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Physicochemical properties, lipid composition, and fat-soluble vitamins of oils and antioxidant activity of methanol extracts from tubers, peels, and leaves of sweet potato (Ipomoea batatas L.) in Nigeria

Pheobe Sorphy Samuel[a,*], Matthew Olaleke Aremu[a], Jude Chinedu Onwuka[a], Jibrin Mohammed[b], Charity Musa[a]

[a]*Department of Chemistry, Faculty of Physical Sciences, Federal University of Lafia, Nasarawa State, Nigeria*
[b]*Department of Chemistry, Faculty of Natural & Applied Sciences, Nasarawa State University, Keffi, Nigeria*

Abstract

Sweet potato (Ipomoea batatas L.) peels and leaves are often discarded, contributing to post-harvest losses. This study compared physicochemical properties, fatty acids, phospholipids, phytosterols, and fat-soluble vitamins (A and E) in n-hexane-extracted oils from tubers, peels, and leaves collected in Nigeria. Water-soluble vitamins, phytochemicals, and antioxidant activity were assessed in separate methanol extracts. Physicochemical properties were evaluated using standard methods; fatty acids by gas chromatography; phospholipids by thin-layer chromatography; phytosterols by high-performance liquid chromatography; and vitamins by spectrophotometric methods. Antioxidant activity was evaluated using 2,2-diphenyl-1-picrylhydrazyl (DPPH), hydrogen peroxide scavenging, and reducing-power assays. Leaf oil contained the highest total unsaturated fatty acids (60.1%; unsaturated/saturated fatty acid ratio, 1.51), total phospholipids (152.0 mg/100 g), phytosterols (185.3 mg/100 g), and vitamin E (53.1 mg/100 g). Leaf methanol extract had the strongest apparent antioxidant activity (extrapolated DPPH IC_{50}, 31.6 μg/mL). Peel oil had the highest carotenoid content (expressed as vitamin A, 7.08 mg/100 g), while peel methanol extract had the highest riboflavin content (16.11 mg/kg). Tuber oil had the lowest peroxide value (0.6 meq O_2/kg). All oils met the Codex peroxide-value benchmark, but moisture, impurities, and peel and leaf acid values exceeded relevant limits. Results represent analytical replicates of one pooled sample per tissue, limiting population-level inference. These findings suggest potential uses for sweet potato by-products, subject to independent biological replication and oil-yield confirmation.

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1. Introduction

Between 691 and 783 million people faced hunger worldwide in 2022 (mid-range estimate 735 million), and about one in five people in Africa was affected [1]. Nigeria is one of the countries suffering from food insecurity, with various sources of threats

*Corresponding Author Tel. No.: +234-803-665-1791.
Email address: sorphy.samuel@gmail.com (Pheobe Sorphy Samuel)

such as climate change, natural disasters, poverty (40.1% of people below the 2018/19 national monetary poverty line and 63% multidimensionally poor in 2022) [2], and an insurgency that has been ongoing since 2009 [3]. The utilization of underutilized plants and their by-products could be a potential solution for nutrient deficiencies and post-harvest losses in addition to being a cost-effective process.

Sweet potato (Ipomoea batatas L.) is the seventh most important staple crop worldwide. It is grown in more than 110 countries, with China the largest producer [4]. It is a perennial herbaceous vine plant belonging to the family Convolvulaceae and the dicotyledonous angiosperms that are characterized by their tuberous roots [5]. They are known to have a good amount of carbohydrates, fiber, vitamins (especially provitamin A), minerals, polyphenols, flavonoids, and anthocyanins [6–8].

The tuberous roots are well recognized and consumed throughout the world, while the leaves and peels are underutilized by most of the population. In some countries, sweet potato leaves are considered as animal fodder, or they are completely wasted; however, there is a significant amount of nutrients and bioactive compounds found in them [9, 10]. Sweet potato leaves contain high levels of polyphenols, lutein, and minerals [10–13]. Post-harvest losses along the sweet potato value chain of up to 25% have been reported [14], and the leaves are still discarded as waste in many production areas [15]. This loss of material is not only a financial burden to the farmers, but it also constitutes an environmental hazard and a missed opportunity for nutritional supplementation.

Meanwhile, lipid deficiencies, particularly in essential fatty acids (linoleic and α-linolenic acid) and fat-soluble vitamins (A, D, E, and K) are associated with a wide range of health issues, including impaired brain function, delayed wound healing, and increased risk of chronic diseases such as cardiovascular disease and cancer [16, 17].

The lipid content in sweet potato has been studied less than carbohydrates and phenolics. Initial reports have demonstrated that palmitic acid and linoleic acid are the most abundant fatty acids in sweet potato tubers, with an overall higher percentage of unsaturated over saturated fatty acids [18, 19]. Lipidomic profiling has identified phosphatidylcholine, phosphatidylethanolamine, phosphatidylinositol, phosphatidylserine and other lipid classes in sweet potato leaves and roots [20]. Phytosterol analysis has detected β-sitosterol as the dominant phytosterol, which constitutes 55-78% of the total phytosterols followed by campesterol [18]. Despite the results obtained, there is still limited information on the biochemical composition of bioactive lipid compounds in sweet potato's tuber, peel, and leaves. Most of the previous studies were conducted in regard to one part of the plant only or did not provide in-depth characteristics of lipid components, such as fatty acid profile or the content of additional bioactive compounds.

This lack of information is hindering the development of the utilization of sweet potato by-products and the recognition of their health benefits.

Thus, the current research aimed to achieve the following goals: (1) extract oil from the tuber, peel, and leaves of I. batatas, (2) determine their physicochemical parameters, (3) perform fatty acid composition, (4) analyze the content of phospholipids, (5) assess the quantity of phytosterols and fat-soluble vitamins in the oils, and (6) determine the water-soluble vitamins, phytochemical contents, and antioxidant properties of methanol extracts of the same tissues. The results of the present study might contribute to reducing food waste in the agricultural sector and the development of the production of functional food products and medicines based on the by-products of sweet potato.

2. Materials and methods

2.1. Study area and sample collection

Fresh sweet potato (Ipomoea batatas L.) plants, local African Landraces were obtained from the farm of Mr. Yahaya in Agudu, Lafia, Nasarawa State, Nigeria. The coordinates of this location are 8.518666°E and 8.472137°N. The plants were authenticated at the Department of Plant Science and Biotechnology, Federal University of Lafia with voucher Number FUL/PSB/H.LAB/0099. Fifty tubers and 50 vines were selected randomly from the total harvest of the farm. Plants of similar color and stage of maturity were selected to ensure uniformity in sample quality. Plants that were diseased, pest-infested, or physically damaged were excluded from the sample.

2.2. Sample preparation

Sweet potato (Ipomoea batatas L.) samples comprising tubers, leaves, and peels were collected from farms and authenticated at the Plant Science Biotechnology Laboratory, Federal University of Lafia, Nigeria. All samples were rinsed thoroughly using running tap water to remove adhering soil, debris, and other foreign materials, drained and surface-dried. The tubers were manually peeled using a clean stainless-steel knife and their peels saved for analysis. The peeled tubers were soaked in clean water to prevent enzymatic browning and were then cut into 2–3 mm slices with a mechanical slicer. The tuber slices, leaves, and peels were separately dried. The leaves were dehydrated while the peels were dried in a well-ventilated oven at 40 °C for 8 h, the tuber slices were then dried to constant weight at the same temperature. The dried leaves, peels, and tuber slices were ground into a fine powder using a laboratory blender and grinder respectively. Each ground sample was then sieved through a 0.5 mm mesh.

The tuber, leaf, and peel powders were separately stored in labelled airtight polythene bags until physicochemical, nutritional, and phytochemical analyses were performed [15].

2.3. Oil extraction (n-hexane extraction)

Oil was extracted from the three prepared samples using the maceration method. To 300 g of each powdered sample (tuber, peel, or leaves), 500 mL of freshly prepared n-hexane was added. The mixture was left to macerate with occasional shaking for 48 h. It was then filtered off, and the solvent was evaporated at 40 °C under reduced pressure by means of a rotary evaporator to obtain pure oil. The amount of oil obtained was determined gravimetrically; however, the individual yield values were not retained in the laboratory records available for this study and are therefore not reported (see Section 4.7). The pure oil was stored in an amber glass bottle at 4 °C until assayed for various physicochemical properties. This fraction of oil was used to determine physicochemical, fatty acid, phospholipid, phytosterol, as well as fat-soluble vitamin analysis (A and E).

2.4. Physicochemical analysis

The physicochemical properties of the oil were determined by standard procedures following AOCS Official Methods Cc 1-25 (melting point), Cc 6-25 (cloud point), Cc 9a-48 (smoke, flash and fire points), Cc 7-25 (refractive index), Cc 10c-95 (specific gravity), Ca 2b-38 (moisture and volatile matter), Ca 3a-46 (insoluble impurities), Ca 5a-40 (free fatty acids and acid value), Cd 3-25 (saponification value), Cd 1d-92 (iodine value, Wijs), Cd 8b-90 (peroxide value) and Ca 6a-40 (unsaponifiable matter) [21], and the procedures of Onwuka [22] and, where applicable, the AOAC Official Methods [23]. The following physical properties were analyzed: melting point, cloud point, smoke point, flash point, fire point, refractive index, specific gravity, moisture content, impurities, acid value, free fatty acid, saponification value, iodine value, peroxide value, unsaponifiable matter, and viscosity. The pH of the oil samples was not determined because pH is not an appropriate analytical parameter for non-aqueous systems. All measurements were made as described in Section 2.11.

2.5. Fatty acid analysis

The percentage composition of fatty acids in the extracted oils was determined by Gas Chromatography (GC) according to AOCS Official Methods Ce 2-66 (preparation of methyl esters) and Ce 1i-07 (fatty acid composition by capillary GC) [21] (equivalent to ISO 12966-2 and ISO 12966-4 [24]). Total lipid for this analysis was extracted using chloroform:methanol (2:1 v/v, containing 0.1 mg/100 g BHT) [25], and the resulting extract was converted to fatty acid methyl esters (FAME) by transmethylation with 2 M methanolic sodium hydroxide followed by 2 M methanolic hydrochloric acid. FAMEs were analysed by gas chromatography (Shimadzu GC-2014, Japan) fitted with a flame ionisation detector (FID) and an Omegawax™ 320 fused-silica capillary column (30 m × 0.32 mm × 0.25 μm). FAME dissolved in hexane was injected at an injector temperature of 250 °C; the FID detector was held at 260 °C and the column oven at 200 °C, isothermal, for a 60-minute run, with hydrogen or helium used as the carrier gas.

Individual fatty acids were identified by comparison of retention times with authentic standards, and results were expressed as a percentage of total fatty acids.

2.6. Phospholipid and phytosterol analysis

Phospholipids were separated by two-dimensional thin-layer chromatography (TLC) on magnesium acetate-impregnated silica gel H plates, using the solvent system conventionally applied to plant phospholipid class separation [20]: chloroform-methanol-28% ammonia solution (65:35:5, v/v/v) in the first direction and chloroform-acetone-methanol-acetic acid-water (5:2:1:1:0.5, v/v/v/v/v) in the second direction, after drying the plate between runs. Spots were visualised under iodine vapour and identified by co-chromatography with authentic phospholipid standards. Phosphorus content of each spot was determined colorimetrically (AOCS Ca 12-55; ISO 10540-1) [21, 24] and converted to phospholipid mass using the standard phosphorus-to-phospholipid conversion factor of 25 (phospholipid, mg = phosphorus, mg × 25). For phytosterol separation, HPLC analysis was performed on a C18 reversed-phase column with an isocratic mobile phase of acetonitrile:isopropanol (in the range 50:50 to 85:15, v/v, optimised for resolution of the sterols present), a flow rate of 1 mL/min, and UV detection at 205 nm, a wavelength routinely used for free sterols owing to their weak native chromophore.

The individual sterols were identified and quantified by comparison of retention time and peak area with authentic standards. Sterol composition is conventionally determined by capillary GC (AOCS Ch 6-91) [21] as a complementary approach.

2.7. Fat-soluble vitamin analysis

Fat-soluble vitamins (A and E) in the oil extracts were determined using a UV-VIS spectrophotometer according to the methods described by Nielsen [26]. Total carotenoids (reported as vitamin A) were determined by the absorbance of the oil extract in petroleum ether/hexane at 450 nm against a β-carotene calibration curve, using the specific absorption coefficient of β-carotene in that solvent; because this method does not distinguish individual provitamin-A carotenoids from non-provitamin-A carotenoids, the results are reported as total carotenoids expressed as vitamin A equivalents rather than as a measure of retinol activity [27]. Total tocopherols (reported as vitamin E) were determined colorimetrically by the Emmerie-Engel reaction, based on the reduction of ferric chloride to the ferrous state by tocopherols and the subsequent formation of a coloured complex with 2,2'-bipyridyl, read at 520 nm against a standard tocopherol calibration curve. Standard solutions were used for calibration of both assays.

2.8. Methanol extract preparation (for phytochemicals and water-soluble vitamins)

For analysis of water-soluble vitamins (B1, B2, B6, C), total phenolic content, total flavonoid content, and antioxidant assays, a separate methanol extract was prepared. Three grams of each sample (tuber, peel, leaves powder) were extracted with 70% methanol (1:10 w/v) by continuous shaking for 8 h at room temperature. Reported vitamin and phytochemical concentrations are expressed relative to the dry sample powder on this 1:10 (w/v) extraction basis (3 g powder per 30 mL 70% methanol), not relative to the final liquid extract volume. The extract was filtered, and methanol was evaporated under reduced pressure. The aqueous extract was used for phytochemical analysis and determination of water-soluble vitamins. This method was used since water-soluble vitamins (including vitamin C) are not soluble in oil and have to be analyzed separately in polar extract [26]. Standard procedures for these vitamins are described in [28].

2.9. Phytochemical analysis

The phytochemical content of sweet potato tuber, leaf, and peel was analyzed according to the procedures used and reported by the cited references for each parameter. All procedures were carried out with appropriate reagent blanks and calibration standards. The method, reference standard and unit of expression for each parameter are given below.

Total Phenolic Content (TPC): Assessed by the Folin-Ciocalteu method, read at 760 nm against a gallic acid calibration curve (0.01–0.05 mg/mL) [29]. Results expressed as μg gallic acid equivalent (GAE) mg^{-1} dry extract.

Total Flavonoid Content (TFC): Assessed by the AlCl3 method, read at 510 nm against a quercetin calibration curve [30]. Results expressed as μg quercetin equivalent (QE) mg^{-1} dry extract.

Alkaloids: Determined by the 1,10-phenanthroline method, read at 510 nm against a quinine calibration curve [31].

Tannins: Determined by the FeCl3/potassium ferrocyanide method of Van-Buren and Robinson, read at 605 nm [31].

Phytates, Oxalates, Steroids: Determined by standard colorimetric and gravimetric methods, read at 519 nm, 420 nm and 780 nm respectively [32–35].

2.10. Antioxidant activity assays

DPPH Radical Scavenging Assay: The method was followed according to the method of Brand-Williams *et al.* [36]. The various concentrations of the methanol extracts (50–800 μg/mL) were added to the DPPH solution. The reaction mixture was left in the dark for 10 min at room temperature, and the absorbance (A) was measured at 515 nm. Thereafter, the percentage inhibition was calculated, and IC_{50} values were evaluated. Because the tested concentration range (50–800 μg/mL) did not extend below the resulting IC_{50} estimates for any tissue, the IC_{50} values reported in Table 7 are extrapolated estimates from the fitted dose-response curve rather than directly interpolated values, and should be treated as approximate rankings rather than precise inhibitory concentrations.

Hydrogen Peroxide (H_2O_2) Scavenging Assay: The scavenging activity for hydrogen peroxide (H_2O_2) was carried out according to the procedure of Ruch *et al.* [37]. A 40-mM hydrogen peroxide solution was prepared using phosphate buffer (0.1 M, pH 7.4). Mixtures of the methanol extracts at various concentrations with the hydrogen peroxide solution were incubated at room temperature for 10 min. The absorbance (A) was measured at 230 nm against phosphate buffer (0.1 M, pH 7.4), whereas ascorbic acid was used as a reference standard.

Reducing Power Assay: The reducing power of the extracts was analyzed according to the procedure described by Oyaizu [38]. The various concentrations of the methanol extracts were mixed with phosphate buffer (0.2 M, pH 6.6) and 1% potassium ferricyanide. The resulting mixture was incubated at 50 °C for 20 min. Thereafter, trichloroacetic acid was added, and the mixture was centrifuged at 3000 rpm for 10 min. The obtained supernatant fluid was mixed with $FeCl_3$ solution (0.1%) and was measured for absorbance at 700 nm. Because this assay measures an increase in absorbance (reducing activity) rather than inhibition, results are expressed as EC_{50}, defined as the extract concentration producing an absorbance equal to 50% of the maximum absorbance increase observed across the tested concentration range, rather than as IC_{50}.

2.11. Statistical analysis

For each tissue, a single pooled sample was prepared as described in Sections 2.3 and 2.8, and the triplicate values reported in Tables 1, 5A, 5B, 6, and 7 are repeated analytical measurements on that pooled sample rather than independent biological replicates; fatty acid methyl esters were likewise prepared from pooled lipid extracts. Data are reported as mean ± standard deviation of these analytical replicates (n = 3 unless otherwise stated). Statistical analysis was carried out using GraphPad Prism version 9. The statistical significance of differences between the three samples (tuber, peel, leaves) was estimated by a one-way Analysis of Variance (ANOVA). Tukey's Honest Significant Difference (HSD) post-hoc test was used to evaluate differences between the means of the various samples. The value of statistical significance used was $p < 0.05$. Within a row, values sharing the same superscript letter are not significantly different (Tukey [39]).

Because the replicates are not independent biological samples, these ANOVA and Tukey's HSD comparisons describe differences between the specific pooled tuber, peel and leaf samples analysed, and should not be read as inferences about the broader populations of tuber, peel and leaf tissue; this is stated explicitly as a limitation in Section 4.7.

Table 1: Physicochemical Properties of Oils from Tubers, Peels, and Leaves of I. batatas

Parameter	Tuber	Peel	Leaves
Melting point (°C)	32.4 ± 0.3^{b}	34.1 ± 0.4^{a}	31.2 ± 0.3^{c}
Cloud point (°C)	5.2 ± 0.2^{b}	6.8 ± 0.3^{a}	4.5 ± 0.2^{c}
Smoke point (°C)	185.3 ± 1.2^{b}	178.6 ± 1.5^{c}	192.4 ± 1.3^{a}
Flash point (°C)	215.6 ± 1.8^{b}	208.3 ± 2.0^{c}	221.5 ± 1.6^{a}
Fire point (°C)	228.4 ± 1.5^{b}	221.7 ± 1.8^{c}	235.2 ± 1.4^{a}
Refractive index (40°C)	1.462 ± 0.002^{b}	1.465 ± 0.002^{ab}	1.468 ± 0.002^{a}
Specific gravity	0.915 ± 0.005^{b}	0.928 ± 0.006^{ab}	0.938 ± 0.006^{a}
Impurities (%)	0.32 ± 0.03^{b}	0.45 ± 0.04^{a}	0.28 ± 0.02^{b}
Moisture content (%)	1.24 ± 0.08^{b}	1.56 ± 0.10^{a}	1.08 ± 0.07^{b}
Acid value (mgKOH/g)	3.30 ± 0.05^{c}	5.43 ± 0.05^{a}	4.22 ± 0.05^{b}
Saponification value (mgKOH/g)	170.4 ± 1.8^{c}	190.7 ± 2.1^{b}	246.8 ± 2.3^{a}
Iodine value (gI_2/100g)	15.99 ± 0.12^{b}	16.50 ± 0.15^{a}	16.75 ± 0.13^{a}
Peroxide value ($meqO_2$/kg)	0.6 ± 0.1^{c}	2.2 ± 0.2^{a}	1.4 ± 0.1^{b}
Unsaponifiable matter (%)	0.39 ± 0.02^{a}	0.43 ± 0.03^{a}	0.39 ± 0.02^{a}
Free fatty acid (% as oleic acid)	1.65 ± 0.08^{c}	2.72 ± 0.10^{a}	2.11 ± 0.09^{b}
Viscosity (mPa·s)	199.8 ± 2.5^{c}	362.7 ± 3.2^{b}	588.1 ± 4.1^{a}

Values are expressed as mean ± standard deviation of three analytical replicates on a single pooled sample per tissue (n = 3; see Section 2.11). Different superscript letters within a row indicate significant differences among the samples analysed ($p < 0.05$; one-way ANOVA with Tukey's HSD).

3. Results

3.1. Physicochemical properties of oils

The physicochemical properties of oils extracted from sweet potato tuber, peel, and leaves are presented in Table 1.

3.2. Fatty acid composition

The fatty acid profiles of oils from sweet potato tuber, peel, and leaves are presented in Table 2.

3.3. Phospholipid and phytosterol composition

The phospholipid content of oils from sweet potato parts is presented in Table 3.

3.4. Vitamin composition

The fat-soluble vitamin content of oil extracts is presented in Table 5A.

3.5. Phytochemical composition

The phytochemical composition of methanol extracts from sweet potato parts is presented in Table 6.

3.6. Antioxidant activity

The antioxidant activities of methanol extracts from sweet potato parts are presented in Table 7.

4. Discussion

4.1. Physicochemical properties and oil quality

Physicochemical properties of oils that were obtained from sweet potato parts are presented in Table 1, where a tissue-dependent distribution pattern was found. Hence, melting points varied significantly from 31.2 ± 0.3 °C (leaf oil) to 34.1 ± 0.4 °C (peel oil), with tuber oil recording an intermediate value of 32.4 ± 0.3 °C. Likewise, cloud point followed the same trend, varying between 6.8 ± 0.3 °C (peel) > 5.2 ± 0.2 °C (tuber) > 4.5 ± 0.2 °C (leaf). While melting and cloud points varied only marginally (3-4 °C), higher values were recorded for peel oil, which suggests that it contains higher quantities of higher-melting components like saturated fatty acids, waxes, and other high-molecular constituents [22]. On the other hand, leaf oil showed a comparatively lower cloud point, presumably indicating a higher amount of unsaturated lipids [22].

The smoke points varied in the range of 178.6 ± 1.5 °C (peel) to 192.4 ± 1.3 °C (leaf); the same trend was observed for flash points, varying between 208.3 ± 2.0 °C (peel) and 221.5 ± 1.6 °C (leaf). Fire points showed a similar pattern, varying from 221.7 ±

Table 2: Fatty Acid Composition of Oils from Tubers, Peels, and Leaves of I. batatas (% of total fatty acids)

Fatty Acid	Tuber	Peel	Leaves
Saturated Fatty Acids	Saturated Fatty Acids	Saturated Fatty Acids	Saturated Fatty Acids
Lauric acid (C12:0)	1.2 ± 0.1^{b}	1.5 ± 0.1^{a}	1.0 ± 0.1^{b}
Myristic acid (C14:0)	1.8 ± 0.1^{a}	2.1 ± 0.2^{a}	1.4 ± 0.1^{b}
Palmitic acid (C16:0)	32.4 ± 0.8^{b}	35.6 ± 0.9^{a}	28.9 ± 0.7^{c}
Stearic acid (C18:0)	4.8 ± 0.2^{a}	5.1 ± 0.2^{a}	4.2 ± 0.2^{b}
Arachidic acid (C20:0)	2.9 ± 0.1^{b}	3.7 ± 0.2^{a}	2.4 ± 0.1^{c}
Behenic acid (C22:0)	2.0 ± 0.1	ND	2.0 ± 0.1
Total Saturated (TSFA)	45.1 ± 1.2^{a}	48.0 ± 1.4^{a}	39.9 ± 1.1^{b}
Monounsaturated Fatty Acids	Monounsaturated Fatty Acids	Monounsaturated Fatty Acids	Monounsaturated Fatty Acids
Palmitoleic acid (C16:1)	1.2 ± 0.1^{b}	1.0 ± 0.1^{b}	1.5 ± 0.1^{a}
Oleic acid (C18:1)	39.1 ± 1.0^{a}	35.2 ± 0.9^{b}	40.8 ± 1.1^{a}
Gadoleic acid (C20:1)	ND	1.1 ± 0.1	1.0 ± 0.1
Total Monounsaturated (TMUFA)	40.3 ± 1.1^{b}	37.3 ± 1.0^{c}	43.3 ± 1.2^{a}
Polyunsaturated Fatty Acids	Polyunsaturated Fatty Acids	Polyunsaturated Fatty Acids	Polyunsaturated Fatty Acids
Linoleic acid (C18:2, ω-6)	12.6 ± 0.5^{b}	13.2 ± 0.5^{ab}	14.5 ± 0.6^{a}
α-Linolenic acid (C18:3, ω-3)	2.0 ± 0.1^{b}	1.5 ± 0.1^{c}	2.3 ± 0.1^{a}
Total Polyunsaturated (TPUFA)	14.6 ± 0.6^{b}	14.7 ± 0.6^{b}	16.8 ± 0.7^{a}
Summary Indices	Summary Indices	Summary Indices	Summary Indices
Total Unsaturated (TUFA)	54.9 ± 1.4^{b}	52.0 ± 1.3^{b}	60.1 ± 1.5^{a}
UFA/SFA Ratio	1.22	1.08	1.51
PUFA/SFA Ratio	0.32	0.31	0.42
MUFA/SFA Ratio	0.89	0.77	1.08
Oleic/Linoleic (O/L) Ratio	3.10	2.67	2.81

Values are expressed as mean ± standard deviation of analytical replicates on pooled lipid extracts per tissue (n = 3; see Sections 2.5 and 2.11). ND = Not detected. Different superscript letters within a row indicate significant differences among the samples analysed ($p < 0.05$; one-way ANOVA with Tukey's HSD).

Table 3: Phospholipid Content in Oils from Tubers, Peels, and Leaves of I. batatas (mg/100g oil)

Phospholipid	Tuber	Peel	Leaves
Phosphatidylcholine (PC)	48.2 ± 1.5^{c}	52.6 ± 1.6^{b}	68.4 ± 2.0^{a}
Phosphatidylethanolamine (PE)	32.4 ± 1.2^{c}	35.8 ± 1.3^{b}	45.2 ± 1.5^{a}
Phosphatidylinositol (PI)	18.6 ± 0.8^{b}	20.4 ± 0.9^{b}	25.8 ± 1.0^{a}
Phosphatidylserine (PS)	8.4 ± 0.4^{b}	9.2 ± 0.5^{b}	12.6 ± 0.6^{a}
Total Phospholipids	107.6 ± 3.5^{c}	118.0 ± 3.8^{b}	152.0 ± 4.5^{a}

Values are expressed as mean ± standard deviation of analytical replicates (n = 3; see Section 2.11). Different superscript letters within a row indicate significant differences among the samples analysed ($p < 0.05$; one-way ANOVA with Tukey's HSD).

1.8 °C (peel) to 235.2 ± 1.4 °C (leaf). Thus, leaf oil revealed the maximum smoke, flash, and fire points, followed by tuber, and the lowest were recorded for peel oil. The high flash and smoke points of leaf oil suggest its high thermal stability and relatively lower volatility of its components [22]. The refractive index of leaf oil was slightly higher than that of tuber oil at 40 °C (1.468 ± 0.002 vs. 1.462 ± 0.002), and the difference was statistically significant ($p < 0.05$; Table 1).

Refractive index of the studied oils was within the typical range for vegetable oils reported in the literature as 1.46-1.52 [22]. However, the difference in the specific gravity was significant, which increased from 0.915 ± 0.005 in tuber oil to 0.938 ± 0.006 in leaf oil. Such a difference may be explained by the different content of fatty acids and non-triglyceride constituents in tuber and leaf oils [22]. Finally, peel oil had the highest content of impurities of 0.45 ± 0.04%, followed by tuber (0.32 ± 0.03) and leaf (0.28 ± 0.02%) oils. The high content of impurities in the peel oil could be justified by the larger surface contacted by the soil particles, cellular debris, pigments, waxes, and other non-lipid materials. This is an essential consideration as the crude oils extracted from plant tissues can comprise phospholipids, pigments, proteins, carbohydrates, waxes, and particulate materials in addition to

Table 4: Phytosterol Content in Oils from Tubers, Peels, and Leaves of I. batatas (mg/100g oil)

Phytosterol	Tuber	Peel	Leaves
β-Sitosterol	85.4 ± 2.5^{c}	92.6 ± 2.8^{b}	118.5 ± 3.2^{a}
Campesterol	22.6 ± 1.0^{b}	24.8 ± 1.1^{b}	32.4 ± 1.3^{a}
Stigmasterol	15.8 ± 0.7^{b}	17.2 ± 0.8^{b}	21.6 ± 0.9^{a}
Δ5-Avenasterol	8.4 ± 0.4^{b}	9.6 ± 0.5^{b}	12.8 ± 0.6^{a}
Total Phytosterols	132.2 ± 4.2^{c}	144.2 ± 4.5^{b}	185.3 ± 5.0^{a}

Values are expressed as mean ± standard deviation of analytical replicates (n = 3; see Section 2.11). Different superscript letters within a row indicate significant differences among the samples analysed ($p < 0.05$; one-way ANOVA with Tukey's HSD).

Table 5A: Fat-Soluble Vitamin Content (Oil Extract)

Vitamin	Tuber	Peel	Leaves
Carotenoids, expressed as vitamin A (mg/100g)	0.35 ± 0.02^{c}	7.08 ± 0.30^{a}	0.84 ± 0.04^{b}
Vitamin E (tocopherol) (mg/100g)	41.1 ± 1.5^{c}	47.2 ± 1.8^{b}	53.1 ± 2.0^{a}

Table 5B: Water-Soluble Vitamin Content (Methanol Extract)

Vitamin	Tuber	Peel	Leaves
Vitamin B1 / Thiamine (mg/kg)	420.3 ± 5.2^{b}	442.0 ± 5.8^{a}	364.6 ± 4.5^{c}
Vitamin B2 / Riboflavin (mg/kg)	0.93 ± 0.05^{c}	16.11 ± 0.80^{a}	13.44 ± 0.70^{b}
Vitamin B6 / Pyridoxine (mg/kg)	4.29 ± 0.20^{a}	3.95 ± 0.20^{a}	4.43 ± 0.20^{a}
Vitamin C / Ascorbic acid (mg/kg)	255.5 ± 8.5^{c}	297.5 ± 9.2^{b}	615.0 ± 12.5^{a}

Values are expressed as mean ± standard deviation of analytical replicates (n = 3; see Section 2.11). Note: Water-soluble vitamins were analyzed in methanol extracts, not in the oil fraction, as they do not partition into oil [26]. Different superscript letters within a row indicate significant differences among the samples analysed ($p < 0.05$; one-way ANOVA with Tukey's HSD).

Table 6: Phytochemical Composition of Methanol Extracts from I. batatas Parts

Phytochemical	Tuber	Peel	Leaves
Total Phenolic Content (μg GAE/mg)	0.705 ± 0.03^{c}	0.990 ± 0.04^{b}	1.370 ± 0.05^{a}
Total Flavonoid Content (μg QE/mg)	12.77 ± 0.8^{a}	7.06 ± 0.5^{b}	14.02 ± 0.9^{a}
Alkaloids (mg/L)	1.126 ± 0.05^{a}	1.132 ± 0.05^{a}	1.138 ± 0.05^{a}
Tannins (mg/L)	6.34 ± 0.3^{b}	10.72 ± 0.5^{a}	5.87 ± 0.3^{b}
Phytates (mg/L)	44.51 ± 1.5^{a}	34.14 ± 1.2^{b}	42.03 ± 1.4^{a}
Oxalates (mg/L)	165.63 ± 5.2^{b}	116.25 ± 4.5^{c}	392.50 ± 8.5^{a}
Steroids (mg/L)	0.1384 ± 0.005^{a}	0.1380 ± 0.005^{a}	0.1393 ± 0.005^{a}

Values are expressed as mean ± standard deviation of analytical replicates (n = 3; see Section 2.11). GAE = Gallic Acid Equivalent; QE = Quercetin Equivalent. Different superscript letters within a row indicate significant differences among the samples analysed ($p < 0.05$; one-way ANOVA with Tukey's HSD). A Total Triterpenoid Content row reported in an earlier version of this table was found, on review, to duplicate the Tannins measurement under a different unit label and has been removed; no independent triterpenoid determination was performed in this study.

Table 7: Antioxidant Activity of Methanol Extracts from I. batatas Parts

Assay	Tuber	Peel	Leaves	F-value	p-value
DPPH Radical Scavenging IC_{50} (extrapolated) (μg/mL)	42.5 ± 1.2^{a}	38.2 ± 1.0^{b}	31.6 ± 0.8^{c}	88.1	<0.001
H_2O_2 Scavenging IC_{50} (μg/mL)	45.3 ± 1.3^{a}	41.5 ± 1.1^{b}	35.8 ± 0.9^{c}	55.5	<0.001
Reducing Power EC_{50} (μg/mL)	48.6 ± 1.4^{a}	44.2 ± 1.2^{b}	38.5 ± 1.0^{c}	52.5	<0.001

Values are expressed as mean ± standard deviation of analytical replicates (n = 3; see Section 2.11). IC_{50} = half maximal inhibitory concentration (lower values indicate stronger apparent antioxidant activity); the DPPH IC_{50} values are extrapolated below the lowest tested concentration (50 μg/mL) and should be read as approximate rankings (Section 2.10). EC_{50} = concentration producing 50% of the maximum observed absorbance increase in the reducing-power assay (Section 2.10). Different superscript letters within a row indicate significant differences among the samples analysed ($p < 0.05$; one-way ANOVA with Tukey's HSD).

triacylglycerols [22].

Similarly, regarding the moisture content, the highest value (1.56 ± 0.10%) was detected for the peel oil followed by the tuber oil (1.24 ± 0.08%) and the leaf oil (1.08 ± 0.07%). The relatively high moisture content of the peel oil might partly account for its high acid and peroxide values [22]. However, low moisture in the product is usually desired to maintain good oil quality and stability during storage [22]. The acid value was significantly different in all samples ($p < 0.05$). The tuber oil had the lowest acid value (3.30 ± 0.05 mg KOH/g), while the peel and leaf oils had a higher acid value (5.43 ± 0.05 and 4.22 ± 0.05 mg KOH/g, respectively). The same pattern was observed for free fatty acids (FFA, % as oleic acid) in all oils, which ranged from 1.65 ± 0.08% in tuber oil to 2.72 ± 0.10% in peel oil and 2.11 ± 0.09% in leaf oil.

The acid value was closely related to the FFA content, most likely because the acid value is a measure of the amount of free fatty acids in an oil sample. The high content of the peel and leaf oils could be an indication of higher hydrolysis of triglycerides as compared to the tuber oil [22]. The saponification values increased from 170.4 mg KOH/g for the tuber oil to 246.8 mg KOH/g for the leaf oil. High saponification of the leaf oil might be an indication of having a greater proportion of short-chain fatty acids, or other saponifiable substances [22]. The measured iodine values (15.99-16.75 g I_2/100 g) are, however, lower than the values of about 59-68 g I_2/100 g calculated from the fatty acid profiles in Table 2 (AOCS Cd 1c-85), which show 52-60% unsaturated fatty acids dominated by oleic and linoleic acids. The measured values should therefore be interpreted with caution, and no conclusion on unsaturation or oxidative stability is drawn from them.

Several factors could plausibly contribute to this gap between the measured and GC-derived iodine values. First, the Wijs titrimetric method measures halogen uptake at reactive double bonds under the specific conditions of the assay; uptake can be suppressed by steric hindrance, conjugation, or by oxidation products (hydroperoxides, polymerised material) that no longer react stoichiometrically with iodine, and such oxidation could have occurred during the 48 h maceration, solvent evaporation, or subsequent storage of the unrefined oil. Second, the GC-based estimate (AOCS Cd 1c-85) is calculated from the FAME profile and implicitly assumes this profile represents the whole triacylglycerol fraction, whereas co-extracted non-triglyceride material consistent with the appreciable impurity and unsaponifiable-matter content of these crude oils (Table 1) would dilute the iodine-reactive lipid per gram of crude oil without being captured by the FAME-based calculation. Third, because the oils were unrefined and not stabilised against oxidation, some loss of titratable unsaturation between extraction and analysis cannot be excluded.

These remain plausible rather than confirmed explanations, and the measured iodine values should continue to be interpreted with caution.

Peroxide value, which is an indicator of primary oxidation of lipids, was found to be lowest in tuber oil (0.6 ± 0.1 meq O2/kg) and highest in peel (2.2 ± 0.2 meq O2/kg) and leaf (1.4 ± 0.1 meq O2/kg) oils. The peroxide values of all three oils are below the Codex maximum for virgin and cold-pressed oils (15 meq O_2/kg) [40]. Tuber oil therefore contained the least primary oxidation products at the time of analysis and peel oil the most; because oxidative stability under storage or heating was not tested (e.g., by Rancimat/OSI), no ranking of stability is claimed.

In contrast, the acid values of peel oil (5.43 mg KOH/g) and leaf oil (4.22 mg KOH/g) exceed the Codex limit of 4.0 mg KOH/g for virgin and cold-pressed oils (0.6 mg KOH/g for refined oils), and the impurity (0.28-0.45%) and moisture (1.08-1.56%) contents exceed the Codex limits for insoluble impurities (0.05% m/m) and volatile matter at 105 °C (0.2% m/m) [40], assuming they were determined as such. Sweet potato oil is not a named oil in the Codex standard for named vegetable oils, so these limits serve only as a quality benchmark, and the crude oils would need refining before food use. The content of unsaponifiable matter in the three types of oil was found to be similar, ranging from 0.39 ± 0.02% in tuber and leaf oils to 0.43 ± 0.03% in peel oil. In addition, no significant differences were found in the content of unsaponifiable matter for the three types of oil.

This result indicates that the amount of lipid components in these oils that are not subject to saponification is similar [22]. Finally, the viscosity of the three oils varied the most, ranging from 199.8 mPa·s for tuber oil to 588.1 mPa·s for leaf oil. Apparently, the difference in the viscosity of leaf oil from the other two types is due to the difference in the content of high-molecular-weight lipids [22].

4.2. Fatty acid composition

Table 2 presents the fatty acid composition of the oils isolated from the tubers, peels, and leaves of the I. batatas. The results showed that palmitic, oleic and linoleic acids were the major fatty acids detected in the three oils. This finding is in agreement with previous studies reported in the literature [18, 19]. With regard to the saturated fatty acids (SFA) palmitic acid (C16:0) was the major component found in all samples which contributed to 32.4 ± 0.8% of tuber oil, 35.6 ± 0.9% of peel oil, and 28.9 ± 0.7% of leaf oil, respectively. The peel value was significantly higher whereas the leaf value was significantly lower than that of the tuber. The total saturated fatty acid (TSFA) content was highest in peel oil (48.0 ± 1.4%), followed by tuber oil (45.1 ± 1.2%) and finally leaf oil (39.9 ± 1.1%). The significantly lower TSFA level in leaf oil may indicate that its fatty acid profile was less dominated by the saturated fatty acids.

This might have nutritional significance as excessive dietary intake of certain saturated fatty acids are generally associated with elevated LDL-cholesterol concentrations and cardiovascular risk [16].

The monounsaturated fatty acids (MUFA) were dominated by oleic acid (C18:1) which was found to be 39.1 ± 1.0% of tuber oil, 35.2 ± 0.9% of the peel oil and 40.8 ± 1.1% of the leaf oil. The level of this acid was lower in the case of the peel oil when

compared to oil from the tuber. Another component of monounsaturated fatty acids is palmitoleic acid, which was present in low amounts (about 1.0-1.5%). No gadoleic acid (C20:1) were found in tuber oil, whereas 1.1 ± 0.1% and 1.0 ± 0.1% were detected in peel and in leaf oils respectively. Therefore, the total content of monounsaturated fatty acids was estimated to be 40.3 ± 1.1%, 37.3 ± 1.0% and 43.3 ± 1.2% for tuber, peel and leaf oils respectively. It can be seen that the leaf oil content is high in monounsaturated fatty acids, which contributes to its overall unsaturation; dietary MUFA are generally associated with favourable health outcomes [41].

For the polyunsaturated fatty acids (PUFA), linoleic acid (C18:2, ω-6) was the major component, varying from 12.6 ± 0.5% in tuber oil to 13.2 ± 0.5% in peel oil and 14.5 ± 0.6% in leaf oil, with the highest content being in leaf oil. α-Linolenic acid (C18:3, ω-3) was present in lower quantities from 2.0 ± 0.1% in tuber oil to 1.5 ± 0.1% in peel oil and 2.3 ± 0.1% in leaf oil. The content in peel oil was significantly lower, whereas that in leaf oil was significantly higher, than in tuber oil. Published data indicate that the total lipids of sweet potato leaves are dominated by α-linolenic acid [42], in contrast to the low proportion found in the leaf oil here, which may reflect differences in cultivar, extraction solvent, tissue drying or storage. Furthermore, the presence of these acids in the diet is essential, as the human body is unable to synthesize linoleic and α-linolenic acids in sufficient quantities [17, 43].

Therefore, the total contents of PUFA in tuber and peel oils were 14.6 ± 0.6% and 14.7 ± 0.6%, respectively, whereas for leaf oil it was significantly higher and equal to 16.8 ± 0.7%. For this oil, the contents of linoleic and α-linolenic acids were the highest among the tested samples, indicating the potential nutritional value of leaf oil. The total content of individual fatty acids is presented as the total unsaturated fatty acid (TUFA) content. According to the results, leaf oil had the highest TUFA content (60.1 ± 1.5%) among the tested samples, followed by tuber (54.9 ± 1.4%) and peel oils (52.0 ± 1.3%). As such, the content of TUFA was the highest in leaf oil, with the lowest content of saturated fatty acids and the highest content of monounsaturated fatty acids, which indicates the potential of leaf oil for use as an unsaturated oil with a balanced fatty acid profile.

The UFA/SFA ratios were 1.22, 1.08 and 1.51 for tuber, peel and leaf oils, respectively. The highest value was found in leaf oil, which indicates a more prominent presence of unsaturated than saturated fatty acids. The PUFA/SFA ratio, recalculated from the total PUFA and SFA values in Table 2, was 0.31 (peel), 0.32 (tuber) and 0.42 (leaf); PUFA thus remained well below SFA in all three oils, although the ratio was highest in leaf oil. Likewise, the MUFA/SFA ratio also showed the same pattern of values: 0.77 (peel), 0.89 (tuber), and 1.08 (leaf). Together, the obtained indices demonstrated that leaf oil had a better profile of unsaturated/saturated fatty acids than the other oils. The oleic/linoleic (O/L) ratio was found to be highest in tuber oil, followed by leaf and then peel (Table 2). The relatively high O/L ratios suggested that oleic acid was substantially abundant compared to linoleic acid in all the oils analyzed.

A higher O/L ratio is also beneficial to oxidative stability since oleic acid is not as susceptible to oxidation as polyunsaturated fatty acids [17].

4.3. Phospholipids and phytosterols: bioactive potential

The data in Tables 3 and 4 revealed differences in the phospholipid and phytosterol content of the oils extracted from the tubers, peels, and leaves of I. batatas. As shown in the data presented in Table 3, phosphatidylcholine (PC) was the prevailing phospholipid in all the tested variants of oil. Its contents ranged from 48.2 ± 1.5 mg per 100 g in tuber oil, 52.6 ± 1.6 mg per 100 g in peel oil, and 68.4 ± 2.0 mg per 100 g in leaf oil, with the content in leaf oil being the highest among the tested variants ($p < 0.05$). The prevalence of PC may be related to its essential role in the construction of cell membranes, as well as its involvement in the processes of membrane cellular respiration [44].

Phosphatidylethanolamine (PE) was the second most prevalent phospholipid in all the variants of oil with contents of 32.4 ± 1.2, 35.8 ± 1.3, and 45.2 ± 1.5 mg per 100 g in tuber, peel, and leaf oils, respectively. The higher content of PE in the leaf oil compared to the other two variants was probably due to the higher content of membranes in the leaves, which is consistent with the more intense life activity of leaf cells [20]. Similarly, phosphatidylinositol (PI) increased from 18.6 ± 0.8 mg/100 g in tuber oil to 20.4 ± 0.9 mg/100 g in peel oil and 25.8 ± 1.0 mg/100 g in leaf oil. Thus, leaf oil contained significantly more PI than tuber oil. Although present at lower concentrations than PC and PE, PI is biologically important since it provides structural and signaling functions within cells [20].

Phosphatidylserine (PS) was found to be the least abundant of the phospholipids analyzed, with values of 8.4 ± 0.4, 9.2 ± 0.5, and 12.6 ± 0.6 mg/100 g for tuber, peel, and leaf oils, respectively. Again, leaf oil exhibited a significantly higher concentration than tuber oil. Despite its relatively low abundance, PS is an important membrane constituent and is involved in several cellular processes [20]. The combined effect of these individual phospholipids was reflected in the total phospholipid content, which increased from 107.6 ± 3.5 mg/100 g in tuber oil to 118.0 ± 3.8 mg/100 g in peel oil and 152.0 ± 4.5 mg/100 g in leaf oil.

The total phospholipid concentration of leaf oil was significantly higher than that of tuber oil. The overall pattern suggests that leaf oil has a greater phospholipid density than the other two oils and may reflect the greater abundance of cellular membranes in photosynthetically active tissue [20]. Table 4 shows similar tendencies for phytosterols. The prevailing phytosterol was β-sitosterol, which was present in all three oils with contents of 85.4 ± 2.5 mg/100 g in tuber oil, 92.6 ± 2.8 mg/100 g in peel oil, and 118.5 ± 3.2 mg/100 g in leaf oil. For leaf oil, the content was significantly increased compared to tuber oil. The prevalence of β-sitosterol is explained by the fact that it is one of the major plant sterols typically found in vegetable oils. Furthermore, as part of the diet, they reduce intestinal cholesterol absorption.

Another prevalent phytosterol was campesterol, which was seen to increase from 22.6 ± 1.0 mg/100 g in tuber oil, 24.8 ± 1.1 mg/100 g in peel oil, to 32.4 ± 1.3 mg/100 g in leaf oil. Again, the leaf oil was significantly richer in this substance than the other two.

Campesterol, similarly to other phytosterols, is involved in the formation of cell membrane structures and may have a beneficial impact on health when consumed in the diet [45]. Stigmasterol contents were relatively lower, varying from 15.8 ± 0.7 mg/100 g in tuber oil to 21.6 ± 0.9 mg/100 g in leaf oil. Thus, leaf oil was richer in this substance as well. Finally, Δ5-Avenasterol was the least prevalent phytosterol, being present at levels of 8.4 ± 0.4; 9.6 ± 0.5; and 12.8 ± 0.6 mg/100 g for the three oils. Its content was also relatively higher in the leaf oil than in the tuber oil. The total phytosterol content followed a similar pattern, increasing from 132.2 ± 4.2 mg/100 g in tuber oil to 144.2 ± 4.5 mg/100 g in peel oil and 185.3 ± 5.0 mg/100 g in leaf oil. Thus, leaf oil had the highest total phytosterol concentration and differed significantly from tuber oil.

Higher phytosterol content of the leaves may be related to their active membrane biosynthesis and metabolic activity [45].

4.4. Vitamin composition

Tables 5A and 5B show significant variation in the vitamin content of Ipomoea batatas tuber, peel, and leaves. Indeed, the content of fat-soluble vitamins A and E and water-soluble vitamins B1, B2, and C was considerably different, which means the nutritional value of the different parts of this plant is not the same. Firstly, the fat-soluble vitamin A represented as carotenoids showed a considerable variation in the oil extracts of I. batatas. Notably, the tuber's oil had a considerably lower amount of carotenoids (0.35 ± 0.02 mg/100 g) than the peel (7.08 ± 0.30 mg/100 g) and leaves (0.84 ± 0.04 mg/100 g) oils. The high carotenoid content of the peel indicates that it may be considered a valuable reservoir of pigment compounds [5].

Vitamin E (tocopherol) was found to be present at higher concentrations than vitamin A in all three oils. Specifically, the content of this vitamin was in the range of 41.1 ± 1.5 – 53.1 ± 2.0 mg/100 g, with the maximum amount detected in the leaf oil. The significantly higher tocopherol content in the peel and leaves suggests the increased accumulation of this vitamin in these tissues as compared to the tuber. Moreover, the relatively high tocopherol content of the leaf oil implies its enhanced nutritional and functional value [13]. The content of water-soluble vitamins was found to vary depending on the parts of the plant. For instance, the amount of thiamine (vitamin B_1) was 420.3 ± 5.2 mg/kg in tuber, 442.0 ± 5.8 mg/kg in the peel, and 364.6 ± 4.5 mg/kg in leaves. Thus, the content of thiamine in leaves was found to be significantly lower than that in the tuber, while in the peel, it was slightly higher than in the tuber.

This vitamin is essential for the body since it is involved in carbohydrate metabolism; hence, it contributes to the normal energy supply, and its presence in all three parts of the plant indicates their nutritional value [5]. Unlike thiamine, the content of riboflavin (B_2) was found to be significantly different between plant parts ($p < 0.05$). Specifically, while there was 0.93 ± 0.05 mg/kg of B_2 in tubers, the amount of this vitamin was much higher in the peel (16.11 ± 0.80 mg/kg) and leaves (13.44 ± 0.70 mg/kg). Thus, the content of B_2 in leaves and peel was significantly higher than that in tubers, which further suggests their potential nutritional value. This vitamin is also of particular importance since it is essential for cell energy metabolism and plays a key role in the biosynthesis of reduced coenzymes [5].

Vitamin B6 (pyridoxine) showed little variation, which was 4.29 ± 0.20, 3.95 ± 0.20 and 4.43 ± 0.20 mg/kg for tuber, peel and leaves, respectively. No significant differences were observed among the tested samples. This similarity may indicate comparable pyridoxine content among the tested tissues. Pyridoxine plays an important role in the metabolism of amino acids, the synthesis of neurotransmitters, and is involved in numerous enzymatic reactions. Concerning water-soluble vitamins, the content of vitamin C was the highest. It was 255.5 ± 8.5, 297.5 ± 9.2, and 615.0 ± 12.5 mg per 1 kg of tuber, peel, and leaves, respectively. Thus, the contents of vitamin C in leaves exceeded those in tuber more than twice. This may be related to the importance of ascorbic acid for many metabolic processes, including photosynthesis [13].

4.5. Phytochemical composition

Table 6 revealed that methanol extracts of the tuber, peel, and leaves of I. batatas differed substantially in their phytochemical composition. The total phenolic content (TPC) rose significantly from 0.705 ± 0.03 μg GAE/mg in the tuber to 0.990 ± 0.04 μg GAE/mg in the peel and 1.370 ± 0.05 μg GAE/mg in the leaves. The significantly higher phenolic concentration in the leaves suggests that these tissues have exceptionally high concentrations of phenolic compounds. Similarly, the total flavonoid content increased with little difference between the plant parts, except for the peel. The leaf extract was the richest (14.02 ± 0.9 μg QE/mg) followed by the tuber (12.77 ± 0.8 μg QE/mg) in terms of flavonoid content, while the peel contained a significantly lower flavonoid concentration of only 7.06 ± 0.5 μg QE/mg. The high flavonoid concentration of the leaves may be due to their exposure to sunlight and protect the photosynthetic tissues from oxidative damage [46].

The tannin content was highest in the peel (10.72 ± 0.5 mg/L), followed by the tuber (6.34 ± 0.3 mg/L) and leaves (5.87 ± 0.3 mg/L). The content of these secondary metabolites in the peel might be explained by their protective role, since the epidermis accumulates various metabolites for a barrier against herbivores, pathogens, and environmental stress [46]. The concentration of alkaloids was relatively similar for the three plant parts, varying from 1.126 ± 0.05 mg/L (tuber) to 1.138 ± 0.05 mg/L (leaves). No significant differences were found among the samples.

A similar trend was observed for the steroid content, which varied slightly between the examined plant parts: 0.1384 ± 0.005, 0.1380 ± 0.005, and 0.1393 ± 0.005 mg/L for the tuber, peel, and leaves, respectively. No significant differences were found for this group of secondary metabolites as well. The highest phytate content was detected in tubers (44.51 ± 1.5 mg/L), while the leaves contained 42.03 ± 1.4 mg/L and the peel had the lowest content (34.14 ± 1.2 mg/L). The higher phytate content in the tuber and leaves may pose a negative impact on the nutritional value of these plant parts, since phytates reduce the bioavailability of minerals [32]. The content of oxalates was found to be the highest in the leaves (392.50 ± 8.5 mg/L) as compared to that in the tuber (165.63 ± 5.2 mg/L) and peel (116.25 ± 4.5 mg/L).

The significantly higher oxalate content in the leaves can have a negative impact on the nutritional value of leafy vegetables, since oxalates decrease the mineral content, in particular calcium, due to the formation of insoluble calcium oxalate [32].

4.6. Antioxidant activity

Leaf extract showed the strongest apparent antioxidant response in all three assays (Table 7). Because the DPPH IC_{50} values are extrapolated below the tested concentration range, these rankings should be treated as indicative rather than as precisely established inhibitory concentrations; confirmatory measurements at lower concentrations would be needed to establish the IC_{50} values directly. The DPPH radical scavenging IC_{50} of leaf extract (31.6 μg/mL) is comparable, within this caveat, to values reported for several well-known plant extracts with known antioxidant properties [47, 48]. The leaf extract, which had the highest phenolic content, also had the strongest apparent antioxidant activity, suggesting a possible association between the two. However, no correlation analysis was performed (n = 3 analytical replicates per tissue does not allow a meaningful correlation), and other constituents such as flavonoids and vitamin C may also contribute [10, 47, 48].

4.7. Limitations of the study

Several limitations should be acknowledged. The first limitation is that the samples were collected from a single geographical location (Agudu, Lafia) and therefore the results may not be generalizable to sweet potatoes grown in different conditions. The second limitation is that the identification of the fatty acids was done by GC alone; future studies should use GC-MS to identify individual fatty acids. The third limitation is that the antioxidant assays were performed in vitro; future studies should evaluate the effects of consuming sweet potatoes in vivo. The fourth limitation is that the stability of the samples during storage was not assessed, which is crucial for commercial applications. Lastly, the analysis was carried out on a single variety of white-fleshed sweet potato; orange-fleshed and purple-fleshed varieties may have different bioactive constituents such as carotenoids and anthocyanins.

In addition, the oils were crude (unrefined), their oxidative stability and behaviour during heating were not evaluated, and the phenolic, water-soluble vitamin and antioxidant data refer to methanol extracts rather than to the oils.

Finally, oil yield (mass of oil recovered per unit mass of starting material) was not quantified in the present study; the conclusions drawn here about the potential utilisation of peel and leaf by-products as oil sources should therefore be read as indicative of oil composition rather than of the quantity recoverable from each tissue, pending yield determination on a defined dry-mass basis in future work. In addition, the triplicate values reported throughout Tables 1, 5A, 5B, 6, and 7 represent repeated analytical measurements on a single pooled sample per tissue rather than independent biological replicates (Section 2.11); the statistical comparisons among tissues should accordingly be read as descriptive differences between the specific samples analysed rather than as inferences about tuber, peel and leaf populations in general, and should be confirmed in a future study using independently collected biological replicates per tissue. The results apply to the samples analysed and need confirmation across cultivars, growing seasons and independent collections.

5. Conclusion

To the best of the authors' knowledge, this is among the first studies to compare the major bioactive lipidic compounds in sweet potato's tuber, peel, and leaves. The three tested parts of sweet potato have different but considerable compositions of potentially bioactive compounds. Leaf oil had the highest proportions of unsaturated and essential fatty acids and the highest phospholipid, phytosterol and vitamin E contents of the three oils, and the leaf methanol extract had the highest phenolic content and the strongest apparent in vitro antioxidant activity; nutritional value, bioavailability and in vivo effects were not assessed. Peel oil, obtained from a material generally considered waste, had the highest carotenoid content (expressed as vitamin A), and the peel methanol extract had the highest riboflavin (vitamin B2) content; the water-soluble B vitamins were determined in the methanol extracts and not in the oil. Whether these carotenoids are bioavailable and can help to address vitamin A deficiency in Sub-Saharan Africa requires further study.

Tuber oil had the lowest peroxide value, indicating the least primary oxidation at the time of analysis; its oxidative stability and suitability for high-temperature processing were not evaluated and require further study (e.g., Rancimat/OSI and heating tests). Compared with Codex quality limits for virgin oils, the peroxide values of all three oils were acceptable, whereas the acid values (peel and leaf oils), impurity and moisture contents of the crude oils exceeded the limits, indicating that refining would be required before food use.

Pending oil-yield and independent-replicate confirmation (Section 4.7), these research findings can help to promote the whole utilization of sweet potato to reduce the amount of agricultural waste and ensure the food and nutrition security of the region. The present study contributes to the comprehension of the potential application of sweet potato by-products in the food, nutraceutical, and pharmaceutical industries.

Data availability

The data supporting this study are available from the corresponding author upon reasonable request.

Declaration of competing interest

The authors declare no competing interests.

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