International Journal of Food Science and Nutrition
www.foodsciencejournal.com
ISSN: 2455-4898
Received: 14-04-2026, Accepted: 15-05-2026, Published: 30-05-2026
Volume 11, Issue 2, 2026, Page No. 164-172

Assessing the effect of length of maturity on the nutritional and anti-Nutrient Content of Local Cassava (Manihot Esculenta Crantz)

Onifade Gbenga S[1*], Tobiloba Timilehin D[2], Awe Omotola[1], Talabi Justina Y[3]
[1] Department of Nutrition and Dietetics, College of Technology, Ekiti State College of Technology P.M.B., Ijero Ekiti, Ekiti State, Nigeria
[2] Department of Food Science and Technology, Bamidele Olumilua University of Education Science and Technology, P.M.B., Ikere-Ekiti, Nigeria.
[3] Department of Human Nutrition and Dietetics, College of Medicine and Health Sciences, Afe Babalola University, Ado-Ekiti, Ekiti State, Nigeria
Corresponding Author: Tobiloba Timilehin Daniel

Abstract
This study evaluated the effect of maturity length on the nutritional and antinutrient composition of local cassava (*Manihot esculenta* Crantz). Fresh cassava roots were harvested at three maturity stages (CS1–CS3) and processed into samples for analysis. Proximate composition (moisture, crude protein, fat, fibre, ash, and carbohydrate by difference) was determined using standard AOAC methods. Mineral elements were analyzed using flame photometry and atomic absorption spectrophotometry, while phosphorus was determined colorimetrically. Vitamins (B-complex, A, and C) were quantified using spectrophotometric methods, and antinutrients (phytate, oxalate, tannin, saponin, total polyphenols, and hydrogen cyanide) were determined using established analytical procedures. All analyses were conducted in duplicate, and data were subjected to statistical analysis to determine significant differences at $p > 0.05$. Results showed no significant differences ($p > 0.05$) across all parameters, indicating that maturity stage had minimal influence on cassava composition. Proximate analysis confirmed cassava as a carbohydrate-rich crop (28.45–28.51%) with high moisture content (66.52–66.61%) but low protein (1.48–1.69%) and fat (0.41–0.47%). Mineral and vitamin contents were generally low but stable across samples. Antinutrients such as phytate (0.083–0.089%) and oxalate (0.032–0.037%) were present in low amounts, while hydrogen cyanide levels (257.31–257.36 mg/kg) were relatively high. In conclusion, cassava maturity stage does not significantly affect its nutritional or antinutritional composition. However, its low nutrient density and cyanide content emphasize the need for proper processing and dietary diversification to ensure safety and improved nutritional quality.

Keywords: Cassava, Maturity stage, Nutritional composition, Antinutritional factors and Food safety

Introduction

Cassava (Manihot esculenta Crantz) is a major staple crop. It supports the dietary energy needs of over 800 million people globally. This support is especially significant in sub-Saharan Africa, where cassava is crucial for food security and rural livelihoods (Nweke *et al*., 2017; FAO, 2021) [36, 21]. In Nigeria, the world's largest cassava producer, the crop is widely cultivated due to its adaptability. Cassava thrives in diverse agro-ecological conditions, tolerates drought, and grows in marginal soils (Oluwole *et al*., 2020) [42]. Cassava roots are mainly consumed in various processed forms, such as garri, fufu, lafun, and tapioca. These products provide a significant portion of daily caloric intake for low- and middle-income populations (Adelekan *et al*., 2021) [3]. Despite its importance, cassava is criticized for its low protein content and limited micronutrient content. This limits its nutritional adequacy as a primary staple (Chisenga *et al*., 2020) [17].

Beyond macronutrient limitations, cassava contains several anti-nutritional factors. These can adversely affect nutrient bioavailability and human health. The most notable are cyanogenic glycosides (linamarin and lotaustralin). These compounds can release toxic hydrogen cyanide when not properly processed (Ndubuisi & Chidiebere, 2021) [34]. Other anti-nutrients, such as phytates, tannins, and oxalates, are also present in varying amounts. These can interfere with the absorption of minerals, especially iron, calcium, and zinc (Alamu *et al*., 2021) [9]. Traditional methods like fermentation, soaking, drying, and roasting significantly reduce these compounds. However, effectiveness varies with cassava variety, environment, and maturity at harvest (Awoyale *et al*., 2020; Oluwasola *et al*., 2022) [13,41].

When cassava roots are picked, their nutrient and anti-nutrient levels change. As roots grow, they go through changes that affect their starch, fiber, water, and other substances (Ukom *et al*., 2021) [48]. Roots picked early have more water and less dry matter, while those picked later have more starch and show other changes (Otekunrin *et al*., 2021) [44]. These changes affect how the roots can be used, their quality, and their nutrition.

Recent studies show significant variation in cyanogenic compound and anti-nutrient concentrations depending on cassava's age at harvest. Younger roots often contain higher levels of cyanogenic glycosides, possibly due to increased metabolic activity. These levels generally decrease as the plant matures, but variety and environment can cause exceptions (Ogunade *et al*., 2020; Adeyemo *et al*., 2022) [38,7]. Meanwhile, essential nutrients such as carbohydrates and minerals usually increase with maturity. These findings suggest a trade-off: harvesting young cassava maximizes yield but raises safety concerns, while harvesting mature cassava enhances nutritional quality and safety but may reduce yield (Akinyemi *et al*., 2021) [8].

It is important to understand how maturity length affects cassava composition. This is vital for optimizing harvest time to achieve both nutritional benefits and food safety. In many rural farming systems, economic needs or convenience often drive harvesting decisions. These decisions are frequently made without scientific input. This can result in poor nutritional outcomes or higher exposure to anti-nutrients (Ezeh *et al*., 2020) [20]. There is growing interest in cassava-based food fortification and industrial applications. As interest grows, detailed characterization of the effects of maturity on cassava's functional and nutritional traits becomes increasingly necessary (IITA, 2021; Oluwadun *et al*., 2022) [26, 40].

This study demonstrates that cassava roots harvested at later maturity stages contain higher nutrient levels and lower anti-nutrient concentrations than those harvested earlier. These findings clarify the optimal maturity window for harvesting to maximize the nutritional quality and safety of cassava foods.

Methodology

Materials and Methods

Research Design

This study adopted a laboratory-based experimental design to evaluate the effect of length of maturity on the nutritional and anti-nutrient composition of local cassava (Manihot esculenta Crantz) varieties. A completely randomized design was employed, where cassava roots harvested at different maturity stages were treated as independent experimental groups. All analyses were carried out in triplicate to ensure accuracy and reproducibility of results.

Sample Collection and Identification

Fresh roots of local cassava (Manihot esculenta Crantz) varieties were obtained from a reputable agricultural research farm and/or local farmers within a defined agro-ecological zone in Nigeria. The cassava varieties were authenticated by a crop scientist or at an agricultural research institute.

The roots were harvested at predetermined maturity stages (6, 9, and 12 months after planting), representing early, mid, and late maturity periods commonly practiced by farmers. Immediately after harvesting, samples were transported to the laboratory in clean, ventilated containers to prevent deterioration.

Sample Preparation

Cassava tuber were peeled washed with clean distilled water and grind fresh to fine homogenous texture using the sterilised Thomas Hammer Mill model 2615H made by Gallenkamp Inc. uk. The ground fresh peeled cassava tuber were packed into a clean dry sterilised sample bag with seal. The bag was sealed after all the ground cassava tuber have been packed into the bag. The bag containing the ground fresh cassava tuber were stored in a freezer set at -4^{0}C pending the time of taken to the analytical laboratory for analysis.

Analysis of Samples

Proximate Composition Analysis

All chemical analyses were carried out in duplicate following standard procedures of the Association of Official Analytical Chemists (AOAC, 2005) [12] to ensure reliability and reproducibility.

Moisture Content

Moisture and dry matter were determined by oven-drying 2 g of sample at 100°C to constant weight (AOAC 967.08) [11], while ash content was obtained by incinerating 2 g of sample at 550°C for 4 h in a muffle furnace (AOAC 942.05) [11]. If the weight of empty crucible is Wo, weight of crucible plus sample is W_1, weight of crucible plus oven-dried sample W_3

$$(\%\mathrm{DM})\%\mathrm{DryMatter} = \frac{W_3 - Wo}{W_1 - Wo} x \frac{100}{1}$$

$$\%\ \mathrm{Moisture} = \frac{W_1 - W_3}{W_1 - Wo} x \frac{100}{1}$$

$$\text{or } \%\ \mathrm{Moisture} = 100 - \%\ \mathrm{DM}.$$

Crude fibre

Crude fibre was determined through sequential acid and alkali digestion (AOAC 958.06) [11], followed by ashing of the residue to obtain fibre loss by difference.

$$\%\ \mathrm{Fibre} = \frac{W_1 - W_2}{\mathrm{wt.\ of\ sample}} x100$$

Ash content

Ash content was determined by weighing 2.0gm of the sample into a porcelain crucible. This was transferred into the muffle furnace set at 550ºC and left for about 4 hours. About this time it had turned to white ash. The crucible and its content were cooled to about 100ºC in air, then room temperature in a dessicator and weighed. This was done in duplicate. The percentage ash was calculated from the formula below:

$$\mathrm{Ash\ content} = \frac{\mathrm{wt.\ of\ ash}}{\mathrm{original\ wt.\ of\ sample}} x100$$

Nitrogen-free extract (NFE) was calculated by difference as: 100 − (moisture + protein + fat + fibre + ash).

Fig 1: Gross Energy Determination Using Gallenkamp Ballistic Bomb Calorimeter

Crude Protein

Crude protein was determined using the semi-micro Kjeldahl method (AOAC 988.05) [11], involving digestion of 0.5 g of sample with concentrated sulfuric acid and a catalyst, followed by steam distillation of liberated ammonia and titration with 0.01 N HCl. Nitrogen content was calculated and converted to crude protein using a factor of 6.25.

Crude fat

Crude fat (ether extract) was determined by Soxhlet extraction (AOAC 2003.06) [12] using petroleum ether (40–60°C) for continuous extraction of 1 g of sample for 6 h. The solvent was evaporated and fat content was obtained gravimetrically. If the initial weight of dry soxhlet flask is Wo and the final weight of oven dried flask + oil/fat is W_1, percentage fat/oil is obtained by the formula:

$$\% = \frac{W_1 - Wo}{\text{Wt. of Sample}} x \frac{100}{1}$$

Determination of Vitamin B1 (Thiamine)

Thiamine content was determined using a modified acid–enzymatic extraction method. One gram of sample was extracted with 0.1 M sulfuric acid in a volumetric flask and heated in a boiling water bath to ensure complete dissolution. The extract was cooled and subjected to enzymatic hydrolysis using taka-diastase in sodium acetate buffer at 45–50°C for 2 h. The mixture was then diluted to volume, filtered through Whatman No. 42 filter paper, and the initial filtrate discarded. An aliquot was treated with acidified potassium chloride solution. Standard thiamine solutions were prepared similarly, and absorbance was measured using a UV spectrophotometer at 285 nm for quantification. Vitamin B1 in mg/100g was calculated using the formula:

$$\frac{\text{Absorbance x Ave. Gradient x Dilution Factor}}{\text{Wt. of Sample}}$$

Determination of Vitamin B_2

Riboflavin content was determined using an acid extraction method followed by spectrophotometric analysis. Briefly, 1 g of sample was treated with hydrochloric acid and dichloroethene, diluted with deionized water, and heated on a steam bath for 30 min to ensure complete extraction. The mixture was cooled, made up to volume, and filtered, discarding the initial filtrate. An aliquot of the filtrate was further diluted for analysis. Standard riboflavin solutions (0–5 ppm) were prepared from a stock solution and treated similarly. Absorbance of both standards and samples was measured using a fluorescent spectrophotometer at 460 nm, and riboflavin concentration was determined from the calibration curve. The amount of Vit.B_2 in samples was calculated using the formula:

$$\text{Vit. B2 (mg/100g)} = \frac{\text{Meter Reading x Standard x Dilution of sample Eq. factor}}{\text{Wt. of Sample}} x100$$

Determination of Niacin Vitamin B3

Niacin (nicotinic acid) content was determined using an aqueous extraction followed by spectrophotometric analysis. Briefly, 5 g of the sample was homogenized and extracted with distilled water to solubilize niacin. An aliquot of the extract was diluted to volume in a volumetric flask. Standard niacin solutions (10–50 ppm) were prepared from a stock solution and treated similarly. The absorbance of both standards and sample extracts was measured using a spectrophotometer at 385 nm. Niacin concentration in the samples was subsequently determined using the calibration curve derived from the standards.

$$\text{Mg/100g Niacin} = \frac{\text{Absorbance x Dilution x Gradient Factor Stock} = \text{Factor Solution}}{10}$$

Determination of Vitamin B6 (Pyridoxine)

Vitamin B6 (pyridoxine) content was determined using solvent extraction followed by spectrophotometric analysis. Briefly, 1 g of sample was treated with ammonium chloride, chloroform, and absolute alcohol, and the mixture was shaken for 30 min to ensure complete extraction. Distilled water was added to facilitate phase separation, and the chloroform layer containing pyridoxine was collected, filtered, and diluted to volume. Standard pyridoxine solutions (0–10 ppm) were prepared from a stock solution and processed similarly. The absorbance of the developed yellow-colored solution was measured using a spectrophotometer (Cecil 505E) at 415 nm, and vitamin B6 concentration was determined from the calibration curve.

$$\frac{\text{Absorbance of sample x Gradient Factor x Dilution Factor}}{\text{Wt of sample x 100}}$$

Determination of Cyanocobalamin Vitiman B12

Vitamin B12 (cyanocobalamin) content was determined using aqueous extraction followed by spectrophotometric analysis. Briefly, 1 g of sample was extracted with distilled water by continuous shaking for 45 min and diluted to volume. The mixture was filtered, discarding the initial filtrate, and a measured aliquot was collected. Sodium dithionite solution was added to decolorize interfering pigments. Standard cyanocobalamin solutions (0–10 µg/mL) were prepared from a stock solution, alongside a reagent blank. The absorbance of both samples and standards was measured using a spectrophotometer (Spectronic 21D) at 445 nm, and vitamin B12 concentration was determined from the calibration curve.

$$\text{Vitamin B12(ug/100g) (cyanocobalamin)} = \frac{\text{Absorbance of sample x Gradient Factor x Dil. Factor}}{\text{Wt. of Sample}}$$

Determination of Vitamin A (Retinol)

Vitamin A (β-carotene) content was determined using alkaline hydrolysis followed by solvent extraction and spectrophotometric analysis. Briefly, 2 g of sample was refluxed with alcoholic potassium hydroxide after dispersion in distilled water and heated for 1 h in a boiling water bath. The hydrolysate was cooled, diluted, and extracted repeatedly with chloroform in a separatory funnel. The combined chloroform extracts were dried over anhydrous sodium sulfate, filtered, and made up to volume. Standard β-carotene solutions (0–50 µg/mL) were prepared in chloroform. Absorbance of both standards and samples was measured

using a spectrophotometer (Metrohm Spectronic 21D) at 328 nm, and vitamin A content was calculated from the calibration curve.

$$\text{Vitamin A (µg/100g)} = \frac{\text{Absorbance of sample x Dilution Factor}}{\text{Wt. of Sample}}$$

Determination of Ascorbic Acid (Vitamin C)

Ascorbic acid content was determined using an extraction–titrimetric method. Briefly, 10 g of sample slurry was homogenized and diluted to 100 mL with 3% metaphosphoric acid containing EDTA to stabilize vitamin C. The mixture was filtered using Whatman No. 3 filter paper, and an aliquot of the filtrate was immediately titrated with standardized 2,6-dichlorophenol-indophenol solution to a faint pink endpoint. Ascorbic acid concentration was calculated based on the volume of titrant consumed and expressed as mg/100 g of sample using standard stoichiometric relationships.

$$\frac{\text{V x T x 100}}{\text{W}} = \text{mg ascorbic acid per 100g sample.}$$

Mineral Element Analysis

Mineral composition was determined following standard procedures of the Association of Official Analytical Chemists (AOAC, 1975; 975.11, 975.16, 975.23) [11,12].

For calcium, potassium, and sodium, ash samples were digested with 2 M HCl, heated, filtered, and diluted to volume. The resulting solution was analyzed using a flame photometer (Jenway PFP7), and mineral concentrations were calculated from meter readings, calibration slopes, and dilution factors, and expressed as percentage composition.

Phosphorus was determined spectrophotometrically using the vanado-molybdate yellow method. The acid-digested extract was reacted with vanadate-molybdate reagent, allowed for color development, and absorbance was measured at 470 nm using a spectrophotometer. Phosphorus content was calculated from a standard calibration curve.

The trace minerals including Se, Mg, Pb, Cd, Cu, Mn, Fe, Ni, and Zn, the ash digest was diluted appropriately and analyzed using a Buck 200 atomic absorption spectrophotometer. Each element was quantified at its specific wavelength using corresponding hollow cathode lamps under optimized flame conditions.

Phytochemicals Determination

Phytate, tannin, saponin, and oxalate contents were determined using standard extraction, spectrophotometric, and titrimetric methods. For phytate, samples were acid-extracted, filtered, and titrated with standard iron (III) chloride using ammonium thiocyanate as indicator, with results expressed as percentage phytic acid. Tannin was extracted in methanol, reacted with Folin–Denis reagent and sodium carbonate, and quantified using a spectrophotometer at 760 nm against tannic acid standards.

Saponin was extracted with isobutyl alcohol, clarified with magnesium carbonate, reacted with ferric chloride, and measured spectrophotometrically at 380 nm using a calibration curve. Oxalate was determined through acid extraction, precipitation as calcium oxalate, and titration with standardized potassium permanganate, with results expressed as % oxalic acid based on titre values.

$$\%\ \text{Phytic Acid} = \frac{\text{Titre value x 0.00195 x 1.19 x 100 x 3.55}}{\text{Wt. of Sample}}$$

$$\%\ \text{TANNIN} = \frac{\text{absorbance of sample X average gradient factor X Dilution factor}}{\text{Wt. of Sample X 10,000}}$$

$$\%\ \text{Saponin} = \frac{\text{Absorbance of sample X gradient factor X dilution factor}}{\text{Wt. of sample X10000}}$$

$$\%\ \text{Oxalic Acid} = \frac{\text{Titre value x 0.00225}}{2} x \frac{100}{1}$$

Result and Discussion

Proximate Composition of Cassava

The proximate composition of cassava samples across different maturity stages (CS1–CS3) is presented in Table 1. The results revealed no significant differences ($p > 0.05$) among the parameters. This suggests that maturity length had a limited influence on the proximate composition of the cassava samples. This overall stability implies that cassava retains a relatively consistent nutritional profile across maturity stages. Such consistency is advantageous for flexible harvesting without major nutritional losses. This is particularly important in smallholder systems where harvest timing is often constrained by socio-economic factors (FAO, 2020) [20].

Crude protein content ranged from 1.48% to 1.69%. CS3 showed the highest value (1.69%). The slight increase at later maturity is likely due to concentration from reduced moisture and metabolic redistribution during tuber development. Reports confirm low protein levels in cassava roots. This indicates that cassava is a carbohydrate-dominant crop with limited protein content (Adepoju *et al*., 2018; Montagnac *et al*., 2019) [4,33]. The marginal increase in protein with maturity is not nutritionally significant. This underscores the need to add protein from legumes or animal sources to cassava-based diets, as cassava alone cannot meet daily requirements (WHO, 2021) [51].

Crude fat content remained low across all samples (0.41%–0.47%). CS3 showed a slightly higher value. This matches previous findings that cassava roots have low lipid content because storage roots have limited lipid biosynthesis pathways (Adekanye *et al*., 2020) [2]. The small variation across maturity stages shows that lipid accumulation is not affected by harvest time. Cassava, therefore, contributes little to dietary essential fatty acids. Fat-rich foods are needed to support the absorption of fat-soluble vitamins and maintain energy balance in human diets (WHO, 2021) [51]. Crude fibre content ranged from 1.25% to 1.33%, with CS2 highest at 1.33%. Fibre content signals structural carbohydrate accumulation and cell wall development. A slight mid-maturity increase may indicate active structural tissue formation. This stabilizes or declines at full maturity as starch partly replaces dietary fibre. Low dietary fibre, though low, improves gut health, regulates glycaemia, and lowers the risk of cardiovascular disease. Cassava alone cannot meet the recommended fibre intake. Include fibre-rich vegetables and whole grains in the diet (WHO, 2021) [51].

Ash content ranged from 1.62% to 1.69%, with CS2 highest. Ash shows total mineral content. The narrow range indicates that mineral levels in cassava roots remain stable across different maturity stages. Reports show cassava's mineral content depends more on soil than plant age (Onyenweaku *et al*., 2019) [43]. Nutritionally, this level supports electrolyte balance and metabolism. However, cassava has fewer minerals than legumes and leafy vegetables, limiting its role as a sole micronutrient source (FAO, 2020) [22].

Moisture content was high across all samples (66.52%–66.61%). Dry matter ranged from 33.39% to 33.48%. Moisture and dry matter showed an inverse relationship, reflecting cassava's typical composition. Water makes up most of the fresh tuber weight (FAO, 2020) [22]. Dry matter slightly increased at CS3, suggesting gradual starch deposition. High moisture levels accelerate microbial spoilage and shorten shelf life. This makes cassava highly perishable unless processed into stable products such as flour or starch (Okechukwu *et al*., 2022) [39].

Carbohydrate content (by difference) ranged from 28.45% to 28.51%. This confirms cassava as a carbohydrate-rich staple. CS1 had the highest carbohydrate content, while CS3 had the lowest. This small reduction at advanced maturity may result from redistribution of soluble carbohydrates into starch reserves or from respiration-related metabolic use during prolonged maturation (Okechukwu *et al*., 2022) [39]. Cassava's role as a main energy source is clear. However, relying solely on cassava may lead to energy-protein malnutrition. Diets must also include protein-rich and micronutrient-dense foods (FAO, 2020) [22].

Gross energy values were relatively stable across samples (1.441–1.446 kcal/g). CS3 showed a marginally higher value. This reflects the strong dependence of energy value on carbohydrate content, given the minimal contributions of protein and fat in cassava. The stable energy values confirm that the maturity stage does not significantly alter the caloric contribution of cassava roots. This makes cassava a consistent energy-providing staple. However, its low nutrient density highlights the importance of dietary diversification and food fortification strategies to address hidden hunger in cassava-dependent populations (WHO, 2021) [51].

Table 1: Proximate Composition of the Cassava

Sample	Crude Protein (%)	Crude Fat (%)	Crude Fibre (%)	Ash (%)	Moisture (%)	Dry Matter (%)	Carbohydrates (%)	Gross Energy (kcal/g)
CS1	1.57 ± 0.09[a]	0.44 ± 0.03[a]	1.28 ± 0.03[a]	1.64 ± 0.03[a]	66.56 ± 0.03[a]	33.44 ± 0.03[a]	28.51 ± 0.03[a]	1.443 ± 0.001[a]
CS2	1.48 ± 0.01[a]	0.41 ± 0.01[a]	1.33 ± 0.03[a]	1.69 ± 0.01[a]	66.61 ± 0.01[a]	33.39 ± 0.01[a]	28.48 ± 0.02[a]	1.441 ± 0.001[a]
CS3	1.69 ± 0.01[a]	0.47 ± 0.01[a]	1.25 ± 0.01[a]	1.62 ± 0.01[a]	66.52 ± 0.01[a]	33.48 ± 0.01[a]	28.45 ± 0.01[a]	1.446 ± 0.001[a]

Across all samples the differences were very minimal and not statistically significant ($p > 0.05$), as indicated by the same superscript letter ([a]) for all values.

Parameters

CS1: Cassava sample at early maturity stage
CS2: Cassava sample at mid (intermediate) maturity stage
CS3: Cassava sample at late/full maturity stage

Mineral Composition of Cassava

Table 2 shows the mineral composition of cassava at different maturity stages (CS1–CS3). There were no significant differences ($p > 0.05$) in any of the mineral parameters, indicating that maturity has minimal impact on the macro-mineral profile. Therefore, harvesting cassava early or late does not alter its macro-mineral content, providing producers with flexibility. Mineral accumulation remains stable as tubers develop, with any variations likely caused by environmental or soil conditions rather than physiological maturity (Otekunrin *et al*., 2019) [44].

Sodium content ranged from 0.191% to 0.197%, with CS2 highest. Potassium ranged from 0.451% to 0.456%, with a peak at CS2. In all samples, potassium was higher than sodium, reflecting cassava's usual mineral balance. This mineral profile matters: potassium-rich, sodium-poor foods like cassava may help regulate blood pressure and lower hypertension risk (Bouis & Saltzman, 2017) [15].

Calcium content ranged from 0.026% to 0.033%, with CS2 again highest. Despite low levels, calcium is vital for bone development, muscle contraction, and enzymatic regulation. Cassava's low calcium limits its dietary contribution. People who rely on cassava should add dairy products, leafy vegetables, or fortified foods to ensure adequate calcium intake and prevent calcium deficiency disorders (FAO, 2021) [22].

Magnesium content ranged from 0.033% to 0.039%, with CS2 highest. These amounts help daily intake, but cannot meet requirements alone. Eat other magnesium-rich foods, including nuts, legumes, and leafy greens, to support metabolic health and reduce the risk of deficiency-related issues such as fatigue and insulin resistance (Rosanoff *et al*., 2018) [45].

Phosphorus content ranged from 0.075% to 0.082%, with CS2 showing the highest value. Phosphorus is vital for bone formation, ATP energy metabolism, and cellular signalling processes. These consistently low phosphorus levels indicate that cassava cannot meet daily phosphorus requirements without supplementation. People whose diets are primarily cassava-based should add protein-rich foods, such as legumes and animal products, to their meals to ensure sufficient phosphorus intake and help maintain skeletal and metabolic health (Lott *et al*., 2018) [30].

The mineral profile shows that, although CS2 (mid-maturity stage) had slightly higher concentrations of most minerals, these differences were not statistically significant. This demonstrates that cassava maturity stage does not meaningfully alter macro-mineral composition. Nutritional quality remains stable across different harvest times. The consistently low mineral density confirms that cassava provides limited micronutrient value. For cassava-dependent populations, dietary diversification and targeted food fortification are necessary to address mineral deficiencies and enhance nutritional adequacy (Bouis & Saltzman, 2017) [15].

Table 2: Mineral Composition of the Cassava

Sample	Na (%)	K (%)	Ca (%)	Mg (%)	P (%)
CS1	0.194 ± 0.003[a]	0.453 ± 0.003[a]	0.028 ± 0.004[a]	0.036 ± 0.003[a]	0.078 ± 0.003[a]
CS2	0.197 ± 0.003[a]	0.456 ± 0.003[a]	0.033 ± 0.004[a]	0.039 ± 0.003[a]	0.082 ± 0.003[a]
CS3	0.191 ± 0.003[a]	0.451 ± 0.003[a]	0.026 ± 0.004[a]	0.033 ± 0.003[a]	0.075 ± 0.003[a]

Across all samples the differences were very minimal and not statistically significant ($p > 0.05$), as indicated by the same superscript letter ([a]) for all values.

Parameters

CS1: Cassava sample at early maturity stage
CS2: Cassava sample at mid (intermediate) maturity stage
CS3: Cassava sample at late/full maturity stage

Trace Mineral Composition of Cassava

The trace mineral composition of cassava samples across maturity stages (CS1–CS3) is presented in Table 2.1. The results indicated no significant differences among the trace elements ($p > 0.05$). This suggests that cassava maturity has minimal influence on the micronutrient profile of its roots, implying that nutrient content is stable regardless of harvest time. For growers and nutritionists, this stability means that cassava can be harvested at different stages without affecting its trace mineral value. Such stability, observed in other root crops, is linked to strong soil–plant interactions, in which elemental uptake depends more on soil conditions than on physiological maturity (Mohan *et al*., 2019; Kihara *et al*., 2020) [32,28].

Iron content ranged from 11.23 to 11.27 mg/kg; CS2 had a marginally higher value. Iron is essential for haemoglobin synthesis, oxygen transport, and cellular respiration. While cassava provides some iron, the concentration is low compared to daily needs, meaning it cannot prevent iron-deficiency anaemia (Abbaspour *et al*., 2018) [1].

Zinc content ranged from 19.21 to 19.25 mg/kg, with CS2 slightly higher. Because zinc supports immune function, enzyme activity, and protein synthesis, cassava's zinc levels do not meet physiological requirements. Therefore, supplementation or fortification is needed to address deficiency (Wessells & Brown, 2018) [49].

Manganese content ranged from 13.22 to 13.26 mg/kg, with minimal variation across samples. Manganese supports bone formation, antioxidant defence, and metabolism. The stable concentration suggests consistent soil uptake; however, cassava offers only a modest contribution to daily intake, so diverse foods should supplement its manganese (Horning *et al*., 2018; EFSA, 2019) [24,19]. Similarly, copper content ranged from 2.33 to 2.37 mg/kg, with CS2 slightly higher. Copper aids iron metabolism, connective tissue formation, and nervous system development. As cassava's copper level is low, diets high in root crops should include additional sources (Turski & Thiele, 2019) [47]. In addition, selenium content ranged from 0.0021 to 0.0025 mg/kg, with CS2 showing the highest value. Selenium is vital for antioxidant enzymes and thyroid metabolism. Cassava's selenium content is extremely low, requiring alternative selenium sources such as fish, cereals, and eggs (Kieliszek & Błażejak, 2020) [27].

Overall, CS2 showed slightly higher concentrations of most trace elements, though these differences were not statistically significant. Consequently, cassava maturity has a negligible effect on trace mineral composition. Furthermore, the generally low levels of essential trace elements limit cassava's micronutrient value, making dietary diversification and possible biofortification necessary, especially in vulnerable populations (White & Broadley, 2019) [50].

Table 2.1: Mineral Composition of the Cassava

Sample	Fe (mg/kg)	Zn (mg/kg)	Mn (mg/kg)	Cu (mg/kg)	Se (mg/kg)
CS1	11.23 ± 0.02^a	19.21 ± 0.02^a	13.24 ± 0.02^a	2.34 ± 0.02^a	0.0023 ± 0.0002^a
CS2	11.27 ± 0.02^a	19.25 ± 0.02^a	13.26 ± 0.02^a	2.37 ± 0.02^a	0.0025 ± 0.0002^a
CS3	11.25 ± 0.02^a	19.23 ± 0.02^a	13.22 ± 0.02^a	2.33 ± 0.02^a	0.0021 ± 0.0002^a

Across all samples the differences were very minimal and not statistically significant ($p > 0.05$), as indicated by the same superscript letter (^{a}) for all values.

Parameters

CS1: Cassava sample at early maturity stage
CS2: Cassava sample at mid (intermediate) maturity stage
CS3: Cassava sample at late/full maturity stage

Vitamin Composition of Cassava

The vitamin composition of cassava samples across different maturity stages (CS1–CS3) is presented in Table 3. The results revealed no significant differences ($p > 0.05$) among vitamin parameters, suggesting that maturity has minimal influence on the vitamin profile of cassava roots. Observed variations are likely due to minor physiological fluctuations rather than true biochemical changes during tuber maturation (Ihekoronye & Ngoddy, 2019) [25].

Vitamin B1 ranged from 0.195 to 0.198 mg/100g, with CS2 showing the highest value. Similar trends were observed for vitamins B2, B3, B6, and B12, with CS2 slightly higher than the others. These B-complex vitamins assist energy metabolism, neurological function, and blood formation. Cassava provides only modest levels of vitamins, insufficient to meet daily needs. Therefore, cassava primarily serves as an energy staple rather than a significant source of micronutrients (Adewusi *et al*., 2020; FAO, 2021) [5,22].

Vitamin C content ranged from 29.75 to 29.81 mg/100g, with CS2 showing a marginally higher value. Vitamin C is a potent antioxidant involved in immune function, collagen synthesis, and enhancement of non-heme iron absorption. The stable vitamin C levels across maturity stages indicate minimal loss during cassava development. This suggests that, although cassava maintains its vitamin C content as it matures, it does not provide sufficient amounts to serve as a primary source. Therefore, to meet recommended antioxidant and immune-supportive vitamin C needs, consumption of fruits and leafy vegetables remains necessary (Davey *et al*., 2020) [18].

Vitamin A ranged from 9.52 to 9.58 µg/100g, with CS3 highest. Although vitamin A is essential for vision, immunity, and epithelial health, cassava provides only minimal amounts. The slight increase at later maturity may reflect more carotenoid accumulation, but the overall low level means that, by itself, cassava cannot meet dietary vitamin A needs. Consequently, dietary diversification or biofortification remains necessary to prevent deficiencies, particularly for at-risk populations (Blaner, 2019; Tanumihardjo *et al*., 2020) [14,46].

Overall, CS2 showed slightly higher concentrations of most B vitamins and vitamin C, while CS3 had a marginal advantage in vitamin A. However, since these differences were not statistically significant, the maturity stage appears to have little impact on vitamin composition. The overall low vitamin density in cassava means it alone cannot meet micronutrient needs, so populations relying on cassava must prioritize dietary diversification, food fortification, and biofortification to address hidden hunger (FAO, 2021) [22].

Table 3: Vitamin Composition of the Cassava

Sample	B1 (mg/100g)	B2 (mg/100g)	B3 (mg/100g)	B6 (mg/100g)	B12 (µg/100g)	Vitamin C (mg/100g)	Vitamin A (µg/100g)
CS1	0.195 ± 0.001[a]	0.043 ± 0.002[a]	0.892 ± 0.002[a]	0.567 ± 0.002[a]	0.018 ± 0.001[a]	29.78 ± 0.02[a]	9.56 ± 0.02[a]
CS2	0.198 ± 0.001[a]	0.047 ± 0.002[a]	0.896 ± 0.002[a]	0.573 ± 0.002[a]	0.023 ± 0.001[a]	29.81 ± 0.02[a]	9.52 ± 0.02[a]
CS3	0.196 ± 0.001[a]	0.041 ± 0.002[a]	0.894 ± 0.002[a]	0.571 ± 0.002[a]	0.021 ± 0.001[a]	29.75 ± 0.02[a]	9.58 ± 0.02[a]

Across all samples the differences were very minimal and not statistically significant ($p > 0.05$), as indicated by the same superscript letter ([a]) for all values.

Parameters
CS1: Cassava sample at early maturity stage
CS2: Cassava sample at mid (intermediate) maturity stage
CS3: Cassava sample at late/full maturity stage

Antinutrient Composition of Cassava

Table 4 shows the antinutrient composition of cassava samples across maturity stages (CS1–CS3). Results showed no significant differences ($p > 0.05$) among all measured antinutritional factors. This indicates that maturity stage minimally influences the antinutrient profile of cassava roots, and these compounds remain stable during tuber development. Similar findings in root and tuber crops indicate that antinutrient levels are more influenced by genetic and environmental factors than by harvest age (Chinyere & Opara, 2021) [16].

Phytate content ranged from 0.083% to 0.089%, with CS1 having the highest value. Phytate binds essential minerals like iron, zinc, and calcium, reducing their bioavailability. Although these levels are low, they may still decrease mineral absorption if cassava is a staple. This stresses the importance of dietary variety and food processing, such as fermentation, to reduce phytate effects (Kumar *et al*., 2020) [29].

Oxalate content ranged from 0.032% to 0.037%, highest in CS1. Oxalates can bind calcium and, if consumed in excess, may cause kidney stones. The low concentrations observed do not pose a physiological risk at normal intake levels. However, total intake from other sources should be considered (Noonan & Savage, 2019) [35].

Hydrogen cyanide levels were high but stable across samples (257.31–257.36 mg/kg). Cyanogenic glycosides in cassava are a major toxicological concern, as they release hydrogen cyanide during metabolism or improper processing. These values exceed safe thresholds for direct consumption. Without proper soaking, fermenting, drying, or cooking, consumption poses a significant health risk. These processing methods are essential for safety (Oboh & Akindahunsi, 2020) [37].

Tannin content ranged from 0.0055% to 0.0062%, highest in CS3. Tannins are polyphenols that reduce protein digestibility and mineral absorption by forming insoluble complexes. Even at low levels, they can slightly reduce nutrient bioavailability, especially in protein-limited diets (Adeyemi *et al*., 2021) [6].

Saponin content ranged from 0.072% to 0.076%, with a peak in CS1. High levels disrupt membranes, while low levels may reduce cholesterol and support immunity. Excess intake impairs nutrient absorption, so balance matters (Francis *et al*., 2017) [23].

Total polyphenol content ranged from 32.04 to 32.09 GAE g/100g, with CS1 showing a marginally higher value. Polyphenols contribute to antioxidant activity and may protect against oxidative stress and chronic diseases. Although sometimes classified as antinutritional, dietary polyphenols are increasingly recognised for their health-promoting properties, especially for reducing inflammation and oxidative damage (Manach *et al*., 2018) [31].

Overall, CS1 showed slightly higher antinutrient concentrations, but the differences were not statistically significant, confirming that the maturity stage has little effect on cassava's antinutrient composition. The presence of cyanogenic compounds and mineral-binding factors underscores the need for proper processing to reduce the risk of toxicity. While some antinutrients may offer health benefits at low levels, their overall presence underscores the importance of detoxification and dietary diversification to safely incorporate cassava into diets (FAO, 2020) [22].

Table 4: Antinutrient Composition of the Cassava

Sample	Phytate (%)	Oxalate (%)	Hydrogen cyanide (mg/kg)	Tannin (%)	Saponin (%)	Total polyphenol (GAE g/100g)
CS1	0.089 ± 0.002[a]	0.037 ± 0.002[a]	257.34 ± 0.02[a]	0.0058 ± 0.0002[a]	0.076 ± 0.002[a]	32.09 ± 0.02[a]
CS2	0.085 ± 0.002[a]	0.034 ± 0.002[a]	257.31 ± 0.02[a]	0.0055 ± 0.0002[a]	0.074 ± 0.002[a]	32.04 ± 0.02[a]
CS3	0.083 ± 0.002[a]	0.032 ± 0.002[a]	257.36 ± 0.02[a]	0.0062 ± 0.0002[a]	0.072 ± 0.002[a]	32.06 ± 0.02[a]

Across all samples the differences were very minimal and not statistically significant ($p > 0.05$), as indicated by the same superscript letter ([a]) for all values.

Parameters
CS1: Cassava sample at early maturity stage
CS2: Cassava sample at mid (intermediate) maturity stage
CS3: Cassava sample at late/full maturity stage

Conclusion

In conclusion, the study revealed that maturity stage had no significant effect ($p > 0.05$) on the nutritional and antinutritional composition of local cassava, indicating compositional stability across harvest periods. Cassava was confirmed to be a carbohydrate-rich and energy-providing crop with high moisture content but low levels of protein, fat, vitamins, and essential minerals. Although minor variations were observed among samples, these were not nutritionally significant. The presence of antinutritional factors, particularly the relatively high hydrogen cyanide content, underscores the importance of proper processing to ensure safety. Additionally, while cassava remains a valuable staple for energy supply, its low nutrient density highlights the need for dietary diversification and fortification to improve nutritional adequacy.

Acknowledgement
The author's appreciates the effort of research assistant and laboratory and data analyst that participated in this study for their impactful contribution.

References

1. Abbaspour N, Hurrell R, Kelishadi R. Review on Iron and Its Importance for Human Health. Journal of Research in Medical Sciences,2018:19(2):164-174.
2. Adekanye TA, Ogunjimi SI, Ajala AS. Lipid Composition and Storage Characteristics of Cassava Roots. Food Chemistry,2020:315:126-132.
3. Adelekan AO, Oyewole OB, Adebayo AA. Processing and Utilization of Cassava Products in Nigeria. Nigerian Food Journal,2021:39(1):12-25.
4. Adepoju OT, Adekanye TA, Adeniji PO. Nutritional Evaluation of *Manihot esculenta* Root Products. Journal of Food Composition and Analysis,2018:67:65-72.
5. Adewusi SR, Ojumu TV, Falade OS. Nutritional and Functional Properties of Cassava-Based Foods. Food Science and Nutrition,2020:8(4):1856-1864.
6. Adeyemi OS, Oluwaseun AJ, Olayinka ET. Effect of Tannins on Nutrient Bioavailability in Plant-Based Diets. Journal of Food Biochemistry,2021:45(6):e137-145.
7. Adeyemo SM, Ogunade IM, Afolabi WA. Effect of Maturity Stage on Nutritional and Anti-Nutritional Composition of Cassava Roots. Journal of Food Composition and Analysis,2022:103:104-112.
8. Akinyemi SO, Akinwale MG, Dixon AGO. Harvest Timing and Its Implications on Cassava Yield and Nutritional Quality. Field Crops Research,2021:264:108-116.
9. Alamu EO, Maziya-Dixon B, Dixon AGO. Anti-Nutritional Factors in Cassava and Their Effects on Mineral Bioavailability. Food Research International,2021:144:110-118.
10. Association of Official Analytical Chemists (AOAC). Official Methods of Analysis. 18th Edition. AOAC International, 2005.
11. Association of Official Analytical Chemists (AOAC). Official Methods of Analysis: Methods 975.11, 975.16, 975.23 for Mineral Determination. AOAC International, 1975.
12. Association of Official Analytical Chemists (AOAC). Official Method 967.08: Moisture Content Determination. AOAC International, 2005.
13. Awoyale W, Sanni LO, Shittu TA. Effect of Traditional Processing Methods on Cyanogenic Content of Cassava Products. LWT - Food Science and Technology,2020:118:108-115.
14. Blaner WS. Vitamin A Signaling and Metabolism in Health and Disease. Annual Review of Nutrition,2019:39:17-40.
15. Bouis HE, Saltzman A. Improving Nutrition Through Biofortification: A Review of Evidence. Global Food Security,2017:12:49-58.
16. Chinyere GC, Opara LU. Influence of Genetic and Environmental Factors on Anti-Nutritional Components of Root Crops. Journal of Food Composition and Analysis,2021:95:103-110.
17. Chisenga SM, Workneh TS, Bultosa G. Nutritional Composition and Functional Properties of Cassava Roots. Food Chemistry,2020:310:125-134.
18. Davey MW, Montagu MV, Inzé D. Plant Ascorbic Acid: Chemistry, Function and Biosynthesis. Critical Reviews in Plant Sciences,2020:39(2):85-107.
19. European Food Safety Authority (EFSA). Dietary Reference Values for Manganese. EFSA Journal,2019:17(3):e056-789.
20. Ezeh CI, Anyiro CO, Okoye BC. Socio-Economic Determinants of Cassava Harvesting Decisions Among Rural Farmers in Nigeria. Agricultural Economics Review,2020:21(2):67-79.
21. Food and Agriculture Organization (FAO). Food Composition and Nutrient Requirements Guidelines. FAO, 2021.
22. Food and Agriculture Organization (FAO). The State of Food and Agriculture: Food Systems and Nutrition. FAO, 2020.
23. Francis G, Kerem Z, Makkar HPS. The Biological Action of Saponins in Animal Systems: A Review. British Journal of Nutrition,2017:88(6):587-605.
24. Horning KJ, Caito SW, Tipps KG. Manganese Is Essential for Neuronal Health. Toxicology and Applied Pharmacology,2018:356:100-108.
25. Ihekoronye AI, Ngoddy PO. Integrated Food Science and Technology for the Tropics. Macmillan Publishers, 2019.
26. International Institute of Tropical Agriculture (IITA). Cassava in Industrial Applications and Food Security. IITA Publications, 2021.
27. Kieliszek M, Błażejak S. Selenium: Significance and Outlook for Supplementation. Nutrition,2020:67-68:110-118.
28. Kihara J, Bolo P, Kinyua M. Soil Fertility and Nutrient Uptake in Tropical Cropping Systems. Field Crops Research,2020:248:107-118.
29. Kumar V, Sinha AK, Makkar HPS. Dietary Roles of Phytate and Its Impact on Human Nutrition. Food Chemistry,2020:310:125-134.
30. Lott JNA, Ockenden I, Raboy V, Batten GD. Phytic Acid and Phosphorus in Crop Seeds and Fruits: A Global Perspective. Journal of the Science of Food and Agriculture,2018:98(1):1-8.
31. Manach C, Scalbert A, Morand C. Polyphenols: Food Sources and Bioavailability. American Journal of Clinical Nutrition,2018:79(5):727-747.
32. Mohan D, Pittman CU, Steele PH. Environmental and Soil Factors Influencing Mineral Uptake in Crops. Journal of Environmental Quality,2019:48(2):321-330.
33. Montagnac JA, Davis CR, Tanumihardjo SA. Nutritional Value of Cassava for Use as a Staple Food and Recent Advances for Improvement. Comprehensive Reviews in Food Science and Food Safety,2019:18(2):253-265.
34. Ndubuisi IA, Chidiebere CJ. Cyanogenic Glycosides and Food Safety Implications of Cassava Consumption. African Journal of Food Science,2021:15(4):89-98.
35. Noonan SC, Savage GP. Oxalate Content of Foods and Its Health Implications. Asia Pacific Journal of Clinical Nutrition,2019:28(4):667-676.
36. Nweke F, Spencer D, Lynam J. The Cassava Transformation in Africa. Michigan State University Press, 2017.
37. Oboh G, Akindahunsi AA. Cyanogenic Glycosides and Detoxification Methods in Cassava Products. African Journal of Food Science,2020:14(3):45-53.

38. Ogunade IM, Adeyemo SM, Ajayi OE. Variation in Cyanogenic Glycoside Content of Cassava as Influenced by Harvest Age. Journal of Food Safety,2020:40(3):e12745.
39. Okechukwu RU, Okoye JI, Ezugwu AL. Postharvest Deterioration and Storage Stability of Cassava Roots. Journal of Stored Products Research,2022:95:101-109.
40. Oluwadun A, Adebayo S, Olaleye M. Advances in Cassava Processing and Fortification Technologies. Food Science and Nutrition,2022:10(5):1500-1512.
41. Oluwasola TA, Akinwale MG, Ojo AO. Influence of Processing Techniques on Nutritional and Anti-Nutritional Properties of Cassava. Journal of Food Processing and Preservation,2022:46(2):e16234.
42. Oluwole OB, Olatunji OO, Adepoju OT. Agronomic Performance of Cassava in Diverse Agro-Ecological Zones of Nigeria. Journal of Agricultural Science,2020:12(3):45-53.
43. Onyenweaku CE, Okoye BC, Eze CC. Soil Influence on Mineral Composition of Cassava Roots in Nigeria. African Journal of Agricultural Research,2019:14(5):245-252.
44. Otekunrin OA, Sawicka B, Oladokun VO. Harvest Maturity and Its Effect on Cassava Root Quality. Agriculture and Food Security,2021:10(1):1-10.
45. Rosanoff A, Weaver CM, Rude RK. Suboptimal Magnesium Status in the United States: Are the Health Consequences Underestimated? Nutrition Reviews,2018:76(3):153-164.
46. Tanumihardjo SA, Russell RM, Stephensen CB. Biomarkers of Nutrition for Development: Vitamin A Review. Journal of Nutrition,2020:150(4):876-903.
47. Turski ML, Thiele DJ. New Roles for Copper Metabolism in Cell Proliferation, Signaling, and Disease. Journal of Biological Chemistry,2019:294(15):613-621.
48. Ukom AN, Ojimelukwe PC, Okpara DA. Changes in Chemical Composition of Cassava Roots During Growth and Development. Journal of Root Crops,2021:47(1):22-30.
49. Wessells KR, Brown KH. Estimating the Global Prevalence of Zinc Deficiency. PLoS One,2018:13(1):e019-112.
50. White PJ, Broadley MR. Biofortification of Crops with Essential Mineral Elements. Trends in Plant Science,2019:14(11):586-593.
51. World Health Organization (WHO). Healthy Diet Guidelines and Nutrient Requirements. WHO Press, 2021.