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Sustainable recovery of bioactive compounds from coffee silverskin

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Bioactive compounds play a crucial role in promoting human health and food preservation, driving current research toward their sustainable recovery from agri-food by-products. The aim of this research was the optimization of an eco-friendly extraction methodology of bioactive compounds from coffee silverskin, the only by-product of the coffee roasting process. Looking towards a transition to a circular economy of the food sector, food-grade green solvents (ethanol and water) were used to compare conventional (maceration) and innovative (ultrasound-assisted) extraction techniques and the variables that support the extraction process (time, temperature and solvent ratio). The data suggested that the extraction by maceration favored the best recovery of phenolic compounds, with chlorogenic and caffeic acids among the most representative ones. In particular, maceration with a hydroalcoholic solvent of ethanol/water (30 : 70), carried out for 60 minutes at 60 °C, improved the extraction efficiency of total polyphenols (44.15 mg GAE per g), flavonoids (32.36 mg ECE per g), and chlorogenic (3.34 mg g^{-1}) and caffeic (1.37 mg g^{-1}) acids, which was associated with a high antioxidant activity shown by DPPH (33.62 μM TE per g) and ABTS (98.13 μM TE per g) assays. The obtained results are encouraging for future applications of the coffee silverskin extract as a sustainable antioxidant ingredient across different sectors.

Sustainability spotlight

The research related to the manuscript focused on silverskin, the by-product from the widespread coffee industry, which produces it in large quantities. The article highlights the effectiveness of a maceration extraction method for bioactive compounds from this matrix using green solvents (ethanol and water in a 30/70 ratio). This allows for added value to be obtained from this by-product, whose extract, with a good antioxidant content, can be used in cosmetic, pharmaceutical, and food production, according to UN's Sustainable Development Goals 3 and 12.

1. Introduction

Coffee is considered one of the world's most appreciated and consumed drinks, with Brazil and Vietnam being the largest producers and exporters of green coffee beans worldwide.[1,2] Nowadays, its use in food formulations and the abundant marketing of capsules compatible with espresso coffee machines have involved an increase in the world's production of roasted coffee and, as a result, to a greater generation of by-products connected with the roasting process.[3,4] Coffee silverskin (CS), a thin tegument of green coffee beans, is separated from the beans during the roasting process[5,6] and is the sole by-product derived from the coffee roasting industries. Globally, the intensification of coffee processing associated with this production

increase has led to a considerable accumulation of CS, with estimated quantities ranging from 0.2 to 0.4 million tons annually, equivalent to approximately 18–36 g (1.8–3.6%) of silverskin per kg of roasted coffee beans.[5] The safe disposal issues of CS are a serious problem since their organic load, and the high content of phenolic acids and caffeine represent a hazard for groundwater contamination, alteration of soil microbiota, and greenhouse gas production.[7] For this reason, new strategies for managing this by-product are urgently needed. Recent studies have reported that CS is a valuable source of many bioactive compounds, such as dietary fiber, protein, and phenolics (especially chlorogenic acids)[6,8,9] able to promote healthy effects by protecting against oxidative damage, carbonyl stress, and accumulation of advanced glycation end-products (AGEs) and prebiotic activity.[10–12] Therefore, CS has already been widely recognized as a functional ingredient in various food categories[5,13–18] to encourage their reuse and valorization in the food sector with a view to a rapid transition towards a circular economy.

The efficient recovery of bioactive substances from by-product extraction is the preliminary step to obtaining high-

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value compounds. The extraction process is a critical sequential separation phase in which target compounds migrate from the raw material to the extractor based on their distribution coefficients[19,20] where extraction methods and the chemical–physical extraction variables play an important role in optimizing and maximizing the extraction process.

Maceration in the solid–liquid system has been the conventional extraction technique for over a century to recover phenolic fractions from plant matrices. Despite the ease and extreme versatility of use, its execution is critical owing to the enormous use of solvent and time that can make the execution long and expensive. On the other hand, the maceration extraction technique using passive diffusion has been recognized as an ideal extraction technique to improve the recovery of antioxidant compounds and to preserve the integrity of the recovered compounds, because they do not undergo mechanical stress as is the case with other extraction methods. Nevertheless, with the critical issues discussed, research testing the possibility of using innovative extraction techniques considers that the recovery efficiency is strongly influenced by the composition of the substrate of interest as well as the variables used during the extraction process.[21]

Ultrasound-assisted extraction represents a valid replacement for conventional methods to maximize the yield of extract, as well as to reduce the time and use of solvents. The sound waves generated by ultrasounds (frequency greater than 20 kHz) and the associated phenomena of compression and rarefaction cause the establishment of cavitation bubbles which, after growing by coalescence, collapse, generating shock waves that cause cell lysis and rapid diffusion of the solvent in the matrix with improvement in mass transfer.[22] However, this method presents criticalities mainly related to the variables of the process (amplitude, frequency, temperature, time, tempering, solvent/raw material ratio). They can lead to unstable cavitation phenomena which cause the development of radical species and the resulting accelerated loss of compounds' susceptibility to oxidation.[23]

In both reported extraction methods, the physicochemical variables (solvent, time, and temperature) play a key role in promoting the diffusion rate of bioactive compounds from the by-products to the solvent.[24,25] The determination of the correct combination time–temperature during the extraction is crucial to preserve the structural integrity of the recovered compounds and to minimize the oxidative damage.[22–24] The temperature reduces the viscosity and surface tension of the solvent, resulting in an improvement in diffusivity in the matrix enhancing the solvation of the target compounds, while a suitable extraction time avoids prolonged exposure of phenolic compounds to oxygen and/or light that aids their degradation process.[23,24] At the same time, the polarity of the extraction solvent allows for maximization of the recovery of target compounds by molecular affinity.[25] Aliphatic alcohols (methanol and ethanol), as well as polar organic solvents, are generally used for the recovery of phenolic compounds since they are predominantly polar. However, given the complexity of food by-products' chemical composition, aliphatic alcohols and organic polar solvents are often mixed with different portions of water to maximize the yield of the extraction.[22–25]

In this context, the use of GRAS (Generally Recognized as Safe) and food-grade solvents as hydroalcoholic mixtures consisting of ethanol and water is a winning choice in the view of green extraction and to produce extracts that can be used for food applications.

This research aimed to identify an eco-friendly and easily replicable extraction technique in industrial realities for the recovery of bioactive compounds from coffee silverskin with a view towards future use of the extract obtained in food applications. To achieve this purpose, this study tested the influence of (i) food-grade extraction solvents (ethanol–water alone or as a mixture); (ii) extraction methods (maceration and ultra-sound-assisted); (iii) extraction variables (temperature–time combination). The best extract was chosen considering the total amount of bioactive compounds (polyphenols, flavonoids, and tannins), chlorogenic and caffeic acids (the most abundant phenolic acids in CS), and antioxidant activity (ABTS and DPPH). Time spent, temperature, and solvent amount were also considered for this evaluation.

This study can give a shred of new evidence on the direct use of CS extracts in the food industry as ingredients in food formulation to improve functional properties and preserve their quality during storage.

2. Materials and methods

2.1 Chemicals

For the extraction process, food-grade ethanol (96% v/v) was supplied by ITW Reagents (Barcelona, Spain), and distilled ultrapure water was obtained using a Milli-Q purification system (Millipore, Bedford, MA, USA).Formic acid (≥98%), methanol (UHPLC-MS grade), and ultrapure water (UHPLC-MS grade) were supplied by Carlo Erba Reagents (Milan, Italy). Standards of chlorogenic acid (≥95%) and caffeic acid (≥98%) were purchased from Sigma-Aldrich (Merck, Darmstadt, Germany). Folin–Ciocalteu reagent, 2,2-diphenyl-1-picrylhydrazyl (DPPH), and 2,2′-azino-bis(3-ethylbenzothiazoline-6-sulfonic acid) diammonium salt (ABTS) were obtained from Sigma-Aldrich (Merck, Darmstadt, Germany).

2.2 Raw material

Caffè Mauro, Gruppo Gimoka S.p.A. roasting industry provided a Coffee Silverskin (CS) sample, a blend (50 : 50) of *Coffea arabica* L. and *Coffea canephora* Pierre ex A. Froehner. The roasted coffee beans, originating from Brazil, were subjected to a medium roasting process (215 ± 5 °C), corresponding to the colour range in the Agtron values of 55–65 (L* range 26–30), using a slow drum roasting process (about 11 min) under controlled airflow conditions.

Immediately after transfer to the FoodTec Laboratory of the Mediterranean University of Reggio Calabria (Reggio Calabria, Italy), the moisture content of CS was determined using a thermal balance (Sartorius Moisture Analyzer MA37, Germany), operating at 70 °C, following the official AOAC method.[26]

The initial moisture content was 13.00 ± 0.40% therefore the raw material was dried (50 °C for 2 h) in a ventilated oven to reduce the moisture content to 10.00 ± 0.30%. Then CS was finely ground using a laboratory grinder, sieved using a 450 μm stainless-steel mesh, to obtain a narrow particle size distribution, and manually mixed in order to ensure heterogeneity and representative sampling. Finally, the CS powder was stored at environmental temperature (25 ± 5 °C) in vacuum-sealed polyethylene bags to prevent oxidative degradation and moisture reabsorption.

The CS powder thus obtained was subjected to the experimental plan within 7 days of its preparation.

2.3 Experimental plan

The experimental plan was divided into two stages. The first stage focused on identifying the best conditions for the method (conventional or innovative) and variables (time/temperature combination) to maximize the bioactive compound extraction process. In the second stage, the selected best conditions were used to evaluate different solvents to select the best one for the maximum recovery of bioactive compounds from CS.

2.3.1 I stage: identification of the optimal conditions to recover bioactive compounds from CS. Conventional (maceration) and innovative (ultrasound-assisted) extraction techniques were compared using the variables that most affect the chemical diffusion rate in the solvent: time (30, 60, and 120 minutes) and temperature (40 and 60 °C).

The mixture (1 : 1) of food solvents, ethanol and water, was selected in stage I of CS extraction based on previous studies demonstrating its effectiveness in recovering bioactive compounds from different food by-products, including coffee by-products (Table 1).

The extraction procedures were performed as reported in Fig. 1 by mixing 1 g of CS powder with 10 mL of solvent. Solid–liquid extraction was carried out on a heating magnetic stirrer (AREX Digital, VELP Scientifica, Italy) at a stirring speed of 800 rpm. The ultrasound-assisted extraction (UAE) was carried out with an ultrasound bath (Flac Instrument, Treviglio, Italy) at the frequency of 59 kHz and set to 80% of its maximum absorbed power (135 W), corresponding to an input of 108 W. The bath temperature was controlled as a function of the applied extraction temperatures (Fig. 1) using a thermostatic recirculating system (Crioterm, GTR 90, ISCO S.r.l., Milan, Italy)
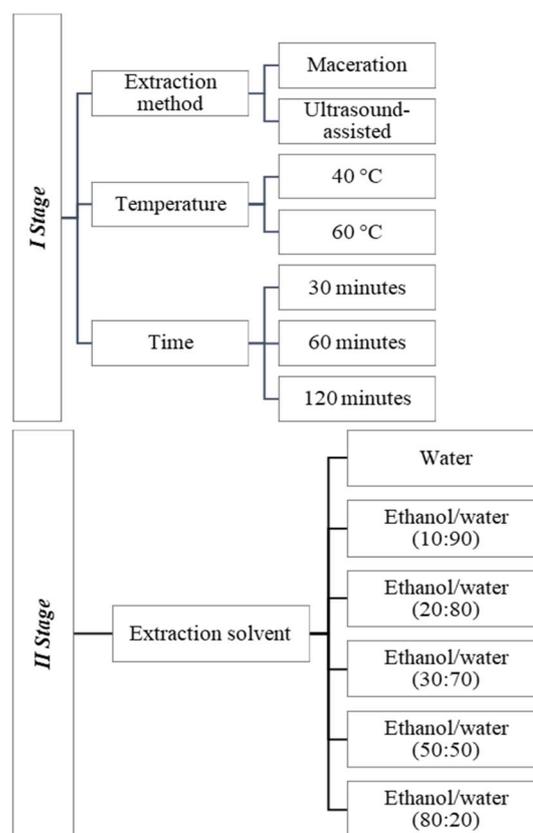

Fig. 1 Schematic representation of tested coffee silverskin extraction.

connected to the ultrasonic bath. Later, the obtained extracts (CSE) were centrifuged for 10 min, 20 °C and 6000 rpm in an NF 1200 R, Nuve centrifuge (Ankara, Turkey), filtered using a Buchner funnel with 0.45 μm filter paper, and stored at −21 °C until successive analyses.

All extractions and analyses were carried out in triplicate.

2.3.2 II stage: testing the effect of food-grade solvents on the recovery of bioactive compounds from CS. The best extraction conditions selected in stage I (method, time, and temperature) were used to test the impact of the polarity of the solvent on the recovery of bioactive compounds from CS. In this stage, six different mixtures of food-grade solvents ethanol/water including, 0 : 100 (CSW), 10 : 90 (CSE10), 20 : 80 (CSE20), 30 : 70 (CSE30), 50 : 50 (CSE50), and 80 : 20 (CSE80) (v/v) were evaluated. The extractions were carried out by mixing CS powder and solvents (1 : 10 raw material/solvent ratio) on a heating plate with constant stirring at 60 °C for 60 minutes. The produced extracts were centrifuged for 10 min at 20 °C and 6000 rpm. Then they were filtered with a Buchner funnel with 0.45 μm filter paper and finally stored at −21 °C until further analyses. Fig. 1 shows the two stages of the antioxidant compound extraction from CS.

2.4 Total phenolic content (TPC), total tannin content (TTC), and total flavonoid content (TFC)

TPC was quantified by a slightly modified method proposed by Alves et al.[34] About 0.3 mL of extract (dilution 1 : 10 to 1 : 50) was

Table 1 Bibliographic report of ethanol–water (1 : 1) use in the extraction of various by-products

By-product	Solvent	Extraction methods	References
Bergamot	Ethanol 50%	Maceration	Gattuso et al.[27]
Olive leaf	Ethanol 50%	Ultrasound	Şahin et al.[28]
Coffee	Ethanol 50%	Maceration	Machado et al.[9]
Pineapple skin	Ethanol 50%	Microwave	Nor et al.[29]
Chestnut shell	Ethanol 50%	Maceration	Vázquez et al.[30]
Aloe vera rind	Ethanol 50%	Ultrasound	Ioannou et al.[31]
Grape	Ethanol 50%	Maceration	Lafka et al.[32]
Passion fruit	Ethanol 50%	Maceration	Sai-Ut et al.[33]

combined with 2.5 mL of the Folin–Ciocalteu (1 : 10) reagent (Sigma Chemical Co., USA) and 2 mL of 7.5% (w/v) Na_2CO_3. The mixture was let to react for 15 minutes at 45 °C, then cooled to ambient temperature for about 30 min, and finally the absorbance at 765 nm was detected against a reagent blank (without the sample) using a PerkinElmer UV-Vis λ2 spectrophotometer (Waltham, Massachusetts, USA). The quantification was carried out using the calibration curve of gallic acid (2–10 mg L^{-1}) and the results are reported as mg of gallic acid equivalents per g of coffee silverskin dry weight (mg GAE per g d.w.).

TTC and TFC were quantified following Costa and colleagues.[35] For TTC, 0.5 mL of the suitably diluted CSE was added to a mixture with 2.5 mL of the Folin–Ciocalteu reagent (1 : 10), and 2 mL 7.5% (w/v) Na_2CO_3 was included after 2 minutes. After 2 hours of incubation in the dark and at room temperature, the mixture was submitted to spectrophotometric reading (λ 725 nm) against a blank and results are reported as mg of tannic acid equivalents on 100 g^{-1} of coffee silverskin dry weight (mg TAE per 100 g d.w.) using the calibration curve of tannic acid (1–20 mg L^{-1}).

Regarding TFC, 1 mL of extract was mixed with 4 mL of distilled water and 0.3 mL of 25% (w/v) $NaNO_2$ and incubated for 5 minutes in a test tube (10 mL). 0.3 mL of 10% (w/v) $AlCl_3$ was then added, and after 1 min at room temperature, 2 mL NaOH (4% w/v) was included in the mixture, which was finally made up to 10 mL with ultrapure water. The reaction was carried out in the dark and at ambient temperature for 10 minutes, then the absorbance of the samples was recorded at 510 nm *versus* a blank (without the sample) and the results are reported as mg of epicatechin equivalents on g of coffee silverskin dry weight (mg ECE per g d.w.).

2.5 Quantification of chlorogenic and caffeic acids

Individual concentrations of chlorogenic and caffeic acids in coffee silverskin extracts were quantified following Brzezińska *et al.*[36] with some modifications. 5 μL of suitably diluted samples were injected into a UHPLC PLATIN blue system (Knauer, Berlin, Germany), coupled with a Photo Diode Array Detector – PDA-1 PLATINblue (Knauer, Germany) and a C18 column (1.8 mm, 100 × 2 mm, Knauer blue orchid). Chlorogenic acid was detected at 330 nm while caffeic acid at 280 nm using formic acid 0.1% (A) and methanol (B) as elution solvents. The chromatographic separation was conducted at 30 °C under the conditions reported in Table 2. The quantification of chlorogenic and caffeic acids was carried out using external standards, and the results are reported as mg per g d.w.

2.6 Antioxidant activity of coffee silverskin (DPPH and ABTS assays)

DPPH and ABTS assays were used for antioxidant activity determination in CS extracts. A methanolic solution 6×10^{-5} M of DPPH (2,2-diphenyl-1-picrylhydrazyl) was prepared and stabilized at room temperature by dilution with methanol until an absorbance of 0.80 ± 0.05 at 515 nm was achieved. Subsequently, 4 μL of CSE adequately diluted was mixed with 2960 μL of a DPPH methanolic radical solution for 30 minutes in the

Table 2 Elution program used to detect chlorogenic and caffeic acids in coffee silverskin

Time (minutes)	Eluent A (%)	Eluent B (%)	Flow rate (mL min^{-1})
Initial	98.00	2.00	0.40
3.00	80.00	20.00	0.40
9.00	50.00	50.00	0.40
14.00	50.00	50.00	0.40
16.00	80.00	20.00	0.40
18.00	95.00	5.00	0.40
20.00	95.00	5.00	0.40

dark at 25 °C. The absorbance was read at 515 nm against methanol with a PerkinElmer UV-Vis λ2 spectrophotometer (Waltham, Massachusetts, USA).[14]

The radical solution of ABTS was generated by reacting equal volumes of 7 mM of ABTS (2,2-azino-bis 3-ethylbenzothiazolin 6-sulfonic acid) and 2.45 mM of potassium persulphate and leaving the resulting mixture to react at 25 °C in a dark environment. After a reaction time of 16 h, the radical solution $ABTS^+$ was then stabilized by dilution with ethanol to an absorbance of 0.70 ± 0.02, measured at 734 nm in a Perkin-Elmer UV-Vis k2 spectrometer (PerkinElmer Inc., Waltham, MA, USA) using ethanol as the blank. Thus, the antioxidant activity of the sample was established by the reaction between 40 μL of suitably diluted extracts and 2960 μL of $ABTS^+$ solution. After 6 min of reaction in the dark at ambient temperature, the absorbance was read at 734 nm, using ethanol as the blank.[37,38]

The results of the antioxidant assay (ABTS and DPPH) are reported as μM TE (Trolox Equivalents) per g of coffee silverskin, compared with a Trolox calibration curve (2–30 μM L^{-1}).

2.7 Microbiological analyses

Microbiological analyses were performed to evaluate the contamination of coffee silver skin extract. First, serial dilutions of CSE were prepared by mixing appropriate aliquots of CSE with a Ringer solution in ratios of 1 : 1, 1 : 10, 1 : 100, and 1 : 1000. Then the dilutions were inoculated in a selective microbiological culture medium consisting of Plate Count Agar (PCA) and Rose Bengal Chloramphenicol (DRBC) for detecting the total bacterial count (TBC) and the presence of yeasts and moulds (Y&M) respectively. The enumerations were carried out after incubating at 25 °C for 48 h in the case of TBC and 120 h in the case of Y&M.[18–38] The results were quantified as Log10 CFU mL^{-1} of CSE.

2.8 Statistical analysis

Statistical analyses were carried out using SPSS Software (Version 15.0, SPSS Inc., Chicago, IL, USA); Tukey's *post hoc* test at $p < 0.05$ was used for the multivariate and one-way analysis of variance. Results are expressed as means ± standard deviation of experimental data ($n = 3$).

3. Results and discussion

3.1 Chemical results of I stage: total phenolic content (TPC), total tannin content (TTC) and total flavonoid content (TFC), chlorogenic and caffeic acids and antioxidant activity

The extraction process of valuable compounds from vegetable matrices and their by-products is highly influenced by intrinsic (chemical composition, complexity of the matrix, preservation/pre-treatment of raw material) and extrinsic (solvent, temperature, time, method of extraction) factors.[39–42] Therefore, detecting suitable extraction variables represents an important challenge in the experimental phase.

The results reported in Table 3 suggest that TPC, TFC TTC, and antioxidant activity (ABTS and DPPH) assays of the extracts achieved by maceration (CSE1–6) were significantly ($p < 0.01$) higher than those detected in the extracts obtained by ultrasound-assisted extraction (CSE7–12).

The experimental data in Table 3 show that although the ultrasound-assisted extraction technique is considered efficient to recover valuable compounds from many by-products, its efficiency in the CS extraction was significantly lower than that of maceration following previous studies on the recovery of bioactive compounds from the by-products of bergamot,[27] apples,[43] pears,[44] and grapes.[45]

The drastic loss of antioxidant compounds during the ultrasound-assisted extraction process can be attributed to the synergistic action of wave frequency, power density, chemical structure of target compounds, time, temperature, and solvent, which collectively determined cavitation dynamics and the corresponding reaction pathways.[45,46] Beyond 40 kHz, the acoustic field favored the formation of a high cavitation microbubble number, whose collapses, less energetic but more frequent, triggered a regime of transient or "unstable" cavitation, dominated by sonochemical rather than thermomechanical effects.[46] These micro-collapses resulted in random microenvironments with extremely high temperatures and pressures (up to about 5000 K and 1000 atm inside the bubbles), which ignited the sonolysis of water and led to the formation of reactive oxygen species (ROS), mainly hydroxyl radicals ($^{\bullet}$OH), hydrogen radicals ($^{\bullet}$H) and hydrogen peroxide (H_2O_2).[46–48] On increasing the power density, the frequency and intensity of collapses led to greater radical production up to an equilibrium level, beyond which coalescence and acoustic shielding reduced their overall effectiveness.[49]

Several studies demonstrated that extended sonication also reduced the phenolic yield, despite the constant temperature, due to cumulative radical stress.[50,51] In the extracts of *Corchorus olitorius* leaves, the total phenolic content increased up to approximately 40 minutes of treatment and then gradually decreased to a significant loss after 90 minutes, due to cumulative exposure to radicals and cavitation stress.[50] Analogous trends were also found in *Sorbus intermedia* berries, beyond 60 minutes of UAE, resulting in measurable degradation of flavonoids and phenolic acids, despite the bulk temperature being kept constant, as a consequence of continuous radical generation.[51] These findings were also consistent with the mechanistic analyses of Shen *et al.*[52] and Zhu *et al.*,[53] who indicated that prolonged exposure to ultrasound played a predominant role in radical accumulation kinetics, promoting secondary oxidation and polymerization reactions even under controlled thermal conditions.

Finally, the solvent also played a crucial role in the UAE extraction process and in assisting the degradation of bioactive compounds. In fact, in hydroalcoholic solvents, comparable to those used in this study, it was found that the alcoholic component was actively involved in the propagation of radical chains, acting as a hydrogen donor and forming carbon-centred intermediates that intensified phenoxy couplings and the cleavage of aromatic rings.[54–56]

Related effects were also observed in various vegetable matrices and agri-food by-products, such as apple pomace,[57] pomegranate[58] and grape,[45] where the extraction yield and

Table 3 Chemical characterization of coffee silverskin[a]

Extraction variables				Parameters				
Extraction method	Temperature (°C)	Time (min)	Sample name	TPC (mg GAE per g)	TTC (mg TAE per 100 g)	TFC (mg ECE per g)	DPPH (μM TE per g)	ABTS (μM TE per g)
Maceration	40	30	CSE1	5.15 ± 0.19^{cd}	3.65 ± 0.01^{cde}	1.07 ± 0.03^{b}	16.5 ± 0.79^{bc}	66.92 ± 3.15^{abc}
		60	CSE2	6.56 ± 0.38^{c}	3.22 ± 0.99^{e}	1.01 ± 0.02^{bc}	16.64 ± 0.78^{bc}	69.95 ± 5.37^{ab}
		120	CSE3	5.46 ± 0.23^{cd}	4.90 ± 0.19^{b}	1.25 ± 0.08^{a}	15.91 ± 0.66^{bc}	67.42 ± 5.91^{abc}
	60	30	CSE4	15.86 ± 0.25^{a}	4.95 ± 0.24^{ab}	1.11 ± 0.00^{b}	19.97 ± 1.31^{ab}	69.46 ± 0.49^{ab}
		60	CSE5	16.79 ± 0.25^{a}	5.55 ± 0.83^{a}	1.31 ± 0.01^{a}	23.35 ± 0.04^{a}	79.63 ± 4.12^{a}
		120	CSE6	11.67 ± 0.42^{b}	2.52 ± 0.01^{f}	1.01 ± 0.07^{bc}	18.36 ± 2.83^{bc}	70.72 ± 1.36^{ab}
Ultrasound	40	30	CSE7	5.64 ± 0.05^{cd}	4.05 ± 0.12^{cd}	0.92 ± 0.01^{cd}	14.67 ± 0.75^{c}	57.82 ± 1.89^{bc}
		60	CSE8	5.23 ± 0.64^{d}	3.48 ± 0.22^{de}	0.70 ± 0.00^{ef}	16.72 ± 2.01^{bc}	63.36 ± 3.22^{abc}
		120	CSE9	5.93 ± 0.20^{cd}	4.14 ± 0.23^{c}	0.84 ± 0.04^{d}	15.41 ± 0.38^{bc}	59.92 ± 2.05^{bc}
	60	30	CSE10	4.04 ± 0.19^{ef}	2.20 ± 0.26^{f}	0.81 ± 0.02^{e}	19.61 ± 0.78^{bc}	67.76 ± 7.51^{abc}
		60	CSE11	4.05 ± 0.08^{ef}	2.46 ± 0.11^{f}	0.64 ± 0.02^{f}	17.19 ± 1.76^{bc}	59.92 ± 1.42^{bc}
		120	CSE12	3.87 ± 0.05^{f}	2.45 ± 0.12^{f}	0.84 ± 0.01^{d}	18.65 ± 0.73^{abc}	53.66 ± 3.21^{c}
Sign.				**	**	**	**	*

[a] Extraction solvent: hydroalcoholic mixture of ethanol : water (50 : 50). Significance at $p < 0.01$; * significance at $p < 0.05$. Different letters in the column are significantly different based on Tukey's *post hoc* test.

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stability of phenolic molecules were closely linked to the interaction of ultrasonic irradiation duration, acoustic power, and the solvent used. Indeed, it was noted that increasing the ultrasonic intensity or prolonging the treatment beyond optimal conditions frequently led to a significant reduction in total phenolic content and antioxidant capacity, likely attributable to secondary oxidation processes, radical recombination, and polymerization induced by unstable cavitation.[57,58]

In contrast, extraction by maceration preserved the molecular integrity of the bioactive compounds, promoting their gradual diffusion into the solvent under mild mechanical conditions, thus limiting oxidative stress and avoiding cavitation-induced degradation. The higher concentration of target compounds in CS extracts obtained by maceration was made possible through mass transfer, controlled by diffusion and selective solubilization of target compounds.[54,59] Furthermore, the close interaction between the solvent and the matrix probably facilitated the release of polyphenols bound to the fibrous and melanoidin structures of CS, preserving their stability without radical oxidation.[50,60]

Besides the extraction method, the tests conducted in stage I also hinted that temperature and time play a key role in the efficiency of the bioactive substance's recovery process. The increase from 40 °C to 60 °C significantly improved the

extraction efficiency, assisted by the increase in extraction time from 30 to 60 minutes (Table 3). In contrast, long extraction times from 60 to 120 minutes adversely affected the extraction efficiency in all samples, due to the oxidation and hydrolysis phenomena of antioxidant compounds, as already reported by Garcia-Salas et al.,[61] Carrera et al.,[62] and Baiano et al.[63]

Regarding the quantification of chlorogenic and caffeic acids, the principal phenolic acids in coffee silverskin are reported in Fig. 2. The extraction by maceration denoted a significant ($p < 0.01$) higher recovery efficiency in CS2 (1.25 mg per g caffeic acid and 2.08 mg per g chlorogenic acid) and CS5 samples (1.39 mg per g caffeic acid and 2.14 mg per g chlorogenic acid). In contrast, all other extractions showed a significant decrease consistently with the other detected parameters. Chlorogenic acid and caffeic acid are often more effectively extracted by maceration than ultrasound for several chemical and mechanical reasons linked to the nature of the molecules themselves and the structure of the raw material. In maceration extraction, the slow diffusion of chlorogenic and caffeic acid in the solvent preserves their integrity while the ultrasound induced cavitation generates overheating and a physical force responsible for causing substantial degradation, as just reported by Guglielmetti et al.[64]

The multivariate analysis (Table 4) evidenced the significant effect of extraction variables investigated on the recovery of bioactive compounds. Only the time of extraction (and its single combination with the other variables) did not affect the antioxidant capacity of the samples. The overall results obtained in stage I suggested that the optimal conditions for the maximum recovery of bioactive compounds with high antioxidant activity from CS were a temperature-controlled maceration extraction at 60 °C for 60 minutes.

3.2 Chemical results of II stage: total phenolic content (TPC), total tannin content (TTC) and total flavonoid content (TFC), chlorogenic and caffeic acids, and antioxidant activity

Among the extraction variables, the extraction solvent plays a key role in the successful recovery of precious bioactive compounds from food by-products. Scientific research is constantly searching for new extraction solvents that have characteristics suitable to meet the high extraction yields
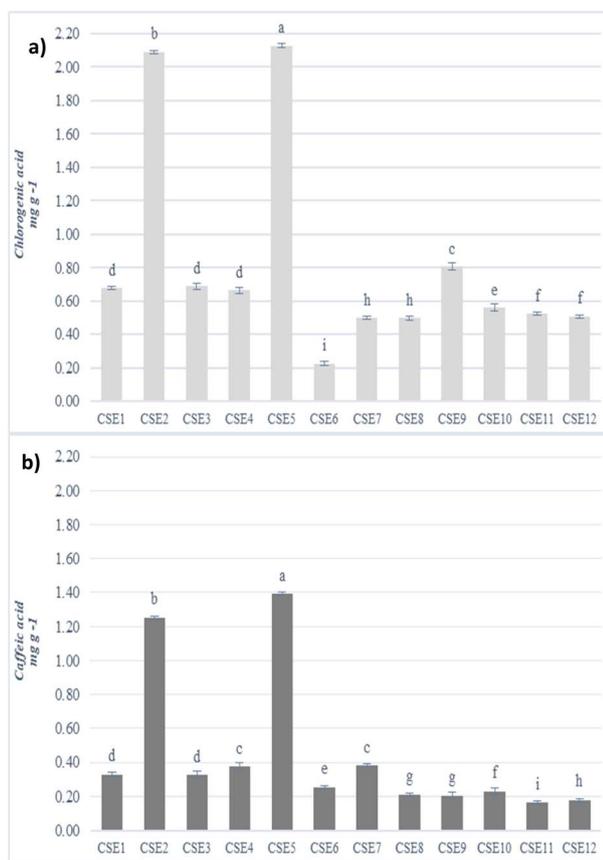

Fig. 2 Quantification of chlorogenic acid (a) and caffeic acid (b) in CS. Different letters are significantly different as assessed by Tukey's *post hoc* test ($p < 0.05$).

Table 4 Multivariate statistical analysis of different extraction methods (*M*), temperature (*T*) and time (*t*) on principal qualitative parameters[a]

	TPC	TTC	TFC	ABTS	DPPH	Caffeic acid	Chlorogenic acid
M	**	**	**	**	*	**	**
T	**	**	**	n.s.	**	**	**
T	**	*	*	n.s.	n.s	**	**
M × *T*	**	*	**	n.s.	n.s.	**	**
M × *t*	**	**	**	n.s.	n.s.	**	**
T × *t*	**	*	*	n.s.	n.s.	**	**
M × *T* × *t*	**	**	**	*	*	**	**

[a] Abbreviations: n.s., not significant; **, *, see Table 3.

required and at the same time with a low or no environmental impact in terms of pollution. Regarding food by-products, new emerging solvents such as ionic and eutectic solvents have been proposed as substitutes for conventional solvents such as acetone, benzene, xylene, isopropanol, methanol, and others with negative impacts on the environment.[65,66] Although in the experimental phase, the characteristics of these emerging solvents have led to promising results regarding the bioactive compound yields, some critical issues are still reported regarding practical feasibility and economic aspects linked to their use.[52,53] In addition, for them to be used for the recovery of bioactive compounds intended for food production, they must be composed of GRAS (Generally Recognized as Safe) components.[52]

The second part of this study centered on the use of food grade solvents which today represent a green and low-cost winning choice for the recuperation of bioactive compounds starting with food by-products: ethanol and water. These solvents have already been recognized as GRAS and meet environmental requirements as they are obtained from renewable and biodegradable sources.[66–68]

These food grade and green solvents are widely used for the recovery of bioactive compounds starting with different food by-products[27–33] and a mixture of them can assist in achieving the recovery efficiency of bioactive compounds, with strong antioxidant activity, from different by-products.[65–68]

The observed results in total phenolic, flavonoid, and tannin contents (Fig. 3) suggested that despite the promising extraction yields obtained in stage I using a hydroalcoholic mixture (ethanol : water 50 : 50) (Table 3), the decrement of ethanol percentage in the mixture ranging from 20% to 30% (CS20 and CS30 samples) involved a significant ($p < 0.01$) increase of bioactive compounds (of 55%). The mixture with a higher ethanol concentration ranging from 50% to 80% (CS50 and CS80 samples) led to a drastic reduction of these valuable compounds (Fig. 3).

The highest TPC was recorded in the sample CSE30 with values of 44.15 mg GAE per g, followed by CSE20 with values of 39.37 mg GAE per g. However, experimental data for TFC and TTC showed that the sample CS50 had a significantly higher ($p < 0.01$) content of these compounds than the sample CSE20 while the value recorded for the sample CSE30 remained higher than that of others. These different recovery trends of the analyzed compounds were related to the complex and varied stereochemistry of the molecules as well as the influence of the solvent in assisting the recovery of different target compounds present in CS such as caffeic acid, chlorogenic acids, quercetin and epicatechin.[69,70] The results of this study showed that hydroalcoholic solutions with 20–50% ethanol (CSE20, CSE30, and CSE50) were particularly effective in extracting the phenolic compounds present in the CS, favoring a more complete recovery of antioxidants than the use of ethanol or water alone or in the mixture at 10% and 80% (CSW, CSE10, and CSE80). Specifically, the hydroalcoholic mixture consisting of ethanol–water (30 : 70) (CSE30) combined with the maceration method for 60 minutes at 60 °C resulted in an appreciable change in the rate of diffusion and dissolution of the compounds in the solvent, resulting in a higher yield of the compounds detected compared to other extraction methods that are more expensive and difficult to apply such as solid-state fermentation with *Penicillium purpurogenum*,[9] and supercritical[39] and ultrasound extraction.[41]

This extraction pattern revealed a biphasic behavior, in which efficiency increased up to 30% ethanol before declining at higher concentrations. These extraction trends (Fig. 3) were attributable both to the polarity of the solvent and to a set of physicochemical factors modulating solvent–matrix interactions. At low ethanol fractions (≤10%), the excessive polarity of the solvent limited the solvation of the non-polar aromatic portions and reduced the permeability of the cell wall.[71] At moderate concentrations (20–30%), however, the lower surface
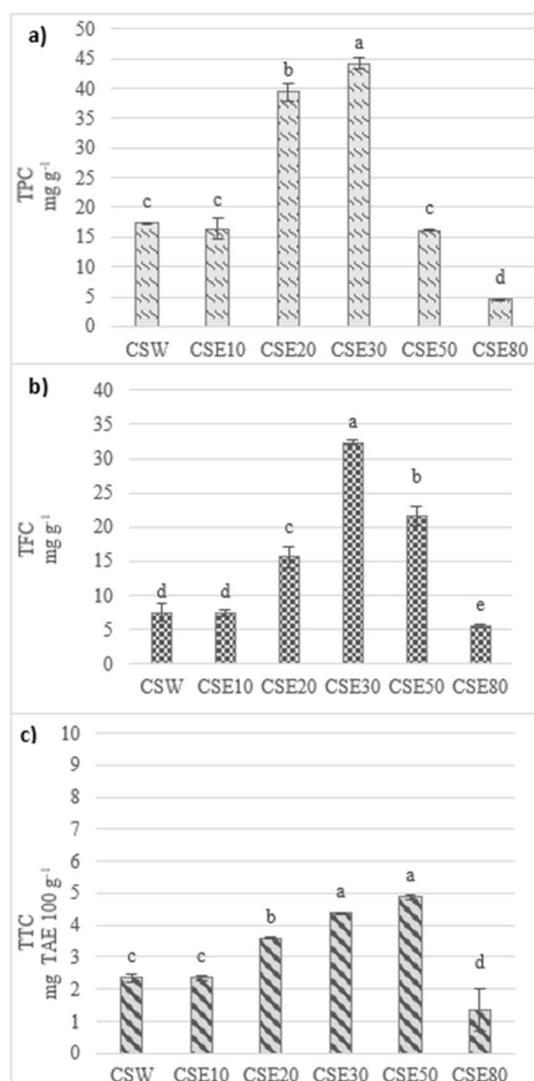

Fig. 3 Quantification of Total Phenolic Content (TPC) (a), Total Flavonoid Content (TFC) (b), and Total Tannin Content (TTC) (c). Different letters are significantly different based on Tukey's *post hoc* test. Abbreviations: CSW (H$_2$O solvent), CSE20 (EtOH 10% solvent), CSE30 (EtOH 30% solvent), CSE50 (EtOH 50% solvent), CSE80 (EtOH 80% solvent).

tension and viscosity of the solvent favoured capillary diffusion, swelling of the vegetable matrix and solvation of amphiphilic compounds, placing it in an optimal polarity range that maximised the mass transfer and solubilisation of bioactive compounds.[54,72,73]

In contrast, once the ethanol fraction exceeded 50%, extraction efficiency decreased dramatically. Besides polarity, this was also due to structural effects in the solvent system caused by reduced water activity and increased viscosity, along with the formation of ethanol clusters linked by hydrogen bonds that altered the solvent–solute H bond network, compromising the hydration of the matrix and reducing its porosity.[56,74] These alterations hindered the effective diffusion of phenolic compounds, resulting in a significant loss of efficiency.[54,75] Similar behaviours were also reported in different food matrices, including olive leaves,[59] apple pomace[57] and citrus[73] or coffee[60] by-products confirming that the extraction efficiency of phenolic compounds depends on a dynamic balance between polarity, viscosity and solvent–matrix affinity.

The influence of the extraction solvent on the main phenolic compounds present in CS was also clearer from the quantification of chlorogenic and caffeic acids performed using the UHPLC system (Fig. 4). The chlorogenic acid and caffeic acid recovery efficiency was directly proportional to the increase in ethanol concentration up to 30% and inversely proportional from 50% to 80% (Fig. 4).Consistent with other studies focusing on the recovery of phenolic compounds from vegetable matrices, moderate concentrations of ethanol allow the creation of an ideal extraction environment, by promoting the equilibrium of the solvent polarity and consequently optimizing the rate of diffusion of the compounds in it. The peculiar chemical structure of phenolic compounds including an aromatic ring (non-polar) linked to one or more hydroxyl groups (polar) determines the change in the polarity of the compounds themselves as already reported by Galanakis et al.[76] The affinity of the extraction solvent (intermolecular forces that occur between phenolic compounds and the solvents) to the stereochemistry (polar and non-polar groups) of phenols leads to a higher recovery efficiency. Hydroalcoholic solutions allow both polar and apolar compounds to be extracted efficiently,

thanks to the balance between water (polar) and ethanol (less polar);[67] the aqueous fraction allowed the solubility of the polar groups present within the molecules while ethanol eased the extraction of the less polar groups.

The factors described above were decisive in the extraction carried out with hydroalcoholic mixtures of 50% ethanol (CSE50) where it was noted that, despite maintenance of high values of caffeic acid, the chlorogenic acid values differed significantly ($p < 0.01$) from samples obtained with hydroalcoholic mixtures composed of 20% and 30% ethanol, samples CSE20 and CSE30, respectively. Vimercati et al.[4] also found a high potential for the extraction of bioactive compounds from coffee by-products in water–ethanol mixtures compared with acetone and isopropanol. They concluded that the differences in extraction yields were mainly due to the polarity relative to the solvents, which for ethanol and water were 0.654 and 1.00 while for acetone and isopropanol it was 0.355 and 0.390, respectively. The data obtained suggested that among the mixtures tested in this study, ethanol–water in the ratio 30 : 70 (CS30) allowed us to get a hydroalcoholic solution with a polarity such as to predispose the recovery of valuable bioactive compounds, of which chlorogenic acid and caffeic acid were the most predominant. Similar to the results obtained for TPC (Fig. 3), the qualification of chlorogenic and caffeic acids results suggest that the optimization of the extraction efficiency was found for the CS30 sample with values of 3.34 mg g^{-1} and 1.37 mg g^{-1} for chlorogenic and caffeic acid respectively. These results were higher than those obtained using another non-food grade solvent such as methanol, methanol/water mixtures (50 : 50),[77] and 60% isopropanol.[78]

The ABTS and DPPH assays were used to obtain more accurate and representative results for the overall antioxidant efficacy of the CS extract. DPPH mainly measures electron transfer whereas ABTS measures electron and hydrogen atom transfer. Therefore, the combined use of these assays allows for the covering of different types of antioxidant mechanisms that are useful for detecting a wider range of antioxidants, including hydrophilic and lipophilic molecules. Increasing levels of antioxidant activity for DPPH and ABTS were found in all extracts obtained using a hydroalcoholic mixture from 10% to 30% ethanol with the ABTS assay being more efficient than the DPPH assay (Table 5). Polyphenols, phenolic acids, and melanoidins
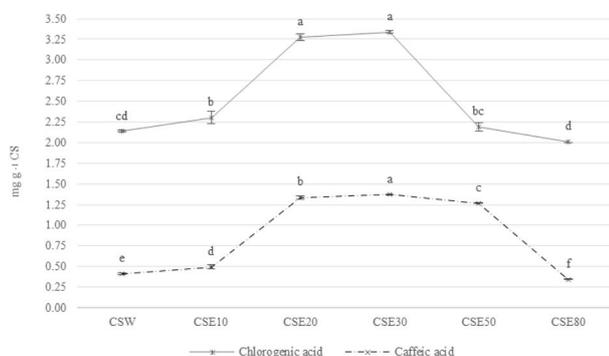

Fig. 4 Quantification of chlorogenic acid and caffeic acid. Different letters are significantly different based on Tukey's *post hoc* test. The sample name abbreviations are reported in Fig. 3.

Table 5 Results of antioxidant activity assays (DPPH and ABTS) for different CS extract samples[a]

Samples	DPPH (μM TE per g)	ABTS (μM TE per g)
CSW	18.31 ± 1.95[c]	71.87 ± 9.89[c]
CSE10	27.47 ± 1.51[b]	92.12 ± 0.52[b]
CSE20	26.11 ± 0.62[b]	99.79 ± 0.49[a]
CSE30	33.62 ± 1.49[a]	98.13 ± 2.99[a]
CSE50	25.03 ± 0.13[b]	78.23 ± 3.10[c]
CSE80	26.92 ± 0.70[b]	42.59 ± 0.11[d]
Sign.	**	**

[a] Abbreviations: ns, not significant; ** significance at $p < 0.01$; * significance at $p < 0.05$ by Tukey's *post hoc* test. The sample name abbreviations are reported in Fig. 3.

are the most reactive compounds in coffee by-products, being particularly efficient in eliminating the free radical ABTS, thanks to their chemical structure that promotes the stabilization of the radicals.[79] In particular, the combination of hydroalcoholic solvents allows for improved recovery of these compounds, since water and ethanol can optimize the extraction of both polar and semi-polar molecules. The experimental data agreed with previous results obtained by Ballesteros *et al.*[80] whereby it was shown that the antioxidant compounds in CS are more soluble in less polar organic solvents of water such as hydroalcoholic mixtures. Indeed, by observing the experimental data it is possible to see that the antioxidant activity is less expressed after extraction with high concentrations of water or ethanol. The experimental data obtained in this study denoted that the CS extract possessed antioxidant activity comparable to a lot of fresh fruits[81] and higher than that of other by-products[82] of the food industry already recognized as a source of antioxidant compounds. This evidence suggests that the CS extract could be effectively used as a natural antioxidant or as an ingredient for the formulation of high-value-added products, following the example of other antioxidant-rich by-products such as olive and citrus by-products already employed in food,[83] cosmetic,[84] and zootechnical applications.[85,86]

Regarding the microbiological analysis in both stage I and stage II, no evidence of microbial presence was found (data not shown): it is plausible that the high temperature of the roasting process, the low moisture content of CS (less than 10%),[6] the storage conditions of the raw material[16] and the extraction conditions allow inhibition of microbial growth.

4. Conclusions and recommendations

This study provided a comprehensive assessment of an eco-friendly extraction approach for recovering bioactive compounds from coffee silverskin, with the aim of promoting its efficient revalorization in view of a rapid transition to a circular economy model of supply chains.

Among the approaches tested, conventional solid–liquid extraction, supported by green solvents and a suitable binomial time–temperature, proved to be most effective compared to techniques considered innovative, such as ultrasound-assisted extraction. It ensured high recovery of bioactive compounds, among which chlorogenic acid and caffeic acid were the most prominent. This evidence confirmed the relevance of the "green" approach as proposed in the present study, based on green food grade solvents, moderate operating conditions and simple process parameters, guaranteeing an optimal balance between extraction performance, safety and eco-sustainability.

In practical terms, the proposed approach appears to be a scalable model that can be replicated in both small-scale artisanal and large-scale industrial production settings, due to its low solvent and tool costs, its simple application, and its coherence with the principles of circular economy. Nevertheless, its full industrial-scale applicability requires further investigation into the raw material's standardisation,

influenced by the botanical origin, roasting degree, and storage conditions, as well as energy and solvent balance management, aspects determining the process's economic and environmental sustainability.

In the future, additional studies should focus on a circular and integrated approach aimed at the synergistic recovery of the coffee silverskin's different fractions (phenolic, fibrous, and lipidic) and their use in several production chains according to co-processing or cascade valorization principles. Within this framework, maceration proves as a well-established, sustainable, and versatile extraction technology, capable of providing a basis for the development of functional ingredients and natural antioxidants, applicable in the food, cosmetic, and zootechnical sectors, while outlining a replicable operating model consistent with the principles of circular economy.

Author contributions

Conceptualization, A. P. (Amalia Piscopo), V. C. (Valerio Chinè), and M. P. (Marco Poiana); methodology, M. A. B. (Miriam Arianna Boninsegna) and T. T. (Tiziana Taglieri); validation, M. A. B., A. D. B. (Alessandra De Bruno) and A. P.; formal analysis, M. A. B. and T. T.; investigation, M. A. B. and A. D. B.; data curation, M. A. B., A. D. B. and A. P.; writing—original draft preparation M. A. B. and A. D. B.; writing—review and editing, M. A. B. and A. P.; supervision, A. P.; V. C. and M. P., project administration, V. C. and M. P.

Conflicts of interest

There are no conflicts to declare.

Data availability

All the data generated or analyzed during this study are included in the manuscript.

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