HUMAN HEALTH & HALAL METRICS

Research article

Journal of Human, Health and Halal Metrics; 2024, 5(2): 33-45
https://doi.org/10.30502/jhhhm.2024.473108.1095

Physicochemical characteristics and fatty acid composition of five selected leafy vegetables grown in southwest Nigeria

Zeenat Lami Usman[1], Mathew Olaleke Aremu[1], Sule Philip Ivom Ogah[1], Hashim Ibrahim[1,2], Stephen Olaide Aremu[3*]

1- Department of Chemistry, Federal University of Lafia, P.M.B. 146, Nasarawa State, Nigeria.
2- Department of Chemistry, University of Nairobi, Nairobi, Kenya.
3- Faculty of General Medicine, Siberian State Medical University, Tomsk, Russian Federation.

Submission: 13 August 2024 ***Revision:*** 27 August 2024 ***Acceptance:*** 17 November 2024

Abstract

Background and Objectives: Vegetable consumption has been linked in different studies to reducing non-communicable diseases, particularly diabetes, cancer, respiratory disease, and cardiovascular disease. This study selected five leafy vegetables, and their physicochemical characteristics, fatty acid compositions, and functional quality indices were evaluated.

Materials and Methods: Foreign particles were removed from the samples (*Cochorus olitorius, Celosia argentea, Crassocephalum crepidioides, Solanum macrocarpon,* and *Launaea taraxacifolia*) by way of cleaning and were air-dried, and the extraction of the oil was done using the Soxhlet technique. Standard analytical methods were used to determine the physicochemical parameters of the extracted oils, while the gas chromatography technique was used to determine the fatty acid profiles.

Results and Conclusion: The physicochemical parameters of the extracted oils were in the range of 1.08–1.31 meq O_2/Kg for peroxide value, 0.14–0.17 mg KOH/g for acid value, 158.92–180.21 mg KOH/g for saponification value, 80.49–102.21 mg I_2/g for iodine value, 0.65–0.88% for unsaponifiable matter, 1.29–1.40 at 40 °C for refractive index, and 0.93–1.10 at 15 °C for specific gravity. The predominant fatty acids in all the studied samples were palmitic (29.32–37.44%), linoleic (19.73–22.54%), γ-Linolenic (13.91–16.29%), and α-Linolenic (16.74–19.64%) acids. PUFAs constitute over two-thirds of the total UFAs, with linoleic acid having the highest abundance of UFA in all the samples. The calculated ratios of ω 6/ω 3 (1.98–2.05), hypocholesterolemic to hypercholesterolemic (h/H) (1.10–1.59), atherogenicity index (AI) (0.44–0.64), thrombogenicity index (TI) (0.40–0.58), the phospholipid composition of the vegetables had a range of 4.36 to 67.96 mg/100 g. The phytosterol concentrations were between 35.4 and 40.6 mg/100 g and were within the acceptable limit of ≤ 10 for edible oils. The evaluated parameters indicated the vegetable oil samples to be of high nutritional quality with preventive potential against chronic degenerative diseases.

Keywords: Fatty acids, Nutritional quality, Phospholipids, Physicochemical parameters, Phytosterols, Vegetables

1. Introduction

Accessing a sufficient, nutritious, and healthy diet is increasingly challenging, resulting in food insecurity, splurging malnutrition burden, deficiency of vital micronutrients, undernourishment, and overnutrition, affecting different societal strata, particularly in developing nations like Nigeria. Micronutrient deficiencies have become nutritional dilemmas involving over two billions people globally [1,2].

*Correspondence to: Stephen Olaide Aremu; E-mail: dr.aresteph@gmail.com

It causes nutritional disorders like weakened immune systems, congenital disabilities, and mental and physical retardation. Leafy vegetables contain several ingredients of great nutritional significance, including vitamins, high dietary fibers, proteins/ essential amino acids, polyunsaturated fatty acids (PUFAs), bioactive compounds, and micronutrients [3]. Hence, there is a need for leafy vegetables that are indigenous, like *Cochorus olitorius, Celosia argentea, Crassocephalum crepidioides, Solanum macrocarpon* and *Launaea taraxacifolia* (Figure 1-5), which are essential and readily available resources by which we can attain food security. Adequate consumption of vegetables is positively correlated with maintaining healthy blood pressure, lowering cholesterol levels in the blood, decreasing the vulnerability to cardiac disease, lowering the risk of congenital disabilities, and maintaining the health of the eyes, skin, teeth, and the gingiva in addition to reducing the mortality rate from cardiovascular diseases [1,4,5].

Figure 1- Leaves and flowers of *Cochorus olitorius*

Figure 2- Leaves and fruits of *Solanum macrocarpon*

Figure 3- Leaves of *Crassocephalum crepidioides*

Figure 4- Leaves and flowers of *Celosia argentea*

Figure 5- Leaves of *Launaea taraxacifolia*

It should be noted that these plants are indeed edible and have been traditionally consumed in various parts of Africa, including Nigeria, for their nutritional and medicinal benefits. *C. olitorius* (jute mallow) and *S. macrocarpon* (African eggplant) are widely recognized as safe for human consump-

tion and are regularly incorporated into local diets. *C. argentea* (Lagos spinach), *C. crepidioides* (Fireweed), and *L. taraxacifolia* (wild lettuce) are also consumed in specific regions. They are valued for their nutritional profiles, which include vitamins, minerals, and antioxidants.

To prevent chronic diseases (mainly cardiovascular diseases (CVDs), oncological diseases, and endocrinological diseases, particularly diabetes) and ensure the availability and accessibility of important micronutrients that are termed "essential" (such as iodine, iron, calcium, zinc, and retinol), a minimum daily intake of four hundred grams of vegetables and fruits is recommended by the World Health Organization (WHO) that is required for a healthy living [6]. Numerous studies have established a link between vegetable consumption and a reduction in non-communicable diseases, primarily diabetes, cancer, chronic respiratory disease, and CVDs [7-10]. The pooled relative risk (RR) for CVDs between the maximum and minimum intake of vegetable categories, for example, shows a decreased CVDs risk with increasing vegetable intake [11]. Edible vegetable oils and leafy vegetables serve as a nutritional source of essential fatty acids, omega-3 and omega-6 fatty acids (PUFAs), with multiple physiological roles in humans, mainly as normal body metabolism. For instance, omega-3 or n-3 fatty acids, are crucial components of cell membranes, and impact the membranes' cell receptors function. They are used as building blocks of hormones in charge of inflammation, vaso-constriction, and -dilation of the wall of the arteries, and hemocoagulation. Additionally, they cleave to receptors on the cell to regulate the activity of the genes [12,13]. Furthermore, several studies have associated with dietary consumption of omega-6, especially linoleic acid, with a lower risk of coronary heart disease [14-18]. The omega-6 fatty acids (also known as n-6 fatty acids) reduce what is known as the bad lipids (triglycerides and low-density lipoprotein cholesterol) while raising the good lipid or high-density lipoprotein cholesterol (HDL-C), resulting in a generally advantageous decrease in the ratio of total to HDL-C [14,19]. The n-6 series of PUFAs are more prevalent in vegetable oils (such as those made from grape, corn, and sunflower seeds) than the n-3 series, which are primarily found in fish oils (such as those from cod, salmon, sardine, and sole). An optimum omega-6:omega-3 ratio indicates a good nutritional quality, and is required for chronic disease prevention and management.

This study aims to determine physicochemical parameters, fatty acid, phospholipids, and sterol compositions of the extracted oils of five selected underutilized leafy vegetables (*C. olitorius, C. argentea, C. crepidioides, S. macrocarpon,* and *L. taraxacifolia*) grown in southwest Nigeria with a view of assessing their health benefits and nutritional quality.

2. Materials and Methods

2.1. Sample collection

The vegetables including *C. olitorius, C. argentea, C. crepidioides, S. macrocarpon,* and *L. taraxacifolia* were collected from the farming village of Iworoko in Ekiti State, southwest Nigeria, in July 2022. The samples were taxonomically identified in the Department of Plant Science and Biotechnology, Federal University of Lafia, Nigeria.

2.2. Sample preparation and treatment

The vegetables underwent a rigorous cleaning process with tap water to eliminate any sand and extraneous particles. Then, they were sliced into segments, left to air-dry at room temperature for a fortnight until reaching a constant weight, and pulverized with a laboratory pestle and mortar. Each powdered sample was individually sealed in a ziplock plastic pouch and stored in a refrigerator at 4 °C until ready for analysis.

2.3. Oil extraction

Every 5 g portion of the air-dried vegetable sample underwent extraction for five hours using a Soxhlet apparatus, employing 250 ml of Analar-grade petroleum ether (boiling range: 40-60 °C) sourced from British Drug Houses, London. The extraction flask was withdrawn from the heating mantle upon

nearing depletion of the petroleum ether, then subjected to oven drying at 105 °C for an hour, followed by cooling in a desiccator prior to subsequent analysis [20,21].

2.4. Determination of physicochemical parameters of the extracted oil

The physicochemical attributes of the extracted oils, encompassing refractive index, peroxide value, acid value, iodine value, specific gravity, unsaponifiable matter, and saponification value were assessed by the methods outlined by AOCS [22–24].

2.5. Fatty acid profile

The oil obtained from each sample underwent conversion to methyl ester following the procedure outlined by Aremu et al. [25]. Specifically, 50 mg of the extracted oil underwent saponification with 3.4 ml of 0.5 M KOH solution in methanol at 95 ºC for 5 min. Neutralization was done with 0.7 M HCl by adding 3 ml of 14% boron trifluoride solution in methanol. The final mixture was heated at 90 ºC for 5 min to complete methylation. Fatty acid methyl esters were analyzed using an HP 6890 gas chromatograph (Bionics Scientific Technologies Ltd., India) equipped with a flame ionization detector, with nitrogen as the carrier gas. Separation of fatty acids was carried out using a polar (HP INNO Wax) capillary column (30 m × 0.53 mm × 0.25 μm), with an initial column temperature of 250 ºC, ramped at a rate of 5 ºC/min until reaching 310 ºC. Injector and detector temperatures were set at 310 ºC and 350 ºC, respectively. Identification of fatty acid methyl esters was done by comparing their retention times with standard FAMEs, and quantitative analysis was conducted by calculating the peak areas. A recovery rate of 0.96 was attained by using heptadecanoic ester as a reference.

2.6. Functional quality of the oil samples

Health status of the oil samples was assessed based on their fatty acid composition. It involved the evaluation of three indexes including hypocholesterolemic/hypercholesterolemic ratio (h/H) (Equation 1) [26,27], atherogenicity index (AI) (Equation 2), and thrombogenicity index (TI) (Equation 3) [28,29].

$$h/H = \frac{C18:1\omega9+C18:2\omega6+C20:4\omega6+C18:3\omega3+C20:5\omega3+C22:5\omega3+C22:6\omega3}{C14:0+C16:0} \quad \text{Equation 1}$$

$$AI = \frac{C12:0+4(C14:0)+C16:0}{\Sigma MUFA+\Sigma\omega6+\Sigma\omega3} \quad \text{Equation 2}$$

$$TI = \frac{C14:0+C16:0+C18:0}{0.5(\Sigma MUFA)+0.5(\Sigma\omega6)+3(\Sigma\omega3)+(\Sigma\omega3/\Sigma\omega6)} \quad \text{Equation 3}$$

Where, C12:0 (lauric acid); C14:0 (myristic acid); C16:0 (palmitic acid); C18:0 (stearic acid); C18:1ω9 (oleic acid); C18:2ω6 (linoleic acid); C18:3ω3 (linolenic acid); C20:4ω6 (arachidonic acid); C20:5ω3 EPA (eicosapentaenoic acid); C22:5ω3 DPA (docosapentaenoic acid); C22:6ω3 DHA (docosahexaenoic acid); MUFA (monounsaturated fatty acids).

2.7. Phospholipid analysis

Determining the phospholipid content provides insights into the emulsifying potency, nutritional benefits, and functional applications of the vegetable samples under analysis. The method employed to determine the phospholipid contents of the samples followed the procedure outlined by Aremu et al. (2017) and Aremu et al. (2021) [21,30]. Initially, 0.01 g of the extracted oils was placed into a test tube, and dried entirely by passing a stream of nitrogen gas. Subsequently, 0.04 ml of chloroform was added to the test tube, followed by addition of 0.1 ml chromogenic solution. The mixture was heated in a water bath at 100 °C for one minute, and then cooled. Next, 5 ml of hexane was added to the test tube, and the contents were gently agitated. The solvent and aqueous layers were allowed to separate. The hexane layer was recovered and concentrated to 1 ml for gas chromatograph

analysis using a flame photometric detector. For the gas chromatographic analysis, a capillary column (DB-5 or equivalent) with a length of 30 m, an internal diameter of 0.25 mm, and a film thickness of 0.25 µm was utilized. Helium was used as the carrier gas at a 1 ml/min flow rate. The oven temperature program started at 250 °C, held for 2 min, then increased to 300 °C at a rate of 3 °C/min, with a final hold at 300 °C for 20 min. The injector temperature was set at 280 °C, and the analysis was conducted in splitless mode, with an injection volume of 1 µl. Detection was carried out using a flame ionization detector (FID) set at 320 °C. The hydrogen flow rate for the FID was 30-40 ml/min, and the airflow rate was 400-450 ml/min. These optimized conditions ensured and accurate separation and identification of phospholipids.

2.8. Phytosterol analysis

The analysis of phytosterols followed the procedure outlined by AOAC [24]. A portion of the extracted oil from each sample was weighed and placed into a screw-capped test tube. Saponification was conducted at 95 °C for 30 min using 3 ml of 10% KOH in ethanol, with the addition of 0.2 ml of benzene to ensure the miscibility of the mixture. Subsequently, 3 ml of deionized water was added, followed by extraction of non-saponified materials using 2 ml of hexane. Three successive extractions with 2 ml of hexane were carried out for one hour, and additional 30 min was consided to ensure thorough extraction of sterols. The hexane was concentrated to 1 ml for gas chromatograph analysis, and 1 µl was injected into the injection port of the instrument. The gas chromatograph condition was the same as described in section 2.7. Sterol peaks were identified by comparing them with standard sterols under the chromatographic conditions.

3. Results and Discussion

3.1. Physicochemical properties of the oil samples

The physicochemical properties of the oils extracted from *C. olitorius, C. argentea, C. crepidioides, S. macrocarpon,* and *L. taraxacifolia* are presented in Table 1. Acid value of the oils ranged from 0.14 in *C. argentea* and *C. crepidioides* to 0.17 mg KOH/g in *L. taraxacifolia.* All the oils were characterized by low peroxide values, so that *C. crepidioides* had the highest peroxide value of 1.31 meq O_2/kg. The lowest peroxide value was observed in *L. taraxacifolia* oil (1.08 meq O_2/kg). The iodine values were below 100 g I_2/100 g, a threshold for oils classification, except for *C. crepidioides* oil, which displayed an iodine value of 102.21 g I_2/100 g. *L. taraxacifolia* oil was found to have the least iodine value (80.49 g I_2/100 g), and the coefficient of variation (CV) between the extracted oils was 10.89%. The mean saponification value for the oil samples and the CV(%) were 172.08 mg KOH/g and 4.98, respectively. *C. argentea* oil exhibited the highest saponification value (180.20 mg KOH/g), closely followed by *C. crepidioides* oil (179.10 mg KOH/g), and the least saponification value was observed in *L. taraxacifolia* oil (158.92 mg KOH/g). The value for unsaponifiable matter ranged from 0.65 to 0.88% for *S. macrocarpon* and *C. argentea* oils, respectively. The extracted oils' refractive index and specific gravity ranged from 1.29–1.40 and 0.93–1.10, respectively. The acid value describes a measure of free fatty acids, and it is a very significant element in the determination of oil freshness and quality, particularly in term of edibility. When the acid value of oil is low, it indicates that the oil has a higher stability per time, and will not get quickly rancid and peroxidized over time, which means the oil is fresh and of good quality. On the contrary, when the acid value is high, it suggests that the suitability of the oil for use in cooking (edibility) is in question. However, it is good to produce paints, shampoos, and liquid soap. The acid values of all the oil samples (0.14–0.17 mg KOH/g) were within the acceptable limit of ≤ 10 for edible oils [31]. Peroxide value is commonly used as a lipid oxidation indicator. The increased formation of reactive oxygen species and secondary oxidation products (such as lipid peroxides and aldehydes) is linked to adverse health effects, including the triggering of inflammatory and cardiovascular diseases. Furthermore, an oil with a high peroxide value

signifies a shorter shelf life and is unsuitable for human consumption [32]. A low peroxide value, on the other hand, indicates the quality and preservation state of the oil. The extracted oil samples with peroxide values 1.08–1.31 meq O_2/kg were safe for consumption because WHO/FAO specified a maximum permitted peroxide level of 10 meq O_2/kg. The iodine value of oil indicates its degree of unsaturation. The extracted oils from *C. argentea*, *C. crepidioides,* and *S. macrocarpon* demonstrated high iodine value (95.5-102.21 g I_2/100 g), comparable to Negash et al. [33] that reported iodine value of 96.40 g I_2/100 g for Niger seed oil. Vegetable oils with high iodine values often contain significant amounts of essential fatty acids like omega 3 and omega 6. These fatty acids are vital for human health as they are crucial to brain function, inflammation management, and heart health. In combination with acid values, saponification values provide information concerning the quantity, type of glyceride, and mean weight of the acid in an oil sample. The samples' saponification value-suggests that the extracted oils contained predominantly long-chain fatty acids (C18 and C16), which have saponification values between 168 to 196 mg KOH/g oil [34]. The unsaponifiable matters of the extracted oils were within the typical range of 0.5–2.5%. Unsaponifiable matter has a variable mixture of alcohols, aldehydes, ketones, fat-soluble vitamins, hydrocarbons, pigments, and sterols, which may be naturally occurring or formed during the degradation or processing of the fat. Sometimes, the unsaponification matter is used to characterize and authenticate the products [35,36].

Table 1- Physicochemical parameters of the oil samples

Parameter	CO	CA	CC	SM	LT	Mean	SD	CV (%)
Acid value (mg KOH/g)	0.16	0.14	0.14	0.15	0.17	0.15	0.01	6.67
Peroxide value (meq O_2/Kg)	1.10	1.12	1.31	1.12	1.08	1.15	0.09	7.82
Iodine value (g I_2/100 g)	81.25	95.50	102.21	97.84	80.49	91.46	9.96	10.89
Saponification value (mg KOH/g)	172.40	180.20	179.10	169.80	158.92	172.08	8.57	4.98
Unsaponifiable matter (%)	0.82	0.88	0.84	0.65	0.80	0.80	0.09	11.25
Specific gravity at 15 °C	1.05	1.08	0.95	0.93	1.10	1.02	0.08	7.84
Refractive index at 40 °C	1.36	1.40	1.39	1.29	1.33	1.35	0.05	3.70

CO: Cochorus olitorius; CA: Celosia argentea; CC: Crassocephalum crepidiodes; SM: Solanum macaropon; LT: Launaea taraxacifoli; SD: standard deviation; CV: coefficient of variation.

3.2. Fatty acid composition of the oil samples

Tables 2 and 3 depict the extracted oils' fatty acid composition. The predominant fatty acids in all the studied samples were palmitic acid (29.32–37.44%), linoleic acid (19.73–22.54%), γ-linolenic acid (13.91–16.29%), and α-linolenic acid (16.74–19.64%). The fatty acid composition of the oils extracted from *C. olitorius, C. argentea, C. crepidioides, S. macrocarpon,* and *L. taraxacifolia* was compaed with the common vegetable oils of sunflower oil, soybean oil, and canola oil. It was observed that our vegetable oils have palmitic acid (C16:0) ranging from 29.32 to 37.44%. Sunflower oil typically contains around 5 to 7%, soybean oil about 10%, and canola oil approximately 4%. The higher palmitic acid content in the studied samples indicates a more significant presence of saturated fatty acids, which might influence the oils' stability and shelf life. Moreover, linoleic acid, an essential polyunsaturated fatty acid, is present in the studied samples at levels between 19.73 and 22.54%. Sunflower oil is known for its high linoleic acid content, typically around 48 to 74%, while soybean oil contains about 50%, and canola oil has about 19 to 26%. The studied oils have a moderate linoleic acid content, which may contribute to their potential health benefits, particularly in reducing cholesterol levels. Furthermore, the oils show γ-linolenic acid content ranging from 13.91 to 16.29%, and α-linolenic acid content from 16.74 to 19.64%. In contrast, common vegetable oils like sunflower, soybean, and canola oil typically contain very low

levels of γ-linolenic acid, with α-linolenic acid content being higher in canola oil (around 8 to 10%), and soybean oil (approximately 7 to 10%). The higher levels of both γ- and α-linolenic acids in the studied samples are significant, as these fatty acids are known for their anti-inflammatory and cardiovascular health benefits.

L. taraxacifoli and *S. macaroon* had the highest composition of saturated fatty acids (SFAs). SFAs may have ties to obesity, heart disease, and other ailments, but in the soap-making industry, SFAs have multiple benefits such as giving soap hardness, which makes the soap last longer. People with sensitive skin have problems with soaps made from tallow, leading them to seek out soaps made from gentler vegetable sources. Polyunsaturated fatty acids constitute over two-thirds of the total unsaturated fatty acids, with linoleic acid being the most abundant unsaturated fatty acid (UFA) among the all oil samples (Tables 2 and 3). PUFAs are reported to control the regulation of the signaling pathway of antioxidants and cause the modulation of inflammatory-related processes. Furthermore, they are also responsible for influencing the lipid metabolism of the liver and other organ responses physiologically, including the central organ of circulation [37]. Linolenic acid is classified into α-linolenic acid (C18:3 n3) and γ-linolenic acid (C18:3 n6), which had two different structures. They are the second and third most abundant UFA in all the oil samples. Linoleic acid, the parent omega 6 fatty acid, promotes skin integrity, cell membrane integrity, the immune system, and the production of eicosanoids. α-linolenic acid, the most common omega 3 fatty acid found in leafy vegetables and vegetable oils, is associated with the valuable effects of minimizing atherogenic lipids and lipoproteins, hypertension, and inflammatory markers, thereby providing the most plausible reason to its physiological benefits in preventing diseases associated with cardiovascular system [13,38]. Observational evidence indicates that higher α-linolenic acid intake is beneficial for maintaining cognitive function and memory with aging [39,40]. Over the years, anthropological and zoological studies have confirmed the anti-inflammatory properties of γ-linolenic acid, a PUFA in the omega 6 series. Evening primrose and hempseed oils containing 8–12 and 6–8% γ-linolenic acid, respectively, treat inflammatory conditions [41]. Since each investigated vegetable oil in this study contains more than 13% γ-linolenic acid (Table 2), the oil samples can treat inflammation. Petroselinic acid, a rare isomer of oleic acid (positionally), and an essential precursor that is renewable was discovered in each oil sample (≤ 2.23%), implying that the oil samples could be used for other industrial purposes.

Table 2- Fatty acid composition (%) of the oil samples

Fatty Acid	CO	CA	CC	SM	LT	Mean	SD	CV(%)
Palmitic acid (C16:0)	29.32	33.40	29.66	37.44	35.54	33.07	3.57	10.80
Palmitoleic acid (C16:1; Cis-9)	1.62	1.52	0.57	1.44	1.13	1.26	0.43	34.13
Stearic acid (C18:0)	4.52	4.40	3.87	4.00	4.48	4.25	0.30	7.06
Petroselinic acid (C18:1; Cis-6)	2.23	2.11	2.09	1.98	2.09	2.10	0.09	4.29
Oleic acid (C18:1; cis-9)	5.21	5.13	4.90	4.61	5.20	5.01	0.26	5.19
Vaccenic acid (C18:1; trans-11)	0.01	0.01	0.03	0.01	0.01	0.02	0.01	50.00
Linoleic acid (C18:2; cis-9,12)	22.29	20.71	22.54	19.73	20.02	21.06	1.29	6.13
Linolelaidic acid (C18:2; trans-9,12)	0.01	0.01	0.02	0.01	0.00	0.01	0.01	100.00
γ-Linolenic acid (C18:3; cis-6,9,12)	15.72	14.87	16.29	13.91	14.54	15.07	0.94	6.24
α-Linolenic acid (C18:3; cis-9,12,15)	18.91	17.71	19.64	16.74	16.87	17.97	1.27	7.08
Arachidic acid (C20:0)	0.02	0.01	0.04	0.01	0.01	0.02	0.01	50.00
Gondoic acid (C20:1; cis-11)	0.03	0.02	0.07	0.02	0.02	0.03	0.02	66.67
Dihomo-γ-linolenic acid (C20:3; cis-8,11,14)	0.02	0.01	0.05	0.02	0.01	0.02	0.01	50.00
Icosatrienoic acid (C20:3; cis-11,14,17)	0.01	0.01	0.02	0.01	0.01	0.01	0.01	100.00

Arachidonic acid (C20:4; cis-5,8,11,14)	0.02	0.01	0.04	0.01	0.01	0.02	0.01	50.00
Eicosapentaenoic acid (C20:5; cis-5,8,11,14,17)	0.01	0.01	0.03	0.01	0.01	0.01	0.01	100.00
Behenic acid (C22:0)	0.01	0.02	0.04	0.01	0.01	0.02	0.01	50.00
Erucic acid (C22:1; cis-13)	0.02	0.02	0.05	0.02	0.02	0.02	0.02	100.00
Docosadienoic acid (C22:2; cis-13,16)	0.01	0.01	0.03	0.01	0.01	0.01	0.01	100.00
Cervonic acid (C22:6; cis-4,7,10,13,16,19)	0.01	0.01	0.02	0.01	0.01	0.01	0.01	100.00
Total	100	100	100	100	100			

CO: Cochorus olitorius; CA: Celosia argentea; CC: Crassocephalum crepidiodes; SM: Solanum macaropon; LT: Launaea taraxacifoli; SD: standard deviation; CV: coefficient of variation.

Table 3- Fatty acid distribution of the oil samples

Parameter (%)	CO	CA	CC	SM	LT
TSFA	33.87	37.82	33.61	41.46	40.04
MUFA	9.13	8.81	7.76	8.08	8.47
PUFA	57.00	53.34	58.67	50.45	51.48
TUFA	66.13	62.15	66.43	58.53	59.95
TEFA	56.99	53.34	58.65	50.37	51.48
TNEFA	43.01	46.66	41.35	49.63	48.52
O/L	0.23	0.25	0.22	0.23	0.26
PUFA/SFA	1.68	1.41	1.75	1.22	1.29

CO: Cochorus olitorius; CA: Celosia argentea; CC: Crassocephalum crepidiodes; SM: Solanum macaropon; LT: Launaea taraxacifoli; TSFA: total saturated fatty acid; MUFA: monounsaturated fatty acid; TUFA: total unsaturated fatty acids; TEFA: total essential fatty acid; TNEFA: total non-essential fatty acid; O/L: Oleic/Linoleic acids ratio; PUFA: polyunsaturated fatty acid; P/S: polyunsaturated fatty acid/saturated fatty acid ratio.

Cunha et al. [26] showed an increased vulnerability of heightened cholesterol levels in the blood if a man consumes a PUFA/SFA ratio of less than 0.45 ladened oils. The value of the PUFA/SFA ratios of all the oil samples in our study was significantly higher than 0.45. The oleic/linoleic (O/L) acids ratio has been associated with high stability and potentiality of the oil for deep frying [42,43]. Due to the low values of O/L level in all the samples (0.22–2.26), the oil samples may not be suitable for frying purposes. Recent human research suggests that the intake levels of omega 6 and omega 3 fatty acids and the ratio between them are crucial in obesity. This effect is linked to the metabolites of arachidonic acid, eicosanoids, and the increased activity of the cannabinoid system. However, it must be noted that an increased intake of docosahexaenoic acid (DHA) and eicosapentaenoic acid (EPA) can cause a reversed pathway [44]. The management and prevention of obesity require a healthy, balanced ratio of omega 6/omega 3, which is also essential for good health. The ω 6/ω 3 ratios of all vegetable oil samples were below the acceptable limit of 4 set by the UK Department of Health [45]. A high value of h/H of the oil directly correlates with enhancing cholesterol metabolism and improving the good lipid of HDL profile. As a result, the more the value in terms of number, the better it is for human intake [27]. Furthermore, previous research has indicated that oils with AI and TI values less than one can be included in a healthy diet [26,28]. Therefore, according to the findings presented in Table 4, these vegetable oils can be considered in a healthy diet.

The best ingredients to balance the SFAs in a soap recipe are UFAs. These vegetables have emollients and moisturizing properties, can offset the drying qualities of SFAs, and create a hard soap and conditioning. Palmitic acid content (37.44%) in *S. macrocarpon* oil was the highest in all the samples. *C. crepidioides* oil contained the lowest level of saturated fatty acids (SFA) (33.61%), while *S. macrocarpon* oil was the highest in SFA, with 41.46%. This was in contrast to the PUFA con-

tent, where *C. crepidioides* and *S. macrocarpon* oils were observed to have the highest and lowest PUFAs of 58.67% and 50.45%, respectively. The concentrations of MUFA in the oil samples ranged from 7.76 to 9.13%, with oleic acid accounting for at least 4.61% of the fatty acids in each sample. The fatty acid profiles revealed that each vegetable oil sample's total essential fatty acid content was greater than 50%. The oleic to linoleic (O/L) and PUFA to SFA ratios ranged from 0.22 to 0.26 and 1.22 to 1.75, respectively. The results of the calculated ratios of omega 6 to omega 3 fatty acids (ω 6/ω 3), h/H, AI, and TI were found in the range of 1.98–2.05, 1.10–1.59, 0.44–0.64, and 0.4–0.58, respectively (Table 4).

Table 4- Functional quality of the oil samples

Quality parameter	CO	CA	CC	SM	LT
ω 6/ω 3	2.01	2.01	1.98	2.02	2.05
h/H	1.58	1.30	1.59	1.10	1.19
AI	0.44	0.54	0.45	0.64	0.59
TI	0.42	0.50	0.40	0.58	0.57

CO: Cochorus olitorius; CA: Celosia argentea; CC: Crassocephalum crepidiodes; SM: Solanum macaropon; LT: Launaea taraxacifoli; h/H: hypocholesterolemic/hypercholesterolemic ratio; AI: atherogenicity index; TI: thrombogenicity index.

3.3. Phospholipid composition of the oil samples

The results of the phospholipid composition in our samples are shown in Table 5. The phospholipid composition (mg/100 g) ranged from 4.36 to 67.96 mg/100 g. The most prominent phospholipid in the samples was 67.96 mg/100 g phosphatidylcholine in *L. taraxacifoli,* followed by 57.73 mg/100 g in *C. crepidiodes,* 54.15 mg/100 g in *C. argentea,* and 53.15 mg/100 g in *S. macaropon.* Lastly, the lowest was 50.81 mg/100 g for *C. olitorius* leaves. Of the phospholipids available in plants and animals, phosphatidylcholine was among the copious as it can take up to almost 40 to 45% of the whole and is responsible for its contribution to the construction of the membrane bilayer. Phosphatidylethanolamine had the second-highest composition after phosphatidylcholine, with *C. crepidiodes* being the highest and *L. taraxacifoli* being the lowest.

Table 5- Phospholipid composition of the oil samples

Phospholipid (mg/100 g)	A	B	C	D	E	Mean	SD	CV (%)
Phosphatidylethanolamine	21.75	20.48	27.93	22.81	27.43	24.08	3.39	14.09
Phosphatidylcholine	50.81	54.22	57.73	53.15	67.96	56.77	6.73	11.85
Phosphatidylserine	1.14	1.71	1.92	1.33	2.09	1.64	0.40	24.28
Phosphatidyglycerol	13.03	16.14	14.78	14.01	13.53	14.30	1.22	8.51
Lysophosphatidylethanolamine	1.13	1.44	2.05	1.10	2.07	1.56	0.48	30.64
Lysophosphatidylcholine	1.52	1.88	2.14	2.15	1.84	1.91	0.26	13.56
Phosphatidylinositol	14.20	16.07	21.07	18.08	16.33	17.15	2.59	15.08
Phosphatidic acid	4.36	5.62	8.80	4.87	7.18	6.17	1.82	29.48

A: Cochorus olitorius; B: Celosia argentea; C: Crassocephalum crepidiodes; D: Solanum macaropon; E: Launaea taraxacifoli; SD: standard deviation; CV: coefficient of variance.

3.4. Phytosterol composition of the oil samples

The most abundant phytosterol in the samples was sitosterol found in *S. macaropon* and *C. crepidiodes* (Table 6). It was followed by campesterol for *C. argentea* and *C. crepidiodes.* Savenasterol concentration ranged from 4.47 to 3.3 mg/100 g for *C. argentea* and *S. macaropon,* respectively. The level of cholesterol was in a minimal amount in each sample, with the highest recorded as 1.92 $\times 10^{-4}$. According to Aremu et al. [46], the average dai-

ly intake of sterols differs from person to person, but usually, it is between 160 to 400 mg/100 g of food. Therefore, consumption of our studied oils can provide the individuals with a part of recommended daily phytosterol in the diet. Importantly, phytosterols can lower blood cholesterol, and also fight and prevent inflammation, oncological diseases, atherogenicity, and oxidation activities. Therefore, they are very important in the pharmaceutical industry [47].

Table 6- Phytosterol composition of the oil samples

Phytosterol (mg/100 g)	A	B	C	D	E	Mean	SD	CV (%)
Cholesterol	$4.16E^{-04}$	$2.72E^{-04}$	$2.09E^{-04}$	$3.69E^{-04}$	$1.92E^{-04}$	$2.92E^{-04}$	$9.82E^{-05}$	33.63
Cholestanol	$4.72E^{-05}$	$8.04E^{-05}$	$6.73E^{-05}$	$5.61E^{-05}$	$6.32E^{-05}$	$6.28E^{-05}$	$1.24E^{-05}$	19.75
Ergosterol	$1.83E^{-03}$	$1.83E^{-03}$	$1.83E^{-03}$	$1.83E^{-03}$	$1.78E^{-03}$	$1.82E^{-03}$	$2.24E^{-05}$	1.23
Campesterol	5.02	6.1	6.01	6.29	5.39	5.76	0.53	9.2
Stigmasterol	2.87	3.28	4.34	3.18	3.59	3.45	0.56	16.23
Savenasterol	3.98	3.79	4.47	3.3	4.26	3.96	0.45	11.36
Sitosterol	23.55	24.61	25.79	25.53	23.61	24.62	1.05	4.26

A: Cochorus olitorius; B: Celosia argentea; C: Crassocephalum crepidiodes; D: Solanum macaropon; E: Launaea taraxacifoli; SD: standard deviation; CV: coefficient of variance.

4. Conclusion

In nature, leafy vegetables are considered to be the wealthiest plant food. A significant amount of fats (both saturated and unsaturated) has been discovered in them. It makes the leafy vegetables very significant contributor to human health in a positive way. The lipid profile of the plants studied in this research will add to the existing body of knowledge, which helps advanced higher research in food industry and industrial chemistry. Fatty acid profile of five leafy vegetables (*C. olitorius, C. argentea, C. crepidioides, S. macrocarpon,* and *L. taraxacifolia*) and their functional quality indices were evaluated in this study. About a score, individual fatty acids were identified in all the vegetable oil samples. The profile revealed that unsaturated fatty acids, particularly PUFA (linoleic, α-linolenic, and γ-linolenic acids), were predominant in all oil samples. The extracted vegetable oil samples displayed satisfactory nutritional quality due to the predominance of PUFA associated with preventing long-term and degenerative diseases. AI and TI indexes and h/H ratio suggested that the oil may have a protective action on the cardiovascular system. The omega 6/omega 3 ratios of all oil samples were within the optimum recommended ranges which indicate that the oils are nutritionally beneficial in terms of good health and preventing and managing obesity. The phospholipids of phosphatidylcholine and phosphatidylethanolamine were also found in the oil samples, which in turn comprise 45-55% and 15-25% of the biological membrane, and play a significant role in anatomical and physiological function and integrity.

5. Acknowledgement

Some of the analyses were carried out in the laboratory of Multi–environmental Management Consultants Ltd., Ikorodu, Lagos, Nigeria. So one of the authors, Zeenat Lami Usman appreciates Honorable Atanda Akeem Bello for the technical assistance rendered during the course of analysis.

6. Conflict of Interest

The authors declare no conflict of interest to this work.

References

1. Chacha JS, Laswai HS. Micronutrients potential of underutilized vegetables and their role in fighting hidden hunger. International Journal of Food Science. 2020. https://doi.org/10.1155/2020/9408315

2. Sultanbawa Y, Sivakumar D. Enhanced nutritional and phytochemical profiles of selected underutilized fruits, vegetables, and legumes. Current Opinion in Food Science. 2022; 46: 100853.

https://doi.org/10.1016/j.cofs.2022.100853

3. Mungofa N, Sibanyoni JJ, Mashau ME, Beswa D. Prospective role of indigenous leafy vegetables as functional food ingredients. Molecules. 2022; 27(22): 7995. https://doi.org/10.3390/molecules27227995

4. Amao I. Health benefits of fruits and vegetables: Review from Sub-Saharan Africa. In: importance of quality vegetables to human health. IntechOpen. London. 2018; 22: 33-53. https://doi.org/10.5772/intechopen.74472

5. Schreinemachers P, Simmons EB, Wopereis MC. Tapping the economic and nutritional power of vegetables. Global Food Security. 2018; 16: 36-45. https://doi.org/10.1016/j.gfs.2017.09.005

6. Food and Agriculture Organization. Promoting fruit and vegetable consumption, Santiago de Chile. 2021. Available at: http://www.euro.who.int/en/health-topics/disease-prevention/nutrition/activities/technical-support-to-member-states/promoting-fruit-and-vegetable-consumption.

7. Frank SM, Webster J, McKenzie B, Geldsetzer P, Manne-Goehler J, Andall-Brereton G, et al. Consumption of fruits and vegetables among individuals 15 years and older in 28 low-and middle-income countries. The Journal of Nutrition. 2019; 149(7): 1252-1259. https://doi.org/10.1093/jn/nxz040

8. Kaur H, Aeri BT. Protective impact of fruits and vegetable intake on cardiovascular risk factors-a review. Journal of Clinical & Diagnostic Research. 2019; 13(5): 6-9. https://doi.org/10.7860/jcdr/2019/41330.12884

9. Smith L, López Sánchez GF, Veronese N, Soysal P, Oh H, Barnett Y, et al. Fruit and vegetable intake and non-communicable diseases among adults aged ≥ 50 years in low-and middle-income countries. The Journal of Nutrition, Health & Aging. 2022; 26(11): 1003-1009. https://doi.org/10.1007/s12603-022-1855-z

10. Nyanchoka MA, van Stuijvenberg ME, Tambe AB, Zuma MK, Mbhenyane XG. Fruit and vegetable consumption patterns and risk of chronic diseases of lifestyle among university students in Kenya. International Journal of Environmental Research and Public Health. 2022; 19(12): 6965. https://doi.org/10.1017/s0029665121000021

11. Zhan J, Liu YJ, Cai LB, Xu FR, Xie T, He QQ. Fruit and vegetable consumption and risk of cardiovascular disease: A meta-analysis of prospective cohort studies. Critical Reviews in Food Science and Nutrition. 2017; 57(8): 1650-1663. https://doi.org/10.1080/10408398.2015.1008980

12. Zhao X, Xiang X, Huang J, Ma Y, Sun J, Zhu D. Studying the evaluation model of the nutritional quality of edible vegetable oil based on dietary nutrient reference intake. ACS Omega. 2021; 6(10): 6691-6698. https://doi.org/10.1021/acsomega.0c05544

13. Shen J, Liu Y, Wang X, Bai J, Lin L, Luo F, et al. A comprehensive review of health-benefiting components in rapeseed oil. Nutrients. 2023; 15(4): 999. https://doi.org/10.3390/nu15040999

14. Farvid MS, Ding M, Pan A, Sun Q, Chiuve SE, Steffen LM, et al. Dietary linoleic acid and risk of coronary heart disease: a systematic review and meta-analysis of prospective cohort studies. Circulation. 2014; 130(18): 1568-78. https://doi.org/10.1161/CIRCULATIONAHA.114.010236

15. Harris WS, Poston WC, Haddock CK. Tissue n−3 and n−6 fatty acids and risk for coronary heart disease events. Atherosclerosis. 2007; 193(1): 1-10. https://doi.org/10.1016/j.atherosclerosis.2007.03.018

16. Mazidi M, Shekoohi N, Katsiki N, Banach M. Omega-6 fatty acids and the risk of cardiovascular disease: insights from a systematic review and meta-analysis of randomized controlled trials and a Mendelian randomization study. Archives of Medical Science. 2022; 18(2): 466. https://doi.org/10.5114/aoms/136070

17. Mozaffarian D, Micha R, Wallace S. Effects on coronary heart disease of increasing polyunsaturated fat in place of saturated fat: a systematic review and meta-analysis of randomized controlled trials. Plos Medicine. 2010; 7(3): e1000252. https://doi.org/10.1371/journal.pmed.1000252

18. Mukhopadhyay S, Goswami S, Mondal SA, Dutta D. Dietary fat, salt, and sugar: a clinical perspective of the social catastrophe. In: Preuss HG, Bagchi D. (eds.). Dietary sugar, salt and fat in human health. Academic Press. 2020; 67-91. https://doi.org/10.1016/b978-0-12-816918-6.00003-2

19. Mensink RP, Katan MB. Effect of dietary fatty acids on serum lipids and lipoproteins. A meta-analysis of 27 trials. Arteriosclerosis, Thrombosis and Vascular Biology. 1992; 12(8): 911-919. https://doi.org/10.1161/01.atv.12.8.911

20. Association of Official Agricultural Chemists (AOAC). Official Methods of Analysis. 18th Edition. 2007.

21. Aremu MO, Ibrahim H, Andrew C. Comparative

studies on the lipid composition of blood Plum (Haematostaphis barteri) pulp and seed oils. The Open Biochemistry Journal. 2017; 11: 94.
https://doi.org/10.2174/1874091X01711010094

22. American Oil Chemists Society (AOCS). Official methods and recommended practices. Champaign, USA. 1993.

23. American Oil Chemists Society (AOCS). Official methods and recommended practices. Champaign, USA. 2004.

24. American Oil Chemists Society (AOCS). Official methods of analysis Cd 8b-90. Champaign, USA. 2005.

25. Aremu MO, Awagulu MS, Ayakeme EB, Zando C, Bini ME, Omosebi MO, et al. Lipid profile and health attributes of mango (*Mangifera indica L.*) seed kernel and cashew (*Anacardium occidentale L.*) nut kernel: a comparative study. Human, Health & Halal Metrics. 2022; 3(2): 14-22.
https://doi.org/10.30502/jhhhm.2022.364887.1061

26. Cunha VM, da Silva MP, de Sousa SH, do Nascimento Bezerra P, Menezes EG, da Silva NJ, et al. Bacaba-de-leque (Oenocarpus distichus Mart.) oil extraction using supercritical CO_2 and bioactive compounds determination in the residual pulp. The Journal of Supercritical Fluids. 2019; 144: 81-90.
https://doi.org/10.1016/j.supflu.2018.10.010

27. Santos-Silva J, Bessa RJ, Santos-Silva FJ. Effect of genotype, feeding system and slaughter weight on the quality of light lambs: II. Fatty acid composition of meat. Livestock Production Science. 2002; 77(2-3): 187-194.
https://doi.org/10.1016/S0301-6226(02)00059-3

28. Pinto RH, Sena C, Santos OV, Da Costa WA, Rodrigues AD, Junior RC. Extraction of bacaba (*Oenocarpus bacaba*) oil with supercritical CO_2: global yield isotherms, fatty acid composition, functional quality, oxidative stability, spectroscopic profile and antioxidant activity. Grasas y Aceites. 2018; 69(2): e246.
https://doi.org/10.3989/gya.0883171

29. Ulbricht TL, Southgate DA. Coronary heart disease: seven dietary factors. The Lancet. 1991; 338(8773): 985-992.
https://doi.org/10.1016/0140-6736(91)91846-M

30. Ibeto CN, Okoye CO, Ofoefule AU. Comparative study of the physicochemical characterization of some oils as potential feedstock for biodiesel production. International Scholarly Research Notices. 2012.
https://doi.org/10.5402/2012/621518

31. Chew SC. Cold-pressed rapeseed (*Brassica napus*) oil: chemistry and functionality. Food Research International. 2020; 131: 108997.
https://doi.org/10.1016/B978-0-12-818188-1.00007-4

32. Negash YA, Amare DE, Bitew BD, Dagne H. Assessment of quality of edible vegetable oils accessed in Gondar city, northwest Ethiopia. BMC Research Notes. 2019; 12(1): 1-5.
https://doi.org/10.1186/s13104-019-4831-x

33. Ivanova M, Hanganu A, Dumitriu R, Tociu M, Ivanov G, Stavarache C, et al. Saponification value of fats and oils as determined from ^{1}H-NMR data: the case of dairy fats. Foods. 2022; 11(10): 1466.
https://doi.org/10.3390/foods11101466

34. Badifu GI. Unsaponifiable matter in oils from some species of Cucurbitaceae. Journal of Food Composition and Analysis. 1991; 4(4): 360-365.
https://doi.org/10.1016/0889-1575(91)90023-Y

35. Shahidi F. Quality characteristics of edible oils. In: Shahidi F, Spanier AM, Ho CT, Braggins T. (eds.). Quality of fresh and processed foods. Advances in Experimental Medicine and Biology. 2004; 542: 239-249.
https://doi.org/10.1007/978-1-4419-9090-7_17

36. Djuricic I, Calder PC. Beneficial outcomes of omega-6 and omega-3 polyunsaturated fatty acids on human health: An update for 2021. Nutrients. 2021; 13(7): 2421.
https://doi.org/10.3390/nu13072421

37. Sala-Vila A, Fleming J, Kris-Etherton P, Ros E. Impact of α-linolenic acid, the vegetable ω-3 fatty acid, on cardiovascular disease and cognition. Advances in Nutrition. 2022; 13(5): 1584-602.
https://doi.org/10.1093/advances/nmac016

38. Nooyens AC, Van Gelder BM, Bueno-de-Mesquita HB, Van Boxtel MP, Verschuren WM. Fish consumption, intake of fats and cognitive decline at middle and older age: the Doetinchem Cohort Study. European Journal of Nutrition. 2018; 57: 1667-1675.
https://doi.org/10.1007/s00394-017-1453-8

39. Yamagishi K, Ikeda A, Chei CL, Noda H, Umesawa M, Cui R, et al. Serum α-linolenic and other ω-3 fatty acids, and risk of disabling dementia: community-based nested case–control study. Clinical Nutrition. 2017; 36(3): 793-797.
https://doi.org/10.1016/j.clnu.2016.05.011

40. Rezapour-Firouzi S. Herbal oil supplement with hot-nature diet for multiple sclerosis. In: Watson RR, Killgore WDS. (eds.). Nutrition and lifestyle in neurological autoimmune diseases. Academic Press. 2017; 229-245.

https://doi.org/10.1016/B978-0-12-805298-3.00024-4

41. Aremu MO, Ibrahim H, Aremu SO. Lipid composition of black variety of raw and boiled tigernut (*Cyperus esculentus L.*) grown in north-east Nigeria. Pakistan Journal of Nutrition. 2016; 15(5): 427-438. https://doi.org/10.3923/pjn.2016.427.438

42. Branch WD, Nakayama T, Chinnan MS. Fatty acid variation among US runner-type peanut cultivars. Journal of the American Oil Chemists' Society. 1990; 67(9): 591-593. https://doi.org/10.1007/BF02540772

43. Simopoulos AP. An increase in the omega-6/omega-3 fatty acid ratio increases the risk for obesity. Nutrients. 2016; 8(3): 128. https://doi.org/10.3390/nu8030128

44. Valencak TG, Gamsjäger L, Ohrnberger S, Culbert NJ, Ruf T. Healthy n-6/n-3 fatty acid composition from five European game meat species remains after cooking. BMC Research Notes. 2015; 8(1): 1-6. https://doi.org/10.1186/s13104-015-1254-1

45. Aremu MO, Waziri AA, Faleye FJ, Magomya AM, Okpaegbe UC. Lipids profile of bitter melon (*Momordica charantia L.*) fruit and ebony (*Diospyros mespiliformis* Hochst ex A. DC.) tree fruit pulp. Bangladesh Journal of Scientific and Industrial Research. 2019; 54(4): 367-374.

46. Aremu MO, Ajine PL, Omosebi MO, Baba NM, Onwuka JC, Audu SS, et al. Lipid profiles and health promoting uses of carrot (*Daucus carota L.*) and cucumber (*Cucumis sativus L.*). International Journal of Sciences. 2021; 10(07): 22-29.

47. Poli A, Marangoni F, Corsini A, Manzato E, Marrocco W, Martini D, et al. Phytosterols, cholesterol control, and cardiovascular disease. Nutrients. 2021; 13(8): 2810. https://doi.org/10.3390/nu13082810