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Evaluation of the Comparative Quality and Safety of Selected Commercial Vegetable Oils in Lokoja, Kogi State, Nigeria: Integrating Physicochemical, Nutritional, and Microbiological Assessments

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ABSTRACT

The nutritional integrity and safety of commercially available vegetable oils is an important food-safety concern, particularly where regulatory monitoring and market surveillance may be inconsistent. This study evaluated four branded vegetable oils (Activa, Kings, Power, and Terra) marketed in Lokoja, Nigeria, using proximate, physicochemical, microbiological, and heavy-metal analyses. Standard analytical procedures were used for proximate composition, specific gravity, acid value (AV), peroxide value (PV), iodine value (IV), saponification value (SV), microbial load, and concentrations of lead (Pb), cadmium (Cd), and arsenic (As). Fat content ranged from 96.98% to 97.26%, whereas moisture content ranged from 0.42% to 0.86%. PV ranged from 5.95 to 23.76 meq/kg, with Activa exceeding the Codex benchmark of 10 meq/kg. AV ranged from 4.88 to 13.44 mg KOH/g and exceeded the reported benchmark of 0.6 mg KOH/g in all samples. IV ranged from 36.22 to 68.94 g I_2/100 g, while SV ranged from 167.96 to 202.58 mg KOH/g. Total viable counts ranged from 1.1×10^5 to 3.1×10^5 CFU/cm³, and Escherichia coli was isolated from Kings and Power samples. Cadmium and arsenic were not detected (<0.001 ppm), while lead occurred at 0.001-0.002 ppm. Overall, the results indicate that the oils had high lipid content and low detectable heavy-metal contamination but exhibited important quality deficiencies, particularly elevated acid values and, in one sample, peroxide value, together with microbial contamination. These findings support strengthened market surveillance, improved processing and storage hygiene, and continued monitoring of commercial edible oils in Lokoja.

Keywords: Edible Oils; Quality Assessment; Oxidative Rancidity; Microbial Contamination; Food Safety; Nigeria

INTRODUCTION

Edible vegetable oils are very important and widely used in household cooking and food processing. They are important dietary sources of fatty acids and provide essential fat-soluble vitamins, and concentrated energy (Jolayemi & Muritala, 2022) Their quality is influenced by the source oil, refining efficiency, exposure to oxygen, light and heat, storage duration, packaging, and handling conditions (Gunstone, 2011; Mba et al., 2015). In Nigeria, increasing urbanization and the widespread availability of commercially packaged oils make routine quality assessment relevant to consumer protection and food safety (Onyeaka et al., 2022).

Important indicators of edible-oil quality include acid value (AV), peroxide value (PV), iodine value (IV), saponification value (SV), moisture content, and specific gravity. AV provides an indication of free fatty acids generated through hydrolytic deterioration, whereas PV measures primary lipid-oxidation products. IV reflects the degree of unsaturation, while SV provides information related to the average molecular characteristics of the fatty acids present (Choe & Min, 2007; Gunstone, 2011). Oxidation and hydrolysis can reduce sensory and nutritional quality and may be accelerated by inappropriate storage conditions (Akinyemi et al., 2021; Zeb, 2020).

Previous studies have shown considerable variation in the physicochemical quality of edible oils sold or used under different storage and processing conditions. Akinyemi et al. (2021) emphasized that storage conditions can substantially alter oil quality through oxidation and hydrolysis. Similarly, studies of vegetable oils have associated changes in peroxide and acid values with exposure to heat, oxygen, moisture, and prolonged storage (Choe & Min, 2007; Naz et al., 2005). The Nigerian food-safety environment also requires attention because analytical surveillance is important for identifying chemical and microbiological hazards across the food chain (Onyeaka et al., 2022).

Despite these studies, there remains limited recent comparative information on the physicochemical, proximate, microbiological, and heavy-metal quality of multiple branded vegetable oils sold specifically in Lokoja, Kogi State. This geographic gap is important because product quality at the point of retail can be affected by local transportation, storage, market exposure, and handling conditions. In addition, assessing several quality dimensions in the same set of commercial brands provides a more integrated picture than evaluating a single parameter alone. The present study therefore addresses this gap by comparing four commonly available branded vegetable oils in Lokoja using proximate composition, physicochemical indices, microbiological quality, and selected heavy metals, with the findings interpreted against relevant food-quality standards and published literature. This study therefore aimed to conduct a comprehensive comparative analysis of four commercially available vegetable oil brands in Lokoja, Nigeria. Specifically, the study evaluated their proximate composition, physicochemical properties, microbial quality,

and Pb, Cd, and As concentrations and compared the findings with applicable quality benchmarks. The study was designed to provide evidence that can support consumer awareness, food-safety surveillance, and regulatory decision-making in the study area.

MATERIALS AND METHODS

Sample Collection

Four leading branded vegetable oils-Activa (Sample A), Kings (Sample B), Power (Sample C), and Terra (Sample D)-were purchased randomly from retail outlets and open markets within Lokoja Metropolis, Kogi State, Nigeria on June 20th, 2025. The samples were obtained as commercially packaged products; the packaging status at the point of purchase was recorded as part of sample handling. Samples were transferred into sterile, airtight amber glass bottles of 25 cm^3 capacity, labelled appropriately, and transported to the laboratory under ambient conditions while protected from light and heat. Analyses commenced within 48 hours of collection.

Proximate Composition Analysis

Proximate analysis was performed according to the standard methods of the Association of Official Analytical Chemists (AOAC International, 2016).

Percentage Moisture Content

Moisture content was determined by oven-drying 2.5 g of sample at 105 °C to constant weight.

$$\% \text{ Moisture} = \frac{W_1 - W_2}{W_1} \times 100$$

where W_1 is the initial sample mass, W_2 is the mass of the dried sample,

Ash content was determined by incinerating 2.5 g of the moisture-free sample in a muffle furnace at 550 °C for 3 hours.

$$\% \text{ Ash} = \frac{W_a}{W_1} \times 100$$

Where W_1 = Initial mass of sample, W_a = mass of sample after incineration

Percentage Crude Protein

Crude protein was determined by the micro-Kjeldahl method using a nitrogen-to-protein conversion factor of 6.25.

Crude protein (%) = Nitrogen (%) × 6.25

Percentage Crude Fat

Two gram (2.0 g) of each commercial vegetable-oil sample was accurately weighed into a suitable extraction thimble. A clean, dry round-bottom flask was dried, cooled in a desiccator, and weighed (W_1). Petroleum ether (40-60 °C) was added to the flask, and the Soxhlet apparatus was assembled. The sample was continuously extracted under reflux for approximately 6 h. Following extraction, the solvent was recovered by distillation. The flask containing the extracted lipid was dried to constant mass, cooled in a desiccator, and weighed (W_2). The percentage fat was calculated from the mass of lipid recovered relative to the original sample mass (AOAC International, 2016). Percentage fat was calculated using:

$$\% \, Fat = \frac{W_2 - W_1}{W_s} \times 100$$

Where; W_1= mass of the clean, dry extraction flask (g), W_2= mass of the extraction flask containing extracted fat after drying to constant mass (g), W_s= mass of vegetable-oil sample used for extraction (g).

Percentage Crude Carbohydrate

This was calculated by difference using the expression below:

$$\%\text{Carbohydrate by difference} = 100 - (\%\text{Moisture} + \%\text{Ash} + \%\text{Protein} + \%\text{Fat})$$

Physicochemical Analysis

Specific Gravity (SG)

Specific gravity was determined at 25 °C using a 25 cm^3 specific gravity bottle (pycnometer). The clean, dry bottle was weighed (W_0), filled with the oil sample, stoppered, wiped dry, and weighed (W_1). The bottle was then cleaned, filled with distilled water at 25 °C, and weighed (W_2). Specific gravity was calculated as:

$$SG_{25/25} = \frac{W_1 - W_0}{W_2 - W_0}$$

where W_0, W_1, and W_2 are the masses (g) of the empty bottle, bottle with oil, and bottle with water, respectively. Measurements were performed in triplicate and expressed as mean ± SD.

Acid Value (AV)

Acid value was determined according to AOAC Official Method 940.28. 28 (AOAC International, 2023), 5 g of oil was dissolved in 50 mL of neutralized ethanol, warmed, and titrated with standardized 0.1 N KOH using phenolphthalein as indicator until a persistent faint pink colour appeared. The acid value was calculated as the mg of KOH required to neutralize the free fatty acids in 1 g of oil.

$$AV = \frac{V \times N \times 56.1}{W}$$

where V= volume of KOH (mL), N= normality of KOH, W= weight of oil (g) and =56.1 is the molecular mass of KOH (g/mol)

Peroxide Value (PV)

Peroxide value was determined according to AOAC Official Method 965.33. 28 (AOAC International, 2023). Approximately 5.0 g of oil was weighed into a glass-stoppered flask and dissolved in 30 mL of acetic acid - chloroform mixture (3:2, v/v). 0.5 mL of saturated potassium iodide (KI) was added, and the mixture was allowed to stand for about 1 min with occasional shaking. 30 mL of distilled water was added, and the liberated iodine was titrated with standardized 0.1 N sodium thiosulfate ($Na_2S_2O_3$) until the yellow colour nearly disappeared. About 0.5 mL of 1% starch indicator was added, and titration continued until the blue colour disappeared. A reagent blank was determined similarly.

$$PV = \frac{(S - B) \times N \times 1000}{W}$$

where S= sample titre (mL), B= blank titre (mL), N= normality of $Na_2S_2O_3$, and W= sample mass (g).

Iodine Value (IV)

Iodine value was determined by the Wijs method according to AOAC Official Method 993.20 (AOAC International, 2023). A known mass of oil was dissolved in an appropriate solvent and treated with Wijs reagent (iodine monochloride). The mixture was allowed to react in the dark for the specified period. Potassium iodide and water were then added, and the liberated iodine was titrated with standardized sodium thiosulfate using starch as indicator. A reagent blank was determined under identical conditions. The Wijs method is an established AOAC method for determining iodine value in vegetable and animal oils and fats.

$$IV = \frac{(B - S) \times N \times 12.69}{W}$$

where B= blank titre (mL), S= sample titre (mL), N= normality of $Na_2S_2O_3$, and W= sample mass (g).

Saponification Value (SV)

Saponification value was determined according to AOAC Official Method 920.160 (AOAC International, 2023). Approximately 1.5-2.0 g of oil was weighed into a flask and mixed with 25 mL of alcoholic KOH. The mixture was refluxed for approximately 30 min to complete saponification and then cooled. Phenolphthalein was added, and the excess KOH was titrated with standardized HCl. A blank determination was performed using the same procedure without the oil sample.

$$SV = \frac{(B - S) \times N \times 56.1}{W}$$

where B= blank titre (mL), S= sample titre (mL), N= normality of HCl, 56.1= molecular weight of KOH, and W= sample mass (g).

Microbiological Analysis

A 5 cm³ aliquot of each oil sample was emulsified in 45 cm³ of sterile peptone water containing 1% polysorbate 80 (Tween 80). Serial ten-fold dilutions (10^{-1}-10^{-5}) were prepared, and 1 cm³ of appropriate dilutions was pour-plated in duplicate on Nutrient Agar for total viable counts and MacConkey Agar for coliform enumeration. Plates were incubated at 30 °C for 24-48 h. Colonies were counted and expressed as colony-forming units per cm³ (CFU/cm³). Representative colonies were characterized by Gram staining and standard biochemical tests, including catalase, citrate utilization and sugar fermentation tests, following standard microbiological procedures (FDA, 2026; FDA, 2020; ISO 4833-1:2013).

Determination of Heavy Metals by AAS

Oil samples (1.00 g) were accurately weighed into digestion vessels and digested with a 3:1 (v/v) mixture of concentrated HNO_3 and $HClO_4$ until a clear digest was obtained. After cooling, the digest was quantitatively transferred into a 50 mL volumetric flask and diluted to volume with deionized water. The digest was filtered where necessary, and a reagent blank was prepared using the same procedure without the sample. The concentrations of lead (Pb), cadmium (Cd), and arsenic (As) were determined using an Agilent 240FS atomic absorption spectrophotometer (AAS). Calibration standards of each element were prepared from appropriate certified stock standards, and calibration curves were constructed by plotting absorbance against concentration. The digested samples and reagent blank were analyzed under the appropriate instrumental conditions for each element. Samples were analyzed in triplicate, with blank correction, and concentrations were calculated from the corresponding calibration curves and expressed as mg/kg (ppm) of oil. The analytical procedure was based on established elemental-analysis procedures and the manufacturer's operating guidance for the Agilent AA system. Calculation

$$C_s = \frac{C_{AAS} \times V \times DF}{W}$$

where:

C_s= concentration of the element in oil (mg/kg)

C_{AAS}= concentration obtained from the calibration curve (mg/L)

V= final volume of digest (L)

DF= additional dilution factor

W= mass of oil sample (kg)

Results were reported as mean ± standard deviation (SD).

Statistical Analysis

All analyses were performed in triplicate. Data were expressed as mean ± standard deviation. One-way analysis of variance (ANOVA) followed by Tukey's Honestly Significant Difference (HSD) post-hoc test was performed using SPSS version 25.0. Differences were considered statistically significant at $p < .05$.

RESULTS AND DISCUSSION

Proximate Composition

The proximate composition of the four oil samples is presented in Table 1. Crude fat content was consistently high (96.98-97.26%), which is expected for commercial vegetable oils because refining and processing remove most non-lipid constituents. The narrow range among the four brands suggests broadly similar lipid concentration and is consistent with the general composition of refined edible oils described by Gunstone (2011). Crude protein was low (0.36-0.61%), as expected for refined oils, while ash content was also low (0.04-0.13%), indicating limited residual inorganic material after processing. Moisture content, however, ranged from 0.42% to 0.86%. These values are relatively high for refined edible oils and are important because residual water can promote hydrolytic reactions and may contribute to deterioration during storage (Akinyemi *et al*., 2021, O'Brien, 2009). Differences among the brands may reflect variation in refining efficiency, drying, packaging, or storage conditions. The carbohydrate values (1.31-1.94%) were calculated by difference and therefore incorporate the cumulative effect of the other proximate components rather than representing directly measured carbohydrate in the oils.

Table 1: Proximate Composition of Selected Vegetable Oil Samples (% w/w)

Parameter	Sample A (Activa)	Sample B (Kings)	Sample C (Power)	Sample D (Terra)
Moisture	0.86 ± 0.02	0.54 ± 0.03	0.68 ± 0.01	0.42 ± 0.02
Ash	0.13 ± 0.01	0.09 ± 0.01	0.04 ± 0.01	0.06 ± 0.01
Crude Fat	97.26 ± 0.10	96.98 ± 0.20	97.15 ± 0.10	97.22 ± 0.10
Crude Protein	0.44 ± 0.05	0.61 ± 0.06	0.48 ± 0.04	0.36 ± 0.03
Carbohydrate*	1.31	1.78	1.65	1.94

Note: Carbohydrate calculated by difference. Values are mean ± SD (n==3).

Physicochemical Properties

The physicochemical results (Table 2) show marked variation in quality indicators among the four brands. Specific gravity ranged from 0.911 to 0.922, within the general range expected for many edible oils. This relatively narrow range suggests broadly similar bulk density characteristics among the samples and does not, by itself, indicate gross compositional abnormality. However, specific gravity alone cannot establish the absence of adulteration because different vegetable oils may have overlapping density ranges (Gunstone, 2011).

Table 2: Physicochemical Properties of Selected Vegetable Oil Samples

Parameter	Sample A	Sample B	Sample C	Sample D	Standard Limit
SG	0.913 ± 0.001	0.922 ± 0.002	0.911 ± 0.001	0.915 ± 0.002	0.91-0.93[a]
P V(meq/kg)	23.76 ± 0.5[a]	9.28 ± 0.3[b]	5.95 ± 0.2[c]	7.55 ± 0.3[bc]	≤10[b]
IV(g/100g)	68.94 ± 1.2[a]	48.50 ± 0.9[b]	36.22 ± 0.8[c]	44.73 ± 1.0[b]	Varies[c]
SV	185.4 ± 2.1[b]	167.96 ± 1.8[c]	194.2 ± 2.3[a]	202.58 ± 2.5[a]	180-200[a]
AV(mg KOH/g)	7.16 ± 0.1[b]	13.44 ± 0.3[a]	5.85 ± 0.2[c]	4.88 ± 0.1[d]	≤0.6[b]

Note. Values are mean ± SD (n == 3). Means in a row with different superscripts differ significantly (p < .05). [a]Typical range for edible oils (Gunstone, 2011). [b]Codex Alimentarius standard (CXS 210-1999; (Codex Alimentarius Commission, 2024)). [c]Dependent on oil type and source.

Peroxide value (PV) is an indicator of primary lipid oxidation. Sample A recorded the highest PV (23.76 ± 0.5 meq/kg), more than twice the 10 meq/kg benchmark used in the manuscript, whereas Samples B, C, and D recorded 9.28, 5.95, and 7.55 meq/kg, respectively. The relatively low values in B-D suggest less advanced primary oxidation at the time of analysis, while the markedly higher value in A indicates greater oxidative deterioration. Oxidation is promoted by oxygen, light, heat, trace metals, and prolonged storage, and differences among brands may therefore arise from variation in antioxidant protection, packaging, storage duration, or retail exposure (Akinyemi *et al*., 2021; Choe & Min, 2007; Zeb, 2020).

Acid value (AV) ranged from 4.88 to 13.44 mg KOH/g, with all four samples substantially above the reported benchmark of 0.6 mg KOH/g. Sample B had the highest AV (13.44 ± 0.3 mg KOH/g), followed by Sample A (7.16 ± 0.1 mg KOH/g), Sample C (5.85 ± 0.2 mg KOH/g), and Sample D (4.88 ± 0.1 mg KOH/g). The uniformly elevated values indicate considerable free-fatty-acid formation and therefore suggest hydrolytic deterioration. Differences between samples may reflect differences in refining, residual moisture, enzymatic activity, or storage conditions. The coexistence of elevated moisture and AV in the samples is particularly relevant because water availability can facilitate triglyceride hydrolysis (Akinyemi *et al*., 2021; Gunstone, 2011).

Iodine values ranged from 36.22 to 68.94 g I_2/100 g. IV is a measure of unsaturation, so the relatively lower values of Samples B-D compared with Sample A suggest differences in the proportions of unsaturated fatty acids among the commercial products. Such differences can arise from the botanical source of the oil, blending of different oils, and refining practices (Gunstone, 2011). The observed range should not be interpreted as evidence of a particular oil source without fatty-acid profiling. The lower IV values are also relevant to oxidative stability because oils containing greater proportions of polyunsaturated fatty acids are generally more susceptible to oxidation (Naz *et al*., 2005; Zeb, 2020).

Saponification values ranged from 167.96 to 202.58 mg KOH/g. Samples A, C, and D were within or close to the stated 180-200 mg KOH/g reference range, whereas Sample B was lower (167.96 ± 1.8 mg KOH/g). SV is influenced by the average molecular mass of the fatty acids in the oil; lower values may therefore be associated with a greater contribution from longer-chain fatty acids, although the inference cannot be made conclusively without fatty-acid composition data. The variation among the samples indicates that the brands may differ in oil source or blending composition (Gunstone, 2011).

Microbiological Quality

Microbial assessment showed total viable counts of 3.1×10^5 CFU/cm³ in Sample A, 1.1×10^5 CFU/cm³ in Sample B, 3.1×10^5 CFU/cm³ in Sample C, and 2.1×10^5 CFU/cm³ in Sample D. Although no coliform growth was reported in the table, *Escherichia coli* was isolated from Samples B and C, while *Pseudomonas spp*., *Enterobacter spp*., and *Bacillus spp*. were recovered from different samples. The detection of *E. coli* is noteworthy because it is commonly used as an indicator of hygiene-related contamination. The microbial findings may reflect contamination introduced during processing, packaging, transportation, retail handling, or laboratory recovery. Food-safety surveillance is particularly important where multiple points in the food chain may contribute to contamination (Onyeaka *et al*., 2022).

Table 3: Microbial Load and Isolates from Vegetable Oil Samples

Sample	TVC (CFU/cm³)	Coliform Count	Microorganisms Isolated
A	3.1×10^5	None	*Pseudomonas* spp., *Enterobacter* spp.
B	1.1×10^5*	None	*Escherichia coli*, *Enterobacter* spp.
C	3.1×10^5	None	*Escherichia coli*, *Bacillus* spp.
D	2.1×10^5	None	*Pseudomonas* spp., *Enterobacter* spp.

*Note. TVC == Total Viable Count. *Growth observed only on MacConkey Agar.*

Heavy Metal Content

The heavy-metal results showed that cadmium and arsenic were not detected above the analytical detection threshold of 0.001 ppm in any sample, while lead was detected at 0.001-0.002 ppm in all four samples. These concentrations are very low relative to the limits reported in the study's benchmark framework. The low concentrations suggest limited contribution from contaminated raw materials, processing equipment, environmental exposure, or packaging at the time of sampling (Ibrahim, *et al*.,2010) Nevertheless, trace-metal monitoring remains important because metals can enter food systems from environmental and processing sources, and continued surveillance is preferable to relying on a single sampling event (Onyeaka *et al*., 2022).

Table 4: Heavy Metal Content in Vegetable Oil Samples (ppm)

Sample	Cadmium (Cd)	Arsenic (As)	Lead (Pb)
A	ND	ND	0.002
B	ND	ND	0.001
C	ND	ND	0.001
D	ND	ND	0.001

Note. ND == Not Detected (<0.001 ppm).

CONCLUSION

The study demonstrated measurable differences in the quality of the four commercial vegetable oils examined. Moisture content ranged from 0.42% to 0.86%, crude fat from 96.98% to 97.26%, and crude protein from 0.36% to 0.61%. PV ranged from 5.95 to 23.76 meq/kg, with Sample A exceeding the 10 meq/kg benchmark. AV ranged from 4.88 to 13.44 mg KOH/g and exceeded the 0.6 mg KOH/g benchmark in all four samples. IV ranged from 36.22 to 68.94 g I_2/100 g and SV from 167.96 to 202.58 mg KOH/g. Total viable counts ranged from 1.1×10^5 to 3.1×10^5 CFU/cm³, and *E. coli* was isolated from two samples (B and C). Cd and As were below 0.001 ppm in all samples, whereas Pb ranged from 0.001 to 0.002 ppm. Thus, the principal quality concerns identified were elevated AV in all samples, elevated PV in Sample A, and the recovery of indicator organisms from two brands, rather than heavy-metal contamination.

i. The elevated AVs indicate widespread hydrolytic deterioration, while the high PV of Sample A indicates substantial primary oxidation.
ii. The microbial findings, particularly the isolation of *E. coli* from Samples B and C, indicate the need for stronger hygienic controls during production, packaging, transportation, and retail handling.

The results support continued surveillance of commercial vegetable oils in Lokoja, with emphasis on oxidative and hydrolytic stability and microbiological quality.

i. Regulatory agencies should strengthen periodic surveillance of commercial vegetable oils using AV, PV, microbial quality, and heavy-metal analyses.
ii. Manufacturers and distributors should strengthen Good Manufacturing Practices, moisture control, packaging integrity, and protection from excessive heat and light during storage and distribution.
iii. Retailers should minimize prolonged exposure of packaged oils to heat and direct light and maintain hygienic storage conditions.
iv. Consumers should be encouraged to purchase properly sealed products from reputable outlets and avoid oils showing obvious evidence of deterioration.
v. Further studies should include fatty-acid profiling, antioxidant-content determination, polycyclic aromatic hydrocarbon analysis, and broader geographic sampling to establish a more comprehensive quality database for commercial vegetable oils in Nigeria.

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