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Centrifugeless dispersive liquid-liquid microextraction based on salting-out phenomenon as an efficient method for determination of phenolic compounds in environmental samples

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Abstract A new centrifugeless dispersive liquid-liquid microextraction (DLLME) method was applied for the convenient extraction of some phenolic compounds from environmental samples. After dispersing the extracting solvent into the sample solution (10.0 mL), the mixture was passed through a small column filled with 5 g sodium chloride. As a result, phase separation was achieved via the salting-out phenomenon, and the extracting solvent was suspended on top of the sample solution. Using a low-toxic and solidifiable extracting solvent (1-dodecanol), after immersing the column into an ice bath, the extracting solvent was solidified, collected easily, and injected into an HPLC-UV instrument. The overall extraction time was 7 min, consumption of the extracting solvent was efficiently reduced to 50 μL, and the centrifugation step was simply eliminated, which made the automation of the procedure easier than the normal DLLME technique. A series of parameters influencing the extraction were investigated systematically. The optimal experimental conditions were found to be 50 μL of 1-dodecanol as the extracting solvent, a flow rate of 2.0 mL min^{-1}, and a pH value of 4.0 for the sample solution. Under these conditions, the method provided a good linearity in the range of 0.5–800 ng mL^{-1}, low limits of detection (0.1–0.3 ng mL^{-1}), good extraction repeatabilities (RSDs below 9.1%, $n = 5$), and enrichment factors of 100–160.

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Introduction

Despite the unprecedented growth in the analytical techniques over the last few decades, one or more pretreatment steps are necessary before a chemical analysis. These are referred to as sample preparation, whose goal is cleanup, sample enrichment, and signal enhancement. Although liquid-liquid extraction still plays a key role in sample preparation, it suffers from consumption of a large volume of toxic solvents, and it is time-consuming and boring. Other extraction techniques include solid-phase extraction (SPE) [1], flow injection extraction (FIE) [2], supercritical fluid extraction (SFE) [3], and microwave-assisted extraction [4]. SPE consumes a less solvent than LLE but it is expensive and time-consuming. Although the solvent used in FIE is less than that in LLE, consumption of organic solvents in the FIE method is about several hundred microliters per analysis. SFE is very complicated, and it requires some equipment such as a high-pressure delivery system. On the other hand, this method consumes a high-purity carbon dioxide and it is expensive.]Microwave-assisted extraction also requires a dedicated equipment that is safe to operate, and optimization of its exploitation can also be complicated. Therefore, in the recent years, significant investigations have been performed to introduce economic, efficient, environmentally friendly, and miniaturized extraction methods [5]. Miniaturization of the conventional LLE method has led to the introduction of different microextraction modes sub-divided under the term "liquid-phase microextraction" (LPME) [6–8]. Single-drop microextraction (SDME) was the first idea in this field. In what follows, hollow-fiber-protected two-phase microextraction (HF-LLME) [9], hollow-fiber-protected three-phase microextraction (HF-LLLME) [10], and electromembrane extraction (EME) [11–13] are described. In 2006, dispersive liquid-liquid microextraction (DLLME) was

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innovated as a fast two-phase microextraction [14, 15]. Despite the significant advantages of the SDME, HF-LLME, and EME methods, they are time-consuming [16, 17]. On the other hand, DLLME provides an efficient extraction in less than 10 min. In this method, due to the increase in the surface area between the donor and acceptor phases, extraction speeds up. Since the introduction of this method, DLLME has been stunningly considered in the sample preparation field [18] and new versions of it have been innovated [19–23]. However, conventional DLLME consumes toxic and even carcinogenic organic solvents such as chlorinated solvents as the extraction solvent. Also it involves a centrifugation step for phase separation, and this is considered as a bottleneck in the automation of the technique [24]. In order to reduce the toxicity of DLLME, utilization of new solvents, typically ionic liquids (ILs), and expansion of the application scope using low-density organic polar solvents have been raised. Although solvent evaporation is efficiently reduced by utilization of ILs as the extracting agent, if GC is to be used as the final analyzer system after the extraction process, this lack of volatility dirties the GC system and even blocks the column [25]. Low-density solvents have fewer toxic elements. However, non-repeatability and difficulty to collect the small microdroplets floating on the sample solution are the main drawbacks of this approach. Introduction of lower-density solvents with proper melting points that would solidify at a low temperature was an appropriate development for reducing the mentioned disadvantages [26]. In this method, first an organic solvent is dispersed into the aqueous sample, and then a centrifugation step followed by solidifying the organic droplets in an ice bath is performed for phase separation. This new version improved some DLLME problems such as method toxicity and collection of the extractant agent after extraction. However, this method suffers from utilization of a centrifugation step for phase separation.

In this work, a simple, rapid, and centrifugeless dispersive liquid–liquid microextraction based on the counter-current salting-out phenomenon was applied to the extraction of some phenolic compounds including 2-nitrophenol, 2-chlorophenol, 4-chlorophenol, and 3-chlorophenol from the environmental samples. In this method, in the absence of the disperser solvent, using a 10-mL glass syringe, the extracting solvent is dispersed into the sample solution, and the mixture is then passed through a small column filled with 5 g of sodium chloride, used as a separating reagent. In this condition, the centrifugation step is simply eliminated and the phase separation is achieved via the salting-out phenomenon.

Chlorophenols are used for different purposes. For example, they are used as the intermediates in the production of plastics, dyes, and pharmaceuticals. Also chlorination of tap water leads to the generation of chlorophenols from phenols, which causes the unfavorable smell of water [27]. They can present significant health hazards due to their moderate bio-accumulation and high toxicity, which increase with the increment of chlorination. Hence, US Environmental Protection Agency (EPA) issued a list of 11 phenolic compounds considered as highly polluting materials, among which chlorophenols are the most toxic and carcinogenic ones [28].

Experimental

Reagents and chemicals

2-Nitrophenol (≥99.0%), 2-chlorophenol (≥99.0%), 4-chlorophenol (≥98.0%), and 3-chlorophenol (≥98.0%) were supplied from Merck (Darmstadt, Germany). In order to obtain a stock solution (1.0 mg mL^{-1}), a certain amount of each phenolic compound was dissolved in HPLC-grade methanol (Ameretat Shimi, Tehran, Iran). These solutions were protected from light, stored at 4 °C in a refrigerator, and re-prepared every 3 weeks. The organic extraction solvents used including 1-undecanol (≥97.5%), 1-dodecanol (≥98.0%), and n-tetradecane (≥99.0%) were obtained from Merck (Darmstadt, Germany). Ammonia (25%) was obtained from Merck-Schuchardt (Munich, Germany). HPLC-grade acetonitrile and water were obtained from Ameretat shimi (Tehran, Iran). Also analytical-grade H$_3$PO$_4$ (85%), NaCl (≥99.0%), NaOH (≥99.0%), and HCl (37%) were all purchased from Merck (Darmstadt, Germany). The other chemicals utilized were of analytical grade.

Sample preparation

The river water (Shahmirzad, Iran), tap water (Semnan University), and waste water (Semnan, Iran) samples were collected in amber glass containers and maintained in the dark at 4 °C until analysis. No filtration or further treatment was applied to any of the samples before extraction.

Instruments

The separation and detection of the target analytes were performed using a Knauer HPLC instrument (Berlin, Germany) equipped with a D-14163 degasser, a Rheodyne 7725i injection valve with a 20-μL injection loop, a K-1001 HPLC pump, and a K-2600 UV detector. The ChromGate software for the HPLC system (version 3.1) was employed to acquire and process the chromatographic data. The stationary-phase column, which was an ODS III C8 (5-μm particle diameter, 250 mm × 4.6 mm i.d.) was obtained from MZ Analysen technik (Mainz, Germany). A mixture of 0.05 M phosphate buffer (pH 4.0) and acetonitrile (62:38) at a flow rate of 1.0 mL min^{-1} was used as the mobile phase in the isocratic elution mode. The injection volumes were 20 μL for all the samples, and the detection was performed at a wavelength of

220 nm. A LAMBDA CZs.ro multi-flow peristaltic pump (LAMBDA, Switzerland) was used for the phase separation, and the pH values for the solutions were measured using a PHS-3BW model pH meter (Bell, Italy). The absorbance spectra of the analyte solutions were obtained on a Shimadzu UV-1650 PC spectrophotometer (Kyoto, Japan).

Procedure

A schematic diagram of the method is shown in Fig. 1. At first, 10.0 mL of the sample solution (pH = 4.0) was poured into a 15.0-mL screw cap glass test tube, to which 50 μL of 1-dodecanol was added as the solidifiable extracting solvent. The mixture was rapidly sucked into a 10-mL glass syringe and then injected into the tube (for 12 times) via a syringe needle. During each cycle, the solution became turbid, due to the dispersion of fine 1-dodecanol droplets into the aqueous bulk. A 10-mL glass syringe barrel was cleaned with deionized water, and then a filter was placed at the bottom of the barrel. Afterward, 5 g sodium chloride was poured into the barrel and compressed slightly with the syringe plunger. The mixture (aqueous sample and 1-dodecanol) was passed through the barrel (flow rate, 2.0 mL min^{-1}). Due to the salting-out effect, the fine droplets of the extracting solvent went up through the mixture, which were collected as a separate layer on top of the sample solution. After blocking the bottom of the barrel, it was immersed in an ice water bath for 2 min. The extracting solvent was solidified, carefully collected using a spatula, and transferred into a small vial, where it melted immediately. Finally, 25 μL of this solution was injected into the HPLC system for analysis.

Calculation of enrichment factor and extraction recovery

The enrichment factor (EF) for the target analytes was calculated according to the following equation:

$$EF = \frac{C_{a,\,final}}{C_{s,\,initial}} \tag{1}$$

where $C_{s,initial}$ is the initial analyte concentration in the sample (donor) solution, and $C_{a,final}$ is the final concentration of the analyte in the acceptor solution.

Also the percentage extraction recovery (ER%) for the TDLLME procedure was calculated according to the following equation:

$$ER = \frac{n_{a,\,final}}{n_{s,\,initial}} \times 100 = EF \times \frac{V_a}{V_s} \times 100 \tag{2}$$

where V_a is the acceptor solution volume, V_s is the sample volume, and $n_{a,final}$ and $n_{s,initial}$ are the number of moles of the analyte finally collected in the acceptor solution and number of moles of the analyte originally present in the sample, respectively.

Results and discussion

In order to determine the most favorable conditions for the method, the effects of different extraction parameters including the number of extractions (number of aspiration-dispersion cycles), type of extracting solvent, flow rate of sample solution, and volume of extracting solvent were studied in terms of the chromatographic peak areas of the analytes. All the optimization experiments were performed in triplicate.

Effect of pH

Sample pH plays a unique role to transfer the target analytes into the organic phase in many LPME methods. Because of the acid–base properties of phenolic compounds, the pH effect on their extraction is important. The addition of a strong acid to the sample solution can reduce dissolution of weak acids and allow their existence in the neutral molecular form. Hence, the effect of the pH of the sample solution on the extraction efficiency was studied by adjusting the solution pH in the range of 2.0–9.0. According to the results obtained (Fig. 2), the extraction efficiency was almost unchanged by changing the pH values in the range of 2.0–5.0, and it was reduced by increasing the pH value to over 5.0. The pK_a values for the chlorophenols studied were in the range of 7.23–9.41 [29]. Theoretically, a pH value of the donor phase of 5.23 (equal to pK_a −2) would be sufficiently acidic. In fact, decreasing the sample pH can reduce the dissolution of the studied phenols in the aqueous samples and allow their existence in the neutral molecular form. Thus, a pH value of 5.0 was selected as the optimum one for this parameter.

Effect of extraction solvent

Selection of an appropriate extracting solvent is critical for the liquid-phase microextraction methods since its physico-chemical properties govern the extraction efficiency, toxicity of the method, and compatibility of the method with the final analyzer instrument. The developed method is based on the consumption of a solidifiable organic solvent. Hence, the solvent used in this method should meet the following requirements: it must have a density lower than water, a melting point near or below the room temperature, a low solubility in water, a high extraction efficiency, a good chromatographic behavior, and a good stability. Only a few organic solvents fulfill the abovementioned requirements. Among them, 1-dodecanol (density, 0.8309 g mL^{-1}; melting point, 22–24 °C), 1-undecanol (density, 0.8298 g mL^{-1}; melting point, 13–15 °C), and n-tetradecane (density, 0.756 g mL^{-1}; melting point, 4–6 °C) are the most used ones. The phase separation was not well done when n-tetradecane was used. Therefore, 1-undecanol and 1-dodecanol were tested as the extraction solvent. The results obtained (Fig. 3) show that both solvents
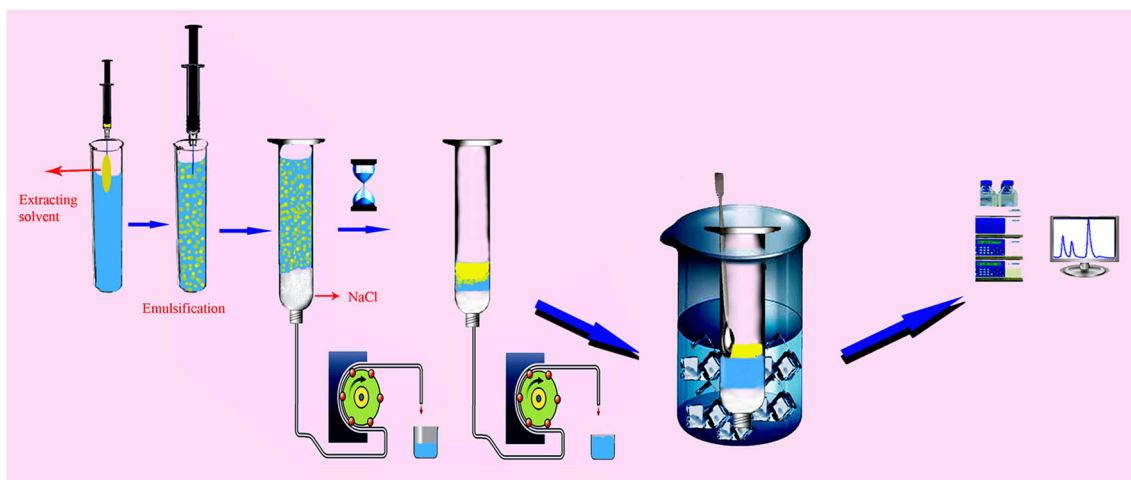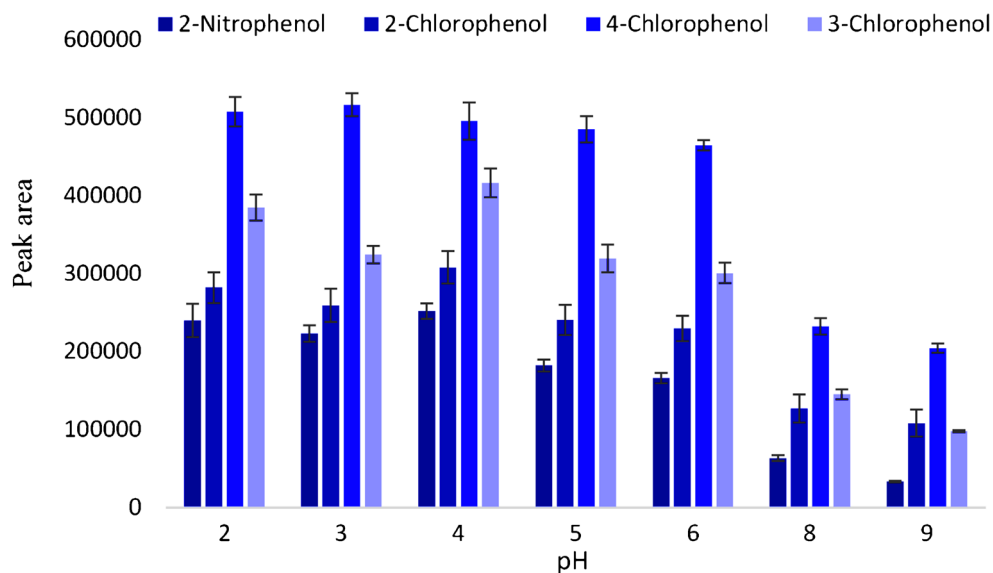
Fig. 1 Schematic diagram of centrifugeless dispersive liquid-liquid microextraction based on counter-current salting-out method

provide almost the same extraction efficiency but since it is easier to work with 1-dodecanol, this solvent was selected as the organic extraction solvent.

Volume of extracting solvent

In a microextraction method, the volume of the extracting solvent is generally selected as low as possible to achieve higher EFs and a lower toxicity for the environment. On the other hand, it should be sufficient for extraction of the maximum amount of analytes, handling the proposed microextraction method, and injection to the final analyzer instrument. The effect of the extractant volume on the extraction efficiency was also investigated. As shown in Fig. 4, the peak area decreases sharply when the extractant volume increases from 50 to 150 μL. It seems that the dilution effect is the main cause for this phenomenon. Therefore, in order to gain a high sensitivity, 50 μL of the extractant was chosen.

Number of extractions (number of aspiration-dispersion cycles)

The extraction numbers play an important role in obtaining the highest extraction efficiency within the least time period. To achieve the best performance, the number of air-agitation cycles was investigated in the range of 1–15. As it can be seen in Fig. 5, with increase in the number of air-agitation cycles, the analytical signals also increased up to the 10th cycle and then remained constant. Hence, to obtain a reasonable precision, 12 times of air-agitations were selected for further studies.

Flow rate of sample solution

The rate of flow of the sample solution through the barrel filled with NaCl affects the extraction recovery and the time of extraction process. It must be high enough to shorten the time reasonably and low enough to perform an effective

Fig. 2 Effect of pH of sample on extraction efficiency of proposed method. Experimental conditions: sample solution, 10.0 mL of 100 ng mL^{-1} of four phenols in distilled water; organic solvent, 1-dodecanol; volume of the organic solvent, 75 μL; 10 times of air-agitation cycles; flow rate of 2 mL min^{-1}. *Error bars* were obtained based on three replicates

Fig. 3 Effect of type of organic solvent on extraction efficiency of proposed method. Experimental conditions: sample solution (pH, 4.0), 10.0 mL of 100 ng mL^{-1} of four phenols in distilled water; 75 μL of each organic solvent; 10 times of air-agitation cycles; flow rate of 2 mL min^{-1}. *Error bars were obtained based on three replicates*
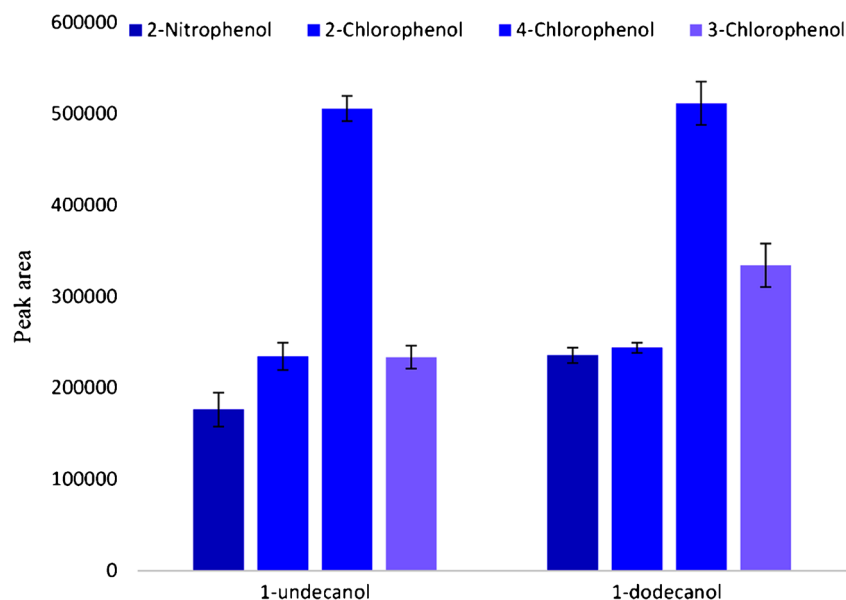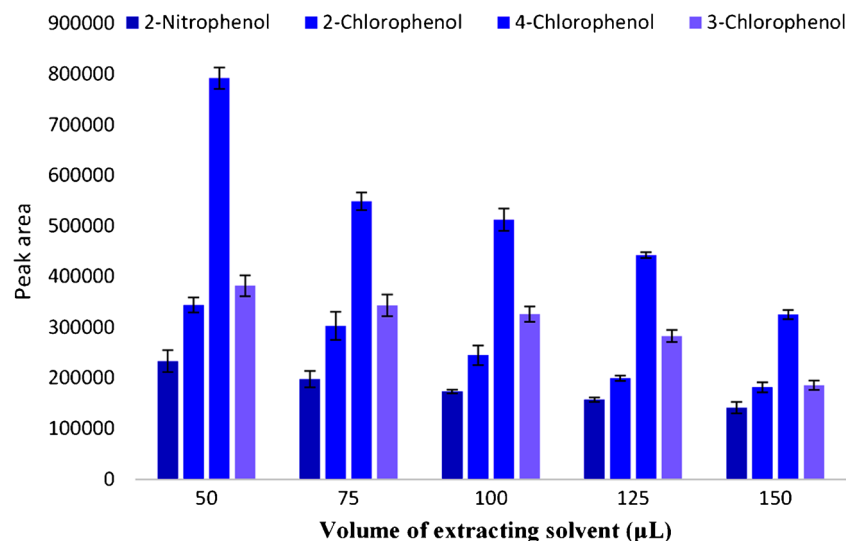

salting-out. The effect of this parameter was investigated in the range of 1–5 mL min^{-1} by loading 10 mL of the sample solution through the column with a peristaltic pump. As it can be seen in Fig. 6, when the flow rate is over 2.0 mL min^{-1}, the extraction efficiency is decreased. It seems that at a flow rate over 2.0 mL min^{-1}, the salting-out effect is incomplete. Hence, a flow rate of 2.0 mL min^{-1} was selected as the optimum flow rate of the sample solution.

Quality assurance/quality control (QA/QC)

In order to ensure an adequate level of quality assurance and a quality control of measurements, the developed method was utilized for the extraction and determination of the understudied phenolic compounds in some real water samples

such as tap, river, and waste water. In order to calibrate the method, eight standard solutions of the analytes were extracted via the extraction method and the final analysis was performed by HPLC-UV. The results obtained revealed that the curves obtained were linear in the range of 0.5–800 ng mL^{-1}, and they showed that the related coefficient of determinations were higher than 0.998. To determine the detection limit of the method for each analyte, initially, a blank sample was analyzed by the method. Then by analysis of the samples containing low concentrations of the understudied analytes and comparing the results obtained, the related LODs were assessed. The limit of detection (LOD) was determined based on a signal-to-noise ratio (S/N) of 3 ([30]. Also the limit of quantification (LOQ) was determined based on S/N = 10. On this basis, LOQs were in the range of 0.5–1.0 ng mL^{-1}, and the LOD values were in the range of 0.1–0.3 ng mL^{-1} for all the

Fig. 4 Effect of volume of organic solvent on extraction efficiency of developed method. Experimental conditions: sample solution (pH, 4.0), 10.0 mL of 100 ng mL^{-1} of four phenols in distilled water; organic solvent, 1-dodecanol; 10 times of air-agitation cycles; flow rate of 2 mL min^{-1}. *Error bars were obtained based on three replicates*

Fig. 5 Effect of number of air-agitation cycles on extraction efficiency of developed method. Experimental conditions: sample solution (pH, 4.0), 10.0 mL of 100 ng mL^{-1} of four phenols in distilled water; 50 μL of 1-dodecanol; flow rate of 2 mL min^{-1}. *Error bars* were obtained based on three replicates
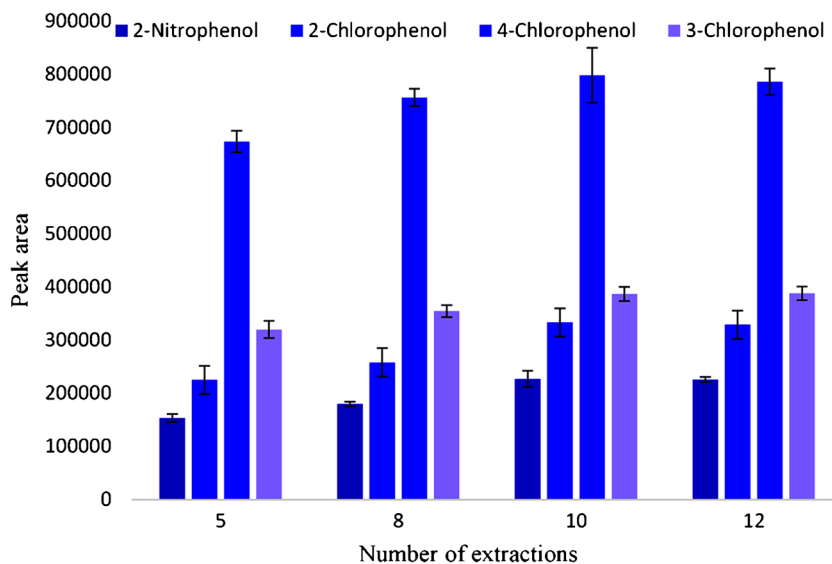
analytes. To evaluate the precision of the developed method, the repeatability of the peak areas obtained was investigated for five replicate extractions and the deionized water sample spiked at a 25-ng mL^{-1} level. This parameter was expressed as the relative standard deviation (RSD) and was provided for both the intra- and inter-day precisions. As it can be seen in Table 1, the relative standard deviations obtained were below 9.1%. To assess the extraction recovery and enrichment factor, the equations presented in "Calculation of enrichment factor and extraction recovery" section were applied. Based on the results obtained, the extraction recoveries were in the range of 39–64%, and enrichment factors in the range of 100–160 were achieved. To investigate the accuracy of the method, the relative recoveries obtained from analysis of real samples spiked with known amounts of the phenols under investigation at low, medium, and high concentration levels were calculated. The analyzed samples were spiked using 10.0, 100.0, and 500.0 ng mL^{-1} of the target analytes. The results obtained revealed that the concentrations of all the phenols present in

the tap and waste water samples were below the detection limit for the method. On the other hand, these results confirmed the existence of 2-nitrophenol, 4-chlorophenol, and 3-chlorophenol in the river water sample. The results obtained showed that the different matrices used for the river, tap, and waste water samples had no significant effect on the extraction efficiency, and obtaining high relative recoveries (from 95 to 105%) approved this fact (Table 2).

The chromatograms for the river water samples for non-spiked and spiking at the concentration level of 25 ng mL^{-1} for the analytes are shown in Fig. 7.

Comparison between the proposed method and other extraction methods

There are a number of methods that are used for the extraction of phenolic compounds from various samples. A comparison of the developed method with some of these methods is

Fig. 6 Effect of sample flow rate on extraction efficiency of developed method. Experimental conditions: sample solution (pH, 4.0), 10.0 mL of 100 ng mL^{-1} of four phenols in distilled water; 50 μL of 1-dodecanol; 12 times of air-agitation cycles. *Error bars* were obtained based on three replicates
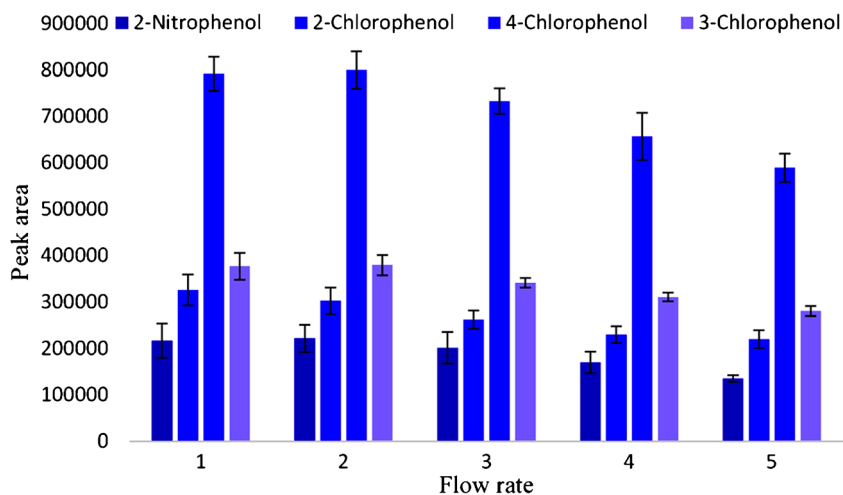
Table 1 Figures of merit for proposed method for target analytes

Analyte	LOD[a]	LDR[b]	R^{2c}	RSD%[d] ($n = 5$) Intra-day (inter-day)	ER%[e]	EF[f]
2-Nitrophenol	0.3	1.0–800	0.999	4.9 (6.8)	39	100
2-Chlorophenol	0.3	1.0–800	0.998	5.1 (7.6)	49	123
4-Chlorophenol	0.1	0.5–800	0.999	7.7 (9.1)	59	148
3-Chlorophenol	0.3	1.0–800	0.999	3.3 (5.4)	64	160

[a] Limit of detection (S/N = 3) (ng mL^{-1})

[b] Linear dynamic range (ng mL^{-1})

[c] Coefficient of determination

[d] Relative standard deviation ($n = 5$, $C = 25$ ng mL^{-1})

[e] Extraction recovery ($C = 25$ ng mL^{-1})

[f] Enrichment factor

provided in Table 3. The information given in this table reveals that after DLLME–GC-ECD and SA-DLLME–HPLC-UV, the proposed method is one of the fastest ones, and it provides a good enrichment factor and extraction efficiency with minimum consumption of organic solvents. Following the steps involved in the developed method and other tabulated methods shows that this method can be one of the simplest ones. The limits of detection for the method in determination of the understudied phenols are comparable with those in the other extraction methods. The proposed method also provides wide LDRs for the determination of analytes. On the other hand, this method uses low-toxic solvents, and there is no need for the centrifugation step, which makes the automation of the procedure easier than the other methods.

Conclusion

A comfortable and centrifugeless dispersive liquid-liquid microextraction was applied for the efficient extraction of some phenolic compounds from the river, tap, and waste water samples. In this method, by utilization of a simple approach, the centrifugation step in the DLLME method is eliminated,

Table 2 Analysis of real samples under optimal conditions

Sample	Initial[a]	2-Nitrophenol			2-Chlorophenol			4-Chlorophenol			3-Chlorophenol		
		N.d[b]			N.d			N.d			N.d		
Waste water	Added[c]	10.0	100.0	500.0	10.0	100.0	500.0	10.0	100.0	500.0	10.0	100.0	500.0
	Found[d]	9.8	103.3	539.9	10.2	99.6	519.0	9.4	94.6	481.1	10.3	104.8	502.2
	RR%[e]	98	103	108	102	100	104	94	95	96	103	105	100
	RSD% ($n = 3$)	5.7	4.5	5.1	6.4	5.0	7.7	7.1	6.7	4.1	6.9	7.7	3.9
	Initial	9.8			N.d			6.7			5.8		
River water	Added	10.0	100.0	500.0	10.0	100.0	500.0	10.0	100.0	500.0	10.0	100.0	500.0
	Found	9.7	102.8	533.3	10.4	99.2	496.8	10.6	96.0	501.0	9.9	95.8	489.7
	RR%	97	103	107	104	99	99	106	96	100	99	96	98
	RSD% ($n = 3$)	6.6	4.3	6.9	3.3	5.6	5.8	8.2	7.1	5.0	5.9	4.9	7.1
	Initial	N.d			N.d			N.d			N.d		
Tap water	Added	10.0	100.0	500.0	10.0	100.0	500.0	10.0	100.0	500.0	10.0	100.0	500.0
	Found	10.1	97.2	533.3	9.7	97.9	507.0	9.1	100.4	479.1	10.0	94.7	501.7
	RR%	101	97	104	97	98	101	91	100	96	100	95	100
	RSD% ($n = 3$)	7.6	7.0	6.8	4.5	2.7	8.4	4.9	6.3	7.2	4.9	7.2	4.7

[a] Concentration of analytes (ng mL^{-1})

[b] Not detected

[c] Spiked concentration (ng mL^{-1})

[d] Concentration of analytes (ng mL^{-1}) in the sample after spiking target analytes
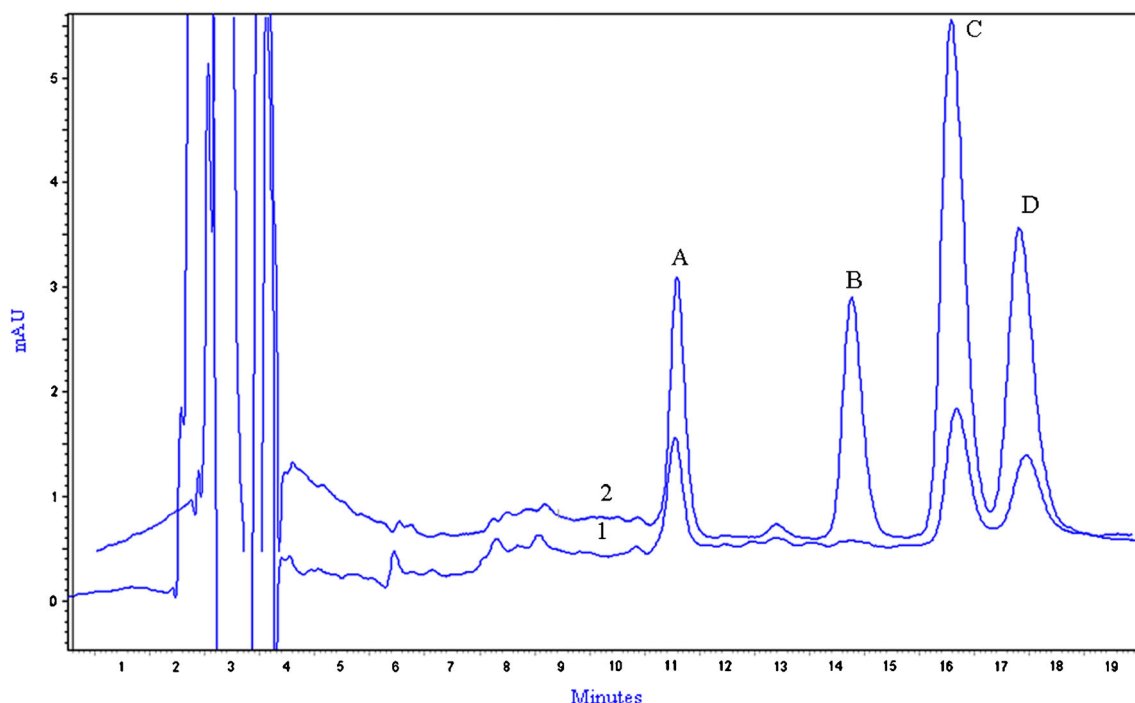
[e] Relative recovery

Fig. 7 Typical chromatograms obtained from river water samples under optimal conditions. (*1*) Non-spiked and (*2*) 25 ng mL^{-1} of each phenol spiked river water sample. (*A*) 2-nitrophenol. (*B*) 2-chlorophenol. (*C*) 4-chlorophenol. (*D*) 3-chlorophenol

and this makes the automation of this method easy. This method is based on the salting-out phenomenon, which makes the approach environmentally friendly. By performing this method, a minimum amount of toxic solvent was consumed, the

Table 3 Comparison between proposed method and other microextraction techniques for determination of target analytes

Analytical method	Analyte	LOD (ng mL^{-1})	LDR (ng mL^{-1})	EF	Analysis time (min)	Ref.
SPE-HPLC	2-Chlorophenol, 4-chlorophenol, 3-chlorophenol	0.1–0.4	1–200	<50	>40	[31]
SFE-LC-ECD	2-Chlorophenol, 4-chlorophenol, 3-chlorophenol	3.0	–	–	>90	[32]
SA-DLLME–HPLC-UV	2-Chlorophenol, 4-chlorophenol	0.1	0.2–200	187–353	6.0	[33]
CPE–HPLC-EC	2-Chlorophenol	3.0	5–200	13	20	[34]
DLLME–GC-ECD	2-Chlorophenol, 4-chlorophenol	0.5–2.0	1–400	287–628	2	[35]
SPME-MD–HPLC-DAD	2-Chlorophenol, 4-chlorophenol	1.6–1.9	1–200	–[a]	20–40	[36]
SDCME–HPLC-UV	2-Chlorophenol	0.1	2–500	–	15	[37]
IL-DLLME-PDA	2-Chlorophenol, 4-chlorophenol	1.2–1.4	10–200	34–37	>10	[38]
DLLME-SFO-HPLC-UV	2-Nitrophenol, 2-chlorophenol, 4-chlorophenol, 3-chlorophenol	0.1	0.5–800	100–160	7.0	This work

SA-DLLME surfactant-assisted dispersive liquid–liquid microextraction, *LC–ECD* liquid chromatography–electrochemical detection, *CPE* cloud point extraction, *SPME-MD* solid phase microextraction micellar desorption, *SDCME* single-drop coacervative microextraction, *IL-DLLME* in situ ionic liquid-dispersive liquid–liquid microextraction

[a] Not reported

overall extraction time was 7 min, and an efficient extraction was achieved.

Compliance with ethical standards

Conflict of interest The authors declare that they have no conflict of interest.

References

1. Junk GA, Richard JJ. Organics in water: solid phase extraction on a small scale. Anal Chem. 1988;60(5):451–4.
2. Karlberg B, Thelander S. Extraction based on the flow-injection principle. Anal Chim Acta. 1978;98(1):1–7.
3. Ndiomu DP, Simpson CF. Some applications of supercritical fluid extraction. Anal Chim Acta. 1988;213:237–43.
4. Ganzler K, Salgó A, Valkó K. Microwave extraction. J Chromatogr A. 1986;371:299–306.
5. Risticevic S, Niri VH, Vuckovic D, Pawliszyn J. Recent developments in solid-phase microextraction. Anal Bioanal Chem. 2009;393(3):781–95.
6. He Y, Lee HK. Liquid-phase microextraction in a single drop of organic solvent by using a conventional microsyringe. Anal Chem. 1997;69(22):4634–40.
7. Jeannot MA, Cantwell FF. Solvent microextraction into a single drop. Anal Chem. 1996;68(13):2236–40.
8. Nerín C, Salafranca J, Aznar M, Batlle R. Critical review on recent developments in solventless techniques for extraction of analytes. Anal Bioanal Chem. 2008;393(3):809–33.
9. Rasmussen KE, Pedersen-Bjergaard S. Developments in hollow fibre-based, liquid-phase microextraction. Trends Anal Chem. 2004;23(1):1–10.
10. Pedersen-Bjergaard S, Rasmussen KE. Liquid–liquid–liquid microextraction for sample preparation of biological fluids prior to capillary electrophoresis. Anal Chem. 1999;71(14):2650–6.
11. Pedersen-Bjergaard S, Rasmussen KE. Electrokinetic migration across artificial liquid membranes: new concept for rapid sample preparation of biological fluids. J Chromatogr A. 2006;1109(2):183–90.
12. Bazregar M, Rajabi M, Yamini Y, Asghari A, Abdossalami asl Y. In-tube electro-membrane extraction with a sub-microliter organic solvent consumption as an efficient technique for synthetic food dyes determination in foodstuff samples. J Chromatogr A. 2015;1410:35–43.
13. Ramos-Payán M, Villar-Navarro M, Fernández-Torres R, Callejón-Mochón M, Bello-López MÁ. Electromembrane extraction (EME)—an easy, novel and rapid extraction procedure for the HPLC determination of fluoroquinolones in wastewater samples. Anal Bioanal Chem. 2013;405(8):2575–84.
14. Berijani S, Assadi Y, Anbia M, Milani Hosseini M-R, Aghaee E. Dispersive liquid–liquid microextraction combined with gas chromatography-flame photometric detection: very simple, rapid and sensitive method for the determination of organophosphorus pesticides in water. J Chromatogr A. 2006;1123(1):1–9.
15. Viñas P, Campillo N, López-García I, Hernández-Córdoba M. Dispersive liquid–liquid microextraction in food analysis. A critical review. Anal Bioanal Chem. 2014;406(8):2067–99.
16. Ghambarian M, Yamini Y, Esrafili A, Yazdanfar N, Moradi M. A new concept of hollow fiber liquid–liquid–liquid microextraction compatible with gas chromatography based on two immiscible organic solvents. J Chromatogr A. 2010;1217(36):5652–8.
17. Hemmati M, Asghari A, Bazregar M, Rajabi M. Rapid determination of some beta-blockers in complicated matrices by tandem dispersive liquid-liquid microextraction followed by high performance liquid chromatography. Anal Bioanal Chem. 2016;408(28):8163–76.
18. Kocúrová L, Balogh IS, Šandrejová J, Andruch V. Recent advances in dispersive liquid–liquid microextraction using organic solvents lighter than water. A review. Microchem J. 2012;102:11–7.
19. Bazregar M, Rajabi M, Yamini Y, Asghari A, Hemmati M. Tandem air-agitated liquid–liquid microextraction as an efficient method for determination of acidic drugs in complicated matrices. Anal Chim Acta. 2016;917:44–52.
20. Bazregar M, Rajabi M, Yamini Y, Saffarzadeh Z, Asghari A. Tandem dispersive liquid–liquid microextraction as an efficient method for determination of basic drugs in complicated matrices. J Chromatogr A. 2016;1429:13–21.
21. Farajzadeh MA, Mogaddam MRA. Air-assisted liquid–liquid microextraction method as a novel microextraction technique; application in extraction and preconcentration of phthalate esters in aqueous sample followed by gas chromatography–flame ionization detection. Anal Chim Acta. 2012;728:31–8.
22. Maya F, Estela JM, Cerdà V. Completely automated in-syringe dispersive liquid–liquid microextraction using solvents lighter than water. Anal Bioanal Chem. 2012;402(3):1383–8.
23. Wu Q, Zhou X, Li Y, Zang X, Wang C, Wang Z. Application of dispersive liquid–liquid microextraction combined with high-performance liquid chromatography to the determination of carbamate pesticides in water samples. Anal Bioanal Chem. 2009;393(6):1755–61.
24. Ebrahimpour B, Yamini Y, Esrafili A. Emulsification liquid phase microextraction followed by on-line phase separation coupled to high performance liquid chromatography. Anal Chim Acta. 2012;751:79–85.
25. Sarafraz-Yazdi A, Amiri A. Liquid-phase microextraction. Trends Anal Chem. 2010;29(1):1–14.
26. Khalili Zanjani MR, Yamini Y, Shariati S, Jönsson JÅ. A new liquid-phase microextraction method based on solidification of floating organic drop. Anal Chim Acta. 2007;585(2):286–93.
27. Rodríguez I, Llompart MP, Cela R. Solid-phase extraction of phenols. J Chromatogr A. 2000;885(1–2):291–304.
28. Rodríguez I, Turnes MI, Mejuto MC, Cela R. Determination of chlorophenols at the sub-ppb level in tap water using derivatization, solid-phase extraction and gas chromatography with plasma atomic emission detection. J Chromatogr A. 1996;721(2):297–304.
29. Lin C-Y, Huang S-D. Application of liquid–liquid–liquid microextraction and ion-pair liquid chromatography coupled with photodiode array detection for the determination of chlorophenols in water. J Chromatogr A. 2008;1193(1–2):79–84.
30. Sulej-Suchomska AM, Polkowska Z, Chmiel T, Dymerski TM, Kokot ZJ, Namiesnik J. Solid phase microextraction-comprehensive two-dimensional gas chromatography-time-of-flight mass spectrometry: a new tool for determining PAHs in airport runoff water samples. Anal Methods. 2016;8(22):4509–20.
31. Liu Q, Shi J, Zeng L, Wang T, Cai Y, Jiang G. Evaluation of graphene as an advantageous adsorbent for solid-phase extraction with chlorophenols as model analytes. J Chromatogr A. 2011;1218(2):197–204.
32. Santos FJ, Jáuregui O, Pinto FJ, Galceran MT. Experimental design approach for the optimization of supercritical fluid extraction of chlorophenols from polluted soils. J Chromatogr A. 1998;823(1–2):249–58.
33. Moradi M, Yamini Y, Esrafili A, Seidi S. Application of surfactant assisted dispersive liquid–liquid microextraction for sample preparation of chlorophenols in water samples. Talanta. 2010;82(5):1864–9.

34. Calvo Seronero L, Fernández Laespada ME, Luis Pérez Pavón J, Moreno Cordero B. Cloud point preconcentration of rather polar compounds: application to the high-performance liquid chromatographic determination of priority pollutant chlorophenols. J Chromatogr A. 2000;897(1–2):171–6.

35. Fattahi N, Assadi Y, Hosseini MRM, Jahromi EZ. Determination of chlorophenols in water samples using simultaneous dispersive liquid–liquid microextraction and derivatization followed by gas chromatography-electron-capture detection. J Chromatogr A. 2007;1157(1–2):23–9.

36. Santana CM, Padrón MET, Ferrera ZS, Rodríguez JJS. Development of a solid-phase microextraction method with micellar desorption for the determination of chlorophenols in water samples: comparison with conventional solid-phase microextraction method. J Chromatogr A. 2007;1140(1–2):13–20.

37. López-Jiménez FJ, Rubio S, Pérez-Bendito D. Single-drop coacervative microextraction of organic compounds prior to liquid chromatography: theoretical and practical considerations. J Chromatogr A. 2008;1195(1–2):25–33.

38. Fan C, Li N, Cao X. Determination of chlorophenols in honey samples using in-situ ionic liquid-dispersive liquid–liquid microextraction as a pretreatment method followed by high-performance liquid chromatography. Food Chem. 2015;174:446–51.