UNIVERSITY OF GHANA, LEGON

COLLEGE OF AGRICULTURE & CONSUMER SCIENCE

DEPARTMENT OF SOIL SCIENCE

THE EFFECT OF PHOSPHORUS SOURCES ON NODULATION AND DRY MATTER YIELD OF PIGEONPEA (*Cajanus cajan*) (L) Millip).

BY

HARRIS YANQUOI

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A DISSERTATION SUBMITTED TO THE DEPARTMENT OF SOIL SCIENCE, COLLEGE OF AGRICULTURE & CONSUMER SCIENCE, UNIVERSITY OF GHANA, LEGON, IN PARTIAL FULFILLMENT OF THE REQUIREMENT FOR THE AWARD OF MASTER OF AGRICULTURE (M. AGRIC.) DEGREE IN SOIL SCIENCE

MARCH, 2014

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MARCH, 2014

DECLARATION

I do hereby declare that this work is the result of my own original research and that, except for references to other peoples work which have been duly cited, this work has neither in whole nor in part been presented for another degree elsewhere.

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March, 2014

ACKNOWLEDGEMENTS

I first and foremost thank the Lord Mighty for his guidance and mercy on my life throughout my study. I wish to express my sincere and profound gratitude to my supervisor Prof. Mark K. Abekoe for his help and understanding throughout the entire period of the dissertation. My deepest gratitude also goes to all the Lecturers of the College especially the Lecturers in the Soil Science Department who in one way or the other contributed immensely to the success of this dissertation. I appreciate all the efforts, suggestions and criticisms which have made this dissertation a successful one.

My final appreciation also goes to the Laboratory Technicians and all my friends who helped and encouraged me in one way or the other. All I have for you is may the Almighty God keep increasing your knowledge. THANK YOU.

HARRIS YANQUOI

DEDICATION

I hereby dedicate this dissertation to my mother, Larwu Gono, my wife Joyce M Yanquoi and all my family members especially my youngest brother J.B Yanquoi who have toiled to see me through my studies.

ABSTRACT

Phosphorus (P) is an essential element which has been identified as a major plant nutrient that limits nitrogen (N_2) fixation in legumes. Different sources of water soluble P have been used to increase growth, nodulation and N_2 fixation in legumes , but there is inadequate information on the effect of phosphate rocks (PRs) on the growth, nodulation, and N_2 fixation of pigeonpea. In this study, Togo rock phosphate (TRP), and water soluble triple superphosphate (TSP) were used to test the effect of P application on dry matter yield and nodulation of pigeonpea. The objectives of the study were to (i) compare Togo rock phosphate with Triple super phosphate with respect to: dry matter yield and nodulation of pigeonpea using two soil series (Toje and Bumbi), (ii) assess the rate of phosphorus application for maximum dry matter yield and nodulation in pigeonpea and (iii) compare the agronomic effectiveness of the two P sources in a green house experiment. The study was carried out in pots with phosphorus application rates of 0, 30, 60, 90, 120, and 150 kg P/ha and pigeonpea was used as a test crop.

The plants were grown and harvested 6 weeks after planting. The shoots were analyzed for dry matter yield and phosphorus uptake and root nodule numbers were counted and weighed. The relative agronomic effectiveness (RAE) of the Togo rock phosphate was calculated using the dry matter yield and P uptake at 120 kg P/ha application rate.

The results of the pot experiment showed that the dry matter yield (DMY) of the pigeonpea crop increased consistently from 0-P kg/ha to 120 kg P/ha and decreased at 150 kg P/ha in both soils. The DMY, nodule numbers and weight in the Toje soil series were significantly higher than in the Bumbi soil despite the relatively higher fertility status of the Bumbi soil series ($p < 0.05$). This suggests that soil fertility is not the only parameter to be considered for growth and development of crops. For soil productivity, both the chemical and the physical properties are equally important since they both contribute to the root development and exploration for nutrients and water in the soil. Dry matter yield,

nodule numbers and weight of the pigeonpea obtained in the TSP treatments were significantly higher than those of the Togo rock phosphate treatments. The agronomic effectiveness of the TRP in the two soils showed that in the Toje soil series, the RAE was 27 % and in the Bumbi soil series, it was 18 % that of the TSP. This could be attributed to the fact that the Togo rock phosphate did not dissolve adequately well to supply the needed P to the pigeonpea during the six weeks growing period.

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CHAPTER ONE

INTRODUCTION

Phosphorus (P) is an essential element for nitrogen fixation and also for the yield of legumes. Like all plants, legumes need nitrogen for their growth and development. Nitrogen and phosphorus are limiting in tropical soils, but fortunately nitrogen-fixing legumes are able to grow well in nitrogen deficient soils since they are capable of fixing their own nitrogen. Phosphorus is needed in N fixation since the element forms part of ATP required for energy transfer in the fixation process. Biological nitrogen fixation (BNF) may be the best way by which N supply to plants can be increased by resource poor farmers. It is estimated that 65% of N input in the Global Agriculture comes from BNF (Giller, 2001). In addition, it is a sound agronomic practice which ensures sustainable use of the environment. Dakora and Mupfte (1996) indicated that a large variety of legumes in Africa has the ability to fix between 151 and 581 kg N ha^{-1}. Nitrogen fixation by using leguminous plants like pigeonpea represents the only means by which net N may increase *in situ*. This process has virtually no economic cost and releases the nitrogen at constant slow rate, which permits its immediate incorporation into the plants system.

Pigeonpea is an important grain crop in the tropics including Liberia. In Liberia the major legumes grown are pigeonpea, cowpea, and groundnut. Apart from the main use as food for man and forage for animals, it is used as green manure. It has nitrogen fixation ability and provides several benefits to the soil in which it grows (Langar and Hill, 1991).

Even though P is needed for nodulation and N fixation in legumes, it is not certain how much P is required for the optimum growth and nodulation in pigeonpea crop. Many farmers do not apply water soluble TSP because of its high cost. The use of rock phosphates as a cheaper source of P has been the subject of much research in many crops

including legumes. Therefore, the use of Togo rock phosphate which is readily available at Wienco, a Fertilizer Company in Ghana, was examined in this study.

Nodulation and yield of pigeonpea may differ in different soil types and for this matter two soils with very contrasting properties but very much cultivated in the coastal savanna zone of Ghana were used in the investigation. Similar soils exist in Liberia and the results of this study can be extrapolated to Liberia where pigeonpea is grown by local farmers.

Therefore, the objectives of the study were to:

1. Compare the effects of two P sources (rock phosphate and water soluble triple super phosphate) on nodulation and dry matter yield of pigeonpea in two coastal savanna soils in Ghana.
2. Assess the critical P rate for dry matter yield nodulation of pigeonpea in these soils.
3. Compare the agronomic effectiveness of the two P sources in a greenhouse experiment.

CHAPTER TWO

LITERATURE REVIEW

2.1: Introduction

This chapter presents a review of studies conducted on the effect of phosphorus on nodulation, dry matter yield and nitrogen fixing ability of pigeonpea. It also includes factors affecting nitrogen fixation, phosphorus status in some West Africa soils, nature of phosphate rock and factors affecting phosphorus levels in soils.

2.2: Pigeonpea plant

Pigeonpea is a diploid belonging to the Cajaninae sub-tribe of the phaseoleae, which also includes soybean, field bean and mungbean (Young et al., 2003). It is the only known cultivated food crop of the 32 species that falls under the Cajaninae sub- tribe. The crop represents about 5% of the world legume production (Hillocks et al., 2000) with more than 70% being produced in India. There is also substantial pigeonpea production in Eastern Africa and Americas. Global annual production of pigeonpea is about 3.6 million tons (Mt) valued at around US$ 1,600 million (FAOSTAT, 2007).

Pigeonpea is a perennial pulse crop that is growing widely throughout the tropic and subtropical regions. Based on its unique characteristic, it plays a major role in the farming system adopted by many smallholder farmers in a large number of developing countries (Nene et al., 1990). It is deep rooted and drought tolerant plant (Troedson et al., 1998). Leguminous food crops are used in several countries, particularly in India as a source of dietary protein (Nene et al., 1990). Pigeonpea is one of the principal dryland crops whose production ranks very low in terms of area cultivated relative to other grain legumes.

The dried roots, leaves, and seeds are used in different countries to treat a wide range of ailments of the skin, liver, lungs and kidney (Nene et al., 1990).

Pigeonpea, like many other legumes, is used to improve the fodder situation and increase the nitrogen content of soils through the Nitrogen fixed. In some West African countries mature seeds are dried and used in making local dishes. The crop is commonly used in rotation in the interior savanna zones of Ghana and Liberia to regenerate soil fertility.

2.2.1: Origin and distribution

The true origin of pigeonpea is still disputable. However, the crop was most likely introduced into East Africa from India by immigrants who moved to Africa in the 19th century to become railway workers and storekeepers (Hillocks et al., 2000). It thereafter moved up the Nile Valley into West Africa and eventually to the Americas. Pigeonpea is increasingly becoming an important subsistence crop in the whole of Africa with production reported in more than 33 countries (Johansen et al., 1993). Due to the subsistence nature of the crop, production area and figures from Africa are gross underestimates (Shanower et al., 1999). The production of pigeonpea in Africa contributes to 9.3% of the world production, which is very little compared to the 74% contribution from India alone. Production trends seem to be increasing with increasing quantity of rainfall in the region. However, the increase in production is largely a result of area expansion rather than increase in yields (Jones et al., 2002). The average yield was reported by FAO to be 718 ± 171 kg/ha and the maximum recorded yield was (1087 kg/ha) (FAOSTAT 2007).

2.2.2: Seeding

When sown under optimal moisture and temperature conditions (29^{0}C–36^{0}C), the seed testa splits open near the micropyle on the 2nd day. The tip of the radical elongates and

emerges from the seed coat. On the 3rd day the hypocotyl appears as an arch and continues to grow upward and the hypocotyl turns light purple. The seeding epicotyl elongates 3 to 7 cm before the first trifoliate leaf emerges (Reddy, 1979).

2.2.3: Pigeonpea as a soil amendment

Pigeonpea (*Cajanus cajan* (L) Millip) is a member of the family Fabaceae and is one of the major legumes of the tropic and subtropics. Pigeonpea has several characteristics that make it valuable as either a production or rotation crop. Some of the benefits of incorporating pigeonpea into the cropping system include its role as soil ameliorant, ability to fix nitrogen, extract phosphorus, and high drought tolerance. The vertical and lateral spread of its root system, enable the crop to have the capacity to break plough pans, thus improving soil structure (Nene and Sheila, 1990). Pigeonpea is more efficient in utilizing iron-bound phosphorus (Fe-P) than several other crop species such as maize, sorghum, soybean, groundnut, and chickpea (Ae et al., 1990). The unique ability of the pigeonpea to utilize Fe-P has been attributed to the solubilizing activity of the root exudates, the deep root system of the crop which allows for optimum moisture and nutrient utilization. Its extensive ground cover reduces soil erosion by wind and water, encourages infiltration, minimizes sedimentation and smothers weeds.

2.2.4: Seed and its germination

The color of the seed ranges from silver, white, cream, fawn, black, pink, or red to purple. They are blotched or speckled. Pigeonpea 100–seed mass ranges from 2.8 to 22.4 g with the cultivated varieties ranging from 7.0 to 9.5 g. Seed shapes are oval, pea–shaped, square, or elongate. The most common is a pea–shaped seed found in large seeded late varieties. The number of seed pods ranges from 2 to 8. The cultivated genotypes possess 3

– 4 seeds per pod. The cotyledons are yellow. Germination is hypogeal and there is no known dormancy. The seedlings emerge from depths of up to 5 cm.

2.2.5: Nodules

Pigeonpea is nodulated by the cowpea group of rhizobia, mainly on the upper 30 cm of the root system. Nodulation starts approximately 15 days after sowing (DAS) and continues up to 120 days. It declines towards pod filling (Kumar Rao et al., 1981). The nodule development is through the meristematic zone, arching around the apical end and the medulla contains many bacteriod–filled cells. The nodules differ in size from 2–4 mm. They may be spherical, oval, elongate, or branched. Nodule formation and development are affected by the soil type, the season, and the duration of the cultivar. Nodulation in pigeonpea is rapid, with about 25 nodules per plant formed in Alfisol within 15 days after sowing (DAS), and about half of these nodules were formed on the primary root. Nodulse formed on the primary root usually have a short life span (<60 days) and continue to form up to 120 DAS on plants grown on both Alfisols and Vertisols (Thompson and Tioeh., 1978). There are many factors such as soil moisture, temperature, soil pH and nutrient supply that may limit the symbiosis under field conditions.

2.3: Soil moisture

Sufficient moisture is required for both normal growth and development of the legume host and endurance of rhizobia. However, rhizobia tend to survive better in dry soils which contain considerable amounts of clay and organic matter**.** The legume rhizobium symbiosis is sensitive to water logging, but can recover when exposed to short-stress periods while prolonged exposure may lead to permanent damage and shedding of nodules (Kumar Rao et al., 1987). Pigeonpea experiences both water deficit (drought) and excess

water (water logging) depending on the season (rainy or dry), intensity and distribution of rainfall. In some soils there is sufficient moisture to meet pigeonpea's requirement, and sometimes drainage is necessary to enhance its growth. Depending on the variety, medium-duration pigeonpea genotypes suffer more from drought stress than long duration genotypes with extensive root system (Nene and Sheila, 1990).

Biological nitrogen fixation is influenced by the availability of moisture in the soil, which varies with texture of the soil, the management practice of the farmer and the kind of vegetation on the soil. Dry and hot conditions and low soil moisture content reduce the growth of the plant and the rate of biological nitrogen fixation (Hungria and Vargas, 2000). Standley et al. (1985) indicated that most agricultural legumes cannot fix N in droughty conditions possibly due to reduction in the growth rate and structure of the rhizobia resulting in increased nodulation failure and while longevity of nodules in the soil is reduced.

Drought conditions also reduce nodule cortical permeability resulting in a decrease in diffusion of oxygen to the N_2-fixing bacteria, reduction in their respiration and also the activity of nitrogenase. The synthesis of leghaemoglobin is reduced and very severe stress from water deficits can lead to the cessation of N_2-fixation altogether (Guerinot, 1991). The transport of nitrogenous compounds from the nodules is also affected by lack of water. This leads to the accumulation of ammonia and other end products of fixation which cause the cessation of N_2-fixing in the nodule (Hungria and Vargas, 2000). Therefore, the process of BNF requires extra amounts of water and that low levels of soil moisture reduce N_2-fixation. Deep-rooted leguminous species such as pigeonpea should therefore be used for BNF where moisture stress conditions are prevalent (Lie, 1981). Symbiotic activity is affected by water deficit to a greater extent than plant dry matter accumulation. The former continued to increase up to 40% soil moisture, whereas dry

matter yields increase at 30-35% soil moisture. Thus, N_2-fixation activity is severely impaired when the soil moisture level falls below field capacity.

2.3.1: Soil temperature

Temperature is one of the major factors considered as imperative factors affecting production and efficiency of nodules. Temperature affects the various biochemical processes including N_2-fixation. In parts of the tropics, the surface soil temperatures can occasionally reach 65-70^{0}C and temperatures above 50^{0}C can be found at 5 cm depth sufficiently high to inhibit germination of seeds and kill many bacteria (Dudeja and Kharana, 1989). Giller and Wilson (1991) reported that many species of cyanobacteria can form spores which are highly resistant to desiccation but most heterotrophic N_2 fixers and rhizobia do not possess this capability. The optimum temperatures for growth and N_2 fixation vary widely between legume species and reflect their environmental adaptation.

Survival of bacteria in soils at high temperature appears to be improved by the presence of clay particles and soil organic matter, but many of the soils where high temperatures are experienced are sandy. Clearly the potential for soil temperature to influence the rate of BNF may be considered. It is worth noting that the optimum temperature ranges for N_2-fixation have been found to be greatly reduced when plants are starved of mineral N (Hungria and Vargas, 2000). The fact that high temperatures may decrease rhizobial survival and establishment in the hot tropical soils means that the benefit of BNF technologies may be greatly reduced in the absence of repeated inoculation or high rate of inoculation of legumes (Hungria and Vargas, 2000).

2.3.2: Soil fertility

Low soil fertility reduces the amount of BNF as plant growth cannot exceed the rate imposed by the most limiting nutrients (exception for legumes of limiting soil N). Danso et al. (1992) and Giller (2001) have suggested that for all soils, the lack of success of rhizobia inoculation may be largely due to soil nutrient deficiencies. The legume-Rhizobium symbiosis imposes additional nutritional requirements for growth. Among nutritional factors that improve performance of legume plants, P fertilization is important. This requires that fertilizer P is either taken up by the crop or efficiently converted into biomass or that it is kept in the soil in a potentially plant available form for a subsequent crop.

2.3.3: Soil pH

The inhibitory effect of low soil pH in most soils of the tropics is a problem for N_2-fixation (Hungria and Vargas, 2000). Several tropical soils are highly weathered and contain predominantly low-activity clay minerals. In addition, they may be highly leached, acidic and infertile and frequently contain toxic concentrations of exchangeable aluminum and manganese (Hungria and Vargas, 2000). Pigeonpea is predominantly grown in neutral to alkaline soils of India, and elsewhere it can be grown in acid soils, in the pH range 4.5 to 5.5 (Dalal and Quilt, 1977; Edwards, 1981; Abruna et al., 1984) but not below pH 4 (Chong et al., 1987). Acid soil conditions are problematic for leguminous plants, N_2-fixing bacteria and the symbiotic process itself (Giller et al., 1998; Danso and Alexandar, 1974). The optimal pH conditions for effective rhizobial growth is between pH 6.0 and 7.0 (Jordan, 1984), and relatively few rhizobia grow well at pH of less than 5.0 (Hungria and Vargas, 2000).

Aluminum decreases the initiation of nodules and hence affects the growth of legumes or functioning of nodules (Dakora and Phillips, 2002). Aluminum toxicity as a result of low soil pH reduces rhizobia survival (Danso and Alexander, 1974). It is possible that essential cytoplasmic process may be extremely sensitive to soil acidity (Hungria and Vargas, 2000). Acidity may also reduce the early stages of the infection process, including the exchange of molecular signals between symbiotic partners and attachment to roots (Hungria and Vargas 2000). Other stages of nodule establishment and function are also affected by acidity (Graham, 1981). Low soil pH often occurs concurrently with increased Al toxicity and reduced calcium (Ca) supply, and these factors can affect the growth of rhizobia.

Soil acidity influences nitrogen fixation by direct and indirect effects on the bacteria and host. It denatures proteins; deactives nitrogenase enzyme and causes aluminum toxicity, thereby reducing nitrogen fixation. A few crop species can tolerate salinity and alkalinity, but not excessive acidity (i.e., pH below 5.0). Depending on the cultivar, pigeonpea as a legume can grow and fix N_2 in acid soils (Nene and Sheila, 1990). However, its N_2-fixation activity seems more sensitive to acidity than plant growth itself (Edwards, 1981). A study to compare the effect of phosphorus on nodulation, and dry matter yield of pigeonpea in a moderately acid and near neutral soil would provide additional information.

2.4: Mineral elements apart from nitrogen and phosphorus

Other elements, apart from N and P such as calcium (Ca), molybdenum (Mo), cobalt (Co), iron (Fe), boron (B) and aluminum (Al) also play important roles in nodulation and hence, rhizobium functioning. Molybdenum is an important element, as it is a constituent of the enzyme nitrogenase and nitrate reductase. Khurana and Dudeja (1981) reported that

application of 0.45 kg Mo ha^{-1} as sodium molybdate significantly increased nodulation, plant dry matter and grain yield of pigeonpea at Hisar in northern India. Seeds dressed with cobalt at a rate of 500 mg^{-1} cobalt nitrate kg^{-1} significantly increased grain yield of pigeonpea, and that this can be interpreted as priming effect of Co in improving nitrogen fixation (Raj, 1987). Cobalt is used in the cobamide electron transport pathways of rhizobia, and thus essential for nitrogen fixation in legumes (Riley and Dilworth, 1985). In South Africa, boron deficiencies were also responsible for reducing BNF in bambara groundnut (Dakora et al., 1997). Boron has been reported to be essential nutrient element for the legume host symbiotic partners but not the rhizobia (Andrew. 1977). Its deficiency in soil inhibits nodulation by legumes.

The importance of calcium to influence rhizobia to infect legumes has been well documented (O' Hara and Dilworth, 1988; O' Hara, 2001). Its functional role is not certain but it has been shown to be of importance in the initial attachment of rhizobial cells to root hair tips (Giller, 2001). Studies by Hernandez and Focht (1985) in Panama, however, indicated that addition of calcium did not increase N_2-fixation. Aluminium tends to accumulate on the roots and impede the uptake and translocation of calcium and phosphorus to the tops (Foy, 1974). Aluminium causes root injury in acid soils and hence reduces plant growth. Aluminium toxicity may thus, cause or accentuate phosphorus deficiency and hence nodulation.

2.4.1: Nutrient deficiency

Most of the nutrients essential for the growth of plants or bacteria play important roles in the nodulation and or N_2 fixation. Failure of these nutrients, or other precise elements for the growth of bacteria or plants, can cause reduction in the quantity and size of nodules formed and the amount of N_2 fixed.

2.5: Nitrogen

There are large differences among species in their ability to fix nitrogen in N-rich soils (Becker et al., 1990). For example, N contribution to rice in the lowland ranges from 90 to about 200 kg/ha (George et al., 1998). Nitrogen fertilization adversely affects the nodulation of many legumes (Franco, 1977). In pigeonpea both nodulation and nitrogenase activity were depressed by soil nitrogen concentration greater than 25 mg N kg^{-1} as NO_3. Quilt and D'alal (1979) found negligible nodulation in plants up to 10 weeks old in soils with 50 mg N kg^{-1}, whereas normal nodule formation occurred at soil – N concentrations of around 20 mg N kg^{-1}. Applications of nitrogen fertilizers at sowing reduced nodule mass per plant by 74% at 20 days after sowing (DAS), but by 60 (DAS) no differences were apparent (Kumar Rao et al., 1981). It is worth noting that the addition of excessive quantities of N to the soil can reduce nodulation and BNF due to the buildup of nitrate in the soil. However, Dakora et al. (1997) pointed out that application of nitrogen fertilizers to 10 groundnut cultivars in Ghana increased nodulation and plant growth by 612% and 453% over a control plot in the savannah zone. Kernel yield was increased by 60% and N_2- fixation by 65% over the control. This finding contradicts the popular knowledge that high nitrogen content of soils reduces N_2-fixation.

2.5.1: Biological nitrogen fixation (BNF)

Since nitrogen is commonly the most limiting plant nutrient in rural farming in the tropics and also the most expensive element as a mineral fertilizer, biological nitrogen fixation (BNF) is probably the second most important process on earth after photosynthesis (Brady, 1990). Biological nitrogen fixation holds great promise for small holder farm in Sub – Saharan Africa. Alley farming systems which use leguminous woody

species as hedges can reduce or eliminate farmers' needs for commercial N fertilizer. Biological nitrogen fixation is the process of capturing atmospheric nitrogen by biological process. It is accomplished by certain microorganism and plant microbe interactions. Legumes are N_2 fixing systems that have long been used for BNF in agriculture. This system is important in the tropics by substituting for fertilizer inputs (Boddey et al., 1995). Biological nitrogen fixation technologies in general, use the symbiotic relationship between higher plants and certain bacteria to reduce the depletion of soil N or to increase the input of soil N. There are several associations involved in this process. Symbiotic fixation may occur in association with legumes. In this case, the bacteria involved are Rhizobium spp. These invade the root hairs and the cortical cells, causing the formation of root nodules. The nodules provide habitat for the rhizobia and energy from carbohydrate for the plant. The bacteria in turn supply the plant with fixed N compounds (Brady, 1990).

2.5.2: System of biological nitrogen fixation

The formation of nitrogen fixation is the reduction of atmospheric nitrogen gas to a biologically valuable N form. Nitrogen gas is very stable and therefore, the reaction is actively costly. It is credibly for this reason that nitrogen fixation ability of legumes is not collective (Giller, 2001). According to Giller (2001), the equation for this effect is as follows:

$$N{=}N + 8H^{+} + 16ATP \longrightarrow 2NH_3 + H_2 + 16DP + 16P_i \text{---------- [1]}$$

Ammonia in turn is combined with organic acids to form amino acids and ultimately protein as follows:

$$NH_3 + \text{organic Acids} \longrightarrow \text{Amino Acids} \longrightarrow \text{Proteins -------- [2]}$$

Nitrogen gas undergoes reduction by an enzyme, nitrogenase, a two protein complex consisting of a larger ion, molybdenum containing member, and a smaller companion containing iron. Nitrogenase is indeed of great significance to mankind (Brady, 1990)

2.5.3: Nitrogen fixation by pigeonpea

In pigeonpea, nodules are produced by a wide spectrum of rhizobia strains belonging to the "cowpea group" (Allen and Allen, 1981). On area basis, pigeonpea is the fifth most important pulse crop in the world, yet work on biological nitrogen fixation in this crop has been relatively limited. Kumar Rao et al. (1987) estimated that the amount of N_2 fixed by pigeonpea genotypes of different maturity groups ranged from 6 to 69 kg N ha^{-1}. It has been reported in Kenya that, under the similar conditions *Cajanus cajan*, C. grahamiana and T. vogelii grew and fixed more N than Sesbania (Gathumbi et al., 2001, Giller, 2001). The N input from the legume fallow to the cropping system may come from both N_2 fixation and capture of N from depth of soil.

2.6: Phosphorus

Nearly all research conducted to investigate the effect of P application on biomass production and nitrogen accumulation by legumes showed significant positive response to these two parameters. These experiments substantiated the importance of phosphorus as a key element in fuelling nitrogen fixation (Cassman et al., 1993). Despite the general response, Tian et al. (1998) and Ankomah et al. (1996) observed differential P responses among legume crop species. The addition of P stimulated pigeonpea nodulation in both Alfisol and Vertisols, while farmyard manure inhibited pigeonpea nodulation (Kumar Rao et al. 1981). Hermandez and Focht (1985) reported that addition of P to infertile acid Oxisol increased shoot and nodule masses.

2.6.1: Phosphorus status in West African soils

Soils of tropical Africa are so heavily weathered and extremely variable that it is unwise to generalize. Fixation of applied P is high in some tropical soils especially those derived from volcanic ash. However, the volcanic soils are of rather minor occurrence in the tropics. Soils of tropical Africa have been formed from Pre-Cambrian parent materials that have been reworked by processes of erosion and deposition (Ahenkorah et al., 1993). Because parent materials in tropical soils are not uniformly distributed and the climate is diverse, inherent fertility of the soils varies considerably. Because of intensive leaching and the degree of weathering, many soils of tropical Africa have relatively low inherent fertility.

After nitrogen, phosphorus is generally the most limiting nutrient element in tropical Africa. In many cases where soils had adequate soil organic matter and nitrogen levels, phosphorus was found to be the most limiting nutrient. Although the total amount of phosphorus in tropical soils is variable, highly weathered soils generally have low total phosphorus (Sanchez et al., 1985). In addition, soils in the tropics sorb phosphorus from the soil solution and thus render it less available for plant uptake. The magnitude of phosphorus fixation is generally related to the soil pH, the iron and aluminium oxides in the soil and exchangeable aluminium content. Based on the crop response to applied P, Sahrawat et al. (1998) concluded that majority of the soils in the humid and sub-humid zones of West Africa were acutely deficient in available P. On acid soils, the problem is more severe as the applied P is converted to unavailable forms due to reactions with iron and aluminum oxides (Mokwunye et al., 1986; Owusu Bennoah et al., 1997; Abekoe and Sahrawat, 2001). Deficiency of phosphate occurs widely in the savannah soils which are high in sesquioxides (Mokwunye et al., 1986).

In some soils of the savannah zone of West Africa, deficiency is so acute that plant growth ceases as soon as the phosphorus stored in the seed is exhausted (Wild and Jones, 1975). Acquaye and Oteng (1972) reported a mean total inorganic P content of 26 savannah soils as 81 mg P kg^{-1}, 93 mg P kg^{-1} for 8 forest/savannah intergrade soils and 124 mg P kg^{-1} for 14 forest soils. Owusu-Bennoah and Acquaye (1989) also reported that the total P values for topsoil of four modal forest soils ranged from 270 to 530 mg P kg^{-1} soil, whereas organic P values ranged from 54 to 243 mg P kg^{-1} soil. Since many tropical soils are inherently low in total phosphorus, shifting cultivation does not really solve phosphorus problems in traditional agricultural systems. In some cases phosphorus deficiencies were observed immediately after clearing (Chien and Menon, 1995). Although banding may be satisfactory in soils with moderate fixation capacities, in high fixing soils requiring high rates of phosphorus fertilizers the band application can limit root development and subject the crop to serious moisture stress (Chien et al., 1987).

2.6.2: Phosphorus status in Ghanaian soils

The total phosphorus of Ghanaian soils is low compared with values reported for soils in most parts of the world. This is because of the age of the parent materials, the rock from which the soil have been derived. Parts of the total P of Ghanaian soils are usually in the organic form, which becomes an important source of P supply to plants only after mineralization (Acquaye and Oteng, 1972). Forest soils in Ghana have more phosphorus than savanna soils in spite of experiencing a more intense weathering. There is a higher percent organic P in forest soils than the savanna soils as a result of higher organic matter from the forest cover. There is an enrichment of the surface soils by the upward translocation through the roots. The forest vegetation is more efficient in returning the leached nutrients from the sub-soil to the surface through the litter than the savanna

vegetation. The total organic and inorganic P content of Ghanaian soils under different vegetative cover follow the order: Forest> Forest- Savanna Intergraded > Savanna (Acquaye and Oteng, 1972). Lack of adequate P not only limits the response of other major nutrients, but also affects the overall fertility and productivity of the soil as well as biological nitrogen fixation in legumes (Becker et al., 1990; Sahrawat et al., 2000). Application of P is essential even for a moderate crop yield. A combination of P efficient crop and P fertilization practices may provide a good strategy for sustainable crop production in soils where P availability is a major constraint. Some findings have demonstrated a varietal difference in P efficiency, which have been reported for rice (Sahrawat et al. 1998; 2000), cowpea (Ankomah et al., 1996), legume cover crops (Vanlauwe et al., 2000) and pigeonpea (Ae et al., 1990).

2.6.3: Phosphate rocks and their chemical nature

Among the inorganic P sources, phosphate rock indigenous to the West African sub region is important. Large deposits of phosphate rock exist in several West African countries (Buresh et al., 1997). The occurrence of this rock in the region has provided a potential outlet and a way to reduce the over dependence on water soluble superphosphates. A series of trials to evaluate the agronomic effectiveness of local phosphate rock sources have been carried out in the sub region (Bationo et al., 1986).

For agronomists and the fertilizer industry, the phosphate rock materials of interest are the complex assembly of materials grouped under a generic name apatite. The phosphate rock can be igneous, sedimentary or metamorphic in origin. Most of the phosphate rock of West African origin is sedimentary in nature. Carbonate apatite or francolite is the most common constituents of sedimentary phosphate rock (Ahn, 1974).

Among the West African countries, Togo (Hahotoe) and Senegal (Taiba and Thies) are significant producers of phosphate rock on the market. In addition, Burkina Faso (Kodjara), Mali (Tilemsi), and Niger (Tahoua) have been exploiting phosphate rock in finely ground form for local use. Many other West African countries like Benin, Cameroon, Ghana, Guinea Bissau, Liberia, Mauritania and Nigeria have phosphate rock deposits that have not been exploited (Johnson, 1995). The P concentration in this phosphate rock ranges from 10-17% P and their agronomic potential is judged by the molar PO_4/CO_3 ratio. Phosphate rocks that have PO_4/CO_3 ratio less than 5 are considered very reactive (Lutz, 1971). Despite a relatively low reactivity, the use of indigenous phosphate rocks appears attractive in terms of their lower cost and the high capital investment for the production of P sources. The use of phosphate rocks as a source of P for crops in different soils and growing environments has been documented (Hammond et al., 1986; Bolan et al., 1990; Buresh et al., 1997). Another important consideration in the use of phosphate rocks is that they have a liming potential due to the release of calcium, which they supply to the soil (Hellums, 1989). The use of phosphate rocks as source of P to crops especially in acid soils appears attractive as the soils have potential acidity to solubilize the phosphate rocks. In some soils such as Ultisols and Oxisols, the use of soluble P is effective in only a short season, but the residual effect is drastically increased, that is, its availability increases with time (Linguist et al., 1997; Sahrawat et al., 1998). The solubility of phosphate rocks may be low initially in acid soils, but the solubility increases with time (Mokwunye, 1995; Hu et al., 1997; Adediran et al., 1998).

2.6.4: Triple superphosphate (TSP)

The main fertilizer constituent of TSP is monocalcium phosphate monohydrate (MCP), formed by the reaction of orthophosphoric with fluorapatite in the rock.

Single superphosphate generally contains 8.7% available P (20 % P_2O_5); whilst triple superphosphate, more concentrated by virtue of the absence of $CaSO_4$ usually contains 20% available P (46% P_2O_5). Owing to the greater water–soluble P content of the superphosphates, they are usually more agronomically effective than acidulated or raw rock phosphates. Both types of phosphate are used in Ghana for direct application but single superphosphate (SSP) is preferred due to the sulphur content. In this study, triple superphosphate was used in the experiment as reference water – soluble P source because it contains negligible amount of sulphur.

Triple superphosphate is known to be highly soluble among other phosphates. The response of crops to added P is common in many soils of West Africa. Greater responses to P applications have been reported for crops grown in the humid regions or zones (Linguist et al., 1997). The rate of application depends on P requirements of the crops and the sorption capacity of the soil (Bationo et al., 1986; Abekoe and Tiessen, 1998). Several studies on P requirement of crops using a soluble P source such as TSP suggest that crops may respond to a rate as low as 10 kg P per hectare. For example, Jama et al. (1998) reported that the application of 10 kg P per hectare as TSP on Alfisol in western Kenya had significant residual effect on the maize crop in the following season. In the study, application of between 10 and 30 kg P per hectare were highly beneficial to the maize crop.

2.6.5: Togo Rock phosphate

Togo rock phosphate is a sedimentary rock deposit. It has about 15 to 17% total P, 59 % CaO, with a molar PO_4^{3-}/CO_3^{2-} ratio of 12.3, 1.5 % Fe_2O_3, I:1 Al_3O_2 , and has a low reactivity in soils (Johnson, 1995). In an experiment to compare the reactivity of TRP and other elements, it was shown that TRP performed very poorly as compared to single super phosphate irrespective of agro- ecological zone (Vanlauwe, 2000). Mokwunye et al. (1980) also confirmed the findings that finely ground untreated Togo rock phosphate gave lower yields than triple super phosphate.

CHAPTER THREE

MATERIALS AND METHODS

3.1: Site characteristics and physiography

The two soils used for this study were the Toje series and Bumbi series. Both soils are located in the Accra Plains.

3.1.1: Toje series

Toje series was obtained at the University of Ghana farm, Legon, near the Legon Botanical Gardens. The site has geographical co-ordinates of 5° 39 N, 0° 539 W in the Greater Accra Region of Ghana, a gentle topography of 0.30%, an annual rainfall between 700-1000 mm and a mean temperature of 26.9°C. The soil type is savanna ochrosol. Toje series has been classified by Eze, (2008) as a Rhodustalf and Rhodic Lixisol according to USDA (1999, 2003) and ISSS-ISRIC-FAO (WRB) (1998) respectively. Toje series is among the most widely cultivated soils of the Accra Plains. Toje is developed on Quartzite schist (Fiagbedzi, 1989).

3.1.2: Bumbi series

The Bumbi series was obtained from Ashaiman on latitude $05^0$41.400, and longitude $00^0$03.018. The land has a bottom slope of 0 to 1%. The Bumbi soil has been classified by Eze, (2008) as Ustic Duraquert under the USDA Soil Taxonomy System. The soil texture is silty clay loam. The area has an annual rainfall between 700-1000 mm and a mean temperature of 26.9°C.

3.1.3: Sampling

A surface soil sample (0-20 cm) was obtained, air dried, crushed and passed through a 2 mm sieve to remove twigs, plant root and ironstone concretions. The soils were bagged for analysis.

3.2: Determination of soil pH

The pH of each soil was measured electrochemically in distilled water at a ratio of 1:1 soil; water, using a Hanna H19017 microprocessor pH meter standardized with aqueous solutions of pH 4 and pH 7. Twenty grams of sieved soil were weighed into a 50 ml beaker and 20 ml distilled water was added to form a suspension. The suspension was then stirred for about 30 min. This was then allowed to stand for about 1 hour, to permit settlement of the entire suspended particles. The pH of the soil was measured after carefully and gently immersing the glass electrodes into the supernatant. The procedure was repeated for the measurement of pH in 0.01 M $CaCl_2$ solution (soil: $CaCl_2$) ratio of 1:2).

3.3: Organic carbon determination

Wet combustion method of Walkley and Black (1934) was used to determine the organic carbon content of the soils. Ten milliliters of 1.0 N potassium dichromate (K_2CrO_7) solution and 20 mL of concentrated sulphuric acid (H_2SO_4) were added to 0.5 g of the soil sample in a conical flask and the content was swirled and the digestion was allowed to proceed for 2 hours. The unreduced $K_2Cr_2O_7$ remaining in the solution after the oxidation of the oxidizable organic material in the soil samples was titrated against 0.2 N Ferrous ammonium sulphate solution after adding 200 mL of distilled water, 10 mL of orthophosphoric acid and 2 mL of barium diphenylamine sulphonate indicator from dirty

brown color to a bright green end point. The percent organic carbon was calculated as follows:

% Carbon = [0.3 (10 – XN) / W]…….. [1]

Where X = ml of ammonium ferrous sulphate used in the titration

N = normality of ammonium ferrous sulphate solution

W = weight of soil sample (g)

3.4: Cation exchange capacity (CEC)

3.4.1: Extraction of Exchangeable bases

Ten grammes of each of the soils were weighed into 200 ml extraction bottles and 100 ml of 1 0 N neutral ammonium acetate (NH_4OAc, pH 7.0) solution added. The suspension was shaken for 1 hour and filtered with Whatman No. 42 filter paper. Suitable aliquots of the extracts were used for the determination of exchangeable cations.

3.4.2: Exchangeable calcium (Ca^{2+}) plus magnesium (Mg^{2+})

10 mL aliquots of the extract were taken into a conical flask and 10 mL of 10% KOH and 1 mL of methylamine were added. Three drops of KCN solution and a few crystals of Cal-red indicator were added. The mixture was then titrated with 0.2 N EDTA using Eriochrome Black T (EBT) as an indicator.

3.4.3: Exchangeable (Ca^{2+}).

Ten ml of the extracts were transferred into a conical flask and titrated with EDTA using Cal-red as an indicator. Exchangeable Mg^{2+} was estimated by difference.

3.4.4: Exchangeable K^+ and Na^+

Exchangeable potassium (K^+) and sodium (Na^+) were determined by flame photometry.

3.5: Exchangeable acidity

Ten grams of the soil were transferred into a dry filter paper in a funnel placed on top of a 100 mL volumetric flask. The soil was successively leached with 10 mL batches of 1.0 N KCl to a total volume of 100 mL. Twenty-five mL aliquots were taken and four drops of phenolphthalein indicator were added. The solution was titrated with 0.02 M of NaOH to the first permanent pink endpoint. A correction for the blank of NaOH titre on 100 mL of KCl solution was made and the KCl extractable acidity (extractable Al^{3+} and H^{+}) were calculated as follow:

$$\text{Meq KCl} = \frac{(\text{ml NaOH sample} - \text{ml NaOH blank}) \times \text{N} \times 100)}{\text{Weight of sample (g)}} \quad \ldots\ldots\ldots\ldots \quad [2]$$

Where N = Normality of NaOH

The effective cation exchange capacity (ECEC) was obtained by summation of exchangeable bases (Ca^{2+}, Mg^{2+}, K^{+} and Na^{+}) and exchangeable acidity (Al^{3+} and H^{+}).

3.6: Total nitrogen determination

The Kjeldahl method was used in the total nitrogen determination. Two grams of soil were weighed into 300 mL Kjeldahl flask and a tablet of a digestion accelerator, selenium catalyst was added. This was followed by addition of 5 ml of concentrated H_2SO_4. The mixtures were digested until the digest became clear. It was cooled and transferred into a 100 mL volumetric flask and topped-up with distilled water to the 100 mL mark. An aliquot of 5 mL was taken into Markham distillation apparatus and 10 mL of 40% NaOH were added. The solution was distilled and the distillate was collected in 5 mL of 2% boric acid (H_3BO_3) to which three drops of methyl red and methylene blue were added and then titrated with 0.01M HCl from green to reddish endpoint. The percentage N was calculated as follows:

$$\% N = \frac{0.01 \text{ x titre x volume of extract x } 0.014\text{x}100}{\text{Weight of sample (g) x volume of aliquot (ml)}} \text{ x } 100 \ldots\ldots\ldots \quad [3]$$

3.7: Total P in the fertilizer materials

The total P in each fertilizer material was determined by weighing 0.1 g of the sample into a clean micro Kjeldahl flask. Five mL of concentrated HCl were added to the sample and warmed gently to dissolve. The flask was allowed to cool. Distilled water was added and the sample shaken after which it was filtered through a Whatman filter paper into 250 mL volumetric flask. The filtrate was diluted to 500 mL and a 2 mL aliquot was used to determine the total P.

3.7.1: Available phosphorus determination

Available P was determined using the Bray 1 method. Ten grams of soil were weighed into centrifuge tubes in duplicate. Bray 1 solution of composition of 0.03 N NH_4F + 0.025 N HCl was added (Bray and Kurtz, 1945). The tubes were shaken on an end-over-end mechanical shaker for five minutes. The solution was filtered through a No.42 Whatman No. 42 filter paper into a 10 mL volumetric flask and made up to the volume. Phosphorus in the filter was determined using the molybdate–ascorbic acid method as follows: A 5 mL aliquot of the filtrate was transferred into a 50 mL volumetric flask containing distilled water in duplicates. The pH was adjusted using P-nitrophenol indicator and neutralized with a few drops of 4 N NH_4OH until the solution turned yellow. The solution was diluted to 40 ml with distilled water after which 8 ml of reagent B (ascorbic acid ammonium molybdate+antimony potassium tartarate + concentrated H_2SO_4 in distilled water) was added and made to volume with distilled water. The solution was mixed thoroughly by shaking and allowed to stand for 50 minutes for the colour to stabilize. A blank was prepared with distilled water and 8 ml of reagent B added as described above. The

intensity of the blue color was measured using the Philips PU 8620 spectrophotometer at a wavelength of 712 nm. The P concentration was calculated as follows:

$$\text{mg P kg}^{-1}\text{ soil} = \frac{(\text{(spectrometer reading – blank reading)x volume of extract})}{\text{Volume of aliquot x sample weight (g)}} \quad \ldots\ldots[4]$$

3.8: Particle size analysis

The particle size distribution was determined by the hydrometer method of Bouyoucous (1962). A 100 mL of 0.01 M Calgon (Sodium hexametaphosphate) was added to 40 g of soil sample and then shaken for four hours on a mechanical shaker. The suspension was transferred into 1000 mL (1L) sedimentation cylinder and the level of the liquid brought to the 1000 mL mark with distilled water. After mixing the suspension with a plunger, a hydrometer was carefully inserted into the suspension and readings of clay and silt were taken after 5 minutes.

After 5 hour, the reading of clay was taken. The sand was washed, oven-dried for 24 hr and then weighed. The percentage of the various soil separates were then determined as follows:

$$\text{Silt (\%) + Clay (\%)} = \frac{\text{corrected hydrometer reading at 5 minutes}}{\text{Oven–dried sample weight (g)}} \text{ x } 100 \ldots\ldots\ldots\ldots \quad [5]$$

$$\text{Clay (\%)} = \frac{\text{corrected hydrometer reading at 5 hours}}{\text{Oven–dried sample weight (g)}} \text{ x } 100 \ldots\ldots\ldots\ldots\ldots\ldots\ldots\ldots \quad [6]$$

Silt (%) = (5) - (6)

Sand (%) = 100 – (5)

The textural class of the soils was determined using the Textural Class Triangle (Figure 1).

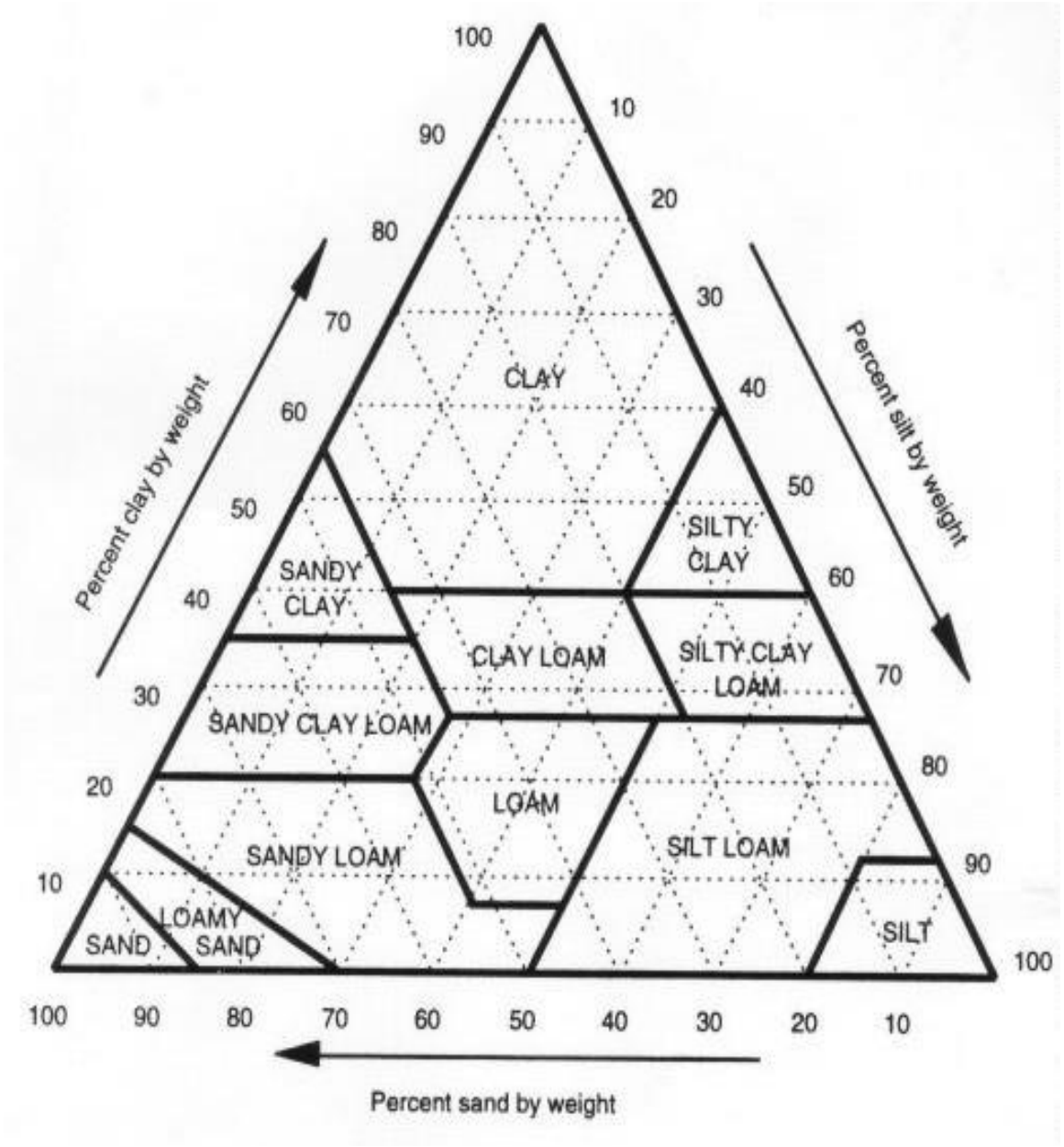

Figure 1 Soil Textural Triangle.

3.8.1: Bulk density determination

For each of the soils, undisturbed soil samples were taken from the field using core samplers. A core sampler was hammered in the soil and then excavated using a cutlass. The core sampler with its content was removed and quickly placed in a polythene bag and sent to the laboratory. A moisture can with its lid was labeled and then weighed empty. The core sampler with its content was unwrapped and the soil pushed into the weighed

empty moisture can. The soil with the moisture can was dried in an even at 105^{o}C for 24 hrs, cooled in a desiccator and later weighed. The volume of the core sampler was determined by measuring the internal diameter and height of the core sampler and the volume calculated from a formula $\pi r^2 h$. The bulk density of the soil was then determined as:

$$\text{Bulk density} = (Oven\ dry\ weight\ of\ soil) \div (volume\ of\ soil) \quad [7]$$

3.8.2: Field Capacity

Five kilograms of Toje and Bumbi soils were weighed into pots perforated at the bottom (10 holes per pot) to allow drainage. The soils were saturated in pots with distilled water and allowed to drain for 48 hours. Five soil samples were taken from each pot and weighed before oven-dring at a temperature of 105°C for 48 hr. The samples were weighed again to determine the moisture content of the soil as follows:

$$\%\text{Water content (Moisture)} = \frac{\text{Weight of wet soil} - \text{Weight of oven-dried soil}}{\text{Weight of oven-dried soil}} \times 100 \quad [8]$$

The moisture content at field capacity thus calculated was noted, pots in the subsequent green house experiment were watered at rates of 60% field capacity.

3.9: Greenhouse experiment

3.9.1: Pot experiment and treatments used

Before the greenhouse experiment, viability test was conducted on the seeds. The viability test showed 80% germination (Appendix Plate 1). Seventy two plastic pots were obtained, 36 pots were each filled with 2 kg each Toje series and then mixed with 1.0 kg acid washed sand. Similarly, each of the other 36 pots was filled with two kg of the Bumbi series and 1.0 kg of acid washed sand added. The sand was added to the soil to make it

easy for root penetration. A control treatment without P addition was included. The two soils were treated with two P sources at six levels and each level was replicated three times. The treatments were: two soils x six P treatments x two types of P fertilizers x three replicates giving a total of seventy-two. In adding the different P fertilizers, the soil in each pot was transferred into a large plastic basin and the weighed amount of the P fertilizer was added. It was thoroughly mixed with the soil and returned to the respective pots. Four pigeonpea seeds were sown in each pot. The soil was maintained at 60% of the respective field capacity throughout the experiment by weighing. The quantities of TSP and TRP applied to the soils in the pots at each treatment level are presented in Table 1. Each treatment had three replicates. They were arranged in completely randomized design (CRD) in the Sinna's Garden located behind the Crop Science Department of the University of Ghana.

Table 1. Application rates of the two phosphorus sources

P Sources	Appl. Rate (kg / ha)	Appl. rate for Toje (g / pot)	Appl. rate for Bumbi (g / pot)	Replicates
Control	0	0	0	3
Togo R.Phosphate	30	0.12	0.10	3
Togo R.Phosphate	60	0.23	0.19	3
Togo R.Phosphate	90	0.35	0.29	3
Togo R.Phosphate	120	0.46	0.38	3
Togo R.Phosphate	150	0.58	0.48	3
Control	0	0	0	3
Triple S. Phosphate	30	0.08	0.07	3
Triple S. Phosphate	60`	0.16	0.13	3
Triple S. Phosphate	90	0.24	0.20	3
Triple S. Phosphate	120	0.32	0.27	3
Triple S. Phosphate	150	0.40	0.33	3

3.9.2: Harvesting, drying and weighing

The plants were harvested by cutting the shoot at the soil level with a sharp knife and the shoots and the leaves were carefully transferred into large brown envelopes. The soil was then poured onto a plastic sheet and the roots carefully removed for nodule count in the laboratory. Each plant was taken and the counting of nodule was carefully done. The weight of the envelopes was taken before and after transferring, then the envelopes were placed in an oven at 70°C for three days. The oven dry materials were then transferred

quickly into desiccators to cool. The plant materials and nodules were weighed again to determine the dry matter yield.

3.9.3: Digestion of plant material

A 0.05 g of the ground plant material was digested with 5 mL of ternary mixture of concentrated HNO_3 and $HClO_4$ in the ratio of 2:3. The digestion was continued until white fumes of $HClO_4$ ceased. The digest was filtered and transferred quantitatively into a 100 mL volumetric flask and then made to volume. Five mL aliquot of each solution was taken for the determination of nitrogen concentration in the plant material as described in 3.6. Phosphorus concentration in the plant was measured from the digest and P uptake of pigeonpea was calculated as:

P uptake = Dry matter yield of Pigeonpea x P concentration in the plant. [9]

3.9.4: Statistical analysis

The effect of treatments on nodule weight, nodule count, P uptake, and dry matter yield of pigeonpea was tested using ANOVA. All statistical analyses were done using Genstat. Unless otherwise indicated, differences were considered to be significant at the 5% probability level. The results of the ANOVA are shown in the Appendix.

CHAPTER FOUR

RESULTS AND DISCUSSION

4.1: Physical properties of the Toje and Bumbi soils

The Toje soil consists of 68% sand, 13% silt and 19% clay (Table 2). Its particle size distribution makes the soil sandy loan. It has a bulk density of 1.21 Mg/m^3 suggesting that water infiltration and root penetration will be good and easy to cultivate. Its field capacity is 34% due to its sandy nature and cannot retain much water. On the other hand, the Bumbi soil is silty clay loam with 25 % sand, 39 % silt and 36 % clay. The soil is compact, clayey, dry and cracking during the dry season. When wet it gets muddy and difficult to cultivate. These properties suggest that the Bumbi soil is vertic in nature and may belong to the Vertisols with 2:1 clay mineral (montmorillonite). Its bulk density is 1.36 Mg/m^3 due to the high clay content. Root penetration in the soil will be rather poor compared to the Toje soil. The field capacity is 43 % and suggests a higher water retention capacity of the Bumbi soil which again may be explained by the high clay content of the soil. Clayey soils have many more micro pores which can retain more water compared to sandy soils.

4.2: Chemical properties

4.2.1: Soil pH

Both Bumbi and Toje soils are slightly acidic with pH of 6.5 in water and 5.9 in 0.01M $CaCl_2$ for Bumbi soil and Toje soil with pH of 6.3 in water and pH of 5.7 in 0.01M $CaCl_2$ (Table 2). The pH values determined by 0.01 M $CaCl_2$ (pH_s) were lower than those determined in water (pH_w), giving a negative ΔpH ($pH_{salt} - pH_{water}$). The negative ΔpH

indicated that the soil surface had a net negative charge and could retain cations on the colloidal complex.

Table 2 Chemical and physical properties of the soils

SOIL PROPERTIES	TOJE	BUMBI
Particle size		
Sand (%)	68	25
Silt (%)	13	39
Clay (%)	19	36
Bulk Density (g/cm^3)	1.21	1.36
Field Capacity (%)	34	43
pH 1:1(H_2O)	6.3	6.5
pH 1:2 $CaCl_2$	5.7	5.9
Organic C (g/kg)	7.45	3.3
Total N (g/kg)	0.56	1.8
Total P (mg/kg)	157	228
Available P (mg/kg)	4.53	33.2
Exchangeable cations ($cmol_c/kg$)		
Ca	1.13	11.6
Mg	0.75	13.5
K	0.61	0.61
Na	0.33	0.39
Exchangeable Acidity ($cmol_c/kg$)	0.37	0.36
Effective cation exchangeable capacity ($cmol_c/kg$)	3.19	26.46

4.2.2: Soil organic carbon and nitrogen

Table 2 shows that the Toje soil had higher organic carbon content (7.45 g/kg) than the Bumbi soil (3.3 g/kg). The higher carbon content of Toje soil was a reflection of its lower bulk density compared to that of the Bumbi soil. This conforms to the fact that organic matter reduces soil bulk density.

The total nitrogen content of the soils was generally low but the Bumbi soil had a higher N content (1.8 g/kg) than the Toje soil (0.56 g/kg^{-1}). Nodulation is found to be lower in soils with high N content and therefore, comparing the two N contents of the soils suggest that nodulation would be higher in the Toje soil than in the Bumbi soil.

4.2.3: Total and available phosphorus

Both total and available P contents were higher in the Bumbi soil than in the Toje soil. The total P content of the Toje soil was 157 mg/kg and that of the Bumbi soil was 228 mg/kg (Table 2).

The available P was generally low in the Toje soil (4.53 mg/kg) as compared to the Bumbi soil of (33.2 mg/kg) even though none of these soils had a history of phosphorus fertilization. In reference to nodulation, it is expected that nodulation would be higher in the Toje soil than in the Bumbi soil (Figure 2, Appendix Table 1) since P is needed for nodulation and N_2-fixation in legumes.

4.2.4: **Exchangeable bases**

The exchangeable Ca^{2+} values in both soils were vastly different. The Toje soil had a lower exchangeable Ca content (1.13 $cmol_c/kg$) than the Bumbi soil (11.6 $cmol_c/kg$). The high Ca^{2+} content of the Bumbi soil is attributed to the presence of calcium carbonate nodules. The nodules were tested for CO_3^{2-} with dilute HCl and effervescence of CO_2 gas

was observed to confirm the presence of carbonates. The presence of $CaCO_3$ in the form of nodules is a property of Vertisols and confirms the vertic nature of the Bumbi soil. The Mg^{2+} in the Toje soil was slightly lower but not significantly different from that of the Bumbi soil. The exchangeable Na^+ content was consistently lower than that of K^+ in both soils. The exchangeable acidity in the Toje was 0.37 $cmol_c/kg$ and 0.36 $cmol_c/kg$ in Bumbi. These values were similar and agreed with the fact that both soils had similar pH values which were near neutral. At these pH levels (pH 6.3 to 6.5) Al^{3+} ions are precipitated and their activity remains very low. Since effective cation exchange capacity (ECEC) is calculated as the sum of the exchangeable bases and exchangeable acidity, the ECEC of the Bumbi soil was higher (26.46 $cmol_c/kg$) compared to that of the Toje soil (3.19 $cmol_c/kg$).

4.3: Effect of phosphorus on the dry matter yield (DMY) of pigeonpea

Figure 2 shows the effect of increasing rate of phosphorus application on dry matter yield (DMY) of pigeonpea. The DMY increased with increase in P application rates from 0-P to 120 kg/ha and decreased at 150 kg/ha in both soils. The trend was similar in both TSP and TRP fertilizers. Dry matter yield at all P treatments in the Toje soil was higher than in the Bumbi soil. However, there was no significant difference ($p < 0.05$) in dry matter yield of the P treatments at 0 and 30 kg P/ha application rates in the two soils.

The dry matter yield in the control experiment (no fertilizer applied) of the two soils was significantly lower than that of the other application rates ($p < 0.05$). Differences in dry matter yield of the pigeonpea occurred at 60, 90, 120 and 150 kg P/ha in both soils ($p < 0.05$). In both TSP and TRP, the two soils recorded the highest dry matter yield at 120 kg P/ha application rate but that of the Toje soil was significantly greater than the dry matter yield of the pigeonpea in the Bumbi soil at 120 kg P/ha. The large differences in

DMY between the two soils may be attributed to the differences in their soil texture and the muddy nature of the Bumbi soil during watering which impeded root development of the

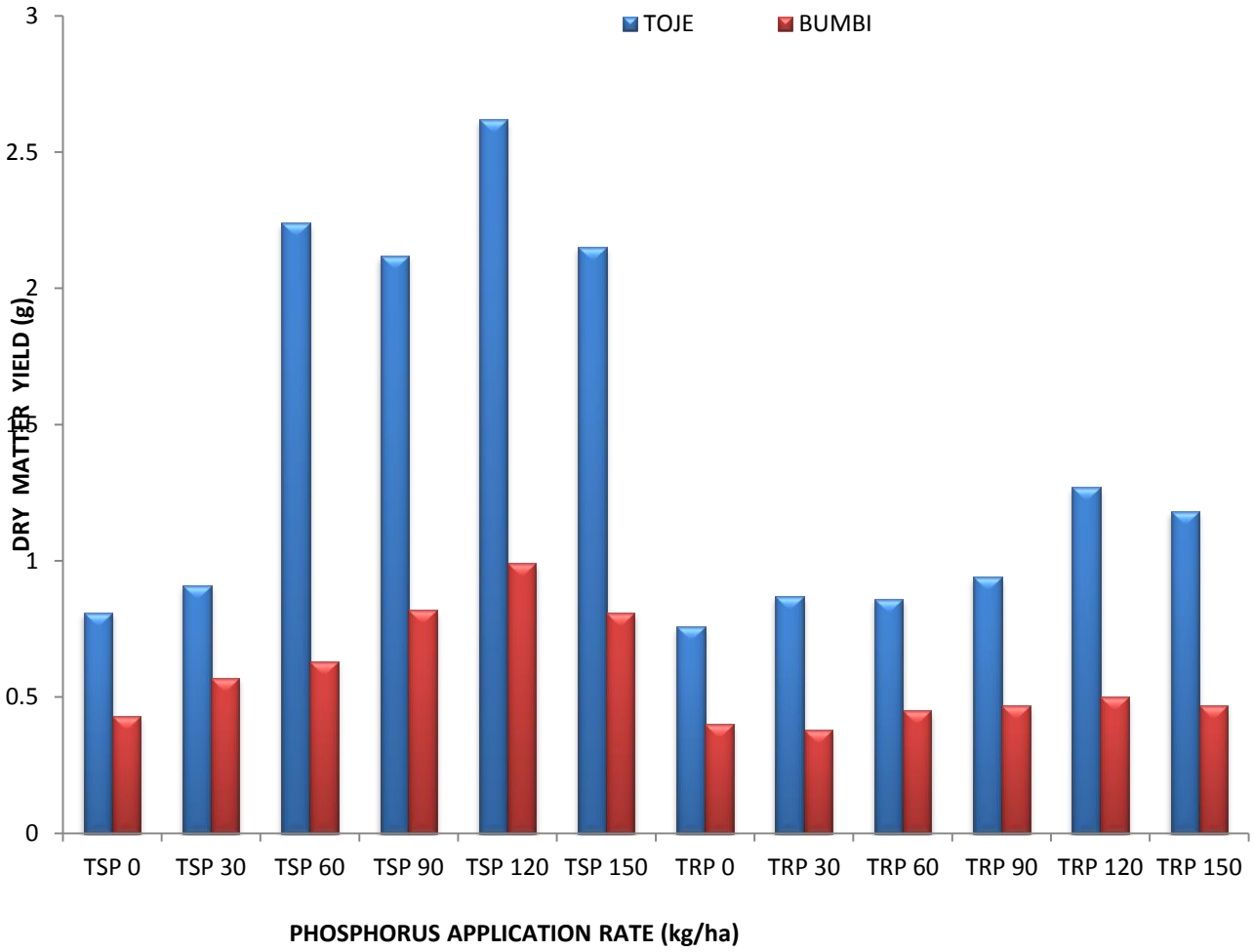

Figure. 2:Dry matter yield of pigeonpea as affected by increasing rates of application of triple superphosphate (TSP) and Togo rock phosphate (TRP).

PIGEONPEA IN TOJE AT CONTROL (Top)

PIGEONPEA IN BUMBI AT CONTROL (Bottom)

Plate 1: Differences in growth of pigeonpea in Toje (top) and Bumbi (bottom) soil

4.3.1: Effect of phophorus on nodule numbers in pigeonpea

Table 3 shows the effect of increasing rate of phosphorus application on the number of nodules counted in the pigeonpea crop. In the Toje soil, the number of nodules ranged from 20 (for the control) to 52 in the TSP_{120} treatment whiles in the Bumbi soil, it ranged from 8 (control) to 28 in the TSP_{150} treatment. The pigeonpea seeds were not inoculated with rhizobium inoculum yet there was nodulation. This shows that both soils contained native rhizobium capable of nodulating pigeonpea.

Generally, the number of nodules in the Toje soil far exceeded the number in Bumbi at all treatment levels. In Toje soil, the highest number of nodules from the TSP and TRP treatments were at 120 kg/ha and were all higher than those of Bumbi which had its highest nodule count in both TSP and TRP treatments at 150 kg/ha. Even though the number of nodules obtained in the Bumbi soil was highest at 150 kg P/ha rate, the nodules were of small sizes and therefore, the nodule weight was lower compared to the nodule weight at 120 kg P/ha. There was no significant difference in nodule numbers at 0-P (control) and 30 kg P/ha. However, there was a significant difference in nodule numbers at the 60 kg P/ha and 90 kg P/ha. Similarly, significant difference occurred between the nodule numbers at 120 kg P/ha and 150 kg P/ha in the two soil series used.

4.3.2: Effect of P on the nodule weight

Figure 3 shows the effect of increasing rate of phosphorus application on nodule weight. The nodule weight in the two soils increased as P application increased. The nodule weight followed a similar trend as the nodule numbers in both soils. In Toje and Bumbi, the highest nodule weight was obtained from the TSP and TRP treatments at 120 kg P/ha. There was a significant difference between the various nodule weights in the two

soils. The nodule weights in the Toje soil at all P treatment levels were significantly different ($p < 0.05$) from the nodule weights in Bumbi soil.

Table 3: Effect of different rates of phosphorus on nodule number of the plant.

Treatment (kg/ha)	Mean Nodule Count of the pigeonpea	
	Toje	Bumbi
TSP_0	20	8
TSP_{30}	24	11
TSP_{60}	35	18
TSP_{90}	40	26
TSP_{120}	52	27
TSP_{150}	47	28
TRP_0	20	8
TRP_{30}	22	9
TRP_{60}	29	12
TRP_{90}	31	10
TRP_{120}	41	18
TRP_{150}	40	22

The Bumbi appears to be richer in available P and Ca which are necessary for nodulation and N_2-fixation but its clayey and muddy nature did not permit the proper growth of the crop.

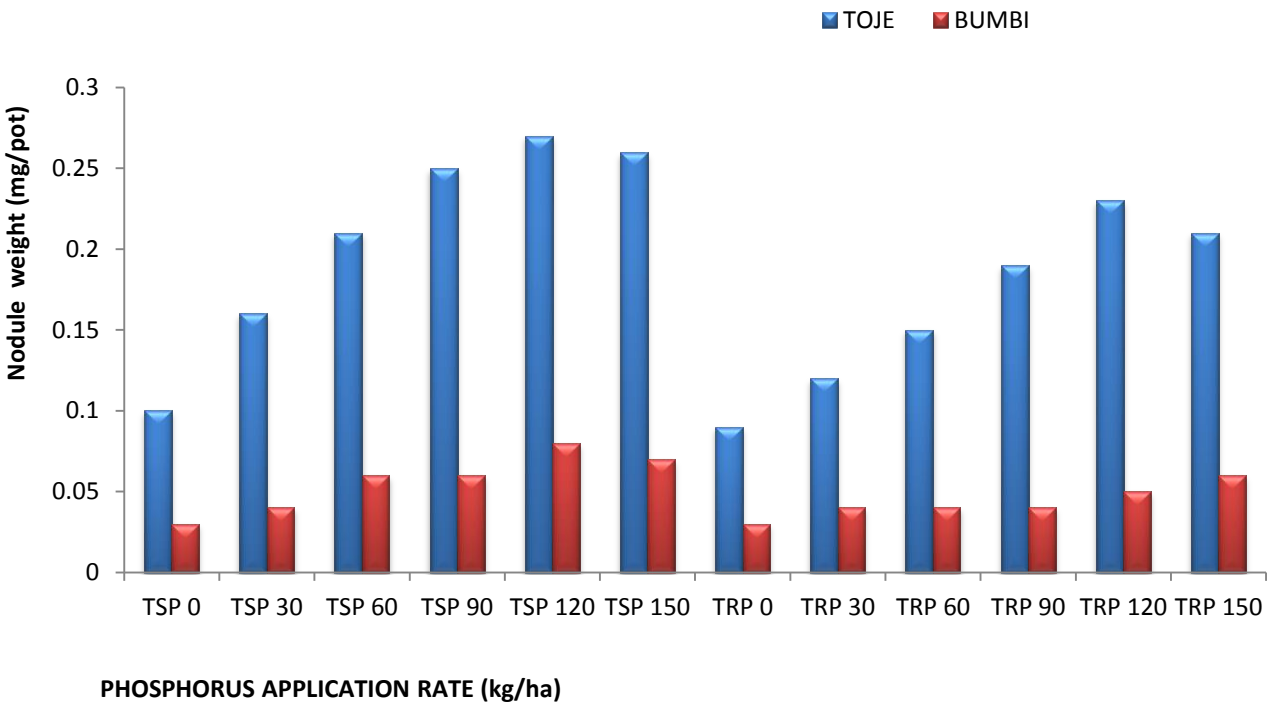

Figure. 3: Effect of increasing rates of P on dry nodules formed on pigeonpeon in Toje and Bumbi soils

4.3.3: The effect of phosphorus on P uptake

Phosphorus uptake by the pigeonpea crop increased with increase in phosphorus application rate (Fig. 4). In the TSP treatment, P uptake increased consistently from the control (58.16 mg/pot) to the TSP_{120} (250.5 mg/pot) and decreased in the TSP_{150} to (184.3 mg/pot) in the Toje soil (Figure 4). A similar pattern of P-uptake was observed in the Bumbi soil where P uptake in the control was the lowest and highest in the TSP_{120} treatment. Togo rock phosphate application did not significantly improve P uptake in both soils as P uptake was very low compared to the TSP treatment. Clearly, the difference in the two P fertilizers could be attributed to the high water soluble P content of the TSP compared with the unreactive TRP which had very little water soluble P (Abekoe and Tiessen, 1998). Also, the soils used had pH values which were moderately to near neutral pH which do not support rock phosphate dissolution since H^+ ions are required for the process to occur.

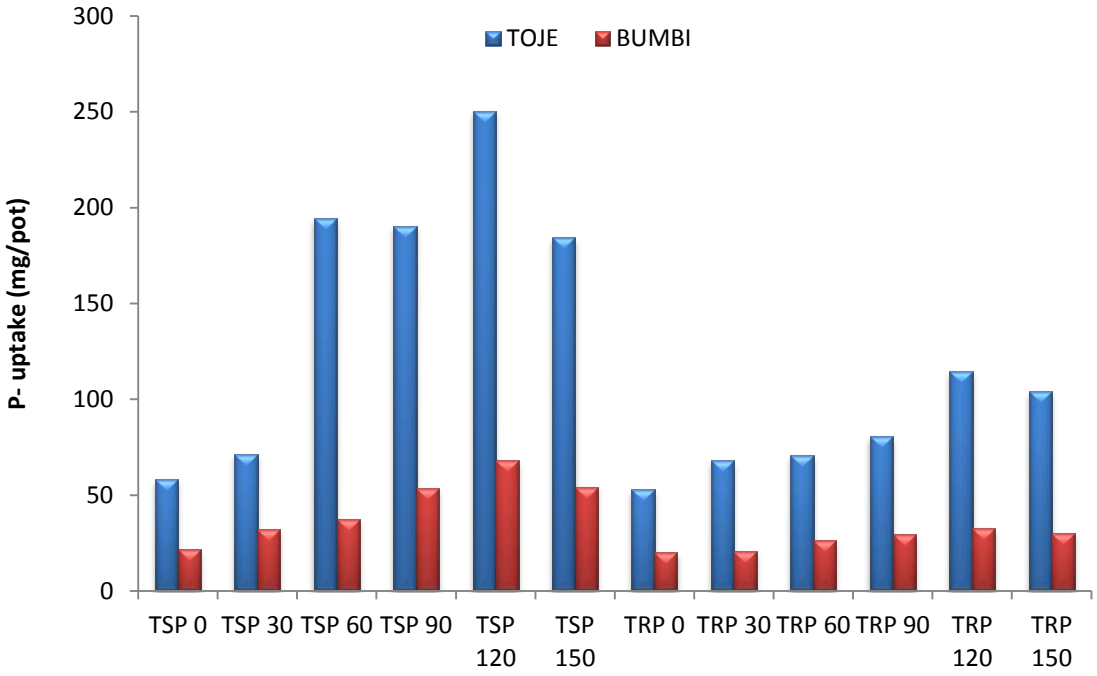

Figure 4: P-uptake by pigeonpea as affected by increasing rates of P application.

4.4: Relative agronomic efficiency (RAE) of triple superphosphate and Togo rock phosphate

In calculating the relative agronomic efficiency (RAE) of the TRP, triple superphosphate was used as a standard fertilizer. The RAE was calculated based on the highest DMY and P-uptake of pigeonpea at 120 kg/ha. The agronomic efficiency of the TRP in the Toje soil relative to TSP in the Toje soil was 27% and that of Bumbi was 17.9%. For P uptake, the RAE of TRP in the Toje soil was 32% compared with 26% in the Bumbi soil (Table 4). The DMY and P-uptake in both soils followed the trend: TSP > TRP. Hence the RAE of the two P sources in the Toje and Bumbi soils with pigeonpea as a test crop was higher in the former soil than in Bumbi.

Table 4: Relative agronomic efficiency of the DMY and P-uptake in Toje and Bumbi soils

P source	DMY	P-uptake	RAE (%)	
	(g)	mg/pot	DMY	P-uptake
TSP $_{(Toje\ 120\ kg/ha)}$	2.62	250.45	100	100
TRP $_{(Toje\ 120\ kg/ha)}$	1.25	114.63	27	32
TSP $_{(Bumbi\ 120\ kg/ha)}$	0.99	68.01	100	100
TRP $_{(Bumbi\ 120\ kg/ha)}$	0.50	32.17	17.9	26
TSP ($_{Toje,\ control)}$	0.81	58.16		
TRP $_{(Toje,\ control)}$	0.76	52.89		
TSP $_{(Bumbi\ control)}$	0.43	21.67		
TRP $_{(Bumbi\ control)}$	0.40	20.08		

$$\text{RAE (\%)} = \frac{(\text{Yield of TRP120}) - (\text{Yield of TRP Control})}{(\text{Yield of TSP120}) - (\text{Yield of TSP Control})} \times 100 \ldots\ldots\ldots\ldots \quad [7]$$

CHAPTER FIVE

5.0: SUMMARY AND CONCLUSION

In this study, two soils, Toje and Bumbi series with different characteristics were fertilized with TSP and TRP at the rates 0, 30, 60, 90, 120 and 150 kg P/ha and were used evaluate the dry matter yield and nodulation of pigeonpea.

The soil properties suggested that the Bumbi soil was more fertile than the Toje soil. The chemical properties such as the effective cation exchange capacity, total N and available P levels in the Bumbi soil were much higher than those of the Toje soil. Despite the relative high fertility of the Bumbi soil, dry matter yield (DMY) and nodulation of the pigeonpea crop were lower than those in the Toje soil. This difference in DMY of the pigeonpea was attributed to the poor physical properties of the Bumbi soil. The soil tended to be muddy after watering, it got dry and cracking during the day. These poor physical properties of the Bumbi soil must have hampered root development and root exploration for nutrients in the soil. The dry matter yield and the total nodule numbers of the pigeonpea crop grown on the Toje soil were significantly higher ($P < 0.05$) than those grown on the Bumbi soil.

The different P sources also showed marked differences in their ability to supply P to the crop. The TSP fertilizer increased the pigeonpea dry matter yield progressively from the 0-P (control) to 120 kg P/ha application rate in both Toje and Bumbi soils but DMY in the Toje soil was significantly higher than in the Bumbi soil. A similar trend in pigeonpea DMY was observed with the Togo rock phosphate although in both soils the dry matter yield was very low compared with those of the TSP-treated soils. In all instances, dry matter yield, nodule count, nodule weight were all significantly higher in the TSP-treated soils than in the TRP-applied soils. The relative agronomic efficiency (RAE) of the TRP

which reflects the availability of P from the Togo rock phosphate to the crop compared to the water soluble TSP was quite low (27% for the DMY in the Toje soil and 17.9% in the Bumbi soil).

5.1: Recommendations

1: For a good growth and nodulation of pigeonpea, the Toje series is more appropriate to use rather than the Bumbi series. Thus, farmers in the coastal savanna agro-ecological zone who intend to grow pigeonpea must use the Toje series instead of Bumbi series. However, organic matter application to the Bumbi soil can improve its soil structure and hence improve water infiltration and make soil better equipped for plant growth.

2: For maximum nodulation and possible N_2-fixation, 120 kg P/ha application rate using TSP as the P fertilizer is recommended.

3: Farmers are advised to use acid soils to grow the pigeonpea crop if rock phosphate is to be used. This practice will maximize the dissolution and therefore, the supply of P from the rock phosphate to the crop.

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APPENDIX

Table 1: Effect of different rates of phosphorus on dry matter yield of the plant.

Treatment	Mean DMY of the pigeonpea	
(kg/ha)	Toje	Bumbi
TSP $_{0}$	0.81	0.43
TSP $_{30}$	0.91	0.57
TSP $_{60}$	2.24	0.63
TSP $_{90}$	2.12	0.82
TSP $_{120}$	2.62	0.99
TSP $_{150}$	2.15	0.81
TRP $_{0}$	0.76	0.40
TRP $_{30}$	0.87	0.38
TRP $_{60}$	0.86	0.45
TRP $_{90}$	0.94	0.47
TRP $_{120}$	1.25	0.50
TRP $_{150}$	1.18	0.47

Table 2: Effect of different application rate of phosphorus on nodule weight of plant

Treatment	Mean Nodule weight of the soils	
(kg/ha)	Toje	Bumbi
TSP $_{0}$	0.10	0.03
TSP $_{30}$	0.16	0.04
TSP $_{60}$	0.21	0.06
TSP $_{90}$	0.25	0.06
TSP $_{120}$	0.27	0.08
TSP $_{150}$	0.26	0.07
TRP $_{0}$	0.09	0.03
TRP $_{30}$	0.12	0.04
TRP $_{60}$	0.15	0.04
TRP $_{90}$	0.19	0.04
TRP $_{120}$	0.23	0.05
TRP $_{150}$	0.21	0.06

Table 3: Effect of different application rates of phosphorus on P-Uptake of the plant

Treatment	Mean P-Uptake of the pigeonpea	
(kg/ha)	Toje	Bumbi
TSP $_{0}$	58.16	21.67
TSP $_{30}$	71.16	32.09
TSP $_{60}$	194.43	37.36
TSP $_{90}$	190.34	53.46
TSP $_{120}$	250.45	68.01
TSP $_{150}$	184.47	54.27
TRP $_{0}$	52.89	20.08
TRP $_{30}$	68.21	20.59
TRP $_{60}$	70.61	26.33
TRP $_{90}$	80.55	29.65
TRP $_{120}$	114.63	32.50
TRP $_{150}$	104.19	30.17

Plate 1: Seed viability test conducted before sowing

Plate 2: Growth of pigeonpea in Bumbi soil series.

Plate 3: Growth of pigeonpea in Toje soil series

Plate 4: Vigorous growth of the pigeonpea in the Toje soil series

Plate 5: Harvested nodules

Analysis of variance

Variate: DRY_MATTER

Source of variation	d.f.	s.s.	m.s.	v.r.
SOIL	1	12.0704	12.0704	67.46
FERTILIZER	1	5.5444	5.5444	30.99
TREATMENT	5	4.7888	0.9578	5.35
SOIL.FERTILIZER	1	1.3889	1.3889	7.76
SOIL.TREATMENT	5	1.7745	0.3549	1.98
FERTILIZER.TREATMENT	5	2.1463	0.4293	2.40
SOIL.FERTILIZER.TREATMENT	5	1.0538	0.2108	1.18
Residual	48	8.5887	0.1789	
Total	71	37.3559		

Analysis of variance

Variate: NODULE_COUNT

Source of variation	d.f.	s.s.	m.s.	v.r.
SOIL	1	5408.00	5408.00	350.16
FERTILIZER	1	684.50	684.50	44.32
TREATMENT	5	4402.67	880.53	57.01
SOIL.FERTILIZER	1	0.50	0.50	0.03
SOIL.TREATMENT	5	216.67	43.33	2.81
FERTILIZER.TREATMENT	5	327.17	65.43	4.24
SOIL.FERTILIZER.TREATMENT	5	47.17	9.43	0.61
Residual	48	741.33	15.44	
Total	71	11828.00		

Analysis of variance

Variate: NODULE_WEIGHT

Source of variation	d.f.	s.s.	m.s.	v.r.
SOIL	1	0.337568	0.337568	87.59
FERTILIZER	1	0.014735	0.014735	3.82
TREATMENT	5	0.082024	0.016405	4.26
SOIL.FERTILIZER	1	0.003612	0.003612	0.94
SOIL.TREATMENT	5	0.035624	0.007125	1.85
FERTILIZER.TREATMENT	5	0.002724	0.000545	0.14
SOIL.FERTILIZER.TREATMENT	5	0.000946	0.000189	0.05
Residual	48	0.185000	0.003854	
Total	71	0.662232		

Analysis of variance

Variate: N_UPTAKE

Source of variation	d.f.	s.s.	m.s.	v.r.
SOIL	1	12.6491	12.6491	124.42
FERTILIZER	1	2.5597	2.5597	25.18
TREATMENT	5	3.0634	0.6127	6.03
SOIL.FERTILIZER	1	1.3472	1.3472	13.25
SOIL.TREATMENT	5	1.5889	0.3178	3.13
FERTILIZER.TREATMENT	5	0.9434	0.1887	1.86
SOIL.FERTILIZER.TREATMENT	5	0.6721	0.1344	1.32
Residual	48	4.8798	0.1017	
Total	71	27.7037		

Analysis of variance

Variate: N_CONCENTRATION

Source of variation	d.f.	s.s.	m.s.	v.r.
SOIL	1	2.622050	2.622050	475.18
FERTILIZER	1	0.000050	0.000050	0.01
TREATMENT	5	0.137333	0.027467	4.98
SOIL.FERTILIZER	1	0.001089	0.001089	0.20
SOIL.TREATMENT	5	0.019317	0.003863	0.70
FERTILIZER.TREATMENT	5	0.006117	0.001223	0.22
SOIL.FERTILIZER.TREATMENT	5	0.002578	0.000516	0.09
Residual	48	0.264867	0.005518	
Total	71	3.053400		

Analysis of variance

Variate: P_CONCENTRATION

Source of variation	d.f.	s.s.	m.s.	v.r.
SOIL	1	93.048	93.048	56.96
FERTILIZER	1	0.648	0.648	0.40
TREATMENT	5	32.524	6.505	3.98
SOIL.FERTILIZER	1	0.006	0.006	0.00
SOIL.TREATMENT	5	1.124	0.225	0.14
FERTILIZER.TREATMENT	5	0.917	0.183	0.11
SOIL.FERTILIZER.TREATMENT	5	0.968	0.194	0.12
Residual	48	78.417	1.634	
Total	71	207.650		

Analysis of variance

Variate: P_UPTAKE

Source of variation	d.f.	s.s.	m.s.	v.r.
SOIL	1	1337.46	1337.46	56.67
FERTILIZER	1	443.55	443.55	18.79
TREATMENT	5	560.66	112.13	4.75
SOIL.FERTILIZER	1	171.38	171.38	7.26
SOIL.TREATMENT	5	238.24	47.65	2.02
FERTILIZER.TREATMENT	5	220.35	44.07	1.87
SOIL.FERTILIZER.TREATMENT	5	129.49	25.90	1.10
Residual	48	1132.91	23.60	
Total	71	4234.05		