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Effects of mineral levels and leaf extracts of some plants on soil pH and growth rate of selected non-leguminous plants

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Abstract. *Njogu MJ, Murungi JI, Wanjau RN. 2021. Effects of mineral levels and leaf extracts of some plants on soil pH and growth rate of selected non-leguminous plants. Cell Biol Dev 5: 76-89.* The purpose of this study was to determine the effect of growth rate on non-leguminous plants such as wheat (*Triticum aestivum* L.), kale (*Brassica oleracea* var *acephala*), and coriander (*Coriandrum sativum* L.) grown on acidic soils treated with leaves and leaf extracts of plants known to have a high buffering capacity and mineralization capacity, i.e., *Jacaranda mimosifolia* D. Don, *Cordia africana* Lam, *Croton macrostachyus* Hochst. ex Delile, *Vitex keniensis* Turril, *Tithonia diversifolia* (Hemsl.) A.Gray, *Manihot esculenta* Crantz, *Carica papaya* L., and *Helianthus annuus* L. For this investigation, the leaves and leaf extracts were applied to the soil with a pH of 4.75. Leaf extracts and leaves of eight species were combined according to the macronutrient content and mineralization of chosen plants discovered in Nyandarua County, Kenya. Wheat, kale, and coriander growth rates were determined by comparing the dry mass of uprooted seedlings from the trial and control studies every fourteen days for 60 days. Flame photometry, atomic absorption spectrometry (AAS), UV/visible spectroscopy, and turbidimetry were used to determine macronutrients in leaf extracts. The analysis of variance (ANOVA) and Student-Newman-Keuls (SNK) tests were used to analyze the data. For 60 days, the mean pH value of leaf extracts ranged from 4.580.27^{a} to 7.440.04^{d}. K^{+}; 189.95±0.17^{c}, Ca^{2+}; 367.33±0.67^{e}, Mg^{2+}; 114.33^{e}±0.33, $PO4^{3-}$; 55.38±0.23^{f}, NO_3^{-}; 322.25±0.40^{f}, and $SO4^{2-}$; 56.48±0.23^{e} had the greatest mean levels in leaves (µg/g). Macronutrient levels were significantly different in soil treated with leaves and leaf extract (Le), commercial NPK fertilizers (Cf), and untreated soil (Us). Wheat (dry mass) grew at a mean rate of 1.27±0.13^{b} g/wk (Le), 1.26±0.12^{b} g/wk (Cf) and 0.32±0.02^{a} g/wk (Us), while in (height) was 7.29±1.43^{b} cm/wk (Le), 6.20±1.95^{b} cm/wk (Cf), 3.98±0.97^{a} cm/wk (Us). For kales 0.20±0.01^{a} g/wk (Le), 0.30±0.02^{a} g/wk (Cf), and 0.03±0.01^{b} g/k (Us), while in (height) 2.15±0.85^{c} cm/wk (Le), 2.57±0.88^{b} cm/wk (Cf), and 1.04±0.02^{a} cm/wk (Us). The mean growth rate for corianders was 0.16±0.01^{a} g/wk (Le), 0.17±0.02^{a} g/wk (Cf), and 0.10±0.01^{a} g/k (Us), while in (height) 1.85±0.56^{b} cm/wk (Le), 1.86±0.58^{b} cm/wk (Cf) and 0.79±0.18^{a} cm/wk (Us). This study's findings show that the plant's leaves and leaf extracts can be used in place of inorganic fertilizers and should be recommended for agroforestry in low-pH environments.

Keywords: Acidic soils, *Brassica oleracea* var. *acephala*, *Coriandrum sativum*, growth rate, *Triticum aestivum*

INTRODUCTION

Today, man is confronted with the difficulty of providing enough food to feed the world's growing population. Thus, food insecurity has been linked to crop productivity being damaged due to excessive use of commercial inorganic fertilizer on tiny plots of land, which are sometimes the only way to sustain high yields of food crops (Supramudho et al. 2012). Excessive usage of inorganic fertilizer has resulted in a low soil pH, reducing crucial nutrient availability to plants (Ge et al. 2018). As a result, organic stuff decomposes slowly. Additionally, it results in the loss of micronutrients, producing nutritionally deficient food (Handayani et al. 2021). Without addressing the issue, the soil will eventually become unproductive. Organic matter is required to boost the soil's buffering capacity and provide plant nutrients.

Liming is used to raise the pH of the soil; however, it is an expensive process that does not contribute to soil fertility. Therefore, there is a need for an alternative way to inorganic fertilizer and liming material application. In addition, the increased human population has increased not just land usage but also low yields, resulting in forest cover encroachment resulting in reduced rainfall (Lepp and Edwards 1998). As a result, an alternative to inorganic fertilizers that also increases forest cover is required.

The inorganic fertilizers that are routinely used and available primarily feed plants with the principal macronutrient. Nitrogen, phosphorus, and potassium percentage compositions are often listed in the order NPK major (macronutrients). Inorganic fertilizers tend to change the soil pH over time due to their acidic nature (Ge et al. 2018). Reduced soil pH has a detrimental influence on the growth of plants and soil organisms, reducing predicted yields. That is because acid deposition causes important elements such as calcium, magnesium, and potassium to be leached (Murungi 1990). Acidic fertilizers may have little effect on soils with significant buffering capacity. Buffering capacity refers to an ecosystem's ability to maintain a consistent pH regardless of the presence of an acid or a base. Buffering is critical for keeping soil pH from rapidly falling.

Soil with a high proportion of limestone and silicate effectively neutralizes hydrogen ions generated by fertilizers that come into contact with water, allowing the pH to remain normal. Soils, particularly those that cover

granite and igneous rocks, are deficient in buffering material. They have a minor acidic flavor and cannot neutralize the acidic effect. The acidic deposition has been demonstrated not to affect places with a high organic composition, and it is assumed that the decaying products of these locations act as a buffer against the effects of acid (Murungi 1990).

Organic farm manure produced on-site is used for a variety of fertilizing purposes. They are significant in part due to their organic composition. All soils require a source of organic matter for various reasons, including as a carrier of usable energy and nutrients for soil organisms (Liu et al. 2013; Qin et al. 2015; Lestari et al. 2017). Organic manure is primarily composed of rotting plants and is utilized on farms. The quality of manure is determined by the plants utilized to produce it. As a result, it is necessary to identify plants capable of producing high-quality manure. However, the decomposition rate of leaves from diverse plants and the concentrations of macronutrients in their leaves have been examined (Murungi 1990; Njagi 2008). Alien plants have replaced the majority of indigenous trees in the area. It has been noted that some of these exotic plants produce a litter that decomposes slowly and produces acidic decomposition products (Murungi 1990). It is considered that organic matter contributes significantly to the chemical soil's buffering ability (Nur et al. 2019; Taberima et al. 2020). Leaves with a high buffering capacity contain a high concentration of important components (Murungi 1990).

Organic farming is based on cultivating biological diversity in the field to disrupt organisms' habitat and on purposeful soil fertility management and replenishment (Dong et al. 2016; Seufert et al. 2017). The growers use no synthetic pesticides or inorganic fertilizers. On the other hand, organic food is more expensive but healthier to consume because it has significantly fewer synthetic residues than regular food (Kriwy and Mecking 2012). Organic farmers use cover crops, compost, and biologically based soil amendments to feed the soil biota and create organic matter (Serrano et al. 2017; Njoroge et al. 2018), producing robust plants that are resistant to disease and insect predation (Zhang et al. 2019).

There is a need to explore the influence of chosen plants with high macronutrient content and high mineralization on the development rate of selected non-leguminous plants and the pH of acidic soils (Njagi 2008). Therefore, this study analyzed eight plants that showed the possibility of using organic manure as a substitute for inorganic fertilizers.

This study has the following precise objectives: (i) To determine the concentrations of K, N, Mg, P, Ca, and S in selected plants' leaves and leaf extracts, as well as their pH; (ii) To measure the pH of acidic soils before and after treatment with selected plant leaves and leaf extracts; (iii) Every two weeks for a period of 60 days, to monitor the height and dry mass of *Triticum aestivum* L. (wheat), *Brassica oleracea* var *acephala* (kale), and *Coriandrum sativum* L. (coriander) cultivated on soil treated with a combination of leaves and leaf extracts.

MATERIALS AND METHODS

Study area

The study took place in Rurii and Kasuku in Kenya's Nyandarua County. The area is between 1,800 and 2,200 meters above sea level, receives an average annual rainfall of 400-2,200 mL, and has a loam soil type. Nyandarua County is bounded on the north by Laikipia County, on the west by Nakuru County, on the east by Nyeri County, and on the south by Kiambu County. The study location was chosen because of the relatively small parcels of land partitioned into parts for fodder crops, non-leguminous plants for food, and potatoes planted twice a year during the long and short rains. Many farmers use inorganic fertilizers at least twice a season to increase yields (for a base and top dressing). However, some farmers cannot buy inorganic fertilizers and rely on organic fertilizers.

Research design

Randomization was used in the experimental design for leaf gathering. It included using leaves from *Jacaranda mimosifolia* D. Don (E1), *Cordia africana* Lam (L1), *Croton macrostachyus* Hochst. ex Delile (L2), *Vitex keniensis* Turril (L3), *Tithonia diversifolia* (Hemsl.) A.Gray (L4), *Manihot esculenta* Crantz (F1), *Carica papaya* L. (F2), and *Helianthus annuus* L. (F3). The rate of mineralization of these leaves, their effect on the pH of acidic soil, and the effect of combined leaves and leaf extracts on the growth rate of *T. aestivum* (wheat), *B. acephala* (kale), and *C. sativum* (coriander) were also investigated in comparison to the same acidic soil treated with inorganic fertilizer NPK in a control experiment using untreated soil. The research design for this study is shown in Figure 1.

The flow chart in Figure 2 illustrates the process of measuring the growth rate of each non-leguminous plant employed in this research. As in untreated soil, soil treated with commercial NPK fertilizers (Cf), and soil treated with leaves and leaves extracts (Le). Each container contained 30 seeds, from which 5 seedlings were removed every 14 days for 60 days to determine the height (cm) and dry mass (g).

Cleaning of apparatus

All apparatus was washed with a liquid detergent, rinsed, and then immersed for 48 hours in 1:1 analytical grade nitric acid. Next, they were rinsed in aqua regia, tap water, and distilled water many times. After that, the glasswares were dried in a hot oven at 120^{o}C. Prior to each test, the cleaning procedure was repeated.

Reagents and solvents

Throughout the research, analytical grade (AR) reagents from Thomas Baker Chemicals Ltd., Mumbai, India, as well as deionized and distilled water, were employed. In addition, the substances were weighed using a research analytical electronic balance (SHIMADZU, model ATY224, Shimadzu Philippines Manufacturing (SPM) Inc.).

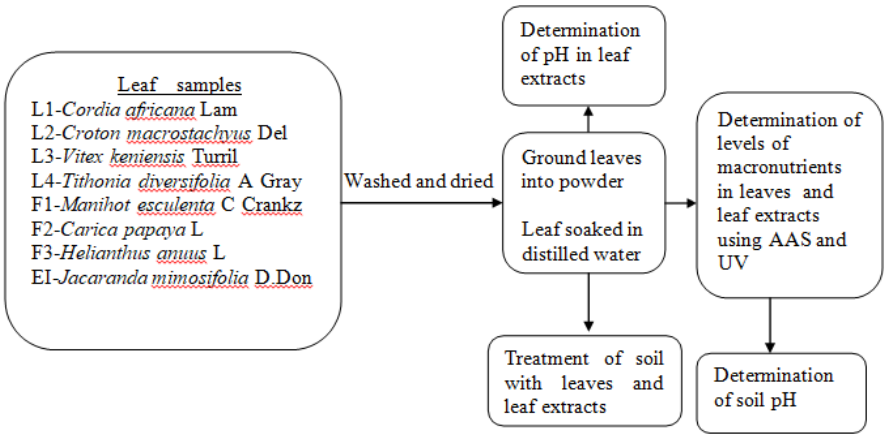

Figure 1. Flow diagram of research design

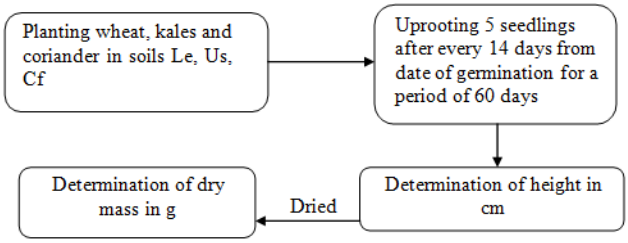

Figure 2. Monitoring of growth rates

Sample collection and sample pre-treatment

Leaves were sampled in April and May 2010 in Nyandarua County. It is rainy, and the plants are vegetative during this season. During this season, trees block sunlight from reaching immature food crops, necessitating their pruning. The leaves and leaf extract acquired for this investigation are used. The sample size was determined using Equation 1 from Gupta and Kapoor (1977):

$$n = \frac{s^2 t_x^2}{d^2 + s t_x^2 / N} \quad \text{.....Eq 1}$$

Where:

n: Sample size

t_x: Critical value of t for (n-1) degrees of freedom and at α: 0:-.05 level of significance, t_x is taken as 1.96.

s: Standard error of the mean

d: Margin of error (the acceptable difference between the sample mean and populationmean), here, taken as 0.04

N: Population size

If *N* is sufficiently large relative to n, Equation 1 reduces to Equation 2

$$n = \frac{s^2 t_x^2}{d^2} \quad \text{..... Eq 2}$$

A systematic sampling design was used to determine the farms from which the plant species would be obtained. First, a random sample technique was utilized to determine six plants from each plant species included in this study. The leaves were then collected from six distinct plants of the same species. Next, the leaves were thoroughly combined from various plants of the same type; sun-dried, placed in clean plastic bags, sealed and labeled suitably. Finally, the leaves plastic bags were placed in nylon sacks and brought to the Kenyatta university's research facility.

In February and March, soil samples were collected in Nyandarua County. After the long dry spell from December to the end of March, the soil is ripe for planting during these months (immediately before the long rains). The farms on which soil samples were gathered were identified through systematic sampling. There are two ways for soil sampling: traverse and zigzag. The traverse approach was used in this investigation. The field's four corners were established, and diagonal sampling was conducted. Twenty soil samples were taken from each farm identified using a soil auger. The soil samples were then properly mixed, and a sub-sample of the mixture was placed in a clean polythene bag for laboratory pH testing. The leftover soil sample was used to monitor the growth rate of the chosen non-leguminous plants. Dry leaves were rinsed with distilled water and sun-dried for three days before being pulverized into a fine powder to determine the macronutrient content. The same powder was used to remediate acidic soils containing non-leguminous plants.

Sample preparation

Preparation of leaf samples for analysis of N, P, K, Ca, and Mg

A bulk of 300 mg was oven dried at 70ºC for four hours. After ground, the leaves were placed in a clean and dry 125 mL "Pyrex" conical flask. A 4 mL quantity of strong sulfuric acid was added, and the flask swirled to soak the sample completely. The flask and contents were heated on an electric hot plate set to "medium" heat for ten minutes. The flask was withdrawn and allowed to cool slightly before adding 10 drops of 30% hydrogen peroxide,

three or four drops simultaneously, to avoid a strong reaction. The flask was spun while the contents remained at the bottom to avoid excessive heating. The flask was allowed to cool before six drops of hydrogen peroxide were carefully added. The flask was then reheated and chilled, and six drops of hydrogen peroxide were added. This procedure was repeated until the solution became colorless. The digest was transferred to a 100 mL volumetric flask and topped off to the desired volume. After that, it was moved to a clean, labeled plastic container and stored until analysis.

Preparation of leaf samples for analysis of sulfur

A quantity of 300 mg grounded leaves was weighed into a dry clean 125 mL "Pyrex" conical flask after being oven dried at 70ºC for 4 hours. A 4 mL quantity of strong nitric acid was added, and the flask swirled to moisten the material completely. The flask and contents were heated on an electric hot plate set to "medium" heat for ten minutes. After cooling the flask, 10 drops of 30% hydrogen peroxide were added, 3-4 drops at a time to avoid a strong reaction. The flask was spun while the contents remained at the bottom to avoid excessive heating. After allowing the flask to cool, six drops of hydrogen peroxide were carefully added and warmed. The chilling process was repeated while adding six drops of hydrogen peroxide until the solution became colorless. The digest was transferred to a 100 mL volumetric flask and diluted to the specified volume. Afterward, the digest solution was transferred to a clean, labeled plastic container and stored in the refrigerator for analysis.

Preparation of stock and standard solutions

Just prior to analysis, stock solutions for various elements were produced from standard solutions. Unless otherwise specified, all reagents used in this investigation were analytical grade. Thomas Baker Chemicals ltd., Mumbai, India, supplied concentrated nitric acid, sulfuric acid, hydrogen peroxide, potassium nitrate, and hydrochloric acid.

The final acid content was kept at roughly 1% throughout the dilutions to maintain the metal in the free ionic condition. Stock solutions were packaged in plastic bottles and clearly labeled. Each time analysis was performed, working standards were produced fresh from stock solutions using serial dilution (Equation 3).

$$C_1V_1 = C_2V_2 \quad \text{.....Eq 3}$$

Where:
C_1: Original concentration
V_1: Original volume
C_2: New concentration
V_2: New volume

A total of six blank samples were digested using the same procedure as the samples to account for background effects from the acids and correct changes resulting from digestion procedures. The elemental concentrations of the elements of interest (K, Ca, and Mg) in each blank sample were determined using an atomic absorption spectrophotometer. According to Christian (2005), their absorbances were measured, and their averages and standard deviations were determined, which were then used to calculate the limit of detection using the formula below:

$$\text{limit of detection} = \frac{3\text{x standard deviation of blank reading}}{\text{Absorbance of standard} - \text{mean absorbance of blanks}} \quad \text{..Eq 4}$$

Magnesium stock and standard solutions

It was necessary to generate a magnesium stock solution (1000 g/g) by dissolving 1.00 g of magnesium ribbon in 300 mL of 1:1 nitric acid and diluting the solution to make it the consistency of a liter with distilled water. A working magnesium standard solution (50 g/L) was prepared by diluting 10 mL of the stock solution to 200 mL and then resolving the solution. The calibration graph was created using a solution containing the following magnesium concentrations: 0, 0.25, 0.5, 1.0, 2.0, 4.0, and 5.0 µg/g of magnesium (in molar concentrations).

Calcium stock and standard solutions

A sum of 2.50 grams of dry calcium carbonate was dissolved in 30 mL of 1 M hydrochloric acid to make the calcium stock solution (1000 µg/g), and the volume was adjusted to 1 L with distilled water at 100ºC for two hours. The calibration curve was generated using standard solutions comprising 0, 5, 15, 20, 30, and 40 µg/g calcium, made by serial dilution of the stock solution.

Potassium stock standard solutions

A potassium stock solution (1000 µg/g) was made by dissolving 1.91 g of dry potassium chloride in approximately 100 mL of distilled water at 100ºC for two hours and diluting to one liter with distilled water. The calibration curve was constructed using potassium standards solutions containing 0, 1, 2, 4, 6, 8, and 10 µg/g potassium.

Nitrogen stock and standard solutions

In order to make a stock solution for nitrogen (with a concentration of 2500 µg/g), 1.179 g of ammonium sulfate was dissolved in water, and the volume was adjusted to one liter with distilled water. The calibration solution consisted of standard nitrogen solutions containing 0, 4, 8, 12, 16, 20, and 24 µg/g nitrogen.

Phosphorous stock and standard solutions

An oven-dried potassium orthophosphate solution (1.10 g) was dissolved in a 250 mL volumetric flask. The volume was then adjusted to the 250 mL mark with distilled water to produce a stock concentration of 1000 µg/g phosphorus. The calibration curve was constructed using standard solutions of phosphorous-containing 0, 1, 2, 3, 4, 5, 6, 7, and 8 µg/g.

Sulfur stock and standard solutions

A stock solution of sulfur (1000 µg/g) was produced by dissolving 2.72 g of potassium sulfate in 500 mL of distilled water and diluting it to 500 mL with distilled water. The calibration curve for sulfur was generated using standard solutions containing 0, 5, 10, 20, 30, 40, and 50 µg/g of sulfur.

Sulfate stock and standard solutions

In order to determine the mineralization of sulfates in a leaf water extract, we prepared a working standard stock solution by dissolving 0.13 g of oven-dried sodium sulfate in distilled water and diluting the solution to one liter with more of the same solution. Sulfates are present in this solution at a concentration of 90 µg/g. The calibration curve was constructed using standard sulfates solutions containing 0, 9, 18, 27, 36, 45, and 54 µg/g.

Sample analysis

Analytical performance of the various instruments to various elements

It was necessary to evaluate the analytical performance of the various equipment used. First, the calibration curves were created by plotting the absorbance of various standards against one another. Next, the slope of the calibration curves was used to determine the sensitivity of the analysis method used. Finally, the detection limit was calculated using the regression equation, as the lowest mean levels obtained by the instrumental signal were equal to the blank signal plus three times the standard deviation of the blank using the regression equation. Tables 1, 2, and 3 present a summary of the findings of this study.

Analytical performance of AAS (Varian spectra AA10) (2005)

Table 1 lists the analytical wavelengths, detection limits, correlation coefficients, and equations of the calibration curves for the measurement of metals in leaves and leaf extracts.

Evaluation of the linearity of the AAS was carried out using the calibration curves that had been established. First, the correlation coefficients (r) were calculated using the absorbance data and the concentration of standards. Then, we estimated the technique detection limits by multiplying the concentration of the provided signals by four times the standard deviations of each of the six blanks. Finally, in order to establish the calibration curves, a plot of absorbance values against the relevant concentration of standards was performed under optimal experimental circumstances.

Analytical performance of the flame photometry

The analytical performance of the flame photometry is shown in Table 2. The linearity of flame photometry was evaluated using the established calibration curves, which were subjected to regression analysis. The correlation coefficients (r) were calculated based on the absorbance values and the concentration of ideal standards employed in the experiments. The concentration of the given signals was computed as the standard deviations of the six blanks times four times the concentration of the given signals. In order to create the calibration curves, a plot of absorbance data against the relevant concentration of ideal standards was performed under optimal experimental conditions.

Analytical performance of T80+ UV/Visible spectrophotometer

The analytical performance of UV/visible spectrophotometry is shown in Table 3. A regression analysis was performed to determine the linearity of the calibration curves that had been established. The calibration curves were plotted using the absorbances and concentrations of ideal standards as input data. To compute the experimental detection limits, we used the regression equation to take the lowest concentration produced by the instrumental signal equal to the blank signal and multiply it by three times the standard deviation of the blank signal. Table 3 displays the findings of the study. The standard solutions created from stock solutions were used to establish UV/visible calibration curves, which were then utilized to determine the amounts of specified analytes in leaves and leaf extracts using the results of the experiments. Based on the data shown in Table 3, it can be stated that the linearity of the calibration curves produced is satisfactory, so accurate measurement can be ensured.

Calibration of pH meter

The pH meter was calibrated using solutions made from pH 4 and 7 tablets.

Table 1. Analytical performance of AAS for the analytes

Analyte	Wavelength (nm)	Range of standards (µg/g)	Sensitivity	Linearity (r2)	Detection limit (µg/g)	Equations for calibration curve
Ca	422.7	0-10	0.0024	0.996	0.038	y=0.01x+0.003
Mg	285.2	0-10	0.0512	0.999	0.032	y=0.084x+0.038

Note: y: absorbance; x: concentration

Table 2. Analytical performance of flame photometry for analytes

Analyte	Range of Standards (µg/g)	Sensitivity	Linearity (r2)	Detection limit (µg/g)	Equations for calibration curve
K	0-10	0.7718	0.996	0.041	y= 0.081x + 0.032

Note: y: absorbance; x: concentration

Table 3. Analytical performance of UV/visible spectrophotometry

Analyte	Range of standards (µg/g)	Sensitivity	Linearity (r2)	Detection limit (µg/)	Equations for calibration curve
Sulfate	0-10	0.0671	0.996	0.038	y = 0.084x + 0.038
Phosphate	0-10	0.5684	0.990	0.003	y = 0.269x + 0.048
Nitrates	0-5	0.2406	0.990	0.048	y = 0.269x + 0.048

Note: y: absorbance; x: concentration

Determination of magnesium by AAS (Varian spectra AA10)

After pipetting a 5 mL aliquot of the wet digested material into a 50 mL volumetric flask, the flask was filled with distilled water. The 285 nm laser beam length was used to nebulize the standards and samples and blank into an atomic absorption spectrophotometer flame to measure absorption. A calibration curve was drawn, and the concentrations of the sample and the blanks were measured (Okalebo et al. 2002).

Determination of calcium by AAS (Varian spectra AA10)

Pipetting a volume of 10 mL of wet digested sample solution into a 50 mL volumetric flask and then filling the flask with distilled water was conducted. A 422 nm laser beam was used to nebulize the standards, samples, and blanks into the flame of an atomic absorption spectrophotometer to measure their absorbance. A calibration curve was drawn, and the concentrations of the sample and the blanks were measured (Okalebo et al. 2002)

Determination of potassium by flame photometry Varian spectra (AA10)

A 5 mL aliquot of the wet digested sample was pipetted into a 50 mL volumetric flask, which was then filled to the top with distilled water. The results are recorded in the table below. A total of 10 mL of the diluted sample and blank were diluted further in a 1:2 ratio, resulting in a final volume of 10 mL. Nebulized into the flame photometer in random order were standards, samples, and a blank sample (Okalebo et al. 2002).

Determination of nitrogen by T80+ UV/Vis spectrometer

Nitrogen determination reagents N1 and N2 were produced. N1 was created by dissolving 34.00 g sodium salicylate, 25.00 g sodium citrate, 25.00 g sodium tartrate, and 0.12 g sodium nitroprusside in one liter of water. In order to make N2, 30.00 g sodium hydroxide was dissolved in water and adjusted to 1 L. Next, sodium hypochlorite was added to a 5 mL aliquot, shaken, and arranged into one liter. The two reagents were employed to treat the samples. First, 5 mL of wet digested samples and blanks were diluted in distilled water at a ratio of 1:9 (v/v). Next, 0.2 mL of diluted wet sample digests, standards, and blanks were pipetted in separate well-labeled test tubes using a micropipette. Each test tube was filled with 5.0 mL of reagent N1 and vortexed, followed by 5 mL of reagent N2. After two hours, the test tubes were allowed to stand, and the absorbance at 650 nm was determined using a PG instruments Ltd T80+ UV/Vis spectrometer (Okalebo et al. 2002).

Determination of phosphorous by T80+ UV/Vis spectrometer

Twelve grams of ammonium molybdate were dissolved in 250 mL warm distilled water (50°C). In a separate experiment, 0.29 g of antimony potassium tartrate was dissolved in 100 mL of distilled water. Both solutions were added to 100 mL of 2.5 M sulfuric acid, well mixed, and diluted with distilled water to two liters. Ammonium molybdate/antimony potassium tartrate was the resultant solution. A 2.11 g of ascorbic acid was dissolved in 400 mL of ammonium molybdate/antimony potassium tartrate solution and thoroughly mixed. A 5 mL sample that had been wet digested was pipetted into a 50 mL volumetric flask. 20 mL distilled water was added, followed by 10 mL ascorbic acid reducing agent and distilled water to make 50 mL. They were then allowed to stand for one hour to allow for complete color development, and their absorbance at 880 nm was determined using a UV/visible spectrophotometer (Okalebo et al. 2002). Additionally, blanks and standards were treated the same way.

Determination of sulfur by UV/Vis spectrophotometry

First, a gelatine-barium chloride solution was prepared by dissolving 0.60 g of gelatin in 200 mL of distilled water and allowing it to stand for 4 to 16 hours in a refrigerator. After bringing the semi-gelatinous fluid to room temperature, 2.00 g of barium chloride was added and well-mixed until dissolved. Next, a 10 mL volume of the digest was pipetted into a 50 mL volumetric flask; 2 mL of gelatin-barium chloride solution was added and diluted with distilled water to the desired concentration. After 30 minutes, absorbance at 420 nm was determined using a UV/visible spectrophotometer (Okalebo et al. 2002). Additionally, blanks and standards were treated the same way.

Determination of mineralization

On the 20th, 40^{th,} and 60th days, 100 mL of deionized water was filtered from the containers where 100 g of leaves had been soaked. It was decided to put the water in a freezer and then analyze it for the presence of potassium, calcium, magnesium, nitrates, phosphorus, and sulfates.

Determination of mineralization of K^+, Ca^{2+}, Mg^{2+}, NO_3^- and PO^{2-}

K^+, Ca^{2+}, and Mg^{2+} concentrations in leaf extracts were determined similarly for wet-digested samples. The concentrations of nitrates and phosphates in leaf extracts were determined in the same manner as for wet-digested

samples, except that the standards for the calibration curve ranged between 0-8 µg/g for nitrates and 0-3.2 µg/g for phosphate.

Determination of mineralization of sulfates by turbidimetry

The following turbidimetric approach was used to determine the quantity of sulfate in leaf water. First, the following solutions were prepared: (i) A glycerol-ethanol combination was prepared by mixing one volume of white glycerol water and two liters of 95% ethanol. (ii) A solution of sodium chloride and concentrated hydrochloric acid was prepared by adding 67.0000 g sodium chloride, and 8 mL concentrated hydrochloric acid to 200 mL water and shaking well.

A 10 mL volume of leaf extract was pipetted into a 50 mL volumetric flask and diluted with distilled water to the desired concentration. After preparation, standards, samples, and blanks (50 mL of each) were placed in clearly labeled beakers. Ten milliliters of glycerol-ethanol and 5 mL of sodium chloride–hydrochloric acid were added. A string bar was inserted, and a beaker was placed on a magnetic stirrer and vigorously swirled. A timer was used to time the addition of 0.02 g of barium chloride. After 60 seconds, the stirrer was stopped, and the absorbance at 420 nm was determined using a UV/visible spectrophotometer. Two samples or standards were run through this procedure once before proceeding to the next, and absorbance was determined within 3-6 minutes of starting the stopwatch (Okalebo et al. 2002).

Calculation of concentrations of elements in the samples

Equation 4 calculated the concentration of critical elements in the samples based on AAS results (readout).

$$\text{Actual concentration } (\mu g/g) = \frac{\text{Concentration } (\mu g/ml) \text{x Volume digested (ml)}}{\text{Weight of dried sample taken (g)}} \quad \text{....Eq 5}$$

When dilution was used, the actual weight was calculated by multiplying the readout values by the dilution factor. The means of the replicate measurements were determined from the actual concentration obtained. The Ca and Mg concentrations in the samples were determined by calculating their means ± standard deviations.

When sample readings were outside the optimum working range, known concentration standards were introduced to bring the sample readings inside this range. The absorbancies of the original sample and the standard were determined. The actual weight of the sample was determined using equation 6 by Skoog et al. (1998).

$$Cx = \frac{A1CsVs}{(A2 - A1)Vx} \quad \text{.....Eq 6}$$

Where:
Cx: Concentration of sample
Cs: Concentration of the standard
A1: Absorbance of the sample before addition of standard
A2: Absorbance of sample after addition of standard
Vs: Volume of standard added
Vx: Volume of sample solution

The pH of leaf extracts analysis

The pH of leaf samples soaked in distilled water was duplicated in the laboratory using a multiline P4 electrochemical analyzer equipped with a suitable multiline pH probe. The pH meter was calibrated using solutions prepared from pH 4 and 7 tablets. Each pill was dissolved in 30 mL distilled water in a 100 mL volumetric flask and agitated until dissolved. Following that, additional distilled water was applied to the spot. Next, a 30 mL sample of the leaf extract was placed in a plastic beaker, immersed with a pH probe, and the pH was determined. The probe was washed multiple times with distilled water following each sample measurement. The pH meter was calibrated again after every ten measurements to guarantee reliable data.

The pH of soil analysis

By weighing 40.00 g of soil into a beaker, the pH of soil treated with powdered leaves and leaf extracts was duplicated. It was then added 100 mL of distilled water mixed for 10 minutes, allowed to stand for 30 minutes, then swirled again for 2 minutes before determining the pH. Next, 30 mL of water was placed in a plastic beaker, the pH probe was immersed, and the pH was determined. After each sample measurement, the probe was washed multiple times with distilled water. Finally, the pH meter was calibrated once again to verify the data was reliable. The identical procedure was performed on soil that had not been treated and soil that had been treated with inorganic fertilizer. The procedure was repeated every 10 days for a total of 60 days.

The analysis of the growth rate of non-leguminous plants

The combined leaves and leaf extract mixture was determined using macronutrient concentrations and their effect on soil pH from this study and an earlier study (Murungi 1990; Njagi 2008). First, to maximize plant nutrition, leaves of *J. mimosifolia* (E1), *M. esculenta* (F1), *C. papaya* (F2), *H. annuus* (F3), *C. africana* (L1), and *T. diversifolia* (L4) were blended in the ratio 3:3:2:2:3:2. Along with treating the acidic soil with mixed leaves, 10 mL of *C. macrostachyus* (L2) and *V. keniensis* (L3) extracts were added at 10-day intervals for 60 days due to their high buffering ability. Next, wheat, kale, and coriander were planted in acidic soil that had been treated with mixed leaves at a mass ratio of 1850:150. In another set, the same plants were grown in acidic soil treated with NPK at a ratio of 1870:130 NPK, and in a third set, the plants were planted in acidic soil that had not been treated.

There were nine replicates of each non-leguminous plant labeled Le with a 2.5 kg capacity for acidic soil treated with combined leaves, nine replicates of each non-leguminous plant labeled FS with a 2.5 kg capacity for soil treated with NPK, and nine containers labeled Us with a 2.5 kg capacity for Us, totaling 27 sets of replicates. On the same day, 30 seeds of each plant were planted 1 cm apart in each container. Five seedlings of each plant were plucked from each container every 14 days from the date of germination for 60 days for each set of duplicates (Le, Cf, and Us). Wheat, kale, and coriander growth rates were

determined using an analytical balance for dry mass and the meter rule for height. Each set of seedlings was washed with tap water and dried under identical conditions, and the average dry mass (g) for each set of experiments was determined. For a total of 60 days, the experiment was reproduced every 14 days.

Data analysis

The data were subjected to statistical analysis, which included the mean, one-way ANOVA, standard deviation, and Student-Newman-Keuls (SNK) test, among other things. The methods are beneficial in determining the dependency of the variables as well as statistically significant differences between the stations (Miller and Miller 1988).

RESULTS AND DISCUSSION

Levels of potassium in leaves and leaf extracts

Through the use of flame photometry, the mean potassium levels in leaves and leaf extracts were determined. Table 4 contains the results of the study.

For potassium, *C. papaya* (F2) had the most potent levels (243.50±0.87 µg/g), while *J. mimosifolia* (E1) had the lowest (71.50±2.29 µg/g). Leaf extracts of *H. annuus* (F3) and *V. keniensis* (L3) showed the highest (18.22±0.38 µg/g) and lowest (10.22±2.38 µg/g) rates of mineralization by the 20th day, respectively. During the 40th day, the leaf extracts of *H. annuus* (F3) and *V. keniensis* (L3) yielded the highest and lowest mean potassium levels, respectively, at 23.43±0.15 and 37.02±1.35. *Helianthus annuus* (F3) 28.76±0.36 and *V. keniensis* (L3) 10.22±2.38 were the highest and lowest, respectively, on the 60th day ($p > 0.05$, SNK test). As the soaking duration increased, potassium mineralization increased in all cases (Table 4).

There have been prior studies showing that *M. esculenta* (F1) 181.67±4.17, *C. papaya* (F2) 243.50±0.87 and *C. africana* (L1) 189.95±5 have high levels of potassium in their leaves, which agree with this study (Lawrence 1990 and Njagi 2008). *Cordia africana* (L1), *M. esculenta* (F1), and *C. macrostachyus* (L2) all had elevated potassium levels in their leaves. That suggests that the leaves of plant species with high mean potassium levels can substitute for expensive bagged inorganic fertilizers. *Carica papaya* (F2) leaves contain 6087 µg/kg of potassium, within the suggested range of 10000-24000 µg/kg for potassium-enriched soil (Lawrence 1990). Plant potassium absorption is 100 g/kg per year (Lawrence 1990). Building protein and photosynthesis, as well as the quality of fruits and the decrease in diseases, are all benefits of potassium. Results show that the combination of leaves and leaf extracts can be employed as a soil supplement for potassium since potassium levels were high in the composite mixture of combined leaves and leaf extracts.

Levels of calcium in leaves and leaf extracts

The mean levels of calcium that were determined are given in Table 5. From 49.33±0.17 µg/g in *J. mimosifolia* (E1) to 367.33±0.67 µg/g *C. africana* (E2), the leaf calcium concentrations differed widely (L1). *Manihot esculenta* (F1) 24.22±0.15, *H. annuus* (F3) 17.16±0.21 and *T. diversifolia* (L4) 28.01±0.24 exhibited the highest rate of calcium mineralization by the 20th day of the extraction. *Helianthus annuus* (F3) was at its maximum level on the 40th and 60th days, whereas *C. africana* (L1) was at its lowest. Soaking time tended to improve calcium mineralization in general (Table 5).

The annual calcium uptake by plants is 170 µg/kg, which is lower than the 1700-24000 µg/kg levels recommended for calcium-enriched soil (Groot et al. 1991). The soil calcium content of *C. africana* (L1), *V. keniensis* (L3), and *M. esculenta* (F1) was found to be 9183.25 µg/kg, indicating that these plants are good organic suppliers of calcium. When calcium levels in soils are too low, leaves can be used in place of more expensive sources such as dolomitic lime or gypsum in order to meet the plant's annual absorption requirements. According to (Njagi 2008), *H. annuus* (F3) had the highest concentration of Ca^{2+}, whereas *C. africana* (L1) had the lowest concentration.

Levels of magnesium in leaves and leaf extracts

Magnesium levels in all leaves and leaf extracts have been analyzed using atomic absorption spectroscopy (AAS). For *T. diversifolia* (L4), the average concentration of Mg^{2+} in leaves was 11.50±0.17 µg/g; for *H. annuus* (L4), it was 114.33±0.33 µg/g (F3). On the 20th day, the leaf extract for *C. africana* (L1) 2.23±0.29 and *H. annuus* (F3) 12.34±0.10 were the lowest and highest, respectively. For *C. africana* (L1), Mg^{2+} levels ranged from 2.43±0.10 to 12.78±0.25 for *V. keniensis* (L3) on day 60. (Table 6).

As a soil supplement for Mg^{2+}, the best trend for *H. annuus* (F3) and *V. keniensis* (L3) was identified throughout the study period. *Helianthus annuus* (F3) and *C. papaya* (F2) showed the highest concentrations of Mg^{2+} in this investigation, which is consistent with the findings of Njagi (2008). In soils where Mg^{2+} deficiency is evident, they were proven to be a good supplement Mg^{2+} concentrations in leaf extracts were nearly identical because it is the only metal found in chlorophyll (Table 6).

Levels of nitrate in leaves and leaf extracts

Nutrient levels in leaves and leaf extracts were measured utilizing UV/visible spectrophotometers. Nitrate levels in leaves ranged from 26.40±0.35 µg/g *H. annuus* (F3) to 322.25±0.40 µg/g *T. diversifolia* (L4) on average (Table 7). *T. diversifolia* (L4) had the highest nitrate-nitrogen 322.25±0.40 levels, whereas leaves from *H. annuus* (F3) had the lowest 26.40±0.35 values, according to the data (Table 4). Leaf extract levels ranged from 0.19±0.20 *V. keniensis* (L3) to 1.37±0.39 *M. esculenta* (F1) during the 20th day of the experiment. Nitrate-nitrogen levels ranged from 0.41±0.18 µg/g *H. annuus* (F3) to 1.39±0.21 µg/g *M. esculenta* (F1) on the 40th day of testing (Table 7).

Table 4. Mean levels (µg/g) of K^+ in leaves and leaf extracts

Plant species	Leaves K^+(mean±SE)	Leaf extracts		
		20th-day K^+(mean±SE)	40th-day K^+(mean±SE)	60th-day K^+(mean±SE)
J. mimosifolia (E1)	71.50±2.29^a	17.32±0.36^f	17.96±0.18^c	19.89±0.31^b
M. esculenta (F1)	181.67±4.17^e	11.20±0.38^d	18.28±0.36^c	18.93±0.15^b
C. papaya (F2)	243.50±0.87^f	13.13±0.36^e	16.99±0.35^c	28.76±0.36^f
H. annuus (F3)	76.23±4.29^a	18.22±0.38^e	23.43±0.15^e	25.04±0.18^d
C. africana (L1)	189.95±5.17^e	13.13±0.28^e	21.12±0.38^d	22.79±0.23^c
C. macrostachyus (L2)	170.83±0.60^d	8.95±0.28^c	16.35±3.33^c	19.25±0.11^b
V. keniensis (L3)	143.83±0.33^c	3.80±0.30^a	7.02±1.35^a	10.22±2.38^a
T. diversifolia (L4)	106.50±0.29^b	6.05±0.37^b	11.52±0.35^b	27.29±0.37^e

Note: Mean values with the same letters within the same column are not significantly different ($p > 0.05$, SNK test)

Table 5. Mean levels (µg/g) of Ca^{2+} in leaves and leaf extracts

Plant species	Leaves Ca^{2+}(mean±SE)	Leaf extracts		
		20th day Ca^{2+}(mean±SE)	40th day Ca^{2+}(mean±SE)	60th day Ca^{2+}(mean±E)
J. mimosifolia (E1)	49.33±0.17^a	4.87±0.32^b	5.23±0.27^b	7.57±0.21^b
M. esculenta (F1)	230.83±0.93^d	24.22±0.15^e	29.66±0.30^d	37.53±0.18^d
C. papaya (F2)	185.50±0.58^c	3.66±0.27^b	5.07±0.27^b	8.74±0.27^c
H. annuus (F3)	316.50±0.50^e	17.16±0.21^d	34.06±0.37^f	39.99±0.18^f
C. africana (L1)	367.33±0.67^e	2.34±0.30^a	2.45±0.21^a	3.42±0.30^a
C. macrostachyus (L2)	193.83±0.73^c	13.86±0.18^c	21.76±0.40^c	38.26±0.27^e
V. keniensis (L3)	114.83±0.67^b	14.21±0.32^c	28.26±0.27^d	38.91±0.40^e
T. diversifolia (L4)	138.00±0.58^b	28.01±0.24^f	31.12±0.24^e	38.33±0.24^e

Mean values with the same letters within the same column are not significantly different($p > 0.05$, SNK test)

Table 6. Levels of Mg^{2+} (µg/g) in leaves and leaf extracts (20^{th}, 40^{th}, and 60^{th})

Plant species	Leaves Mg^{2+}(m±SE)	Leaf extracts		
		20^{th} day Mg^{2+}(m±SE)	40^{th} day Mg^{2+}(m±SE)	60^{th} day Mg^{2+}(m±SE)
J. mimosifolia (E1)	12.67±1.33^a	3.18±0.20^b	3.33±0.25^b	3.35±0.11^b
M. esculenta (F1)	30.17±1.17^c	6.04±0.20^d	6.09±0.33^d	6.12±0.37^d
C. papaya (F2)	41.00±0.17^d	3.75±0.29^b	4.21±0.38^c	4.29±0.20^c
H. annuus (F3)	114.33±0.33^e	12.34±0.10^f	12.38±0.33^f	12.58±0.25^f
C. africana (L1)	28.17±0.17^b	2.23±0.29^a	2.33±0.26^a	2.43±0.10^a
C. macrostachyus (L2)	30.33±1.17^c	5.02±0.26^c	12.23±0.20^f	12.22±0.26^f
V. keniensis (L3)	29.67±0.17^c	11.84±0.11^f	12.69±0.31^f	12.78±0.25^f
T. diversifolia (L4)	11.50±0.17^a	9.17±0.26^e	9.67±0.26^e	9.90±2.33^e

Note: Mean values with the same letters within the same column are not significantly different ($p > 0.05$, SNK test)

Nitrates-nitrogen levels in *T. diversifolia* (L4) were determined to be within the required range of soil enriched with nitrogen 2000-10000 µg/kg since it contained 8056.25 µg/kg. This study found that *T. diversifolia* (L4), a good organic nitrogen source, could supply 100 µg/kg of nitrogen per year to plants. According to what was reported, this agrees with Njagi (2008). Additionally, our findings support the use of *T. diversifolia* (L4) as a top dressing because it decomposes quickly and releases minerals into the water supply (Nancy and Mary 1990).

Levels of phosphate-phosphorous in leaves and leaf extracts

UV/visible spectroscopy was used to estimate the mean amount of phosphate-phosphorous in leaves and leaf extracts; the results are given in Table 8.

Helianthus annuus (F3) to *T. diversifolia* (L4) had mean phosphate-phosphorous levels of 10.60±4.31 µg/g in leaves. Leaf extracts from *C. papaya* (F2) and *H. annuus* (F3) yielded the highest phosphate-phosphorous mineralization levels on the 20^{th} day. Plant leaves of *C. papaya* (F2) had the greatest mean levels of phosphate-phosphorous on the 40^{th} day, whereas those of *H. annuus* (F3) had the lowest. During the 60^{th} day, *M. esculenta* (F1) leaf extract levels averaged 2.03±0.11 µg/g, while *H. annuus* (F3) levels averaged 4.35±0.32 µg/g. Increased soaking time increased the content of leaf extract (Table 8).

Tithonia diversifolia (L4) and *M. esculenta* (F1), which have relatively high phosphorus levels and are readily available as food and fodder crops, can be used to collect phosphorus from the leaves. Phosphorous levels were found to match those of Njagi (2008) closely.

Level of sulfate-sulfur in leaves and leaf extracts

The turbidimetric technique was used to determine the mean concentrations of sulfate-sulfur in leaves and leaf

extracts. Table 9 shows the results. From 12.75±0.40 µg/g in *M. esculenta* (F1) to 56.48±0.23 in *J. mimosifolia* (E1), the leaves of these plants contained various levels of sulfur. By the 20th day, leaf extracts of *V. keniensis* (L3) and *J. mimosifolia* (E1) contained between 0.13±0.24 µg/g and 1.37±0.23 µg/g, respectively. *Helianthus annuus* (F3) varied from 0.27± 0.35 µg/g to 1.49± 0.29 µg/g in *J. mimosifolia* (E1) and 0.52± 0.36 µg/g in *H. annuus* (F3) to 1.99± 0.33 µg/g for *C. macrostachyus* (L2) by the 40th day, respectively (Table 9).

Helianthus annuus (F3) leaves provided 778 µg/kg of sulfur, compared to *C. papaya* (F2) leaves which provided 762 µg/kg. High sulfur demand necessitates higher leaf application because this supply is below recommended amounts of sulfur (1500-1600 µg/kg). Feeding plants with the foliar sulfur deficit is possible by applying leaf extracts. As the soaking duration grew, so did the leaf extract concentration, which is the data in Table 9. This study found that the leaves of *H. annuus* (F3) and *C. papaya* (F2) can be used as an alternative organic source of sulfur. According to Murungi (1990) and Njagi (2008), sulfur levels in this study were extremely low for all species.

Recommended levels of the plant nutrients and plant leaves that are mostsuitable

Detailed macronutrient needs for plant growth and leaf quality are summarized in Table 10. It was shown that *M. esculenta* (F1), *C. papaya* (F2), and *H. annuus* (F3) had the largest occurrences in the supply of macronutrients (P, K and Ca), (Ca, Mg and S) and (K, Mg, and S), sequentially. On the other hand, *Cordia africana* (L1) and *T. diversifolia* (L4) exhibit high quantities of potassium, calcium, nitrogen, and phosphorus. Therefore, the ideal composition is made up of *C. papaya* (F2) for potassium, *H. annuus* (F3) for magnesium and sulfur, *C. africana* (L1) for calcium, and *T. diversifolia* (L4) for nitrogen and phosphorus to ensure a sufficient supply of macronutrients, (Table 10).

Table 7. Mean levels of NO_3^- (µg/g) in leaves and leaf extracts

Plant species	Leaves NO_3^-(m±SE)	Leaf extracts		
		20th day NO_3^-(m±SE)	40th day NO_3^-(m±SE)	60th day NO_3^-(m±SE)
J. mimosifolia (E1)	93.06±0.59^{e}	0.31±0.22^{a}	0.42±0.12^{a}	2.12±0.16^{d}
M. esculenta (F1)	68.17±0.21^{d}	1.37±0.39^{b}	1.39±0.21^{b}	1.84±0.12^{c}
C. papaya (F2)	71.58±0.11^{e}	0.40±0.19^{a}	0.69±0.20^{a}	0.89±0.13^{a}
H. annuus (F3)	26.40±0.35^{a}	0.23±0.11^{a}	0.41±0.18^{a}	1.25±0.17^{b}
C. africana (L1)	40.25±0.02^{b}	0.22±0.22^{a}	0.43±0.19^{a}	2.22±0.27^{d}
C. macrostachyus (L2)	30.47±0.08^{b}	0.21±0.16^{a}	0.43±0.33^{a}	1.23±0.17^{b}
V. keniensis (L3)	50.65±0.24^{c}	0.19±0.20^{a}	0.58±0.23^{a}	0.89±0.19^{a}
T. diversifolia (L4)	322.25±0.40^{f}	0.23±0.26^{a}	0.61±0.16^{a}	2.75±0.29^{e}

Note: Mean values with the same letters within the same column are not significantly different ($p > 0.05$, SNK test)

Table 8. Mean levels (µg/g) of PO_4^{3-} in leaves and leaf extracts

Plant species	Leaves PO_4^{3-} (m±SE)	Leaf extracts		
		20th day PO_4^{3-}(m±SE)	40th day PO_4^{3-}(m±SE)	60th day PO_4^{3-}(m±SE)
J. mimosifolia (E1)	38.90±0.31^{d}	1.42±0.33^{a}	1.83±0.26^{a}	2.08±0.30^{a}
M. esculenta (F1)	49.47±0.29^{e}	1.28±0.14^{a}	1.79±0.17^{a}	2.03±0.11^{a}
C. papaya (F2)	32.37±0.31^{c}	3.05±0.20^{d}	3.45±0.15^{c}	3.72±0.15^{c}
H. annuus (F3)	10.60±4.31^{a}	3.14±0.33^{d}	3.59±0.14^{c}	4.35±0.32^{d}
C. africana (L1)	28.02±1.54^{c}	1.92±0.26^{c}	3.28±0.20^{c}	3.83±0.17^{c}
C. macrostachyus (L2)	18.38±0.31^{b}	1.96±0.30^{c}	2.69±0.33^{b}	3.53±0.14^{c}
V. keniensis (L3)	37.97±0.31^{d}	1.97±0.14^{c}	2.89±0.33^{b}	2.98±0.30^{b}
T. diversifolia (L4)	55.38±0.31^{f}	2.35±0.30^{b}	2.89±0.20^{b}	3.56±0.33^{c}

Note: Mean values with the same letters within the same column are not significantly different ($p > 0.05$, SNK test)

Table 9. Mean levels (µg/g) of sulfate-sulfur in leaf extracts

Plant species	Leaves SO_4^{2-}(m±SE)	Leaf extract		
		20th day SO_4^{2-} (m±SE)	40th day SO_4^{2-} (m±SE)	60th day SO_4^{2-}(m±SE)
J. mimosifolia (E1)	56.48±0.23^{e}	1.37±0.23^{a}	1.49±0.29^{d}	1.94±0.38^{d}
M. esculenta (F1)	12.75±0.40^{a}	0.97±0.40^{a}	1.31±0.33^{c}	1.49±0.18^{c}
C. papaya (F2)	30.49±0.21^{d}	0.68±0.11^{a}	1.33±0.30^{c}	1.94±0.36^{d}
H. annuus (F3)	31.14±0.11^{d}	0.17±0.23^{a}	0.27±0.35^{a}	0.52±0.36^{a}
C. africana (L1)	19.14±0.08^{b}	0.74±0.38^{a}	1.15±0.23^{b}	1.64±0.15^{c}
C. macrostachyus (L2)	19.64±0.24^{b}	0.99±0.11^{a}	1.12±0.29^{b}	1.99±0.33^{d}
V. keniensis (L3)	19.74±0.40^{b}	0.13±0.24^{a}	0.35±0.29^{a}	1.29±0.40^{b}
T. diversifolia (L4)	23.52±0.35^{c}	0.74±0.38^{a}	0.97±0.35^{b}	1.48±0.20^{b}

Note: Mean values with the same letters within the same column are not significantly different ($p > 0.05$, SNK test)

Table 10. Macronutrients and the best sources from the plants studied

Macronutrients	Common range levels in soil µg/kg	Recommended range enriched soil µg/kg	Annual uptake µg/kg	Best leaves for macronutrients
N	2000-4000	2000-10000	100	*T. diversifolia* L4
P	100-5000	400-2000	20	*T. diversifolia* L4, *M. esculenta* F1, *J. mimosifolia* E1 and *V. keniensis* L3
K	400-5000	10000-15000	100	*C. papaya* F2, *C. africana* L1, *M. esculenta* F1, and *C. macrostachyus* L2
Ca	400-35000	1700-24000	170	*C. africana* L1, *H. annuus* F3, and *M. esculenta*
Mg	600-15000	9000-15000	15	*H. annuus* F3 and *C. papaya* F2
S	100-10000	1500-1600	15	*H. annuus* F3 and *C. papaya* F2

Source: Groot et al. (1991)

The pH of leaf extracts

Table 11 shows the pH values of leaf extracts from various plant species, as measured by the researchers. There was no statistically significant difference in the pH of the leaf extract on day 0 ($p > 0.05$, SNK test). On the other hand, by day 15, the value had dropped from 6.67±0.04 to 4.58±0.27. Leaves of *C. africana* (L1) 6.21±0.23, *T. diversifolia* (L4) 6.07±0.23, and *H. annuus* (F3) 6.19±0.32 were extracted on day 15. It was observed on both days 30th and 45th. *Helianthus annuus* (F3) had a pH of 6.25±0.16 while *C. africana* (L1) had a pH of 7.44±0.04. Leaf extracts from *C. africana* (L1) have a pH of 6.21±0.23 to 7.44±0.04, which indicates that they are most suited for treating acidic soils. The pH of *J. mimosifolia* (E1), *C. papaya* (F2), and *M. esculenta* (F1) fluctuated from 4.58±0.27 to 5.49±0.23 throughout the 60 days (Table 11).

By the 60th day, the greatest pH obtained was 7.44 ±0.04 for *C. africana* (L1), and the lowest was 4.58 ±0.27 for *J. mimosifolia* (E1). By the 50th day, the pH of leaf extracts for all species had decreased from 6.67 to below 6.00, except for *H. annuus* (F3), *V. keniensis* (L3), and *T. diversifolia* (L4) (Table 11). The trend in this study for leaf extracts is consistent with that observed in (Njagi 2008; Murungi 1990), where pH decreased and then raised or reduced depending on the type of leaves and the duration of soaking in distilled water.

The pH of *C. africana* (L1) increased over 7.00 over 60 days. The pH of *C. africana* (L1), *J. mimosifolia* (E1), *C. papaya* (F2), and *M. esculenta* (F1) did not increase above 5.50 following the first decline. Several species, including *J. mimosifolia* (E1), *C. africana* (L1), *H. annuus* (F3), *V. keniensis* (L3), *T. diversifolia* (L4), and *C. macrostachyus* (L2), had pH levels rise over 5.50 following the first decline (Figure 3).

Changes in soil pH with the type of soil treatment

It was determined that a pH of 4.75 was used as a control for the acidic soil sample prior to treatment. At the beginning and end of the study, the pH levels of the soil were measured using a pH meter for 60 days. Table 12 lists the pH values of the soils.

It was found that after 50 days, there was a range in the pH of the Us from 4.75±0.02 to 4.02±0.02. pH rose from 4.02 to 4.12 by the 60th day of the study. Leaves and leaf extracts (le) increased the soil's pH from 4.75±0.01 on the 10th day to 6.69±0.01 on the 60th day. SNK test results showed a substantial decrease in pH from 4.75±0.01 to 4.43±0.02 by day 10 and an increase to 4.45±0.02 by day 20 following treatment with NPK. pH decreased from 4.42±0.01 by the 30th day to 4.42±0.01 by the 40th day, and finally declined to 3.98±0.01 by the 50th day and then increased to 4.12±0.01 by the 60th day. Table 12 shows that by the 10th day, the pH of all soils had lowered.

The results corroborate what was stated by (Njagi 2008) that *C. africana* (L1), *C. macrostachyus* (L2), *T. diversifolia* (L4), and *V. keniensis* (L3) increased the pH of the soil to 6.0 or greater for liming. Furthermore, the leaves and leaf extracts of F3, *V. keniensis* (L3), *T. diversifolia* (L4), and *C. africana* (L1) were found to be the most effective in raising the pH of acidic soil, as indicated in Tables 12 and Figure 4.

Table 11. The pH of leaf extracts on days 0, 15, 30, 45 and 60

Plant species	Days				
	0	15	30	45	60
J. mimosifolia (E1)	6.67±0.04^{a}	4.58±0.27^{a}	4.60±0.24^{a}	4.97±0.13^{a}	5.38±0.04^{a}
M. esculenta (F1)	6.67±0.03^{a}	5.40±0.13^{b}	4.87±0.28^{a}	4.90±0.06^{a}	5.12±0.03^{a}
C. papaya (F2)	6.67±0.07^{a}	5.34±0.15^{b}	5.50±0.28^{b}	5.49±0.23^{b}	5.26±0.07^{a}
H. annuus (F3)	6.67±0.08^{a}	6.19±0.32^{d}	6.25±0.16^{c}	5.78±0.06^{c}	6.42±0.08^{c}
C. africana (L1)	6.67±0.04^{a}	6.21±0.23^{d}	7.04±0.25^{d}	7.28±0.05^{f}	7.44±0.04^{d}
C. macrostachyus (L2)	6.67±0.18^{a}	5.49±0.15^{b}	5.85±0.47^{b}	6.27±0.08^{d}	6.00±0.18^{b}
V. keniensis (L3)	6.67±0.05^{a}	5.78±0.24^{b}	5.66±0.06^{b}	5.78±0.28^{c}	6.45±0.05^{c}
T. diversifolia (L4)	6.67±0.20^{a}	6.07±0.23^{d}	6.32±0.47^{c}	6.67±0.12^{e}	6.79±0.20^{c}

Note: Mean values with the same letters within the same column are not significantly different ($p > 0.05$, SNK test)

Table 12. the pH of untreated soil, soil treated with NPK, and soil treated with leaves and leaf extracts

Soils	Days						
	0	10	20	30	40	50	60
Us	4.75±0.02^{a}	4.45±0.01^{b}	4.42±0.01^{a}	4.21±0.01^{a}	4.12±0.01^{a}	4.02±0.02^{b}	4.12±0.01^{a}
Le	4.75±0.01^{a}	4.34±0.02^{a}	4.73±0.01^{c}	5.67±0.02^{c}	5.67±0.02^{c}	6.75±0.01^{c}	6.69±0.01^{b}
Cf	4.75±0.01^{a}	4.43±0.02^{b}	4.45±0.01^{b}	4.42±0.02^{b}	4.42±0.01^{b}	3.98±0.01^{a}	4.12±0.01^{a}

Note: Mean values with the same letters within the same column are not significantly different (p > 0.05, SNK test)

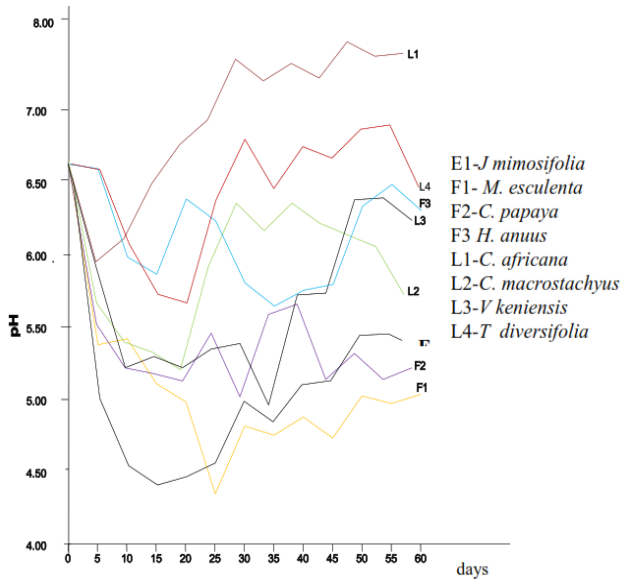

Figure 3. pH trends for leaf extracts

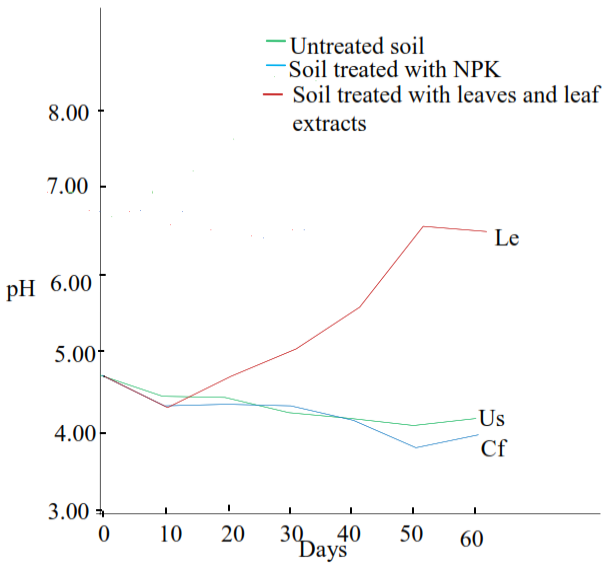

Figure 4. pH trends of soils Le, Cf, and Us

When compared to both NPK-treated and untreated soil, soil treated with leaves and leaf extracts had the highest pH value of 4.75±0.01 after treatment of acidic soil. While NPK treatment had a pH of 3.98 on the 50th day, the soil with mixed leaves and leaf extract had the highest pH. There was a noticeable increase in soil pH after applying leaves and leaf extracts. When NPK was applied, the pH declined to its lowest value of 3.98±0.01 on the 60th day (Table 12 and Figure 4), which suggests NPK was the likely cause of this drop given the Us had a slightly higher pH of 4.02±0.22 by the 50th day (Table 12). The effect of combined leaves and leaf extracts on the growth rate of *T. aestivum* (wheat)

Results of *T. aestivum* (wheat) growth rates in soils treated with leaves and Le, soil treated with Cf, and Us are presented in Table 13. In soil treated with leaves and leaf extracts, *T. aestivum* (wheat) grew at 1.27±0.13 g/wk dry mass and 7.29±1.43 cm/wk height, while in soil treated with Cf, it grew at 1.26±0.12 g/wk dry mass and 6.20±1.9 cm/wk height. The growth rate of *T. aestivum* (wheat) in the Us was 0.32±0.02 g/wk, compared to 1.27±0.13 g/wk in soil treated with leaves and leaf extracts 1.26±0.12 g/wk in soil treated with Cf. Cf leaves and leaf extracts had little effect on *T. aestivum* (wheat) (Table 13). That could occur because the macronutrients in the soil treated with leaves, leaf extracts, and Cf were practically equal, or both contributed appropriate macronutrients.

Those grown in the Us had a mean height of 3.98±0.97 cm/wk, and those grown with Cf had a mean height of 6.20±0.19 cm/wk, respectively (Table 13). The growth rates of *T. aestivum* (wheat) in Us 3.98±0.97 cm/wk, leaves and leaf extracts 7.29±1.43 cm/wk, and Cf 6.20±1.95 cm/wk were significant (p>0.05, SNK test). The macronutrient amounts in the three studies may have contributed to the observed differences in *T. aestivum* height (wheat). The Le included macronutrients that increased *T. aestivum* height (wheat). Figure 5 shows *T. aestivum* (wheat) cultivated on soil with Le, Cf, and Us.

The effect of combined leaves and leaf extract on growth rate *B. acephala* (kale)

For 60 days, researchers tracked the growth of *B. acephala* (kale) in soil treated with Le, Cf, and Us (Table 14). When grown in Cf, *B. acephala* (kale) dry mass increased at a rate of 0.300.02 g/wk, followed by Le, and the least at 0.03±0.01 g/wk when grown in Us. In soil treated with Le, *B. acephala* (kale) grew at a rate of 0.20±0.01 g/week, while Cf at a rate of 0.30±0.02 g/week showed no significant difference (p>0.05 in the SNK test). Growth rates of *B. acephala* (kale) in soil treated with Cf were the fastest, with growth rates of 2.57±0.88 cm/wk, and the slowest, with growth rates of 1.04±0.50 cm/wk in the Us. On the other hand, *Brassica acephala* (kale) grew rapidly in soil treated with Le, with a growth rate of 2.15±0.85 cm/week in soil treated with Le. That is, macronutrient concentrations were high in both the soil treated with Cf and the soil treated with Le, indicating that the soil was supplying enough nutrients to meet plant

demand. According to the findings of this study, *B. acephala* (kale) responded favorably to the leaves and leaf extracts (Table 14), and its heights were comparable to those of plants grown with NPK fertilization. As indicated in Figure 6, *B. acephala* (kale) was grown in soil treated with Le, in Us, and soil treated with nitrogen, phosphorus, and potassium (NPK) fertilizer.

The effect of combined leaves and leaf extract on the growth rate of *C. sativum* (coriander)

To see how *C. sativum* (coriander) grows in soil treated with Le, Cf, and Us, see Table 15. SNK tests showed no statistically significant difference (p>0.05) in the growth rate of the dry mass of *C. sativum* (coriander) growing in soil treated with combined Le 0.06±0.01 g/wk, soil treated with Cf 0.17±0.02 g/wk and Us 0.01±0.001 g/wk. There was a 1.86±0.58 cm/wk growth rate in soil treated with Le and a 0.79±0.57 cm/wk growth rate in the Us (Table 15).

Compared to Us, soil treated with Cf or soil treated with a mixture of Le showed the fastest growth rate for *C. sativum* (coriander) dry mass. For *C. sativum* (coriander), soil treated with Cf grew faster than soil treated with combined Le, because soil treated with combined leaves and leaf extracts took longer to provide macronutrients for the plants. In contrast, Cf delivered them directly to the soil (Figure 2). Therefore, excessive use of Cf on the soil of *C. sativum* plants results in lower yields than using a combination of Cf and leaves and leaf extracts (Figure 7).

Table 13. Mean growth rate of *T. aestivum* (wheat) dry mass (g)

Soil type	Dry mass (Mean±SE) g/wk	Height (Mean±SE) cm/wk
Us	0.32±0.02^{a}	3.98±0.97^{a}
Cf	1.27±0.13^{b}	6.20±1.95^{b}
Le	7.29±1.43^{b}	7.29±1.43^{b}

Note: Mean values with the same letters within the same column are not significantly different (p > 0.05, SNK test)

Table 14. Mean growth rate of *B. acephala* (kale) dry mass (g)

Soil type	Dry mass (Mean±SE) g/wk	Height (Mean±SE) cm/wk
Us	0.03±0.01^{b}	1.04±0.50^{a}
Cf	0.30±0.02^{a}	2.57±0.88^{b}
Le	0.20±0.01^{a}	2.15±0.85^{c}

Note: Mean values with the same letters within the same column are not significantly different (p > 0.05, SNK test)

Table 15. Mean growth rate of *C. sativum* (coriander) in terms of dry mass (g)

Soil type	Dry mass (Mean±SE) g/wk	Height (Mean±SE) cm/wk
Us	0.01±0.01^{a}	0.79±0.57^{a}
Cf	0.17±0.02^{b}	1.86±0.58^{b}
Le	0.06±0.01^{a}	1.85±0.56^{b}

Note: Mean values with the same letters within the same column are not significantly different (p > 0.05, SNK test)

Le **Cf** **Us**

Figure 5. *Triticum aestivum* (wheat) grown in soil treated with Le, Cf, and Us

Le **Cf** **Us**

Figure 6. *Brassica acephala* (kale) grown in soil treated with Le, Cf, and Us

Le **Cf** **Us**

Figure 7. *Coriandrum sativum* (coriander) grown in soil treated with Le, Cf, and Us

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