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Research Article

Nutraceutical potentiality of *Moringa oleifera* and *Senna occidentalis* grown under organic conditions from Burkina Faso

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Abstract

Quality nutrition is required for sustainable human health as well as disease prevention. This study aimed to evaluate the physico-chemical elements sources with nutraceutical value of *Moringa oleifera* and *Senna occidentalis*. *M. oleifera* had benefited from a biological treatment during its cultivation. Physico-chemical parameters analysis showed a remarkable richness in proteins (between $6.38 \pm 0.23\%$ and $26.79 \pm 0.09\%$), in lipids (between $0.49 \pm 0.20\%$ and $7.52 \pm 0.37\%$), in total sugars (between $57.44 \pm 0.29\%$ and $78.77 \pm 0.32\%$) and ashes (between $6.2 \pm 0.06\%$ and $10.71 \pm 0.06\%$). *M. oleifera* exhibits a higher nutritional profile than *S. occidentalis*, with the exception of ash content. This richness is reflected in the abundance of mineral elements. The predominant minerals, sodium, calcium, potassium, and magnesium, exhibited respective concentrations of 195.4 ± 0.28 mg in *M. oleifera* leaves, 95.56 ± 0.33 mg in *S. occidentalis* leaves, 74.35 ± 0.42 mg in *M. oleifera* leaves, and 11.47 ± 0.41 mg in *S. occidentalis* leaves. These two plants are also a good source of amino acids, including essential (threonine, valine, isoleucine, leucine, lysine, methionine, phenylalanine, and histidine) and non-essential (aspartate, glutamate, serine, glycine, arginine, alanine, proline, tyrosine, and cysteine) amino acids.

Keywords

organic plants, superfoods, functional foods, proximate composition, minerals, amino acid composition

Abbreviations

ICP-MS – Inductively Coupled Plasma Mass Spectrometry; *M. oleifera* – *Moringa oleifera*; *S. occidentalis* – *Senna occidentalis*; MOL – *Moringa oleifera* leaves; MOF – *Moringa oleifera* flowers; SOL – *Senna occidentalis* leaves

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Introduction

The term "nutraceutical" was first used in 1979 by Stephen DeFelice to describe any food or food component that provides medical or health benefits and was used for the prevention and treatment of disease. Nutraceuticals are defined as isolated nutrients, encompassing a range of products from food supplements and diets to genetically modified "designer" foods and phytoproducts (Tapas et al. 2008).

A nutraceutical is also defined as a non-toxic dietary extract supplement that offers health benefits and has scientific evidence supporting its use for both the treatment and prevention of disease (Dillard & Bruce German, 2000). However, a functional food was defined as a food that was expected to have a relevant effect on well-being and health, or cause a reduction in the risk of disease. However, it is important to distinguish between nutraceuticals and superfoods. Nutraceuticals are naturally occurring, biologically active compounds extracted from various food sources. Superfoods, on the other hand, are fresh or processed foods claimed to have specific health-promoting properties and/or reduce the risk of chronic diseases beyond their basic nutritional value (Mohammadian, 2021). In this context, the functional component may be an essential macronutrient or micronutrient, a nutrient that is not considered essential, or a non-nutritive component (Shahidi, 2009).

In light of the intriguing attributes of functional foods and the pivotal role of nutraceuticals, it is imperative to undertake a comprehensive examination of locally available flora that can be readily obtained. A plethora of studies have been conducted on *Moringa oleifera* (*M. oleifera*) and *Senna occidentalis* (*S. occidentalis*), demonstrating their substantial potential as functional foods and nutraceuticals (Debela & Tolera, 2013). *Moringa oleifera* was referred to as a "vegetable tree" due to the numerous edible uses to which it is put. *Senna occidentalis* was a legume that contains compounds, which, it has been demonstrated, rival those found in vegetables. Moreover, it was also consumed for therapeutic purposes (Mune Mune et al. 2016).

However, there is a paucity of research evaluating these properties under biological cultivation conditions. Consequently, this study was undertaken to illuminate the nutraceutical potentiality of these two plants. The selection of these plants was further informed by their accessibility, given their proximity to human populations. *M. oleifera* is employed for both nutritional and therapeutic purposes due to its multifaceted characteristics (Anwar et al. 2007; Latif et al. 2011). *S. occidentalis* is a commonly utilized plant in traditional medicine for the treatment of various diseases (Manikandaselvi et al. 2016; Thiaw et al. 2015). These plants are resilient to climatic hazards, which makes them important for investigation and popularization among populations. However, there is currently limited data on the effectiveness of these plants in all aspects, which encourages further research. *M. oleifera* is grown organically, while *S. occidentalis* comes from an environment with organic farming practices.

Refer to the USDA's official organic standards documentation, organic foods are defined as agricultural products that are cultivated and processed without the utilisation of synthetic pesticides, fertilisers, GMOs, irradiation, or sewage sludge. The production of these products is subject to stringent standards that promote ecological balance and biodiversity. Consequently, organic farmers must rely on natural substances and physical, mechanical, or biologically based farming methods.

The main objective of this study is to explore the nutraceutical potential of these two plants.

Materials and Methods

Plant materials. *M. oleifera* and *S. occidentalis* (Fig. 2) grown under biological conditions, were harvested in 2020 and powdered at Bissiri (Fig. 1) by Moringa Faso society. Organs involved were leaves, stem bark and flowers for *M. oleifera*; leaves, stem bark, seeds and seed capsules for *S. occidentalis*.

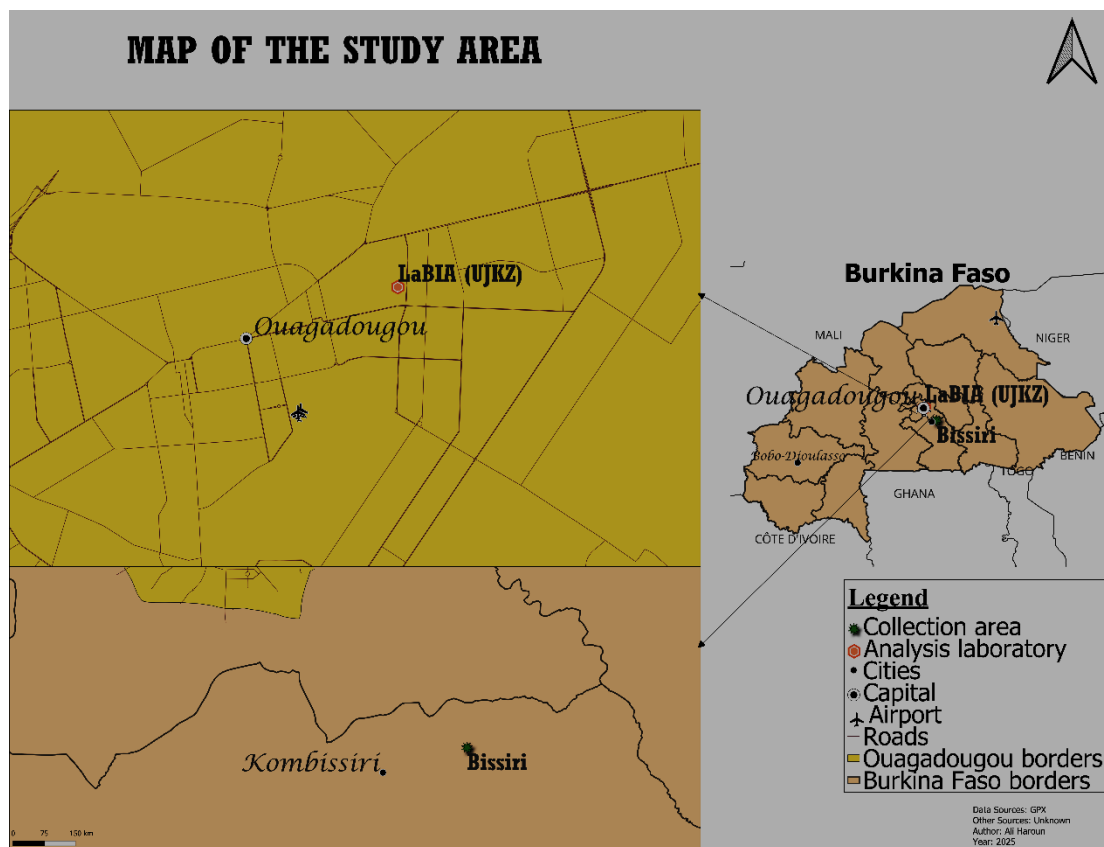


Figure. 1 : Harvesting site and study area for *M. oleifera* and *S. occidentalis* samples

Methods.

Determination of water content. The water content was determined by differential weighing after drying in an oven at 105 ± 2 °C according to standard NF V03-707 (2000) using the overnight method. Five (05) grams of sample (Pe) were weighed in a boat whose empty weight was previously determined (Pv). The boats were placed in an oven at 105 °C overnight (approximately 12 h). They were then removed from the oven and placed in a desiccator for cooling during 30 min, then weighed again (Pf). Each sample was weighed in triplicate for mean calculation. Humidity percentage was obtained by the following formula:

$$\% H = \frac{Pe - (Pf - Pv)}{Pe} * 100$$

Where:

% H - moisture content;

Pe - test sample;

Pf - final weight (basket + dehydrated sample);

Pv - platforms empty weight

Determination of ash content. Ash content was determined by differential weighing after placing the samples in oven overnight according to the French standard (ISO 2171:2007, 2007), used by the physico-chemical laboratory of the Department of Food Technology (DTA). Thus, 5 g of the sample (Pe) were weighed in a crucible whose empty weight was previously determined (Pv).

Crucibles were then placed in the oven at 600°C overnight (approximately 12 h). Calcined samples were then removed from the oven, cooled in a desiccator then the final weight (Pf) was determined by weighing the crucibles. Samples were weighed in triplicate to determine an average value and reduce errors.

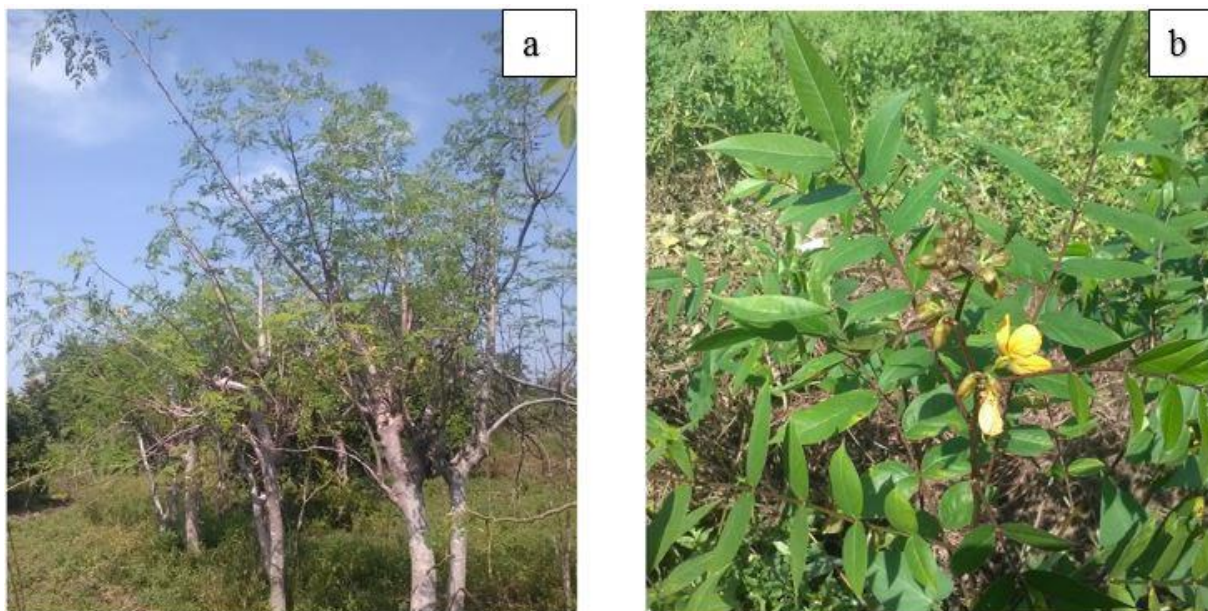


Figure 2: *Moringa oleifera* (a) and *Senna occidentalis* (b)

Ash content was calculated according to the relationship below:

$$\% C = \left[\frac{Pf - Pv}{Pe} * 100 \right]$$

Where:

% C - ash content compared to raw sample;

Pe - test sample;

Pf - final weight (crucible + calcined sample);

Pv - crucibles empty weight.

Ash content in relation to the dry matter was given by the following relationship:

$$\% C/ms = \left[\frac{\% C}{100 - \% H} * 100 \right]$$

Where:

% C - ash content compared to raw sample;

% H - moisture content;

ms - dry matter.

Determination of total sugar content. Total sugar content was determined by sulfuric orcinol method. This method is the method adapted from [Masuko et al. \(2005\)](#).

In a beaker containing 0.2 g of previously ground sample and five (05) mL of distilled water, under magnetic stirring for 10 min. Mixture was transferred to a 100 mL volumetric flask, and the volume was adjusted, then homogenized. In a test

tube, containing one (01) mL of homogenate, two (02) mL of orcinol reagent and seven (07) mL of 60 % sulfuric acid were added. After vortexing, the tube was placed in a boiling water bath for 20 min, in the dark for 45 min, then at room temperature for 10 min. The absorbance was read using a spectrophotometer at 510 nm.

Total sugar content was determined using the standard curve established from several concentrations of 1 mg.mL⁻¹ D-glucose stock solution.

Determination of protein content. Protein determination was carried out using the KJELDHAL method ([Abrams et al. 2014](#)) on an automatic apparatus of the Vadopest 4S Gerhardt type (Germany) in accordance with the French standard “V03-050, September 1970”.

On aluminum foil, 0.2 g of previously dried sample was weighed and introduced into a digestion tube or Kjeldahl matrass, to which was added Kjeltabs catalyst pellet and 10 mL of concentrated sulfuric acid. Prepared samples were mineralized on a heating block at a progressive temperature of 90, 120...400 °C for 4 h until the solution completely discolored.

With a distiller, mineralized material obtained was diluted with distilled water (approximately 50 mL and neutralized with concentrated soda (10 N).

Distillate (150 mL) was collected in a beaker containing 5 mL of a color indicator composed of bromocresol green, methyl red and boric acid. The whole was titrated with 0.1 N sulfuric acid until the indicator changed from green to pink. The volume (Ve) of sulfuric acid (H₂SO₄) at equivalence was noted. Protein content was determined by prior determination of nitrogen (N) content using the following formula:

$$\% N = \frac{0.14 * 0.1 * (Ve - V0)}{PE} * 100$$

Where:

% N - nitrogen rate compared to the raw sample;

Ve - volume of the acid at the equivalent point;

V0 - volume of blank;

Pe - test sample.

Obtained value was multiplied by the conversion factor to obtain the protein content by the formula:

$$\% \text{ Protein} = [\% N * 6.25]$$

Protein content in relation to the dry matter was given by the following relationship:

$$\% P/ms = \left[\frac{\% P}{100 - \% H} * 100 \right]$$

Where:

% P/ms - Protein content in relation to dry matter;

% H - Moisture content.

Determination of lipid content. International standard (ISO 659:2009, 2009) was used to determine samples lipid content by the Soxhlet method. Extraction was done hot (boiling) by soaking followed by rinsing the sample with hexane. The lipid content was determined by weighing after evaporation of the hexane by distillation.

Five g of sample (Pe) were weighed in a cartridge then covered with dehydrated cotton and placed in a Soxhlet. The Soxhlet was mounted in a flask of known weight (Pv) and the cartridge was immersed with hexane (about 200 mL). The extraction was done hot (boiling on a hot plate) for 4 h. The solvent was then evaporated by distillation using a rotary evaporator. The flask containing the fat was then dried in an oven for 1 hour for complete evaporation of the hexane. It was then cooled in a desiccator and then weighed again and the final weight (Pf) was

recorded. The following relationship was used to determine the lipids percentage:

$$\% MG = \left[\frac{Pf - Pv}{Pe} \right] * 100$$

Where:

% MG -fat content of raw sample;

Pe - test sample;

Pf - final weight (balloons + fat);

Pv - empty weight of balloons.

The ratio of fat to dry matter was given by the following relationship:

$$\% MG/ms = \left[\frac{\% MG}{100 - \% H} * 100 \right]$$

Where:

% MG/ms - fat content in relation to dry matter;

% H - moisture content.

Mineral flow chart (wet process). Approximately 0.2009 g of the plant powder was weighed and placed in a glass test tube. Using a pipette, 2 mL of concentrated nitric acid (HNO₃, 65%) was withdrawn and added. The plant powder was left to dissolve completely for approximately one (01) hour.

The test tube was placed in a beaker containing distilled water and the assembly is brought to a hot plate at 150°C under a hood for 2h30 to ensure acid digestion.

The mineralized test tube content was transferred to a 25 mL volumetric flask then topped up with distilled water for rinsing the test tube up to the gauge mark. This solution is filtered with Wattman paper with 0.45 µm diameter.

The preparation is read using a Varian AA240FS flame Atomic Absorption Spectrometer (Chiffolleau & Truquet, 1994).

Determination of amino acid profile. Total amino acids profile was determined by reverse-phase HPLC according to the “Waters Pico-Tag” method described by Bidlingmeyer et al. (1984). A hot acid hydrolysis of the defatted sample was carried out to release the amino acids which were subsequently complexed using phenyl-isothiocyanate (PITC) into amino acid-phenyl-thiocarbamyl, visible by the detector of the HPLC chain.

Acid hydrolysis and filtration of the sample. 0.4 g as test portion of the defatted sample were placed in a glass bottle of approximately 100 mL. To this test portion, a volume of 15 mL of hydrochloric acid (6N) was added and the assembly was slightly closed and placed in a ventilated oven at 105°C for 24 h. After cooling to room temperature, the hydrolysate was transferred to a 50 mL flask and filled with Milli-Q water up to the mark, and then was homogenized and filtered through a Millipore filter with a porosity of 0.45 µm.

Amino acid-phenyl-thiocarbamyl complex derivatization or formation. For each sample, 10 µL aliquot of the filtrate was placed in duplicate in tubes (sample vials), homogenized and then dried for 15 to 20 min using the “Pico.Tag Workstation” coupled to a vacuum pump. The dried sample was then re-dried under the same previous conditions after dissolution by homogenization in 10 µL of re-drying solution composed of a mixture of ethanol, Milli-Q water and triethylamine (2:2:1; v/v/v). A volume of 20 µL of the derivatization solution consisting of a mixture of ethanol, Milli-Q water, triethylamine and phenyl-isothiocyanate (7:1:1:1; v/v/v/v) was again added to the re-dried sample and the whole was homogenized by vortexing then incubated at room temperature for 20 min so that the amino acid-phenyl-thiocarbamyl complex is formed. At the end, a final drying for 45 min to 1 hour, using the “Pico.tag Workstation” was carried out to eliminate all traces of excess PITC in the complex, which was diluted using 100 µL of “Pico.Tag sample diluent”, before being injected.

Injection and HPLC analysis in reverse phase. Samples sequence was created on the computer using the EMPOWER2 software before activating the start button of the analysis. Automatically, HPLC column equilibration was carried out by the device before requesting sample injection. Using a 100 µL syringe, a volume of 40 µL of the diluted solution of the complexed sample was placed in the injector before putting it in the injection position to start sample automatic reading. The device is equipped with a Waters Detector, the signal of which, translated by a chromatograph consisting of peaks as a function of the retention time, has made it possible to identify and quantify, according to the areas of the peaks, the amino acids of the sample.

Statistical analysis. Analysis of variances (one-way ANOVA) was used to verify the statistical influence of the different parameters on each other. The results obtained were expressed in the form of means $\pm$ standard deviation. GraphPad Prism 5 software was used to perform these statistical tests.

Results and Discussion

Determination of physico-chemical properties.

Fig. 3 shows the moisture content of the two plants and the physico-chemical profiles results overview according to the plant dry matter are presented in Fig. 4.

M. oleifera flowers have moisture highest value (7.12) while the lowest value (4.00) is noted in *S. occidentalis* seeds capsules. These results are superior to those reported by Kudre et al. (2013) on isolates of three legumes. However, they are lower than those of Adewumi et al. (2022) on *M. oleifera* protein isolates.

MOL, MOE, MOF, SOL, SOE, SOC and SOG protein content are respectively 26.79 ± 0.09 ; 6.38 ± 0.23 ; 19.18 ± 0.14 ; 22.21 ± 0.05 ; 20.16 ± 0.00 ; 9.78 ± 0.10 and 24.52 ± 0.06 %. These values are significantly different. Our results are inferior to those of Adewumi et al. (2022) on his three legumes protein isolates. The alkaline conditions of the treatment for the analysis could be at the origin of the variation in protein content of the different organs (Adewumi et al., 2022). The relatively lower protein content of MOE and SOG could be explained mainly by their high fat and total sugar content (Kudre et al. 2013). Indeed, fats and sugars serve as essential sources of energy and structural components for the growth of plants. Consequently, in order to facilitate optimal growth, most plants prioritise the synthesis of these metabolites in their metabolism (Kosová et al. 2013).

MOL, MOE, MOF, SOL, SOE, SOC and SOG fat content values are 7.19 ± 0.14 ; 0.49 ± 0.20 ; 7.52 ± 0.37 ; 6.63 ± 0.03 ; 4.24 ± 0.00 ; 0.49 ± 0.19 ; 3.04 ± 0.22 % respectively. These values are significantly different except between MOL and MOF. MOL fat content is higher than the value 2.2 ± 0.2 % reported by Adewumi et al. (2022).

The low values obtained in our results (MOE and SOC) could be explained by a possible saponification undergone at alkaline pH with the NaOH used for solubilisation.

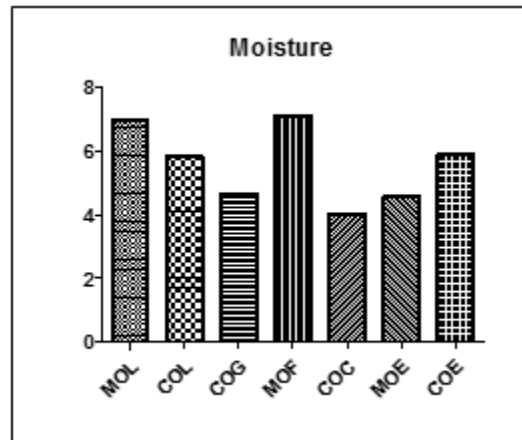


Figure 3. Moisture content of *M. oleifera* and *S. occidentalis* organs
 MOL - *M. oleifera* leaves; MOE - *M. oleifera* bark; MOF - *M. oleifera* flowers; SOL - *S. occidentalis* leaves; SOE - *S. occidentalis* bark ; SOC - *S. occidentalis* shell; SOG - *S. occidentalis* seeds

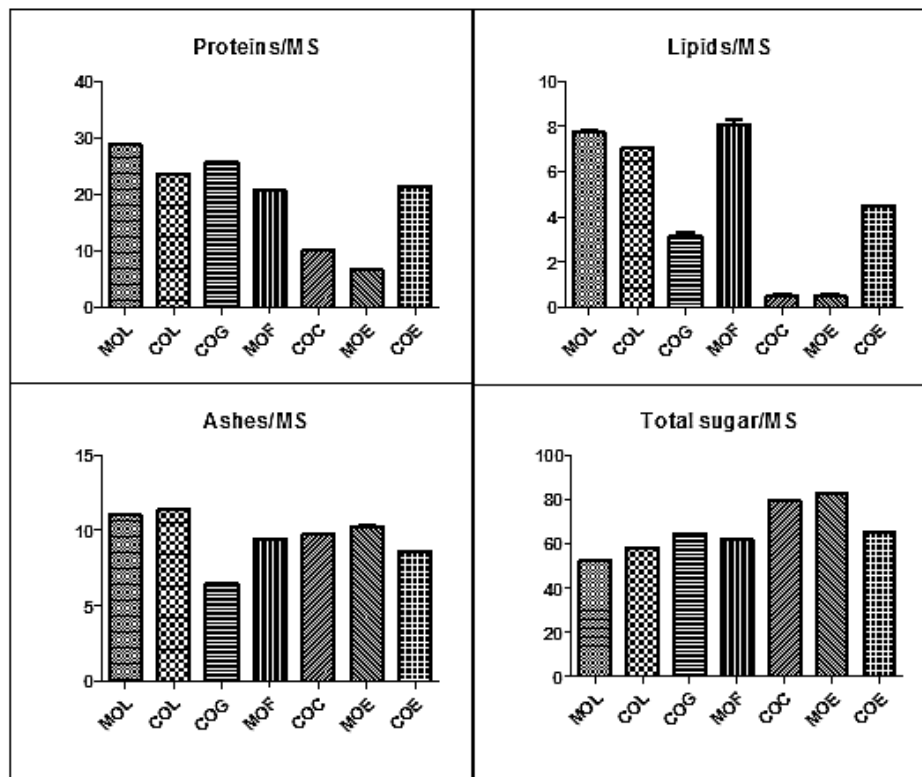


Figure 4. Physico-chemical profiles of *M. oleifera* and *S. occidentalis* organs relative to dry matter
 MOL - *M. oleifera* leaves; MOE - *M. oleifera* bark; MOF - *M. oleifera* flowers; SOL - *S. occidentalis* leaves; SOE - *S. occidentalis* bark ; SOC - *S. occidentalis* shell; SOG - *S. occidentalis* seeds

Ash contents were 10.28 ± 0.05 ; 9.81 ± 0.09 ; 8.75 ± 0.05 ; 10.71 ± 0.06 ; 8.09 ± 0.03 ; 9.37 ± 0.07 ; 6.216 ± 0.06 % for MOL, MOE, MOF, SOL, SOE, SOC and SOG respectively, and they were significantly

different. All of our values are higher than those reported by Kudre et al. (2013) for all legume protein isolates. The highest SOL ash content (10.71

± 0.06 %), indicates that *S. occidentalis* leaves had higher mineral content than other organs.

All the values of total sugars are significantly different except for SOE and SOG. These values are much higher than those found by Kudre et al. (2013) and Adewumi et al. (2022).

Minerals flow chart with wet process. As indicated in Table 1, an assortment of values has been recorded following the determination of mineral content. These values are expressed in milligrams per 100 grams of dry matter.

Table 1. Minerals quantitative values in *M. oleifera* and *S. occidentalis* organs (mean $\pm$ standard deviation)

	MOL	MOE	MOF	SOL	SOE	SOC	SOG	P value
Cu	0.01 $\pm$ 0.00	0.03 $\pm$ 0.00	0.07 $\pm$ 0.00	0.05 $\pm$ 0.00	0.05 $\pm$ 0.00	0.06 $\pm$ 0.00	0.05 $\pm$ 0.00	0.0001
Mg	2.35 $\pm$ 0.41	7.09 $\pm$ 0.41	6.72 $\pm$ 0.41	11.47 $\pm$ 0.41	7.43 $\pm$ 0.41	7.98 $\pm$ 0.41	8.45 $\pm$ 0.41	0.0001
Mn	0.24 $\pm$ 0.03	0.05 $\pm$ 0.03	0.08 $\pm$ 0.03	0.25 $\pm$ 0.03	0.09 $\pm$ 0.03	0.14 $\pm$ 0.03	0.07 $\pm$ 0.03	0.0001
Fe	0.39 $\pm$ 0.23	0.21 $\pm$ 0.23	0.34 $\pm$ 0.23	0.30 $\pm$ 0.23	0.22 $\pm$ 0.23	0.23 $\pm$ 0.23	0.27 $\pm$ 0.23	0.9508
Zn	0.16 $\pm$ 0.07	0.14 $\pm$ 0.07	0.19 $\pm$ 0.07	0.28 $\pm$ 0.07	0.17 $\pm$ 0.07	0.18 $\pm$ 0.07	0.24 $\pm$ 0.07	0.1929
K	45.48 $\pm$ 0.42	46.98 $\pm$ 0.42	74.35 $\pm$ 0.42	68.75 $\pm$ 0.42	35.53 $\pm$ 0.42	57.60 $\pm$ 0.42	36.34 $\pm$ 0.42	0.0001
Na	195.4 $\pm$ 0.28	2.77 $\pm$ 0.28	2.51 $\pm$ 0.28	109.3 $\pm$ 0.28	2.02 $\pm$ 0.28	7.53 $\pm$ 0.28	3.17 $\pm$ 0.28	0.0001
Ca	60.31 $\pm$ 0.33	68.60 $\pm$ 0.33	18.26 $\pm$ 0.33	95.56 $\pm$ 0.33	60.86 $\pm$ 0.33	39.77 $\pm$ 0.33	56.99 $\pm$ 0.33	0.0001

MOL - *M. oleifera* leaves; **MOE** - *M. oleifera* bark; **MOF** - *M. oleifera* flowers; **SOL** - *S. occidentalis* leaves; **SOE** - *S. occidentalis* bark; **SOC** - *S. occidentalis* shell; **SOG** - *S. occidentalis* seeds; **Cu** - copper; **Mg** - magnesium; **Mn** - manganese; **Fe** - iron; **Zn** - zinc; **K** - potassium; **Na** - sodium; **Ca** - calcium

Minerals flow chart have permit to highlight eight mineral in the two plants various organs.

The different minerals content are measured by atomic absorption spectrometry. Bonferroni's multiple comparison test showed that there is a significant difference for all compared values except for seeds and leaves of *S. occidentalis*.

Mainly, four minerals have manifested their presence in quite remarkable proportions. These minerals was sodium (Na), calcium (Ca), potassium (K) and magnesium (Mg). For these minerals, we report 195.4 $\pm$ 0.26 mg (*M. oleifera* leaves); 95.56 $\pm$ 0.33 mg (*S. occidentalis* leaves); 74.35 $\pm$ 0.42 mg (*M. oleifera* flowers) and 11.47 $\pm$ 0.41 mg (*S. occidentalis* leaves) respectively. Peñalver et al. (2022) found in *Moringa oleifera* leaves for sodium, calcium, potassium and magnesium values of 133.11 $\pm$ 20.09 mg/100 g; 1.48 $\pm$ 0.01 mg/100g; 1.75 $\pm$ 0.02 mg/100 g and 301.11 $\pm$ 2.08 mg/100 g respectively. Magnesium value is only higher than ours is. ICP-MS was used for their minerals analysis, method different from ours.

When compared, it was found that, *M. oleifera* leaves are richer in sodium (195.4 $\pm$ 0.26) than *S. occidentalis* leaves (109.3 $\pm$ 0.27).

On the other hand, *S. occidentalis* leaves are remarkably rich (95.56 $\pm$ 0.33) in calcium than *M. oleifera* leaves (60.31 $\pm$ 0.33). Calcium is more abundant in *Moringa* bark than its other organs. Potassium in *M. oleifera* flowers (74.35 $\pm$ 0.42) has the highest concentration value. In *S. occidentalis*, leaves are richer in magnesium (11.47 $\pm$ 0.41) than other organs compared.

Determination of amino acid profile. Seventeen amino acids were detected and identified in *M. oleifera* (leaves, bark and flowers) and *S. occidentalis* (leaves, bark, seeds and seed capsules). Total amino acids in MOL, MOE, MOF, SOL, SOE, SOC and SOG were 44.37; 4.26; 28.7; 44.81; 19.19; 18.8 and 47.02 g/100 g respectively. Amino acids are more concentrated in *S. occidentalis* seeds than compared to other organs. *M. oleifera* and *S. occidentalis* leaves have substantially the same amino acid values. Total amino acids lowest values are found in *M. oleifera* bark (4.26), and *S. occidentalis* bark (19.19) and seed capsules (18.8). Amino acids high concentrations in *S. occidentalis* seeds and leaves would result from the fact that in leaves level elaborated sap is produced. Some of it stays in leaves and a good part is stored in seeds. Methionine and cysteine were not detected in *M. oleifera* bark. An alkaline treatment could cause

Table 2. *M. oleifera* and *S. occidentalis* amino acid contents

Amino acids	Code samples						
	MOL	MOE	MOF	SOL	SOE	SOC	SOG
1 Asparagine	13.51	1.09	8.28	14.32	6.64	6.44	13.82
2 Glutamine	10.35	0.77	6.89	10.01	3.29	3.25	14.01
3 Serine	1.37	0.19	0.97	1.29	0.71	0.72	1.59
4 Glycine	1.23	0.08	0.67	1.09	0.43	0.42	0.84
5 <u>Histidine</u>	0.59	0.11	0.40	0.60	0.24	0.22	0.66
6 Arginine	1.88	0.17	1.63	1.64	0.57	0.56	1.89
7 <u>Threonine</u>	1.37	0.15	0.88	1.20	0.57	0.55	1.15
8 Alanine	1.87	0.14	1.02	1.63	0.57	0.59	1.26
9 Proline	2.33	0.61	2.33	3.92	1.83	1.88	2.89
10 Tyrosine	1.07	0.18	0.61	1.02	0.59	0.53	0.91
11 <u>Valine</u>	1.85	0.20	1.04	1.64	0.80	0.75	1.52
12 <u>Méthionine</u>	0.62	0.00	0.34	0.50	0.26	0.24	0.60
13 Cystine	0.28	0.00	0.16	0.21	0.08	0.16	0.30
14 <u>Isoleucine</u>	1.03	0.11	0.64	1.05	0.46	0.42	0.70
15 <u>Leucine</u>	2.35	0.17	1.28	2.08	0.93	0.86	1.77
16 <u>Phénylalanine</u>	1.42	0.12	0.81	1.36	0.60	0.56	1.26
17 <u>Lysine</u>	1.26	0.19	0.76	1.25	0.73	0.66	1.68
Total	44.37	4.26	28.70	44.81	19.19	18.80	47.02

MOL - *M. oleifera* leaves; **MOE** - *M. oleifera* bark; **MOF** - *M. oleifera* flowers; **SOL** - *S. occidentalis* leaves; **SOE** - *S. occidentalis* bark; **SOC** - *S. occidentalis* shell; **SOG** - *S. occidentalis* seeds; essential amino acids are underlined

these amino acids destruction when starting analysis (Zhu et al. 2006). Aspartic acid is by far the most abundant amino acid in the different organs of both plants except in *S. occidentalis* seeds in which glutamic acid is the most abundant.

The non-essential amino acids (aspartate, glutamate, serine, glycine, arginine, alanine, proline, tyrosine and cysteine) of both plants are represented in high proportion. Founded values for these non-essential amino acids were 14.01; 13.51; 3.92; 1.90; 1.87; 1.37; 1.23; 1.07 and 0.30 respectively for glutamate (SOG), aspartate (MOL), proline (SOL), arginine (SOG and MOL), alanine (MOL), serine (MOL), glycine (MOL), tyrosine (MOL) and cysteine (SOG). The main non-essential amino acids were glutamate (SOG), aspartate (MOL) and proline (SOL). Our results are contrary to those of Adewumi et al. (2022) who instead found glutamine and asparagine as the main non-essential amino acids.

Methionine and cysteine low content in our two plants is similar to other legume seed protein studies reported by Adebawale et al. (2011).

Amino acids are organic compounds, which are precursors of proteins; therefore, they influence proteins quantity and quality (Mune Mune et al. 2016). They are essential for enzymes, immunoglobulins, hormones production, for body tissues growth and repairing and form red blood cells structure. Moreover, they play an important role in glucose formation, acting as a buffer when other precursors are scarce. Amino acids are essential for performing specific functions in the body (Moyo et al. 2011).

These two plants are therefore a good source of essential amino acids, because they are particularly rich in threonine, valine, isoleucine, leucine, phenylalanine, lysine, methionine, phenylalanine and histidine compared to whey, peas, brown rice, soy, hemp and wheat protein (Fig. 12).

Fig. 5, 6 and 7 show the peaks of *Moringa oleifera* different amino acids.

Fig. 8, 9, 10 and 11 show the peaks of *Senna occidentalis* different amino acids.

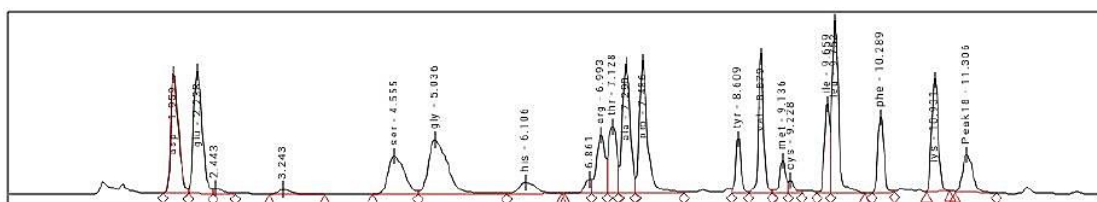


Figure 5. Amino acid peaks of *Moringa oleifera* leaves

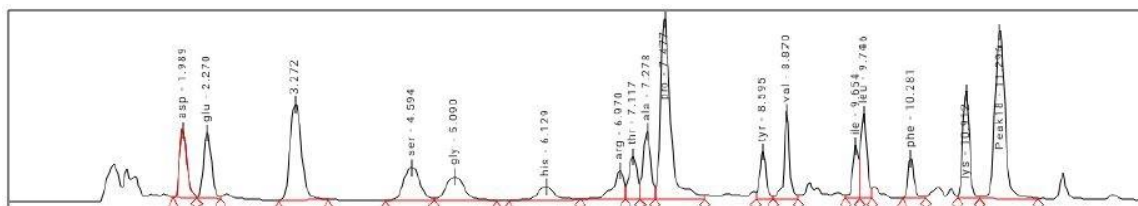


Figure 6. Amino acid peaks of *Moringa oleifera* bark

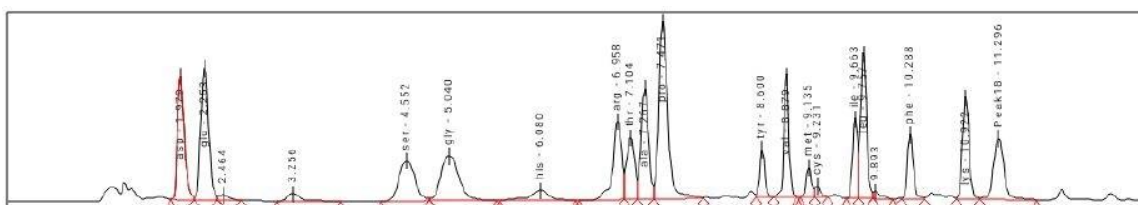


Figure 7. Amino acid peaks of *Moringa oleifera* flowers

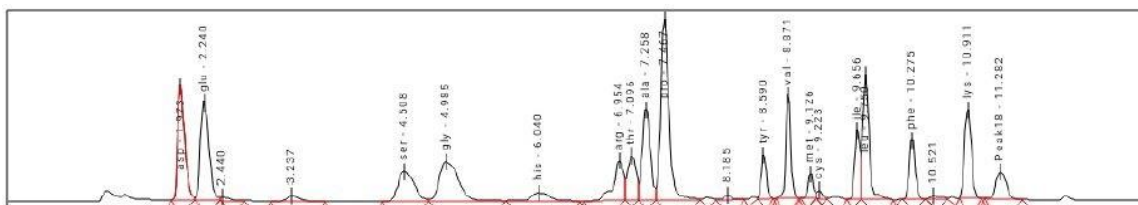


Figure 8. Amino acid peaks of *Senna occidentalis* leaves

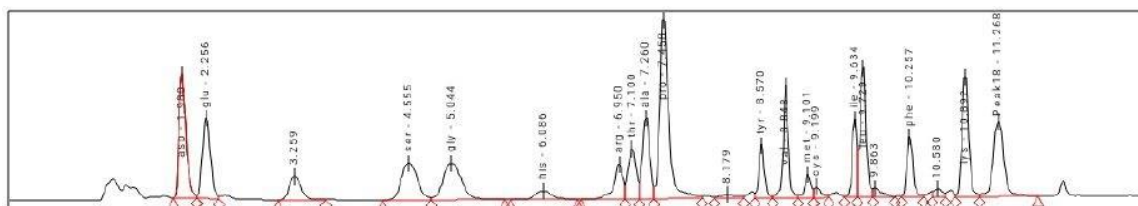


Figure 9. Amino acid peaks of *Senna occidentalis* bark

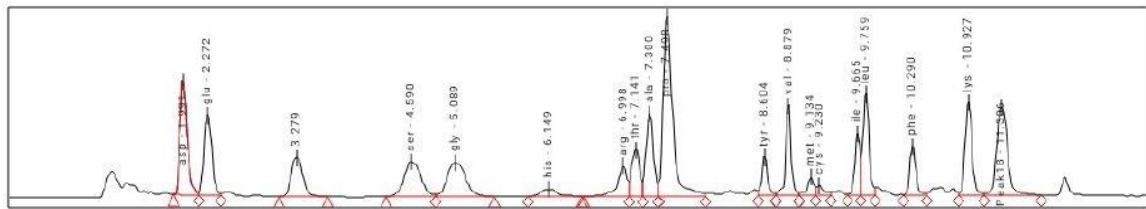


Figure 10. Amino acid peaks of *Senna occidentalis* seed capsules

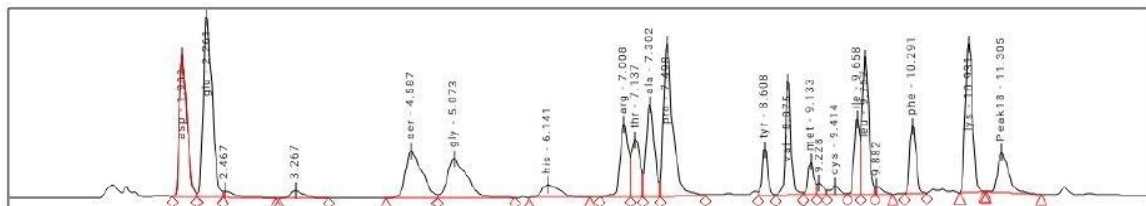


Figure 11. Amino acid peaks of *Senna occidentalis* seeds

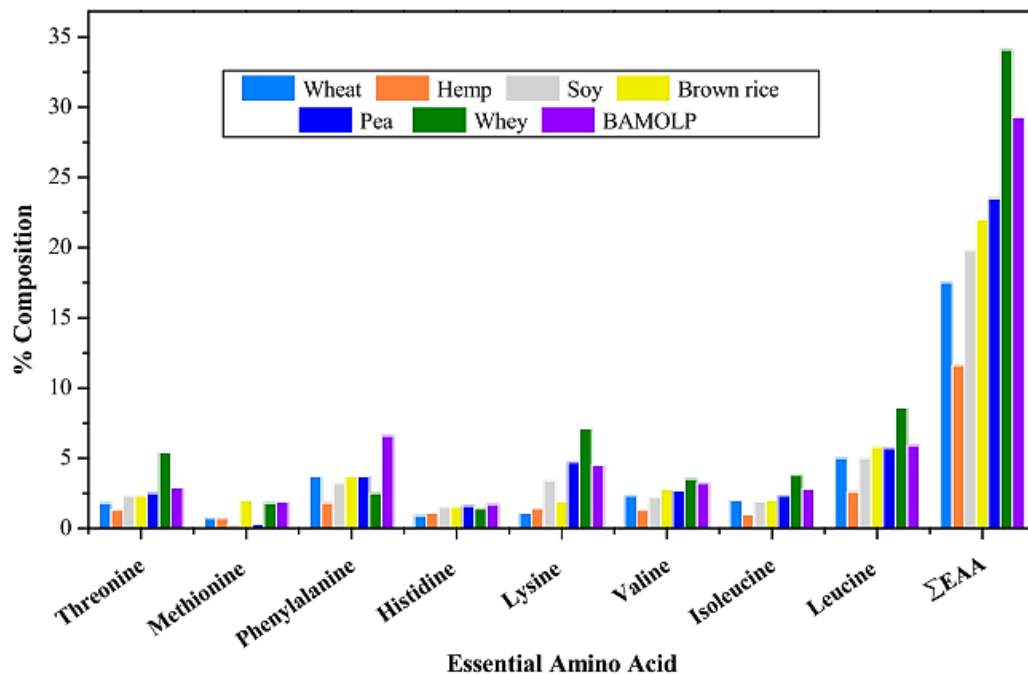


Figure 12. Amino acids comparison in Bambara groundnut and *Moringa oleifera* leaf powder protein complex (BAMOLP) and other protein sources. ΣEAA - total essential amino acid. [Source: (Adewumi et al. 2022)]

Conclusions

The analysis of *M. oleifera* and *S. occidentalis* reveals that these two plants possess notable nutraceutical potential. Indeed, the protein content of *M. oleifera* leaves was found to be significantly higher than that of *S. occidentalis* leaves and the protein content of the other two plants' organs. *M. oleifera* cultivated under organic conditions

represents a valuable source of protein and is rich in essential amino acids. It can be concluded that these two plants are an excellent source of essential amino acids, particularly threonine, valine, isoleucine, leucine, phenylalanine, lysine, methionine, and histidine. This is in comparison to other plants, including whey, peas, brown rice, soy, hemp, and wheat. *Moringa* flowers were found to be

particularly rich in fat. Additionally, both *Moringa* and *Senna* contain high levels of sugars and ash, which indicate a high mineral content. The parameters analysed in this study, including organic and mineral content, have been shown to be effective indicators of the nutraceutical value of these two plants cultivated under organic conditions. This study's findings suggest that these plants could be used as supplements to cereals to address certain nutritional deficiencies.

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Author Contributions

Conceptualization, S.A., T.Y. and Z.C.; methodology, S.A., K.S. and T.F.; formal analysis K.S., S.S. and T.F.; investigation, K.S.; resources, S.A. and T.Y.; data curation, K.S., T.F. and S.S.; writing original draft preparation, K.S. and T.F.; writing – K.S., T.F. and S.A., review and editing, K.S., and T.F., visualization, S.A. and T.Y.; supervision, S.A. and Z.C.; project administration, S.A. and T.Y.; funding acquisition, S.A. All authors have read and agreed to the published version of the manuscript.

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Institutional Review Board Statement

The protocol carried out in this research did not involve the use of animals.

Informed Consent Statement

Not applicable.

Data Availability Statement

The original contributions presented in this study are included in the article. Further inquiries can be directed to the corresponding authors.

Conflicts of Interest

The authors declare no conflicts of interest.

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