Date	Cultivar	Residue	Replicate	Initiai weight (g)	Final weight (g)	Ash (g)	mass loss (%)
03/01/94	DLP-5690	Leaves	1	0.9	0.41	0.17	8.203
		Leaves	2	0.9	0.51	0.17	6.98
		Leaves	3	0.9	0.49	0.17	7.209
		Stems	1	1.1	0.65	0.06	5.647
		Slems	2	1.1	0.55	0.06	6.993
		Stems	3	1.1	0.8	0.06	4.127
		Roots	1	2	1.43	0.44	5.505
		Roots	2	2	126	0.44	6.431
		Roots	3	2	1.42	0.44	6.431
							5.559
	DP-5215	Leaves	1	0.9	0.47	0.16	7.402
		Leaves	2	0.9	0.49	0.16	7.151
		Leaves	3	0.9	0.42	0.16	8.03
		Stems	1	1.1	0.59	0.02	6.293
		Stems	2	1.1	0.67	0.02	5.343
		Slems	3	1.1	0.71	0.02	4.866
		Roots	1	2	1.36	0.1	4.56
		Roots	2	2	1.33	0.1	4.676
		Roots	3	2	1.35	0.1	4.75
	HS-46	Leaves	1	0.9	0.53	0.11	6.32
		Leaves	2	0.9	0.49	0.11	6.647
		Leaves	3	0.9	0.48	0.11	6.979
		Stems	1	1.1	0.84	0.18	4.433
		Stems	2	1.1	0.87	0.16	4.116
		Stems	3	1.1	0.81	0.16	4.75
		Roots	1	2	1.32	0.1	4.94
		Roots	2	2	125	0.1	5.363
		Roots	3	2	1.35	0.1	4.75

Table E. Continued

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
03/29/94	DLP-5690	Leaves	1	0.9	0.54	0.17	6.587
		Leaves	2	0.9	0.54	0.17	6.587
		Leaves	3	0.9	0.67	0.17	4.1371
		Stems	1	1.1	0.75	0.06	4.7
		Stems	2	1.1	0.8	0.06	4.127
		Stems	3	1.1	0.74	0.06	4.615
		Roots	1	2	1.62	0.44	4.469
		Roots	2'	2	128	0.44	6.322
		Roots	3	2	128	0.44	6.322
	DP-521 5	Leaves	1	0.9	0.78	0.16	3.513
		Leaves	2	0.9	0.47	0.16	7.402
		Leaves	3	0.9	0.46	0.16	7.528
		Stems	1	1.1	0.46	0.02	7.6
		Stems	2	1.1	0.42	0.02	8.312
		Stems	3	1.1	0.38	0.02	8.787
		Roots	1	2	1.06	0.1	6.586
		Roots	2	2	1.12	0.1	6.206
		Roots	3	2	1.14	0.1	6.08
	HS-46	Leaves	1	0.9	0.5	0.11	6.715
		Leaves	2	0.9	0.64	0.11	4.872
		Leaves	3	0.9	0.66	0.11	4.608
		Stems	1	1.1	0.54	0.16	7.6
		Stetns	2	1.1	0.63	0.16	6.65
		Stems	3	1.1	0.56	0.18	7.388
		Roots	1	2	12	0.1	5.7
		ROMS	2	2	1.14	0.1	6.08
		Roots	3	2	1.73	0.1	2.343

Table F. Mass loss of peanut residues.

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
01/07/94	Florunner	Leaves	1	0.57	0.35	0.1	11.605
		Leaves	2	0.57	0.41	0.1	9.429
		Leaves	3	0.57	0.37	0.1	10.88
		Stems	1	1.43	1.33	0.07	7.876
		Stems	2	1.43	1.35	0.07	6.95
		Stems	3	1.43	1.29	0.07	9.73
		Roots	1	2	1.68	023	1.585
		Roots	2	2	1.6	023	1.751
		Roots	3	2	1.58	023	1.807'
	NC-7	Leaves	1	0.57	0.45	0.09	8.845
		Leaves	2	0.57	0.4	0.09	10.951
		Leaves	3	0.57	0.39	0.09	11.372
		Stems	1	1.43	1.35	0.07	6.75
		Stems	2	1.43	1.33	0.07	7.65
		Stems	3	1.43	1.36	0.07	6.3
		Roots	1	2	1.35	024	1.867
		Roots	2	2	1.29	024	1.993
		Roots	3	2	1.37	024	1.825
	NC-I 1	Leaves	1	0.57	0.37	0.16	14.498
		Leaves	2	0.57	0.41	0.18	12.887
		Leaves	3	0.57	0.47	0.16	10.471
		Stems	1	1.43	1.3	0.04	7.528
		Stems	2	1.43	1.31	0.04	7.085
		Slems	3	1.43	1.28	0.04	8.414
		Roots	1	2	1.21	0.44	2.772
		Roots	2	2	1.19	0.44	2.817
		Roots	3	2	1.22	0.44	2.75
Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
01/11/94	Florunner	Leaves	1	0.57	0.43	0.1	8.704
		Leaves	2	0.57	0.4	0.1	9.792
		Leaves	3	0.57	0.45	0.1	7.979
		Stems	1	1.43	1.38	0.07	5.56
		Stems	2	1.43	1.4	0.07	4.633
		Stems	3	1.43	1.36	0.07	6.487
		Roots	1	2	1.48	023	2.085
		Roots	2	2	1.48	023	2.085
		Roots	3	2	1.5	023	2.029
	NC7	Leaves	1	0.57	0.31	0.09	14.742
		Leaves	2	0.57	0.35	0.09	13.057
		Leaves	3	0.57	0.41	0.09	1023
		Stems	1	1.43	1.3	0.07	9
		Stems	2	1.43	1.29	0.07	9.45
		Stems	3	1.43	1.2	0.07	13.5
		Roots	1	2	122	024	2.14
		Roots	2	2	1.19	0 2 4	2.203
		Roots	3	2	123	024	2.119

Table F. Mass loss of peanut residues.

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
	NC-11	Leaves	1	0.57	0.31	0.16	113.915
		Leaves	2	0.57	0.35	0.16	15.304
		Leaves	3	0.57	0.4	0.16	13.29
		Stems	1	1.43	1.28	0.04	0.414
		Stems	2	1.43	1.25	0.04	9.742
		Stems	3	1.43	1.29	0.04	7.971
		Roots	1	2	1.12	0.44	2.975
		Roots	2	2	1.16	0.44	2.885
		Roots	3	2	1.02	0.44	3.2
Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
01/18/94	Florunner	Leaves	1	0.57	0.33	0.1	12.331
		Leawes	2	0.57	029	0.1	13.782
		Leaves	3	0.57	0.37	0.1	10.88
		Stems	1	1.43	1.35	0.07	6.95
		Stems	2	1.43	1.34	0.07	7.413
		Stems	3	1.43	1.32	0.07	8.34
		Roots	1	2	1.41	0.23	2.279
		Roots	2	2	1.45	023	2.168
		Roots	3	2	1.38	023	2.363
	NC-7	Leaves	1	0.57	0.28	0.09	16.006
		Leaves	2	0.57	0.28	0.09	18.008
		Leaves	3	0.57	0.32	0.09	14.321
		Stems	1	1.43	1.26	0.07	110.8
		Stems	2	1.43	122	0.07	12.6
		Stems	3	1.43	121	0.07	13.05
		Roots	1	2	0.99	0.24	2.622
		Roots	2	2	1.1	0.24	2.391
		Roots	3	2	0.95	0.24	2.708
	NC-11	Leawes	1	0.57	0.36	0.16	14.901
		Leawes	2	0.57	0.34	0.16	15.706
		Leawes	3	0.57	0.33	0.18	16.109
		Stems	1	1.43	126	0.04	9.3
		Stems	2	1.43	1.22	0.04	111.071
		Stems	3	1.43	1.25	0.04	9.742
		Roots	1	2	0.95	0.44	3.358
		Roots	2	2	0.98	0.44	329
		Roots	3	2	1.02	0.44	3.2
Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
02/01/94	Florunner	Leaves	1	0.57	0.42	0.1	9.067
		Leawes	2	0.57	0.48	0.1	6.891
		Leawes	3	0.57	0.47	0.1	7253
		Stems	1	1.43	1.3	0.07	9.268
		Stems	2	1.43	1.31	0.07	a.803
		Stems	3	1.43	1.35	0.07	6.95
		Roots	1	2	123	0.23	2.78
		Roots	2	2	124	023	2.752
		Roots	3	2	12	023	2.884

Table F. Continued

	NC-7	Leaves	1	0.57	0.21	0.09	18.954
		Leaves	2	0.57	0.3	0.09	15.163
		Leaves	3	0.57	029	0.09	15.584
		Stems	1	1.43	1.37	0.07	5.85
		Slems	2	1.43	1.39	0.07	4.95
		Stems	3	1.43	1.37	0.07	5.85
		Roots	1	2	0.85	0.24	2.916
		Roots	2	2	0.96	0.24	2.665
		Roots	3	2	0.91	0.24	2.79
	NC-I 1	Leaves	1	0.57	0.51	0.16	8.66
		Leaves	2	0.57	0.49	0.16	9.665
		Leaves	3	0.57	0.49	0.16	9.665
		Slems	1	1.43	1.37	0.04	4.428
		Stems	2	1.43	1.39	0.04	3.542
		Stems	3	1.43	1.38	0.04	3.985
		Roots	1	2	1.04	0.44	3.155
		Roots	2	2	1.12	0.44	2.975
		Roots	3	2	1.09	0.44	3.043
Sampling Date	Cuttivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
03/01/94	Florunner	Leaves	1	0.57	0.14	0.1	19.222
		Leaves	2	0.57	6.13	0.1	19.565
		Leaves	3	0.57	0.16	0.1	1 a.497
		Stems	1	1.43	1.18	0.07	14.826
		Stems	2	1.43	124	0.07	12.046
		Stems	3	1.43	1.28	0.07	10.193
		Roots	1	2	1.35	0.23	2.556
		Roots	2	2	1.33	023	2.502
		Roots	3	2	1.34	023	2.474
	NC-7	Leaves	1	0.57	0.49	0.09	7.16
		Leaves	2	0.57	0.47	0.09	8.003
		Leaves	3	0.57	0.48	0.09	7.561
		Stems	1	1.43	123	0.07	12.15
		Stems	2	1.43	1.25	0.07	11.25
		Slems	3	1.43	129	0.07	9.45
		Roots	1	2	121	024	2161
		Roots	2	2	1.39	024	1.783
		Rwts	3	2	126	0.24	2.056
	NC-I 1	Leaves	1	0.57	0.52	0.16	8.457
		Leaves	2	0.57	0.51	0.16	8.86
		Leaves	3	0.57	0.53	0.16	8.054
		Stems	1	1.43	1.33	0.04	62
		Slems	2	1.43	1.37	0.04	4.428
		Slems	3	1.43	1.35	0.04	5.314
		Roots	1	2	1.73	0.44	1.6
		Roots	2	2	1.65	0.44	1.78
		Roots	3	2	1.67	0.44	1.735

Table F. Continued

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
03/29/94	Florunner	Leaves	1	0.57	0.15	0.1	4.268
		Leaves	2	0.57	0.26	0.1	3.365
		Leaves	3	0.57	0.24	0.1	3.529
		Slems	1	1.43	0.66	0.07	3.08
		Stems	2	1.43	0.6	0.07	3.3
		Stems	3	1.43	0.59	0.07	3.336
		Roots	1	2	1.43	0.23	'1.973
		Roots	2	2	1.6	0.23	'1.553
		Roots	3	2	1.64	0.23	'1.455
	NC-7	Leaves	1	0.57	0.4	0.09	2.166
		Leaves	2	0.57	0.31	0.09	2.916
		Leaves	3	0.57	0.32	0.09	2.833
		Slems	1	1.43	0.44	0.07	3.886
		Stems	2	1.43	0.55	0.07	3.483
		Slems	3	1.43	0.54	0.07	3.52
		Roots	1	2	0.7	0.24	3.781
		Roots	2	2	0.73	0.24	3.707
		Roots	3	2	0.69	0.24	3.805
	NC-1 1	Leaves	1	0.57	0.3	0.16	3.239
		Leaves	2	0.57	0.21	0.16	3.917
		Leaves	3	0.57	0.39	0.16	2.561
		Stems	1	1.43	0.56	0.04	3.404
		Stems	2	1.43	0.61	0.04	3.217
		Stems	3	1.43	0.65	0.04	3.068
		Roots	1	2	1.56	0.44	1.983
		Roots	2	2	1.08	0.44	3.065
		Roots	3	2	1.37	0.44	2.41

Table G. Mass loss of sorghum residue.

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
01/07/94	Triumph-266	Leaves	1	0.85	0.66	0.2	13.705
		Leaves	2	0.85	0.7	0.2	12.3
		Leaves	3	0.85	0.82	0.2	8.082
		Stems	1	1.15	0.97	0.07	9.118
		Stems	2	1.15	1.05	0.07	6.2
		Stems	3	1.15	0.81	0.07	14.954
		Roots	1	2	1.72	0.34	4.928
		Roots	2	2	1.69	0.34	5.166
		Roots	3	2	1.72	0.34	4.928
	GW-744BR	Leaves	1	0.85	0.57	0.11	13.467
		Leaves	2	0.85	0.52	0.11	15.216
		Leaves	3	0.85	0.46	0.11	17.291
		Stems	1	1.15	0.98	0.1	12.18
		Stems	2	1.15	1.02	0.1	9.68
		Stems	3	1.15	0.97	0.1	11.76
		Roots	1	2	1.49	0.3	5.036
		Roots	2	2	1.07	0.3	3.916
		Roots	3	2	1.64	0.3	4.103
	NKing-300	Leaves	1	0.85	0.8	0.14	8.047
		Leaves	2	0.85	0.74	0.14	9.141
		Leaves	3	0.85	0.74	0.14	9.141
		Stems	1	1.15	1.05	0.05	5.862
		Stems	2	1.15	1.04	0.05	6.253
		Sterns	3	1.15	1.01	0.05	7.425
		Roots	1	2	1.7	0.2	3.84
		Roots	2	2	1.71	02	3.764
		Roots	3	2	1.5	02	5.377

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
01/11/94	Triumph-266	Leaves	1	0.85	0.6	02	15.814
		Leaves	2	0.85	0.61	02	15.462
		Leaves	3	0.85	0.6	02	15.814
		Stems	1	1.15	1.08	0.07	5.146
		Stems	2	1.15	0.0	0.07	11.672
		Stems	3	1.15	0.91	0.07	11.307
		Roots	1	2	1.54	0.34	8.358
		Roots	2	2	1.61	0.34	5.802
		Roots	3	2	1.71	0.34	5.007
	GW-744BR	Leaves	1	0.85	0.51	0.11	15.562
		Leaves	2	0.85	0.43	0.11	18.329
		Leaves	3	0.85	0.45	0.11	17.637
		Stems	1	1.15	0.77	0.1	20.16
		Stems	2	1.15	0.88	0.1	15.51
		Stems	3	1.15	0.72	0.1	22.26
		Roots	1	2	1.53	0.3	4.787
		Roots	2	2	1.03	0.3	4.165
		Roots	3	2	1.65	0.3	4.041

Table G. Continued

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
	NKing-300	Leaves	1	0.85	0.68	0.14	i1.335
		Leaves	2	0.85	0.6	0.14	14.26
		Leaves	3	0.85	0.6	0.14	14.26
		Stems	1	1.15	1.04	0.05	6.253
		Stems	2	1.15	0.86	0.05	'13.253
		Stems	3	1.15	1	0.05	7.816
		Roots	1	2	1.47	02	5.607
		Rools	2	2	1.49	0.2	5.454
		Roots	3	2	1.56	02	4.916
Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
01/1 MU	Triumph-266	Leaves	1	0.85	0.62	0.2	15.111
		Leaves	2	0.85	0.58	0.2	16.517
		Leaves	3	0.85	0.51	02	18.977
		Stems	1	1.15	0.84	0.07	13.86
		Slems	2	1.15	0.69	0.07	19.331
		Stems	3	1.15	0.83	0.07	14.225
		Roots	1	2	1.41	0.34	7.392
		Roots	2	2	1.65	0.34	5.484
		Roots	3	2	1.53	0.34	6.438
	GW-744BR	Leaves	1	0.85	0.44	0.11	17.983
		Leaves	2	0.85	0.42	0.11	18.875
		Leaves	3	0.85	0.35	0.11	21.095
		Stems	1	1.15	0.8	0.1	18.9
		Stems	a	1.15	0.74	0.1	21.42
		Stems	3	1.15	0.64	0.1	25.62
		Roots	1	2	1.56	0.3	4.478
		Roots	2	2	1.65	0.3	4.041
		Roots	3	2	1.46	0.3	5.222
	NKing-300	Leaves	1	0.85	0.53	0.14	16.82
		Leaves	2	0.85	0.73	0.14	9.507
		Leaves	3	0.85	0.55	0.14	16.068
		Stems	1	1.15	0.37	0.05	8.989
		Stems	2	1.15	0.83	0.05	14.48
		Stems	3	1.15	0.88	0.05	12.506
		Roots	1	2	1.53	02	5.140
		Roots	a	2	1.44	02	5.638
		Roots	3	2	1.43	02	5.915
Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Finalweight (g)	Ash (g)	mass loss (%)
02/01/94	Triumph-266	Leaves	1	0.85	0.44	02	21.437
		Leaves	2	0.85	0.44	02	21.437
		Leaves	3	0.85	0.41	02	22.491
		Stems	1	1.15	0.74	0.07	17.508
		Stems	2	1.15	0.58	0.07	23.344
		Stems	3	1.15	0.74	0.07	17.508
		Roots	1	2	1.34	0.34	7.948
		Roots	2	2	1.4	0.34	7.471
		Roots	3	[illegible]	1.55	0.34	6279

Table G. Continued

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
	GW-744BR	Leaves	1	0.85	0.32	0.11	22.133
		Leaves	2	0.85	0.3	0.11	22.825
		Leaves	3	0.85	0.3	0.11	22.825
		Stems	1	1.15	0.6	0.1	27.3
		Stems	2	1.15	0.63	0.1	26.04
		Stems	3	1.15	0.72	0.1	2226
		Roots	1	2	1.3	0.3	6.217
		Roots	2	2	1.26	0.3	6.499
		Roots	3	2	1.31	0.3	6.155
	NKing-300	Leaves	1	0.85	0.37	0.14	22.67
		Leaves	2	0.85	0.4	0.14	21.573
		Leaves	3	0.85	0.52	0.14	17.185
		Slems	1	1.15	0.81	0.05	15242
		Stems	2	1.15	0.68	0.05	20.323
		Stems	3	1.15	0.81	0.05	15.242
		Roots	1	2	1.71	0.2	3.764
		Roots	2	2	1.43	0.2	5.915
		Roots	3	2	1.21	0.2	7.605
03/01/94	Triumph-266	Leaves	1	0.85	0.48	0.2	9.174
		Leaves	2	0.85	0.61	0.2	7.081
		Leaves	3	0.85	0.48	02	9.174
		Stems	1	1.15	0.69	0.07	7.351
		Stems	2	1.15	0.84	0.07	8.034
		Stems	3	1.15	0.75	0.07	6.51
		Roots	1	2	1.33	0.34	7.204
		Roots	2	2	1.1	0.34	8.955
		Roots	3	2	1.25	0.34	7.872
	GW-744BR	Leaves	1	0.85	0.44	0.11	9.154
		Leaves	2	0.85	0.5	0.11	8.097
		Leaves	3	0.85	0.88	0.11	5.281
		Stems	1	1.15	0.56	0.1	9.328
		Stems	2	1.15	0.41	0.1	11.356
		Stems	3	1.15	0.47	0.1	10.545
		Roots	1	2	1.06	0.3	9.111
		Roots	2	2	1.08	0.3	8.984
		Roots	3	2	1.25	0.3	7.715
	NKing-300	Leaves	1	0.85	0.68	0.14	5.633
		Leaves	2	0.85	0.56	0.14	7.34
		Leaves	3	0.85	0.57	0.14	7.109
		Slems	1	1.15	0.58	0.05	8.731
		Stems	2	1.15	0.68	0.05	7.605
		Stems	3	1.15	0.58	0.05	8.731
		Roots	1	2	127	0.2	7.144
		Roots	2	2	1.22	0.2	7.528
		Roots	3	2	0.95	02	9.602

Table G. Continued

Sampling Date	Cultivar	Residue	Replicate	Initial weight (g)	Final weight (g)	Ash (g)	mass loss (%)
03/29/94	Triumph-266	Leaves	1	0.85	0.54	0.2	8.208
		Leaves	2	0.85	0.54	0.2	8.028
		Leaves	3	0.85	0.67	0.2	6.118
		Stems	1	1.15	0.75	0.07	6.51
		Stems	2	1.15	0.8	0.07	5.810
		Stems	3	1.15	0.74	0.07	6.649
		Roots	1	2	1.62	0.34	52
		Roots	2	2	1.26	0.34	7.655
		Roots	3	2	128	0.34	7.655
	GW-744BR	Leaves	1	0.85	0.78	0.11	3.166
		Leaves	2	0.85	0.47	0.11	8.626
		Leaves	3	0.85	0.46	0.11	8.802
		Stems	1	1.15	0.46	0.1	10.41
		Stems	2	1.15	0.42	0.1	11221
		Stems	3	1.15	0.38	0.1	11.762
		Roots	1	2	1.06	0.3	9.111
		Roots	2	2	1.12	0.3	8.07
		Roots	3	2	1.14	0.3	8.523
	NKing-300	Leaves	1	0.85	0.5	0.14	8.364
		Leaves	2	0.85	0.84	0.14	5.974
		Leaves	3	0.85	0.86	0.14	5.633
		Stetns	1	1.15	0.54	0.05	9295
		Stems	2	1.15	0.83	0.05	8.027
		Stems	3	1.15	0.56	0.05	9.013
		Roots	1	2	1.2	02	7.681
		Roots	2	2	1.14	02	8.142
		Roots	3	2	1.73	02	3.61

Table H. Chage in specific surface area of cotton residue.

Sampling Oak	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
01/07/94	OLP-5690	Leaves	1	1783.964
		Leaves	2	1688.235
		Leaves	3	1723.844
		Stems	1	1031.305
		Stems	2	1035.622
		Sterns	3	940.264
	OP-5215	Leaves	1	1812.851
		Leaves	2	1796.842
		Leaves	3	1842.36s
		Stems	1	853.434
		Stems	2	938.412
		Stems	3	852.915
	HS-46	Leaves	1	1771.404
		Leaves	2	1695.231
		Leaves	3	1668.254
		Stems	1	775.698
		Sterns	2	814.905
		Sterns	3	689.361
Sampling Date	Cuttivar	Residue	Replicate	Specific surface Area (mm^2)
01/11/94	OLP-5690	Leaves	1	1669.529
		Leaves	2	1685.623
		Leaves	3	1704.653
		Stems	1	914.833
		Stems	2	912304
		Slems	3	898.732
	OP-5215	Leaves	1	1653,623
		Leaves	2	1689.874
		Leaves	3	1656.231
		Sterns	1	826.172
		Stems	2	850.168
		Stems	3	812.426
	HS-46	Leaves	1	1599.632
		Leaves	2	1687231
		Leaves	3	1653.966
		Stems	1	794.396
		Stems	2	731.05
		Stems	3	742.116
Sampling Date	Cuttivar	Residue	Replicate	Specific surface Area (mm^2)
01/18/94	OLP-6690	Leaves	1	1564.326
		Leaves	2	1661258
		Leaves	3	1612258
		Stems	1	930.92
		Stems	2	802.536
		Stems	3	759.599
	OP-5215	Leaves	1	1563.256
		Leaves	2	1602.365
		Leaves	3	1699.532

Table H. Continued.

		Stems	1	850.764
		Stems	2	756.851
		Stems	3	730.421
	HS-46	Leaves	1	1498.632
		Leaves	2	1562.358
		Ceaves	3	1586.652
		Stems	1	786.572
		Stems	2	702.305
		Stems	3	728.235
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
02/01/94	OLP-5690	Leaves	1	1532.698
		Leaves	2	1524.832
		Leaves	3	1499.362
		Stems	1	871.182
		Stems	2	805.519
		Stems	3	825.423
	DP-521 5	Leaves	1	1542.632
		Leaves	2	1488.632
		Leaves	3	1586.362
		Stems	1	719.324
		Stems	2	798.262
		Stems	3	711.258
	HS-46	Leaves	1	1423.632
		Leaves	2	1399.865
		Leaves	3	1402.362
		Stems	1	752.282
		Stems	2	663.487
		Stern-s	3	701.589
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
03/01/94	OLP-5690	Leaves	1	1399.851
		Leaves	2	1465.654
		Leaves	3	1423.656
		Stems	1	720.97
		Stems	2	772.329
		Stems	3	805.654
	DP-521 5	Leaves	1	1265.632
		Leaves	2	1356.987
		Leaves	3	1363.52
		Stems	1	775.231
		Stems	2	683.739
		Stems	3	702.532
	HS-46	Leaves	1	1399.12
		Leaves	2	1289.365
		Leaves	3	1352.654
		Stems	1	666.539
		Stems	2	688.379
		Stems	3	674.235

Table H. Continued.

Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
03/29/94	OLP-5690	Leaves	1	1285.657
		Leaves	2	1301.562
		Leaves	3	1289.365
		Stems	1	688.201
		Stems	2	650.816
		Stems	3	683.338
	DP-5215	Leaves	1	1288.741
		Leaves	2	1286.365
		Leaves	3	1198.562
		Stems	1	657.293
		Stems	2	728.534
		Slems	3	709.445
	HS-46	Leaves	1	1285.632
		Leaves	2	1186.235
		Leaves	3	1254.238
		Stems	1	629.381
		Stems	2	640.825
		Stems	3	659.024

Table I. Change in specific surface area of peanut residue.

Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
01/07/94	Florunner	Leaves	1	2250.229
		Leaves	2	2250.942
		Leaves	3	2394.624
		Stems	1	1500.112
		Slems	2	1500.628
		Stems	3	1596.416
	NC-7	Leaves	1	1603.049
		Leaves	2	1590.483
		Leaves	3	1481.607
		Stems	1	1068.699
		Slems	2	1060.322
		Stems	3	987.738
	NC-l 1	Leaves	1	2094.639
		Leaves	2	2161.695
		Leaves	3	2116.055
		Stems	1	1396.426
		Slems	2	1441.13
		Slems	3	1410.704
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
01/11/94	Florunner	Leaves	1	1691.45
		Leaves	2	1633.52
		Leaves	3	1887.548
		Stems	1	1127.833
		Stems	2	1089.013
		Stems	3	1258.365
	NC-7	Leaves	1	1501.869
		Leaves	2	1500.912
		Leaves	3	1481.343
		Slems	1	1001248
		Stems	2	1000.608
		Stems	3	987.562
	NC-11	Leaves	1	2024.577
		Leaves	2	2133.938
		Leaves	3	2035.256
		Stems	1	1349.718
		Stems	2	1422.825
		Stems	3	1356.837
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
01/18/94	Florunner	Leaves	1	1432.442
		Leaves	2	1507.52
		Leaves	3	2110.135
		Stems	1	954.981
		Stems	2	1005.013
		Stems	3	1406.757
	NC-7	Leaves	1	1450.69
		Leaves	2	1343.436
		Leaves	3	1428.367

Table 1. *Continued.*

Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
		Stems	1	967.120
		Stems	2	895.623
		Stems	3	952.258
	NC-11	Leaves	1	1929.202
		Leaves	2	1489.548
		Leaves	3	1538.488
		Stems	1	1286.135
		Stems	2	993.03 1
		Stems	3	1025.659
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
02/01/94	Florunner	Leaves	1	1297.958
		Leaves	2	1368.958
		Leaves	3	1414.867
		Stems	1	865,305
		Stems	2	912.6384
		Stems	3	943.256
	NC7	leaves	1	1405.664
		Leaves	2	1369.728
		Leaves	3	1405.668
		Stems	1	937.122
		Stems	2	926.465
		Stems	3	937.125
	NC-1 1	Leaves	1	1629.843
		Leaves	2	1478.431
		Leaves	3	1501.684
		Stems	1	1066.562
		Stems	2	985.621
		Stems	3	1001.123
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
03/01/94	Florunner	Leaves	1	1297.957
		Leaves	2	1368.957
		Leaves	3	1414.867
		Stems	1	665.305
		Stems	2	912.638
		Stems	3	943.258
	NC-7	Leaves	1	1377.768
		Leaves	2	1349.848
		Leaves	3	1216.284
		Stems	1	918.511
		Stems	2	899.898
		Stems	3	810.856
	NC-11	Leaves	1	1624.731
		Leaves	2	1479.354
		Leaves	3	1494.387
		Stems	1	1063.154
		Stems	2	986.236
		Stems	3	996.256

Table 1. Continued.

Sampling ;Date	Cultivar	Rasidue	Replicate	Specific surface Area (mm^2)
0329194	Florunner	Leaves	1	1254.329
		Leaves	2	1287.852
		Leaves	3	1297.987
		Stems	1	838.219
		Stems	2	845.235
		Stems	3	B65.125
	NC-7	Leaves	1	1254.32
		Leaves	2	1281.192
		L.eaves	3	1216.537
		Stems	1	836.213
		Stems	2	854.128
		Stems	3	811.025
	NC-11	Leaves	1	1591.717
		Leaves	2	1437.391
		Leaves	3	1494.048
		Stems	1	1061.145
		Stems	2	958.261
		Stems	3	996.031

Table J. Change in specific surface area of sorghum residue.

Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
01107194	Triumph-266	Leaves	1	2371.251
		Leaves	2	1294.83
		Leaves	3	1306.202
		Stems	1	1580.634
		Stems	2	836.219
		Stems	3	870.801
	GW-7448R	Leaves	1	1628.68
		Leaves	2	1859.15
		Leaves	3	1377.775
		Stems	1	1085.787
		Stems	2	1239.433
		Stems	3	918.516
	NKing-300	Leaves	1	1921.103
		Leaves	2	1487.885
		Leaves	3	2274.091
		Stems	1	1280.735
		Slems	2	991.923
		Stems	3	1516.061
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
01/11/94	Triumph-266	Leaves	1	1565.75
		Leavw	2	1657.125
		Leaves	3	1581.609
		Stems	1	1057.167
		Stems	2	1104.75
		Slems	3	1054.539
	GW-7448R	Leaves	1	1766.847
		Leaves	2	1641.172
		Leaves	3	1343.953
		Stems	1	1177.898
		Slems	2	1094.115
		Stems	3	895.988
	NKing-300	Leaves	1	1512.66
		Leaves	2	1431.586
		Leaves	3	1487.132
		Slems	1	1008.454
		Slems	2	954.39
		Slems	3	965.421
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
01/18/94	Triumph-266	Leaves	1	1720.478
		Leaves	2	1494.911
		Leaves	3	139201
		Slems	1	1146.985
		Stems	2	996.607
		Stems	3	928.006
	GW-7448R	Leaves	1	1393.95
		Leaves	2	1645.371
		Leaves	3	1371.543

Table J. Contlnued.

Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
		Stems	1	929.3
		Stems	2	1096.914
		Stems	3	914.362
	NKing-300	Leaves	1	1703.716
		Leaves	2	1360.117
		Leaves	3	1290.498
		Stems	1	1135.81
		Stems	2	900.745
		Stems	3	860.332
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
02/01/94	Triumph-266	Leaves	1	1914.823
		Leaves	2	1340.288
		Leaves	3	1307.354
		Slems	1	1278.548
		Stems	2	893.525
		Stems	3	871.569
	GW-744BR	Leaves	1	1399.205
		Leaves	2	1229.792
		Leaves	3	1300.349
		Stems	1	932.803
		Stems	2	819.861
		Stems	3	866.899
	NKing-300	Leaves	1	1664.187
		Leaves	2	1342.443
		Leaves	3	1342.872
		Stems	1	1109.658
		Stems	2	894.962
		Stems	3	895247
Sampling Date	Cultivar	Residue	Replicate	Specific surface Area (mm^2)
03/01/94	Triumph-266	Leaves	1	1231.095
		Leaves	2	1892.18
		Leaves	3	1430.478
		Stems	1	820.73
		Stems	2	1261.453
		Stems	3	953.851
	GW-744BR	Leaves	1	133.533
		Leaves	2	1319.422
		Leaves	3	1208.443
		Stems	1	887.021
		Stems	2	879.632
		Slems	3	805.632
	NKing-300	Leaves	1	1203.354
		Leaves	2	1293.157
		Leaves	3	1366.153
		Stems	1	802235
		Stems	2	862.104
		Stems	3	910.788

Table J. Continued.

Sampling Date	Cuttivar	Residue	Replicate	Specific surface Area (mm^2)
03/29/94	Triumph-266	Leaves	1	1093.314
		Leaves	2	1 507.983
		Leaves	3	1288.835
		Stems	1	728.876
		Stems	2	1005.322
		Stems	3	a59223
	GW-744BR	Leaves	1	1250.348
		Leaves	2	1243.08
		Leaves	3	1231.973
		Stems	1	833.565
		Stems	2	828.719
		Stems	3	821.315
	NKing-300	Leaves	1	1302.674
		Leaves	2	1254286
		Leaves	3	1261.079
		Stems	1	868.449
		Stems	2	838.19
		Stems	3	840.719

Table K. ANOVA for CO2 evolution from no-till and plowed soils amended with peanut residue.

SOURCE	DF	SS	MS	F	Significant
Soil	1	9.060368	9.060368	0.05674475	
Depth	3	1106.07699	368.69233	2.30910625	
- linear	1	26.849355	20.8493..	0.1681565	
- quadratic	1	7.392884	7.392864	0.04630136	
- cubic	1	1071.83475	1071.834751	6.7128609	*
Soil*Dpth	3	4676.36659	1558.788529	9.76263418	**
- linear	1	2472.33803	2472.338032	15.4841605	***
- quadratic	1	1628.7657	1628.765698	10.2008986	***
- cubic	1	575.261857	575.261857	3.60264340	
Error (a)	16	2554.70163	159.6688456		
Time	S	11358.725	2271.744999	72.4864128	***
- linear	1	9058.0113	9058.011295	289.021323	***
- quadratic	1	1907.3793	1907.379302	60.8603006	***
- cubic	1	366.842741	368.842741	11.7689649	**
- quartic	1	17.187227	17.187227	0.54640681	
- quintic	1	7.304431	7.304431	0.23306841	
Soil*Time	S	106.522397	21.3044794	0.67977933	
- linear	1	89.455435	89.455435	2.85432722	
- quadratic	1	15.602814	15.602814	0.49785166	
- cubic	1	0.041369	0.041369	0.00131999	
- quartic	1	0.093946	0.093946	0.00299761	
- quintic	1	1.328833	1.326833	0.04240015	
- linear*linear	1	1.226565	1.226565	0.03913701	
- linear*quadratic	1	4.909641	4.909641	0.1566559	
- linear*cubic	1	286.681271	286.681271	9.15375372	**
- quadratic*linear	1	0.076256	0.076258	0.00243316	
-quadratic*quadratic	1	2.556083	2.556083	0.08155902	
-quadratic*cubic	1	144.013663	144.013833	4.5951673	*
- cubic*linear	1	5.070099	5.070099	0.16177577	
- cubic*quadratic	1	1.866062	1.866062	0.05954196	
- cubic*cubic	1	54.063451	54.063451	1.72504644	
- quartic*linear	1	18.318810	18.318618	0.5845134	
- quartic*quadratic	1	0.42737	0.42737	0.01383644	
- quartic*cubic	1	2.226306	2.226306	0.07103655	
- quintic*linear	1	1.416799	1.416799	0.04520698	
- quintic*quadratic	1	0.233665	0.233885	0.00745638	
- quintic*cubic	1	3.00459	3.00459	0.09588989	

Table K. Continued.

SOURCE	DF	SS	MS	F	Significant
S*D*T	15	1680.73547	112.0490311	3.57523944	**
linear*linear*linear	1	889.677402	869.677402	27.7495032	***
linear*linear*quadrat	1	361.994844	361.994844	11.5504635	***
linear*linear*cubic	1	15.976778	15.976778	0.50978403	
linear*quadratic*line	1	159.618111	159.618111	5.09306471	*
lineafquadratic'qua	1	136.314224	136.314224	4.34948866	*
linear*quadratic*cubi	1	26.63316	26.63316	0.84980587	
linear*cubic*linear	1	8.394755	8.394755	0.26785827	
lineaf cubic*quadrati	1	48.573996	48.573996	1.54988994	
linear*cubic*cubic	1	27.897104	27.897604	0.89013556	
linear*quartic*linear	1	9.84662	9.64662	0.30780254	
linear*quartic*quadra	1	2.809419	2.809419	0.08964241	
linear*quartic*cubic	1	5.47294	5.47294	0.17462954	
linear*quintic*linear	1	7.347722	7.347722	0.23444973	
lineaf quintic*quadra	1	0.212978	0.212978	0.00679588	
linear*quintic*cubic	1	0.165413	0.165413	0.00527797	
Error (b)	80	2507.22298	31.34028725		
Total	143	24525.7012			
PE (a+b)					