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Nutritional Value of Wild and Domesticated *Sanguisorba minor* Scop. Plant

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Abstract: *Sanguisorba minor* Scop. is a wild edible species distributed in the Mediterranean area and present in numerous traditional food recipes. In the present study, the assessment of nutritional value (ash, carbohydrates, fat, proteins, energy, free sugars, organic acids, tocopherols, fatty acid composition, and minerals) of wild and domesticated *S. minor* plants was performed. Results showed an increase in ash, protein, fat, organic acid, and α-tocopherol content after the plant's domestication. Retention of free sugars, especially sucrose, was observed from wild plants to domesticated ones. However, the cultivated plants reported a higher content of polyunsaturated fatty acids than saturated molecules, and both wild collection and domestication maintained a low ω6/ω3 ratio, confirming the role of this species in the prevention of oxidative and inflammatory processes. This aspect is also suggested by the high α-tocopherol content, a vitamin known for its ability to scavenge free-radical species. Nevertheless, a high oxalic acid content was found in domesticated plants. However, the management of fertilization in open field cultivation can be robust in terms of organic acid and mineral (e.g., calcium) content. Indeed, the most representative macrominerals found in domesticated plants were Ca and Mg. The present study suggests a possible introduction of *S. minor* species in the human diet as a functional food or ingredient by virtue of its high nutritional properties and contents. Moreover, the management of fertilization and domestication might be a solution to maintain/enhance the nutritional profile of this wild species.

Keywords: *Sanguisorba minor*; wild harvest; cultivation; nutritional characteristics

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1. Introduction

Sanguisorba minor Scop. is a wild edible species belonging to the Rosaceae family well known for its edibility, its folkloristic and traditional uses, and its nutritional and nutraceutical properties discovered during the last few years [1,2]. *S. minor* is commonly harvested wild in dry and semi-dry grasslands throughout Europe [3]. In Mediterranean traditional food recipes, wild *S. minor* and other species or sub-species of this genus (e.g., *S. minor* spp. *muricata*, *S. officinalis*) are used as boiled vegetables, in traditional soups, and in "misticanza" salad [4]. Nowadays, the rediscovery of ancient food recipes and culinary and medicinal traditions is improving because of the increase in human dietary deficiencies in the undeveloped world [5]. For these reasons, interest in the nutritional and nutraceutical properties of this wild edible herb is increasing, especially regarding its aerial parts and young shoots, thanks to their edibility [6,7]. Viano et al. [8] reported palmitic (29.1%), linoleic (22.6%), and linolenic (21.4%) acids as the main fatty acids and glutamic and aspartic acids as the main amino acids present in wild-collected *S. minor* ssp. *muricata*. Concerning the mineral composition, Pirhofer-Walzl et al. [9] reported a higher concentration of the macrominerals P^{+}, Mg^{2+}, K^{+}, and S and the trace elements Zn^{2+} and B^{+} in wild-collected *S. minor* than in common grasses. Karkanis et al. [7] observed a

high amount of α-tocopherol (vitamin E). This vitamin is well known as an inhibitor of cyclooxygenase 2 (COX-2) involved in inflammatory processes but also as a free-radical scavenger [10–12].

Considering the significant nutritional and nutraceutical properties of *S. minor* species, which are able to diversify the human diet and satisfy the increasing market demands for functional foods as well as enhance nutritional intake, the domestication of *S. minor* plants could be efficient to meet the different requirements of people from different parts of the world [2,7,13]. For this reason, the possibility of domestication of this species is increasingly studied, and the high germination percentage of *S. minor* makes it a promising plant [6]. Therefore, its suitability for different domestications has recently been analyzed in terms of nutritional and bioactive compounds as well as medicinal properties such as antimicrobial or cytotoxic activities [2,7,14,15]. Domestication is usually able to retain chemical compounds present in wild edible species, even though the adopted cultivation technique and the genetic features of the wild edible plants under investigation are the major characteristics affecting the changes in nutritional composition during the domestication [14,16,17].

A deep knowledge concerning the nutritional composition of wild *S. minor* may lead to characterizing the wild nutritional composition of this species and finding an adequate domestication technique able to retain its nutritional composition. Therefore, the aim of this study was to compare and reveal the differences between wild and domesticated *S. minor* plants in terms of nutritional features. In particular, minerals of this species were analyzed for the first time in this experiment in both wild and domesticated *S. minor*, while fatty acids, free sugars, organic acids, and tocopherols were analyzed using proximate analysis for the first time in wild *S. minor* plants.

2. Materials and Methods

2.1. Plant Material and Experimental Design

Seedlings of *S. minor* were collected in the wild (W) in the Tuscany region (43°43′ N, 10°31′ E, Italy) and, at the same time, Tirrenofruit s.r.l., a wholesaler of vegetable food products, provided domesticated *S. minor* plants cultivated on a local farm (F) located in the same area as the wild collection during the spring of 2019. The growing conditions monitored by a weather station near the experimental parcels were: 22 °C average temperature, 50% humidity, and 71.71 lumens cm^{-2} light intensity with 14 h of light. The growth soil was mainly sandy, and 30 kg of manure per ha was used as fertilizer for the soil. *S. minor* young plants were collected by farmers when the shoots were adequate for edibility. Shoots plus leaves were weighed and oven dried at 60 °C until reaching a constant weight, and the dry matter percentage was calculated. Dry samples were used for the analyses described below. All analyses were carried out in triplicate.

2.2. Nutritional Composition Analysis

2.2.1. Proximate Analysis

Samples were analyzed in terms of macronutrients (proteins, fat, carbohydrates, and ash) as reported by the AOAC procedures [18]. The crude protein (N × 6.25) was determined by the Kjeldahl method (978.04) [AOAC, 2005]. The analyte was referred to as "crude" protein because the method determines N, a component of all proteins. In addition, N from sources different from true proteins was also determined. The ash content (AOAC 930.05) was determined by subjecting the sample to incineration at 600 ± 15 °C for 5 h; the crude fat was determined using a Soxhlet apparatus with petroleum ether (AOAC 920.39); and the total carbohydrate content was estimated by difference. The results were expressed in g per 100 g of dry weight (DW). The total energy was calculated using the following equation: energy (kcal) = 4 × (g proteins + g carbohydrates) + 9 × (g fat). Indeed, proteins and carbohydrates provide 4 kcal g^{-1} energy intake, while fats provide 9 kcal g^{-1} energy intake. The results for total energy were expressed in kcal per 100 g DW.

2.2.2. Fatty Acids

The authors previously described the determination of fatty acids using gas-liquid chromatography with flame ionization detection (GC/FID) on a capillary column. Fatty acids were methylated using a transesterification procedure, where 5 mL of methanol:sulfuric acid:toluene in a 2:1:1 (*v*:*v*) ratio was added to the samples, and the mixture was allowed to react for at least 12 h in a bath at 50 °C and 160 rpm. Following this, 3 mL of deionized water were added to obtain phase separation. The fatty acid methyl esters (FAME) were recovered by adding 3 mL of diethyl ether and shaking in a vortex. Then, the upper phase was passed through a microcolumn of anhydrous sodium sulfate to remove water. Resulting samples were recovered in a vial with Teflon, and prior to injection, samples were filtered using a 0.2 µm nylon filter from Millipore [18].

Fatty acid identification was carried out as reported by Barros et al. [19] using gas chromatography coupled with a flame ionization detector (GC-FID/capillary column, DANI model GC 1000, Contone, Switzerland) and a split/splitless injector, using a Macherey–Nagel column (30 m × 0.32 mm I.D. × 0.25 µm d_f). The column temperature in the oven was programmed as follows: starting at 50 °C, it was held for 2 min, then ramped up at a rate of 10 °C per minute to 240 °C and held for 11 min. The carrier gas used was hydrogen, with a flow rate of 4.0 mL/min (0.61 bar), measured at 50 °C. Split injection at a temperature of 250 °C was performed at a ratio of 1:40. For each analysis, 1 µL of sample was injected into the GC. Fatty acid identification was achieved by comparing the relative retention times of the FAME peaks in the samples with the standard (standard mixture 47885-U; fatty acids C4–C24; Sigma, St. Louis, MO, USA). Results were recorded and processed using CSW 1.7 software (DataApex 1.7; Podohradska, Czech Republic) and expressed as relative percentages of each fatty acid.

2.2.3. Free Sugars

Free sugars were determined as previously reported by Barros et al. [19] using the Internal Standard (IS, melezitose, Sigma-Aldrich, St. Louis, MO, USA) method. Dried sample powder (1 g) was extracted in 1 mL of melezitose (5 mg/mL) and 40 mL of 80% (*v*/*v*) aqueous ethanol at 80 °C for 1.5 h, shaking every 15 min. The resulting suspension was centrifuged (Centurion K24OR refrigerated centrifuge, West Sussex, UK) at 15,000× *g* for 10 min. The supernatant was concentrated at 40 °C under reduced pressure and washed three times with 10 mL of diethyl ether, successively. After concentration at 40 °C in the oven, solid residues were dissolved in water to obtain a final volume of 5 mL and filtered through 0.2 µm Whatman nylon filters. After the filtration, the sugar molecules were analyzed by high-performance liquid chromatography coupled to a refraction index detector (HPLC-RI, Knauer, Smartline system 1000). The chromatographic separation was achieved with an Eurospher 100-5 NH2 column (4.6 mm × 250 mm, 5 mm, Knauer) operating at 35 °C (7971R Grace oven). The used mobile phase was acetonitrile/deionized water, 7:3 (*v*/*v*), at a flow rate of 1 mL/min, and the injection volume was 20 µL. Results were recorded and processed using Clarity 2.4 software (DataApex, Prague, Czech Republic). Quantification was performed using internal standards (d-(−)-Fructose, d-(+)-Glucose, d-(+)-Sucrose, and d-(+)-Trehalose; Sigma, St. Louis, MO, USA), and free sugar concentrations were further expressed in g/100 g of DW calculated by internal normalization of the chromatographic peak area.

2.2.4. Organic Acids

Organic acids were determined as previously reported by Barros et al. [19]. Dried sample powder (about 1 g) was extracted with 25 mL of metaphosphoric acid, stirring the solution at 150 rpm at 25 °C for 20 min. Extracted samples were filtered through Whatman No. 4 paper and, successively, through 0.2 µm nylon filters. The Shimadzu 20A series UFLC (Shimadzu Corporation, Kyoto, Japan) was used for the analysis. Separation was achieved on a Phenomenex SphereClone reverse-phase C18 column (5 µm, 250 mm × 4.6 mm I.D.) thermostatically controlled at 35 °C. Elution was carried out using sulfuric acid (3.6 mM)

at a flow rate of 0.8 mL/min. Detection was performed using a PDA at the preferred wavelengths of 215 nm and 245 nm (for ascorbic acid). Detected organic acids were quantified by comparison of the area of their peaks recorded at 215 and 245 nm with calibration curves obtained from commercial standards of each compound: ascorbic acid ($y = 8 \times 10^7x + 55{,}079$; $R^2 = 1$); citric acid ($y = 1 \times 10^6x + 4170.6$; $R^2 = 1$); fumaric acid ($y = 172{,}760x + 52{,}193$; $R^2 = 0.999$); malic acid ($y = 952{,}269x + 17{,}803$; $R^2 = 1$); oxalic acid ($y = 1 \times 10^7x + 96{,}178$; $R^2 = 0.999$); and quinic acid ($y = 601{,}768x + 8853.2$; $R^2 = 1$); all were purchased from Sigma (St. Louis, MO, USA). Results were processed using LabSolutions Multi LC-PDA software (Shimadzu Corporation, Kyoto, Japan), and they were expressed in g/100 g DW.

2.2.5. Tocopherols

Tocopherol determination was carried out as previously reported by Barros et al. [19]. The extraction was performed by adding 100 µL of butyl hydroxy toluene solution in hexane (BHT; 10 mg/mL) and 400 µL of tocol solution (SI; Matreya (State College, PA, USA)) in hexane as an internal standard (50 µg mL) to dried samples (500 mg). The extracted samples were homogenized, first with methanol (4 mL) by vortex mixing (1 min), then with hexane (4 mL) by vortex mixing (1 min), and finally with saturated NaCl aqueous solution (2 mL) by vortex mixing again. The resulting samples were centrifuged at 4000× *g* for 5 min at 10 °C, and the supernatant was transferred to a vial. Samples were re-extracted twice with hexane and then dried under nitrogen steam, dehydrated with anhydrous sodium sulfate, and filtered using 0.2 µm nylon filters from Whatman. Tocopherols were determined using an HPLC system (Knauer, Smartline System 1000, Berlin, Germany) coupled to a fluorescence detector (FP-2020; Jasco, Easton, PA, USA) programmed for excitation at 290 nm and emission at 330 nm. The chromatographic separation was achieved with a Polyamide II normal-phase column (250 mm × 4.6 mm; YMC Waters) operating at 30 °C. The mobile phase used was a mixture of *n*-hexane and ethyl acetate (70:30, *v*/*v*) at a flow rate of 1 mL/min. The identification of α-tocopherol was carried out by chromatographic comparison with an authentic standard and quantification by calibration curve obtained from commercial standards (α-, β-, γ-, and δ-isoforms; St. Louis, MO, USA) using the internal standard methodology. Results were recorded and processed using Clarity 2.4 software (DataApex, Prague, Czech Republic). Results were expressed in mg per 100 g DW.

2.2.6. Minerals

Dried sample powders (0.2 g) were mineralized at 220 °C for 90 min using a solution of HNO_3:$HClO_4$ (2.5:1 *v*/*v*) and an atomic absorption spectrometer (Varian AA 24FS, Australia) was used for the determination of Na^+, K^+, Ca^{2+}, Mg^{2+}, Cu^{2+}, Mn^{2+}, Fe^{2+}, and Zn^{2+}. Results were expressed as mg per g fresh weight (FW) for Na, K, Ca, and Mg and µg per g FW for Cu, Mn, Fe, and Zn.

2.3. Statistical Analysis

Differences between wild (W) and domesticated (F) *S. minor* plants were calculated by the Student's *t*-test ($p < 0.05$) with GraphPad Software (GraphPad, La Jolla, San Diego, CA, USA). The data are expressed as the mean ± standard deviation (±SD) of three replicates.

3. Results and Discussion

3.1. Nutritional Value

Table 1 reports results of nutritional composition in terms of moisture, ash, proteins, fat, carbohydrates, and total energy of W and F *S. minor* plants. The highest content of ash, proteins, and fat was recorded in cultivated plants, whereas the same plants showed the lowest moisture and carbohydrate content than W plants.

Table 1. Proximate composition of wild (W) and domesticated (F) *Sanguisorba minor* plants expressed in dry weight (mean ± SD). Data were analyzed following the Student's *t*-test ($p < 0.05$) between W and F plants.

Proximate Composition			
	W	**F**	***t*-Student *p*-Value**
Moisture (g/100 g)	75.02 ± 0.04	71.20 ± 1.00	0.0054
Ash (g/100 g)	7.80 ± 0.60	10.10 ± 0.40	0.0491
Proteins (g/100 g)	18.80 ± 0.40	23.10 ± 0.30	0.0063
Fat (g/100 g)	2.72 ± 0.04	4.00 ± 0.30	0.0213
Carbohydrates (g/100 g)	70.70 ± 0.20	62.79 ± 0.40	0.0018
Energy (kcal/100 g)	382.00 ± 3.00	379.20 ± 0.50	0.2507

No significant differences were found in terms of caloric energy provided between W and F *S. minor* plants. However, for future steps in the experimentation of the nutritional value of *S. minor* species, the content of soluble and insoluble dietary fibers, consisting of non-starch polysaccharides and other plant components such as cellulose, resistant starch, resistant dextrins, inulin, lignins, pectins, beta-glucans, and oligosaccharides, could be useful to more correctly calculate the caloric value of this species because the energy conversion factor for soluble sugars is 3.75–4.0 kcal/g, whereas that of commonly eaten foods containing a mixture of fermentable and nonfermentable fibers is 0–1.9 kcal/g, according to the Food and Agriculture Organization (FAO). Moreover, other authors analyzed the compositional analysis of Sanguisorba spp. since this plant is considered promising in terms of nutritional and nutraceutical value. Interestingly, Viano et al. [8], analyzing *S. minor* spp. muricata collected as wild in a French park near Marseille, reported a lower content of fat (1.51%) when compared with W or F *S. minor* plants analyzed in the present experiment. However, Tsugkiev et al. [20], analyzing S. officinalis collected as wild at different sea levels, observed lower ash and protein content and higher fat content when compared with W and F plants of the present experiment, even though these authors found many differences between plants collected from different sea levels (e.g., ash content between 5.8 and 7.6 g/100 g, dependent on sea level increase). Thus, different sea levels as well as all other growth environmental parameters might be the reason for different plant chemical compositions. Differences might also be attributed to pedo-climatic growth conditions and genetic features. Moreover, Karkanis et al. [7], analyzing *S. minor* plants whose seeds had been found on wild plants, observed a similar fat and carbohydrate content to the wild or domesticated plants of the present experiment when cultivated with only peat. In this case, the cultivation with only peat when compared with the cultivation with peat and perlite (independently of the percentage of added perlite) resulted more similar to the cultivation in open field or to wild collection, likely due to the presence of perlite, which can lead to the increase of macro- and micropores of plant growth substrate, enhancing the plant nutrient uptake present in the manure used as fertilizer of growth soil, and consequently modify the chemical composition of plant aerial parts [21].

3.2. Sugar Content

The most abundant free sugar identified in both W and F *S. minor* plants was sucrose (Table 2). Wild plants were not significantly affected by domestication in terms of free sugars. Sucrose and raffinose contents were not significantly different in W and F *S. minor* plants, while fructose and glucose in W plants were slightly higher than those in F plants.

Table 2. Sugar content of wild (W) and domesticated (F) *Sanguisorba minor* plants expressed in dry weight (mean ± SD). Data were analyzed following the Student's *t*-test ($p < 0.05$) between W and F plants.

Sugar Content (g/100 g)			
	W	**F**	***t*-Student *p*-Value**
Fructose	0.50 ± <0.01	0.30 ± 0.10	0.0492
Glucose	1.00 ± 0.10	0.30 ± 0.10	0.0182
Sucrose	3.61 ± 0.04	4.10 ± 0.20	0.0825
Raffinose	0.31 ± 0.01	0.28 ± 0.02	0.3491
Total sugars	5.42 ± 0.03	4.98 ± 0.20	0.1068

Differently, Karkanis et al. [7] showed that fructose and glucose were the major identified sugars in domesticated *S. minor* plants. In general, the sugar content found in the present experiment was lower than that found by Karkanis et al. [7]. Conversely, Viano et al. [8] reported a lower content of total free sugars in *S. minor* spp. *muricata* collected as wild than that of the samples under investigation, independent of cultivation or wild collection. These results confirm that free sugar content is obviously affected by plant growth conditions in the environment. However, the *S. minor* plants of the present experiment might also be considered a rich source of carbohydrates when cultivated, making them an important parameter for the introduction of a new functional food or ingredient in the human diet, even though a crude fiber determination is necessary to confirm this statement.

3.3. Organic Acid and Tocopherol Content

Organic acid content was reported in Table 3. Oxalic acid was found to be the most representative organic acid in the F *S. minor* plants, followed by citric acid. Organic acid content was significantly affected by domestication. The domesticated plants showed a significantly higher content of all organic acids than W *S. minor* plants, except for malic acid.

Table 3. Organic acid and tocopherol content of wild (W) and domesticated (F) *Sanguisorba minor* plants expressed in dry weight (mean ± SD). Data were analyzed following the Student's *t*-test ($p < 0.05$) between W and F plants.

Organic Acid Content (g/100 g)			
	W	**F**	***t*-Student *p*-Value**
Oxalic acid	2.64 ± 0.05	11.50 ± 0.70	0.0028
Quinic acid	0.16 ± 0.01	1.70 ± 0.10	0.0007
Malic acid	1.14 ± 0.02	0.79 ± 0.01	0.0027
Citric acid	1.76 ± <0.01	5.94 ± 0.01	<0.0001
Fumaric acid	0.11 ± <0.01	0.50 ± 0.03	0.00256
Total organic acids	5.82 ± 0.06	20.43 ± 0.70	0.0013
Tocopherols (mg/100 g)			
	W	**F**	***t*-Student *p*-value**
α-Tocopherol (vitamin E)	0.50 ± 0.10	11.00 ± 1.00	0.0001

Other authors confirmed that domestication increased the organic acid content, especially that of oxalic and citric acids [7]. Unfortunately, the higher content of oxalic acid in cultivated plants than that of wild plants is a negative aspect since the high intake of this organic acid in the form of oxalate in the human diet can bind with calcium in the body, forming calcium oxalate crystals and leading to kidney stones in human beings [22]. Moreover, high levels of oxalates in the body can interfere with the absorption of certain minerals, such as calcium and iron, leading to mineral deficiencies [23]. However, the management of the domestication and the fertilization (by manure in the present experiment) might be

useful to decrease the content of oxalates in *S. minor* edible parts. Boiling, steaming, or other cooking technologies could also be very useful to reduce oxalic acid content since it is well known that this organic acid is very sensitive to high temperatures [24].

Regarding tocopherol content (Table 3), α-tocopherol was the only isoform observed in both F and W *S. minor* samples. Results reported a higher significant content in F *S. minor* plants when compared with W plants, suggesting the possible role of domestication in enhancing α-tocopherol content. The presence of the only α-tocopherol isoform in *S. minor* species was confirmed by Karkanis et al. [7]. Further studies are necessary to understand the biological mechanism that affects the α-tocopherol content during domestication. A speculation could be performed in terms of oxidative stress induced by domestication and the role of α-tocopherol as a scavenger in the light reactions of the photosynthetic process [25]. The oxidative stress could not be present in the wild collected plants due to the adaptation of plants to wild conditions since the collected plants could be a regrowth of the plant. However, the increase in α-tocopherol content during the cultivation of *S. minor* plants enhances the nutraceutical value of this species as a food or ingredient. Indeed, it has been well known for several years that in the diet, α-tocopherol is a vitamin with excellent biological activity as a free-radical scavenger and that it can serve as a therapeutic drug against free-radical-involved diseases as well as an inhibitor of COX-2 involved in the inflammatory process [10–12].

3.4. Fatty Acids

A few significant differences between W and F *S. minor* plants were found in fatty acid content (Table 4). Specifically, lauric (C12:0), pentadecanoic (C15:0), palmitic (C16:0), stearic (C18:0), and lignoceric (C24:0) acids resulted significantly higher in W *S. minor* plants when compared with domesticated ones, while oleic (C18:1n9) and linoleic (C18:2n6) acids reported higher content in domesticated plants than those collected as wild. However, the main fatty acid identified in the aerial part of *S. minor* was α-linolenic acid (36.28–37.50%), followed by palmitic (25.06–22.03%) and linoleic (11.33–13.40%) acids. Similarly, Karkanis et al. [7] observed α-linolenic acid as the most abundant fatty acid (49.4%) in the aerial part of *S. minor* plants, followed by palmitic (14.6–15.6%) and linoleic (12.9–13.1%) acids. In addition, Viano et al. [8] reported α-linolenic, palmitic, and linoleic acids as the most abundant fatty acids in the aerial part of *S. minor* spp. *muricata* plants collected as wild, confirming our findings despite the different species utilized in both experiments. The differences in terms of fatty acid content can be attributed to many factors, such as different environmental growth conditions, cultivation techniques, or harvest periods [14]. However, the similarities in terms of fatty acid varieties with other experiments suggest that the fatty acid composition is also determined by the plant genus.

Saturated fatty acids (SFA) were mainly found in W *S. minor* plants, while polyunsaturated fatty acids (PUFA) were the predominant fatty acid class present in F *S. minor* plants. Clearly, this result affected the PUFA/SFA ratio. Indeed, the wild plants reported a higher PUFA/SFA ratio when compared with domesticated plants. Due to these differences in fatty acid composition, F plants had a higher nutritional value than W plants, as the PUFA/SFA ratio was higher than 1.0, indicating a predominance of PUFA. In the latter case, both analyzed samples had ω6/ω3 ratios less than 0.45, highlighting a predominance of the ω3 family over the ω6. Guil et al. [26] and Simopoulos [27] reported the importance of both of these ratios since they are associated with the beneficial effects of *S. minor* on the cardiovascular system. Indeed, a very high ω6/ω3 ratio promotes the pathogenesis of many diseases, including cardiovascular diseases, cancer, and inflammatory and autoimmune diseases, while increased levels of ω3 PUFA exert suppressive effects [27].

Table 4. Fatty acids of wild (W) and domesticated (F) *Sanguisorba minor* plants expressed in dry weight (mean ± SD). Data were analyzed following the Student's *t*-test ($p < 0.05$) between W and F plants.

	Fatty Acids (%)		
	W	**F**	***t*-Student *p*-Value**
C8:0	0.25 ± 0.02	0.16 ± 0.03	0.0691
C10:0	0.46 ± 0.04	0.37 ± 0.02	0.1027
C12:0	1.24 ± 0.04	0.74 ± 0.04	0.0062
C13:0	nd	1.10 ± 0.04	-
C14:0	3.00 ± 0.10	nd	-
C15:0	0.49 ± 0.02	0.35 ± 0.03	0.0212
C16:0	25.10 ± 0.30	22.23 ± 0.05	0.0069
C16:1	2.60 ± 0.20	2.00 ± 0.40	0.2196
C17:0	0.87 ± 0.03	0.60 ± 0.10	0.1447
C18:0	6.30 ± 0.10	5.00 ± 0.10	0.0048
C18:1n9	5.40 ± 0.10	11.40 ± 0.10	0.0003
C18:2n6	11.33 ± 0.03	13.40 ± 0.10	0.0009
C18:3n3	36.30 ± 0.30	37.50 ± 0.40	0.0739
C22:0	1.90 ± 0.40	1.03 ± 0.04	0.087
C24:0	1.79 ± 0.03	0.96 ± 0.01	0.0004
SFA	41.40 ± 0.10	34.50 ± 0.10	0.0003
MUFA	7.95 ± 0.10	13.40 ± 0.30	0.0017
PUFA	47.61 ± 0.30	50.90 ± 0.50	0.0136
PUFA/SFA	1.15 ± <0.01	1.47 ± 0.02	<0.0001
SFA/MUFA	5.21 ± 0.003	2.58 ± 0.002	<0.0001
ω6/ω3	0.31 ± <0.01	0.36 ±< 0.01	0.0037

nd: not detected. Caprylic acid (C8:0); Capric acid (C10:0); Lauric acid (C12:0); Tridecyclic acid (C13:0); Myristic acid (C14:0); Pentadecanoic acid (C15:0); Palmitic acid (C16:0); Palmitoleic acid (C16:1); Heptadecanoic acid (C17:0); Stearic acid (C18:0); Oleic acid (C18:1n9); Linoleic acid (C18:2n6); α-Linolenic acid (C18:3n3); Behenic acid (C22:0); Lignoceric acid (C24:0). SFA—Saturated fatty acids; MUFA—Monounsaturated fatty acids; PUFA—Polyunsaturated fatty acids; PUFA/SFA is the ratio between PUFA and SFA values; ω6/ω3 is the ratio between C18:2n6 and C18:3n3.

3.5. Macromineral and Trace Element Content

Macrominerals and trace elements useful in the diet found in W and F *S. minor* plants were reported in Table 5. Many significant differences were observed between the F and W plants. Among macrominerals, Na^+ and K^+ were higher in W plants, whereas Ca^{2+} and Mg^{2+} were higher in F plants. Among trace elements, the highest contents were observed in W plants. The most representative trace element was Fe.

Table 5. Macromineral and trace element composition of *Sanguisorba minor*, wild-collected (W) and provided by a local farm (F). Each value is the mean (±SD) of three replicates. Macrominerals (Na^+, K^+, Ca^{2+}, and Mg^{2+}) were expressed in mg/g fresh weight, while trace elements (Cu^+, Mn^{2+}, Fe^{2+}, and Zn^{2+}) were expressed in µg/g fresh weight. Data were analyzed following the Student's *t*-test ($p < 0.05$) between W and F plants.

	Na	**K**	**Ca**	**Mg**	**Cu**	**Mn**	**Fe**	**Zn**
W	1.48 ± 0.13	8.30 ± 0.45	1.59 ± 0.03	0.51 ± 0.02	7.09 ± 0.33	37.06 ± 1.90	56.33 ± 3.42	16.97 ± 0.17
F	0.34 ± 0.02	5.51 ± 0.16	4.52 ± 0.26	1.44 ± 0.07	4.55 ± 0.38	13.23 ± 1.59	39.21 ± 5.68	16.39 ± 0.31
t-Student *p*-value	<0.0001	0.0005	<0.0001	<0.0001	0.0010	<0.0001	0.0107	0.0469

The results of the present experiment were confirmed by Pirhofer-Walzl et al. [9], who reported the high content of K in *S. minor* plants collected in the wild. In addition, Lenzi et al. [28] reported similar results for Ca^+, Mg^{2+}, Fe^{2+}, Cu^{2+}, Mn^{2+}, and Zn^{2+} in domesticated *S. minor* plants, confirming our findings.

The lower amount of trace elements in F plants when compared with the W ones suggested the inefficiency of domestication to maintain the trace element content of the wild plants and, consequently, to consider domestication and the management of fertilization as methods to contribute as new sources of macrominerals in the diet of undeveloped countries.

4. Conclusions

The affect of domestication demonstrated positive results, with an increase in some organic acids, α-tocopherol, and macrominerals.

The present work provides valuable results about the characterization of a little-known plant called *Sanguisorba minor*, which could represent a food plant in the future. For this reason, the use of this species in the human diet could be an introduction to a pool of nutritional elements to be considered, especially by people at risk, in the prevention of cardiovascular and chronic diseases. Given its high nutritional value and its edibility, further research about the use of this species as a food or ingredient in the human diet might be needed.

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