Polar organic chemical integrative sampler (POCIS) allows compound specific isotope analysis of substituted chlorobenzenes at trace levels

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Abstract

Compound specific isotope analysis (CSIA) is an established tool to demonstrate *in situ* degradation of traditional groundwater contaminants at heavily contaminated sites, usually at mg/L range aqueous concentrations. Currently, an efficient preconcentration method is lacking to expand CSIA to low aqueous concentration environmental samples. This work demonstrated the compatibility of polar organic chemical integrative sampler (POCIS) with CSIA of C, H, and N isotopes for four NH_2- and NO_2-substituted chlorobenzenes at low μg/L. Diffusion and sorption showed insignificant carbon isotope fractionation (<0.7‰) in laboratory experiment, except for a reproducible shift of 1.6‰ for 3,4-dichloronitrobenzene. A similar constant reproducible shift of 0.8-2‰ was evident for N-CSIA. Whereas, the compatibility of POCIS for H-CSIA seems to be analyte specific possibly reflecting the adsorption mechanism to POCIS by H-bonding. Performance of the POCIS-CSIA method was evaluated in a pilot constructed wetland where comparable C- and N-CSIA results were obtained from grab sampling and POCIS. This work opens the potential of CSIA application to the low concentration polar emerging contaminants in the environment, such as pesticides, pharmaceuticals, and flame-retardants.

Keywords: POCIS; CSIA; isotope analysis; chloronitrobenzenes; passive sampling

1. Introduction

Substituted chloronitrobenzenes, chloroanilines, and nitrotoluenes are common source materials to produce various pesticides, dyes, explosives, pharmaceuticals, personal care products, preservatives, and antioxidants.[1] Chloroanilines can be formed in the natural environment by biotic and abiotic processes during the reductive transformation of chloronitrobenzenes,[2,3] and

transformation of several phenylurea herbicides, such as diuron, linuron, neburon, and monuron.[4] Microbial oxidation of chloroanilines by soil fungus can also form chloronitrobenzenes.[5] Nitrotoluenes are primary by-products of explosives and found extensively at former munition sites.[1] Many of these compounds are acutely toxic, mutagenic, carcinogenic, and listed on the United States Environmental Protection Agency's list of priority pollutants.[6] Due to their persistence in the environment,[7] they are detected in industrial wastewaters and natural water bodies in various regions.[8,9]

Conventionally, contaminant fate in the environment is evaluated by monitoring the changes in their *in situ* concentrations. However, concentration-based approaches cannot provide unequivocal evidence of *in situ* degradation as non-destructive processes, e.g., sorption, volatilization, and dilution, can also affect concentration. Compound specific isotope analysis (CSIA) has the potential to distinguish between destructive and non-destructive processes *in situ* by measuring the changes in the ratio of a heavy (e.g., ^{13}C) and a light (e.g., ^{12}C) stable isotopes of an element (e.g., C) in a molecule.[10] Physical processes do not involve bond cleavage, typically resulting in negligible isotope fractionation.[11–13] CSIA has been used as a direct line of evidence for *in situ* degradation of many organic compounds.[10] However, due to the high instrumental detection limits of isotope ratio mass spectrometers (IRMS), CSIA has been mainly limited to contaminated sites with high µg/L to mg/L range concentrations.[10] Recent applications of CSIA to µg/L to higher ng/L concentrations have been made after extracting 10 to 100 L of water for each analysis using solid-phase extraction (SPE).[14,15] Using large volume SPE is susceptible to preconcentrate co-contaminations and background matrix. Besides, significant isotope fractionation (up to 6‰) was observed for the extraction of more than 10 L.[14] Thus, an efficient preconcentration technique for trace contaminants in water free from method-induced

isotope fractionation is necessary to expand CSIA for low concentration environmental samples, such as long-term monitoring groundwater sites and surface water environments where concentrations are often more diluted.

Combining passive integrative sampling with CSIA could be an effective *in situ* preconcentration technique that requires little effort during sample preparation. Although various *in situ* passive sampling techniques are widely accepted, limited research, to date, has explored their compatibility with CSIA. Wang et al. (2004) reported negligible C and H isotope fractionation of several polycyclic aromatic hydrocarbons using semi-permeable membrane devices under different exposure conditions.[16] Another permeation-based passive sampler known as the Waterloo Membrane Sampler was evaluated for sorption- and desorption-associated C isotope fractionation for gas-phase hydrocarbon sampling[17] and was successfully applied at contaminated sites.[17,18] A type of diffusion passive sampler, called peeper, also showed no significant C isotope fractionation for aromatic and chlorinated aromatic compounds,[19] and was successfully deployed in sediments to track pore water benzene and chlorobenzene natural attenuation by CSIA.[20,21] Until today, no passive sampling technique has been validated for CSIA targeting trace polar organic compounds, such as pesticides, pharmaceuticals, and personal care products. The polar organic chemical integrative sampler (POCIS), introduced by Alvarez et al. in 2004, consists of a sorbent phase sandwiched between two polyethersulfone membrane filters.[22] POCIS involves multiphase analyte mass transfer from the water column to the sampler receiving phases via diffusion and adsorption, which can potentially cause isotope fractionation. Thus, a careful evaluation of the sampling method is critical to ensure that the original isotope signature of the analytes are conserved in POCIS.

The main objective of this work was to evaluate the compatibility of POCIS with CSIA to monitor *in situ* transformation of the target compound groups at a contaminated industrial site. Because POCIS has never been applied for most of the target compounds, we first evaluated whether POCIS can accumulate enough analyte mass for CSIA application. Second, we evaluated method-induced isotope fractionation during sampling and post-processing phases using laboratory experiments. Third, we demonstrated the performance of the POCIS-CSIA method in at a field pilot constructed wetland system.

2. Materials and methods

2.1 Chemicals and materials

All chemical details are given in Supporting Information (SI) Section S1. Selected analytes have been abbreviated as follows: 2-nitrotoluene (2-NT), 2- and 4-chloronitrobenzene (2-CNB and 4-CNB), 4-chloroaniline (4-CA), 2,3- and 3,4-dichloroaniline (2,3-DCA and 3,4-DCA), and 2,5- and 3,4-dichloronitrobenzene (2,5-DCNB and 3,4-DCNB). 2,5-DCNB was used as an internal standard to correct for potential losses during sample processing.

All POCIS were assembled using OASIS HLB bulk sorbent (30 μm particle size, divinylbenzene N-vinyl-pyrrolidone, Waters, Massachusetts, USA) and polyethersulfone (PES) microporous membrane disk filters (0.1 μm pore size, 90 mm diameter, PAL Corporation, California, USA) following standard configuration.[23] Details on POCIS preparation and pre-treatment are given in Suchana et al (modeling paper) and briefly descried in Section S2.

2.2 POCIS laboratory experimental systems

Three experiments were performed statically without renewal of the analytes in large glass jars (diameter: 19 cm, height: 26 cm, total capacity: 7.5 L) as detailed in Suchana et al. (modeling paper). Static experimental setup was chosen over continuous flow-through systems to avoid isotope dilution.

First, a preliminary exposure experiment was conducted to verify the potential of POCIS to accumulate the target compounds for CSIA. For that, 7 L of autoclaved ultrapure water (resistivity of 18.2 MΩcm) was spiked at a target initial concentration of 1 mg/L for each 4-CNB, 3,4-DCNB, 4-CA, and 3,4-DCA using methanol stock (<0.5% v/v of methanol). The system was mixed and left untouched for 24 h to allow for analyte dispersion. Then, one POCIS was suspended vertically in each jar from the top (membrane exposure level 4.5-5.0 cm from bottom) using a stainless-steel wire loop. The deployment of the POCIS defined the start of the experiment, i.e., time 0. On day 44 since the start of the experiment, water sample was collected for concentration analysis, and POCIS HLB and PES phases were extracted separately to quantify the accumulated analytes.

Second, a kinetic experiment was performed using a sacrificial setup with one POCIS per jar, each filled with 6 L of autoclaved ultrapure water. Each jar was spiked with an initial target concentration of 10 mg/L for each analyte as before. Note that 2-NT was spiked instead of 4-CA in the second experiment to obtain a complete separation in the isotopic trace in the IRMS. A relatively high initial concentration was used to ensure sufficient mass accumulation in POCIS for CSIA at six sampling times. After 24 h of spiking, water from the top and bottom of each jar was analyzed immediately to confirm the system was well mixed and determine the initial spiking concentration in each setup. One POCIS was deployed in each setup (i.e., time 0), and duplicate POCIS were sacrificed at days 7, 14, 21, 31, 45, and 60. At each time point, water

samples were also collected for concentration and isotope analyses. Analyte mass balance in sacrificial setup was calculated at each time point (Section S3).

Third, another kinetic experiment was conducted identical to the second experiment, except that no HLB sorbent was placed inside the two PES membranes of a POCIS configuration. This experiment was performed to quantify analyte isotope fractionation induced by diffusion through and adsorption on the PES membrane. On day 60, the PES membranes were extracted for both concentration and isotope analyses.

2.3 POCIS deployment in the field

POCIS were deployed at a pilot surface flow constructed wetland system for 60 days. The wetland system was continuously fed by partially-treated groundwater from an industrial site contaminated with the target compounds. Six POCIS were deployed including three POCIS protected with conventional stainless-steel mesh and three POCIS protected with a copper mesh to act as an antimicrobial metal.[24] In addition, two separate POCIS, one in each casing material, were deployed to evaluate the potential of biofilm growth on the outer membrane surface during the exposure period (referred to as "biofilm POCIS"). Isotope signatures from POCIS were compared with that of the 4 L grab sample preconcentrated by SPE using the method described in Suchana et al. SPE/CSIA paper. After retrieval, all POCIS were wrapped separately and stored at −20°C until further processing.

2.4 Controls and blanks

For laboratory experiments, duplicate negative controls were prepared by deploying one POCIS in an analyte-free jar to assess contamination during deployment and sample processing. Duplicate positive controls were prepared with ultrapure spiked water and POCIS metal parts only (i.e., no membrane and no sorbent) to monitor contaminant loss due to processes other than POCIS uptake, such as volatilization and sorption to the experimental setup. Duplicate POCIS fabrication blanks (i.e., not deployed in water but that underwent the same preparation steps) were also used to evaluate potential contamination during POCIS preparation. For field POCIS, an additional trip blank (i.e., taken to the field during the deployment and retrieval but never opened), and separate deployment and retrieval blanks (i.e., taken to the field and exposed to the air during the deployment and retrieval, respectively) were used to evaluate potential contamination during sampling.

2.5 Sample processing for concentration and isotope analyses

Water samples from Experiment 1 were analyzed after filtration through 0.22 μm syringe filter (PTFE, Chromspec, ON, Canada) without further processing. For Experiment 2, water was extracted at each time point by OASIS HLB SPE cartridge (6 mL, 30 μm particle size, 200 mg sorbent, Waters) for both concentration and isotope analyses following the method described in Suchana et al. SPE-CSIA paper. Extraction of PES phase was performed following Suchana et al. modeling paper.

For concentration analysis using SPE, HLB, and PES extracts (ethyl acetate), a suitable aliquot (dilution factor 100 to 1000) was mixed with ultrapure water to obtain concentrations within the instrumental calibration range. Due to the limited water solubility of ethyl acetate (<8% v/v),

samples were prepared by adding <5% (v/v ethyl acetate:water) of the final extract to ultrapure water. For isotope analysis, ethyl acetate extracts were injected directly or diluted, if necessary, for a splitless injection.

For field POCIS, the outer membrane surface was first washed thoroughly using ultrapure MQ water and then both the HLB and PES phases were processed as before for isotope analysis. Extraction of field water collected using grab sampling was performed by SPE (Suchana et al. SPE/CSIA paper). All extracts were stored at –20°C until analysis. All extraction methods are briefly described in Section S4.

2.6 DNA extraction and scanning electron microscopy of field POCIS

The two PES membranes of the biofilm POCIS were collected at day 60 of the field exposure, rinsed with ultrapure water to remove large unattached particles, and then processed for both DNA extraction (one membrane) and scanning electron microscopy (the other membrane).

The central exposed circle of the membrane and the donut shaped outer part that was under the metal ring were cut into small pieces using a sterile slide, and transferred to a microcentrifuge tube for DNA extraction following the PowerSoil™ Kit protocol (Mo Bio Laboratories Inc. Carlsbad, CA, USA). DNA extracts were sequenced for the 16SrRNA gene using the MiSeq Sequencing System equipment (Illumina Inc., USA). Details are given in Section S5.

The morphologies attached to the PES membranes of POCIS were observed by scanning electron microscopy (JEOL®6390LV, JEOL Ltda., Tokyo, Japan), with 10 kV of acceleration voltage. Similarly as DNA extraction, both the exposed and outer part of the membrane were collected

separately, dried at room temperature, and coated with a fine gold layer for 5 minutes, using the Denton Vacuum IV metallizer device, before visualization.

2.7 Analytical methods

Concentration analysis

A Dionex Ultimate 3000 series High-Performance Liquid Chromatography (HPLC) connected with a diode array detector (DAD) was used for concentration determination of laboratory samples (Suchana et al. modeling paper). Concentration analysis of the field samples was performed using xxx. Details are given in Section S6.

Compound specific carbon isotope analysis

Isotope signatures ($\delta^{13}C$, $\delta^{15}N$, and $\delta^{2}H$) were measured using a TRACE 1300 gas chromatograph (GC) coupled with an isotope ratio mass spectrometer (IRMS, Delta V plus, Thermo Scientific). Conversion of analytes to the measurement gas for C- and N-CSIA (e.g., CO_2 and N_2) was performed in a commercial NiO/CuO/NiO-tube reactor (Isomass Scientific Inc., Alberta, Canada) operated at 1000°C. Measurement of H isotope was performed using custom-made chromium-based high-temperature conversion reactor (Cr/HTC) operated at 1200°C. Details on chromatographic parameters are given in Suchana et al. (SPE paper).

The isotope signature of the pure compounds was determined in an elemental analyzer (EA) coupled with an IRMS at the Helmholtz Centre for Environmental Research (UFZ) in Leipzig, Germany. Isotope measurement of the experimental samples was bracketed with that of the characterized pure standards. All reported values were normalized using a multi-point calibration

of in-house reference standards. The measured $\delta^{13}C$ values were considered accurate if the bracketed standards were within ±0.5‰ from their characterized values, considering a total analytical uncertainty of ±0.5‰.[25] A total analytical uncertainty of ±0.5‰ and ±10‰ was accepted for N and H isotopes, respectively (Suchana et al. SPE paper).

The change in isotope signature of the experimental samples was expressed as the differences of the measured δ values by GC/IRMS from that of the reference standards measured in EA/IRMS, i.e., $\Delta\delta_{Sample} = \delta_{EA/IRMS} - \delta_{GC/IRMS}$. All values were reported in per mill (‰), equivalent to MilliUrey (mUr)[26], and relative to the international standards of Vienna PeeDee Belemnite, Vienna standard mean ocean water, and air for carbon, hydrogen, and nitrogen, respectively.

2.8 Method quantification limit

The method quantification limit of the POCIS-CSIA method depends on the sampler-water partition of analytes (equilibrium sampling regime) or the sampling rate (kinetic sampling regime) in POCIS and the instrumental detection limits of the IRMS. Details are provided in Section S7.

3. Results

3.1 Analyte accumulation in POCIS for CSIA

Overall, a significant accumulation of all target analytes was observed in both HLB and PES phases from Experiment 1 (Figure S1, Section S8). Between 1580 (3,4-DCNB) to 4860 nmol (4-CA) accumulated in the HLB sorbent; whereas, the PES membranes accumulated on average

236 2430 nmol of 4-CA, 4-CNB, and 3,4-DCNB and 5180 nmol of 3,4-DCA. Accumulation in the
237 HLB sorbent was 1.7 to 1.9 times higher than that of the PES membranes for monochlorinated
238 compounds, i.e., 4-CA and 4-CNB. Dichlorinated compounds, i.e., 3,4-DCA and 3,4-DCNB,
239 accumulated preferentially in the PES membranes. For a final 1.5 mL extract volume and
240 standard splitless 1-μL solvent injection in the GC, between 6 (3,4-DCNB, HLB sorbent phase
241 alone) and 20 nmol of C (3,4-DCA, PES membrane phase alone) can be injected on column. If
242 the two phases are pooled and the extracts prepared and injected under the same conditions, even
243 higher amounts of C can be obtained from one POCIS, i.e. from 16 (3,4-DCNB) to 36 nmol (3,4-
244 DCA) of C. In the field, larger analyte accumulation can be expected due to advection
245 processes.[22]

246

247 **3.2 POCIS kinetic experiments**

248 Figure 1 shows the analyte mass and corresponding $\delta^{13}C$, $\delta^{15}N$, and $\delta^{2}H$ profiles over the course
249 of the experiments in the water and POCIS sorbent and membrane phases (Experiment 2).

250 *Analyte mass profile*

251 A decrease of analytes from the water phase and accumulation in POCIS was observed over time
252 from each sacrificial setup (Figure 1a-1d). After 60 days, the initial spiked mass of 2-NT, 4-
253 CNB, and 3,4-DCA decreased by 21-22% and that of 3,4-DCNB decreased by 44% in the water
254 phase. We considered that adsorption equilibrium was reached when less than 2% of the analyte
255 mass in the water phase changed between two consecutive sampling times. While concentration
256 equilibrium was apparent at day 45 for 2-NT, 4-CNB, and 3,4-DCA, the mass of 3,4-DCNB in
257 water decreased by 6% between day 45 and 60, indicating non-equilibrium adsorption on

POCIS. However, this 6% decrease in 3,4-DCNB mass was mainly due to the HLB phase (93% of the total mass decrease) while additional adsorption on PES was negligible (Figure 1d). Thus, over a 2-month deployment period, adsorption of 3,4-DCNB might reach an equilibrium only on the PES phase.

Carbon, nitrogen, and hydrogen isotope profiles

For 2-NT, the $\delta^{13}C$ values from all three phases, and the $\delta^{15}N$ value from the HLB phase remained constant and within ±0.5‰ of the characterized isotopic signature throughout the exposure time (Figure 1e, 1i). Although the $\delta^{15}N$ from the PES phase showed an average shift of 0.8‰ (i.e., $\delta^{15}N_{EA/IRMS} - \delta^{15}N_{sample} = 0.8$ ‰) for the entire deployment time (Table 1), it became stable with an average shift of 0.4‰ between 31 and 60 days. The $\delta^{2}H$ values were depleted in ^{2}H throughout the deployment time (Figure 1m) compared to the EA/IRMS characterized value, with a constant shift of 19 and 24‰ for the HLB and PES phases, respectively. However, overall the average shifts from both PES and HLB phases were similar, i.e., within a band of 5‰ (Table 1).

For 4-CNB, the $\delta^{13}C$ values from both the HLB and PES phases were relatively depleted in ^{13}C compared to the characterized value (Figure 1f). A similar average shift of 0.6 and 0.7‰ was observed in the HLB and PES phases (Table 1), respectively, over the course of the experiment. Changes in $\delta^{13}C$ from the water phase were insignificant. The obtained $\delta^{15}N$ values were relatively enriched in ^{15}N (Figure 1i) with an average shift of −1.3 and −1.6‰ for the HLB and PES phases, respectively (Table 1). However, the magnitude of the shift for both C and N was constant and independent of the deployment time. The obtained $\delta^{2}H$ values from both the HLB and PES phases showed some instability up to 14 days (Figure 1n) but the overall shift remained within ±5‰ for both phases (Table 1).

For 3,4-DCA, the average shift in $\delta^{13}C$ values from the water and PES phases was <±0.5‰ (Table 1), although a slight enrichment in ^{13}C in water was noticeable after 31 days (Figure 1g). The $\delta^{13}C$ values from the HLB phase were enriched in ^{13}C during the initial uptake phase (day 0 to 31) and eventually reached isotopic equilibrium after day 31 (Figure 1g). If only the $\delta^{13}C$ values from day 31 onward were considered, the average HLB phase $\delta^{13}C$ value became –26.6 ± 0.5‰ with a <0.5‰ shift from the characterized pure standard. The $\delta^{15}N$ values from the HLB and PES phases were enriched with ^{15}N over the course of the deployment time (Figure 1k), and reached a constant shift of −1.7 and −1.5‰, respectively if averaged between 31 to 60 days. A similar trend was observed for $\delta^{2}H$ where both phases were enriched with heavier ^{2}H and an apparent isotopic equilibrium was only obtained at day 60 (Figure 1o). However, it should be noted that the day 0 isotope signature of the water phase had a significant shift of −23.8‰ from the EA/IRMS value (Table 1).

For 3,4-DCNB, the $\delta^{13}C$ values were enriched with ^{13}C (Figure 1h) and showed a constant and reproducible average shift of –1.6 and –1.4‰ for the HLB and PES phases, respectively, throughout the exposure time (Table 1). However, $\delta^{13}C$ values from the water phase remained within analytical uncertainty, i.e., ±0.5‰ (Figure 1h). A similar trend was observed for $\delta^{15}N$ and the average shift from the both phases were also comparable (Figure 1l, Table 1). The $\delta^{2}H$ values were stable for 60 days with negligible average shift form the characterized value (<±5‰) (Figure 1p, Table 1).

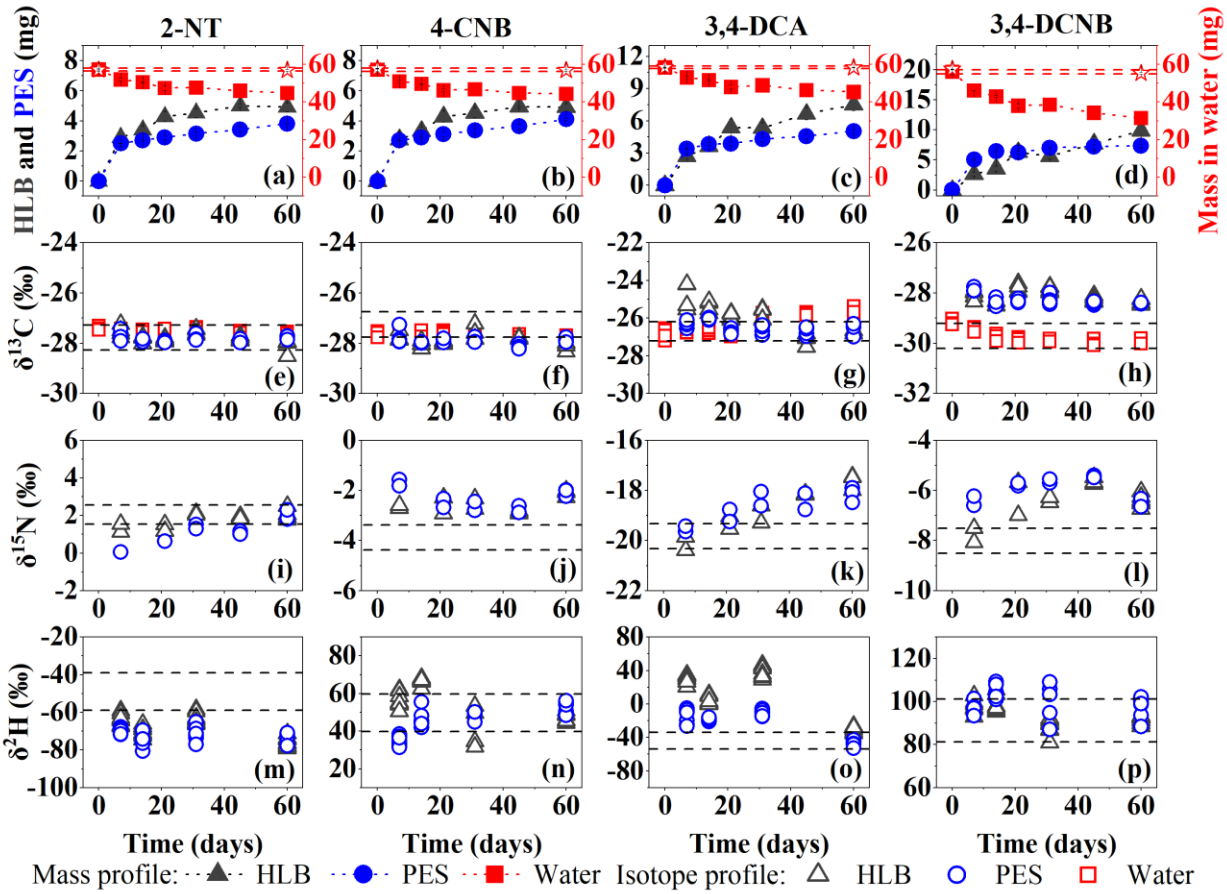

Figure 1 Analyte mass (a-d) in HLB (▲), PES (●), and water (■) phases, and the corresponding $\delta^{13}C$ (e-h), $\delta^{15}N$ (i-l), and $\delta^{2}H$ (m-p) values in HLB (△), PES (○), and water (□) phases over time for: 2-NT, 4-CNB, 3,4-DCA, and 3,-4-DCNB. In (a) to (d), the horizontal red dashed lines represent ±1 standard deviation of the measured initial spiked mass from all sacrificial setups, and the open red stars represent the average mass measured from positive controls at days 0 and 60. The error bars are ±1 standard deviation from duplicate experiments, and the dotted lines are added to guide the observed trend (a-d). Isotope values of duplicate injections from duplicate setup are reported without averaging (note that these 4 data points sometimes overlapped for $\delta^{13}C$ and $\delta^{2}H$; whereas $\delta^{15}N$ are reported for one replicate). The horizontal black dashed lines

represent the total analytical uncertainty of ±0.5‰ for $\delta^{13}C$ and $\delta^{15}N$, and ±10‰ for $\delta^{2}H$ around the characterized values by EA/IRMS.

Table 1 Average δ (‰) values calculated from all sampling points for different phases and the corresponding average shift (Δδ) from the EA/IRMS-characterized values

Analyte	EA	HLB		PES		Water	
		$\delta^{13}C$*	$\Delta\delta^{13}C$**	$\delta^{13}C$*	$\Delta\delta^{13}C$**	$\delta^{13}C$*	$\Delta\delta^{13}C$**
2-NT	–27.8 ± 0.1	–27.8 ± 0.2	0	–27.8 ± 0.1	0	–27.5 ± 0.1	–0.3
4-CNB	–27.3 ± 0.1	–27.9 ± 0.2	0.6	–27.9 ± 0.2	0.7	–27.6 ± 0.1	0.4
3,4-DCA	–26.7 ± 0.2	–26.0 ± 0.8	–0.7	–26.5 ± 0.3	–0.2	–26.3 ± 0.5	–0.4
3,4-DCNB	–29.7 ± 0.03	–28.1 ± 0.3	–1.6	–28.3 ± 0.2	–1.4	–29.7 ± 0.3	0
		$\delta^{15}N$*	$\Delta\delta^{15}N$**	$\delta^{15}N$*	$\Delta\delta^{15}N$**	$\delta^{15}N$	$\Delta\delta^{15}N$
2-NT	2.1 ± 0.03	1.8 ± 0.4	0.2	1.4 ± 0.7	0.7	nd	nd
4-CNB	–3.9 ± 0.02	–2.6 ± 0.3	–1.3	–2.3 ± 0.4	–1.6	nd	nd
3,4-DCA	–19.8 ± 0.1	–18.7 ± 1.0	–1.1	–18.6 ± 0.6	–1.2	nd	nd
3,4-DCNB	–8.0 ± 0.03	–6.7 ± 0.7	–1.6	–6.0 ± 0.5	–2.0	nd	nd
		$\delta^{2}H$*	$\Delta\delta^{2}H$**	$\delta^{2}H$*	$\Delta\delta^{2}H$**	$\delta^{2}H$***	$\Delta\delta^{2}H$**
2-NT	–48.9 ± 0.3	–67.8 ± 4.6	18.9	–73.2 ± 4.0	24.1	–53.2 ± 1.9	4.3
4-CNB	49.8 ± 0.6	54.4 ± 8.2	–4.6	45.9 ± 6.0	3.9	47.3 ± 1.7	2.5
3,4-DCA	–43.7 ± 1.5	11.0 ± 28.2	–54.7	–23.4 ± 12.7	–20.3	–19.9 ± 1.8	–23.8
3,4-DCNB	91.2 ± 0.5	93.0 ± 4.8	–1.8	100.5 ± 3.6	–9.3	92.1 ± 3.4	–0.9

* Average of all measurement ± 1 standard deviation
** $\Delta\delta = \delta^{h}X_{EA/IRMS}$ – Average $\delta^{h}X$ obtained by GC/IRMS for the corresponding phase (where "h" is for heavy isotope, and X is for an element, i.e., C, N, or H)
*** Day 0 isotope signature in water
nd: not determined

3.3 Field evaluation of POCIS and grab sampling

We compared *in situ* isotope signatures obtained by SPE and POCIS from a pilot constructed wetland to evaluate the performance of the POCIS-CSIA method under environmental conditions. Only DCA isomers were present at that site; therefore, the comparison is based on 2,3- and 3,4-DCA. The $\delta^{13}C$ obtained by SPE was −31.4±0.1‰, and −27.3±0.2‰ for 2,3- and 3,4-DCA, respectively (Table S3, Section S10). The $\delta^{13}C$ values of 2,3- and 3,4-DCA from POCIS sorbent and membrane phases showed no significant difference (<0.5‰) from that of the SPE, expect for a small offset of 0.6‰ from the membrane phase of 3,4-DCA (Table S3, Section

S10). The obtained $\delta^{15}N$ value of 2,3-DCA was 3.9±0.2, 2.9±0.5, and 2.5±0.2‰ from SPE, and POCIS sorbent and membrane phases, respectively (Table S4, Section S10). Unlike lab Experiment 2 where the N isotope signature of 3,4-DCA (a comparable isomer to 2,3-DCA) was slightly enriched with heavier ^{15}N in both phases (~1‰) compared to the EA/IRMS signature, the $\delta^{15}N$ values of 2,3-DCA from the field POCIS were slightly enriched with lighter ^{14}N compared to SPE. However, the observed differences in $\delta^{15}N$ values between grab sampling and POCIS were ~1‰, which is two times the analytical uncertainly for N and thus likely to be considered indistinguishable from the SPE-obtained isotope signatures in the field. Finally, no significant differences were observed in the $\delta^{13}C$ values of the stainless-steel and copper casings (within ±0.5‰); no data were available for H or N isotopes.

3.4 Quality assurance and quality control (QA/QC)

No background contamination was observed from the controls and blanks. Total mass loss of each analyte from the water phase was <5% during laboratory experiments (positive controls). The standard deviations from replicate injections of the same sample and the duplicate experimental setups were within the analytical uncertainly of each isotope. The only exception was for the HLB phase of 3,4-DCA at day 7 for C and 3,4-DCNB at day 21 for N (Figure 1g, 1l). Compared to the overall data, it is likely an artefact related to the initial rapid adsorption on POCIS. The method-induced isotope fractionation for the PES phase extraction (Table S5, Section S11) and for the SPE method used for the laboratory and field water extractions were negligible (Suchana et al. SPE paper).

4. Discussion

4.1 Compatibility of POCIS with C-, N-, and H-CSIA

Carbon isotope equilibrium of 2-NT was observed within 7 days (Figure 1e), although concentration equilibrium needed approximately 31 days (Figure 1a). Initially, a temporal trend in $\delta^{13}C$ values was observed for 3,4-DCA; nevertheless, the obtained isotope values were within the characterized signature as POCIS uptake approached near-equilibrium after 31 days (Figure 1c, 1g). Thus, POCIS is compatible with C-CSIA without further considerations for 2-NT and 3,4-DCA. For 4-CNB and 3,4-DCNB, while a significant shift in carbon isotope signature was evident, it was constant and reproducible throughout the deployment (Figure 1f, 1h). A similar constant and reproducible shift in $\delta^{13}C$ was observed for hexane (1.4‰), benzene (1.2‰), and trichloroethylene (0.9‰) in the Waterloo Membrane Sampler.[17] This sampler was successfully applied at contaminated sites for CSIA by correcting the reproducible shift of $\delta^{13}C$ associated with the gas-phase passive sampling technique.[17,18] However, as the carbon isotope fractionation in POCIS was <1‰ for 4-CNB, i.e., 2 times the total analytical uncertainty of $\delta^{13}C$, and <2‰ for 3,4-DCNB, i.e., the recommended maximal isotope shift for field interpretation,[10] we do not recommend isotopic correction for carbon during field application of the POCIS-CSIA technique.

A significant shift was observed for nitrogen isotope for all compounds for both sorbent and membrane phases (0.8 to 2‰), except the sorbent phase of 2-NT (<0.5‰). However, the shifts were constant after 21 days. The total shift was ≤2‰ and thus no isotopic correction is recommended for N isotope during field application.

The average hydrogen isotope signature of 4-CNB and 3,4-DCNB from POCIS differed be less than 10‰ from the EA/IRMS signature, i.e., 2 times the total analytical uncertainty for $\delta^{2}H$

(±5‰) (Table 1). Thus, no correction is recommended for 4-CNB and 3,4-DCNB. However, a significant shift of >10‰ for 2-NT and 3,4-DCA was observed (Table 1). However, the shift for 2-NT was constant throughout the deployment time for both phases (standard deviation <5‰) for which a correction factor, similarly as the Waterloo Membrane Sampler,[17] could be applied for field application. The δ^2H for 3,4-DCA was mostly too variable between the sampling times which might require further mechanistic study to interpret this behaviour.

4.2 Diffusion- and adsorption-induced isotope fractionation

Analyte mass transfer from the water column to POCIS receiving phases involves multistep diffusion, i.e., in the water boundary layer, pore waters in PES and HLB, and in polymer matrix involving intra-particle and interstitial diffusion.[27,28] Thus, three phenomena could lead to isotope fractionation during POCIS preconcentration: (i) aqueous-phase diffusion; (ii) diffusion in PES and HLB; and (iii) adsorption on PES and HLB.

Diffusion and subsequent adsorption of molecules incorporating heavy isotopes are expected to be slower than for those made of light isotopes, resulting in an enrichment with lighter isotopes in the POCIS receiving phases as the overall direction of mass transfer is towards the POCIS. Diffusion typically does not cause significant isotope fractionation[13,19,29] considering typical temporal and spatial sampling regimes in the field. However, the static experimental system used in this study could likely cause a solute gradient in the water boundary layer that might be responsible for diffusion-induced isotope fractionation. Since all target compounds have comparable molecular mass, water diffusivity, $\log K_{OW}$, membrane-water partition coefficient ($\log K_{PES\text{-}water}$), and Freundlich membrane adsorption coefficients ($\log K_F$, n_f) values (Table S1),

similar isotope fractionation due to diffusion in all phases should have been observed for all compounds. Most importantly, when a significant shift was observed, POCIS receiving phases were predominantly enriched with the heavier isotopes, which is unlikely the results of a diffusion-induced process. Therefore, the observed isotope fractionation in POCIS might be explained by the adsorption steps in the PES and HLB phases.

To directly evaluate potential isotope fractionation specifically associated with the PES phase, we compared the results from standard POCIS configuration (Experiment 2, Figure 1) to that of the HLB-free POCIS configuration (Experiment 3, Table S2, Section S9). The $\delta^{13}C$ values of 3,4-DCNB from the PES phase showed no significant shift (<0.5‰) in the HLB-free POCIS (Table S2), thus indicating that the observed carbon isotope shift from Experiment 2 was likely associated with the HLB phase adsorption. Electrostatic interactions between aromatic compounds and PES involve H-bond formation and π-π interactions,[30–33] where additional aromatic staking could occur for compounds having electron-withdrawing substituents, e.g., Cl-, NO_2.[34,35] However, such interactions between (di)chlorobenzenes and polysulfone membrane previously showed negligible carbon isotope fractionation under equilibrium sorption condition.[19] As PES membrane contains similar repeating units as polysulfone membranes, adsorption on PES did not cause carbon isotope fractionation.

Adsorption on HLB is mainly favored by dipole-dipole and hydrogen bond interactions, but electron lone pair interactions with HLB could also be present for polar compounds[36,37]. The lone pair of electrons from Cl- and NO_2- substituents of 3,4-DCNB possibly caused stronger intermolecular interactions with the HLB phase resulting in significant carbon isotope fractionation. Multistep sorption and successive partitioning steps were previously reported to cause preferential accumulation of lighter carbon isotope on the sorbent phase depending on the

specific intermolecular interactions.[38–40] However, the observed shifts in POCIS for most compounds were counter-intuitive as the isotope signature in HLB and PES phases were mostly enriched with heavier ^{13}C (Figure 1h), ^{15}N (Figure 1j, 1k, 1l), and ^{2}H (1o).

Inverse isotope effects during adsorption processes were reported preciously when a molecule binds to a non-biological binding pocket and has been termed as binding isotope effects (BIEs).[41] For example, inverse BIEs were observed for *p*-xylene and carbon tetrachloride while binding in a dimeric capsule molecule.[42] Gibb and coworkers studied the C-H···X-R hydrogen bond interaction between a cavitand sorbent and a variety of halogenated compounds. Depending on the halogen atom and the size of the sorbent, either normal or inverse BIEs were observed.[41,43] It is plausible that the strong C-H···X-R interaction between the benzyl moiety of HLB and the Cl atoms of the compounds could lead to the observed inverse isotope fractionation during the adsorption/binding processes. The role of Cl atom for the inverse isotope effect is somewhat evident from our work as we observed the least inverse effect for 2-NT which does not contain Cl atom.

Similar significant inverse carbon and nitrogen isotope fractionation were observed during direct immersion solid-phase microextraction (SPME) of several NO_2- and NH_2- substituted chlorobenzenes.[44] Although it was suggested as an artefact of reactor oxidation state,[44] it could also be a BIEs due to the C-H···X-R interaction between the polyacrylate SPME fiber and halogenated compounds. Analyte accumulation in direct immersion SPME and POCIS is similar, where aqueous phase compounds diffuse and adsorb on the sorbent phase. However, interactions and associated isotope fractionation are highly dependent on the specific analyte-sorbent pair and, thus, difficult to postulate.

436 **4.3 Potential of CSIA at trace levels using POCIS**

437 We estimated the minimum water concentration ($C_{w, min}$) required for precise and accurate C-, H-
438 and N-CSIA, which ranged from 5 to 2 μg/L for C, 518 to 307 μg/L for H, and 1901 to 418 μg/L
439 for N after a 60-day deployment if the accumulated mass in the HLB phase (conventional POCIS
440 sink) of one POCIS were considered (Table S6, Section S12). The obtained C-CSIA method
441 quantification limits of the target compounds using POCIS were comparable to the 10 L of water
442 extraction using SPE.[15] We also calculated the projected $C_{w,min}$ of C-CSIA for other polar
443 organic compounds ($1.4 \leq \log K_{OW} \leq 4.5$), such as pesticides, fungicides, pharmaceuticals, and
444 anticorrosives, which ranged between 0.1 to 1.5 μg/L for the HLB phase of one POCIS (Table
445 S7, Section S12).

446 Both HLB and PES phases acted as significant sinks for the target compounds (Figure 1a-1d).
447 Besides, differences in average δ values obtained from both POCIS phases were <0.5‰ for C
448 and N and <10‰ for H isotopes, even when a significant shift was observed (except for the 3,4-
449 DCA for H) (Table 1). Thus, the recovered mass from both phases could be combined to further
450 lower the CSIA detection limits without introducing isotopic bias. For example, $C_{w, min}$ for C-
451 CSIA ranged between 1 to 3 μg/L for combined PES and HLB extracts of one POCIS after a 60-
452 day deployment (Table S8, Section S12), which is approximately 50% lower compared to that of
453 the HLB phase alone. POCIS preconcentration also lowered the detection limits for N- and H-
454 CSIA of the target compounds at concentrations down to approximately 200 μg/L and 200-1000
455 μg/L, respectively when sorbent and membrane extracts from one POCIS were combined (Table
456 S3). This concept was demonstrated for the field POCIS that allowed us to obtain $\delta^{13}C$ values at
457 concentration down to <3 μg/L for 2-CNB (Table S3, Section S10) and for $\delta^{15}N$ at 144 μg/L for

458 3,4-DCA (Table S4, Section S10) using the combined sorbent and membrane extracts of one
459 POCIS; whereas, we could not measure the isotope signatures from the 4-L SPE.

460 Moreover, similar to concentration analysis,[23] combining the PES and HLB extracts from
461 multiple POCIS could potentially lower the CSIA detection limits compared to a single POCIS
462 (Table S8, Section S12). However, we attempted to combine both the sorbent and membrane
463 extracts of three POCIS but the obtained isotope traces were compromised due to the increased
464 background from highly preconcentrated matrix interferences.

465 Based on the observed accumulation of the target compounds by POCIS, three suggestions could
466 be offered to explore POCIS preconcentration for CSIA at the ng/L range. First, more HLB
467 sorbent mass can be added to the POCIS to increase overall adsorption capacity. Studies
468 suggested that sampling rates could be doubled using 600 mg HLB instead of 200 mg in the
469 standard POCIS configuration.[45] Second, a modified POCIS design can be used to increase the
470 exposure surface area. A rectangular POCIS configuration mainly used for groundwater[46] has a
471 sampling capacity equivalent to three standard circular POCIS and would be ideal for achieving
472 ng/L range CSIA. Third, selective sorbent can be used in POCIS to increase analyte
473 preconcentration. Various commercially available sorbents, such as OASIS MCX, OASIS MAX,
474 Starta, Chromabond HR-X,[47–49] and molecularly imprinted sorbents[50,51] have been used for
475 increased and selective analyte accumulation in POCIS. Selective sorbents can potentially
476 achieve high *in situ* preconcentration as well as lower background interferences for CSIA.

477

478 **4.4 POCIS conserves isotope signatures in the field**

The long deployment time of POCIS in the field, typically between 1 to 4 weeks, can lead to biofilm growth and sediment deposition on the membrane surface. The deposition of suspended materials might cause diffusion-induced isotope fractionation, whereas the biofilm layer might affect the isotope signature due to increased diffusion barrier and potential microbial degradation of the sampled compounds. Our results from scanning electron microscopy and DNA analysis confirmed negligible biofilm formation and the presence of diverse microbial community (maximum abundance <15% for a single genus) on the exposed membrane surface (Figure S3, Section S13). In addition, we did not observe the dominance of microbes known to degrade the target compounds (Figure S3, Section S13). Thus, it is unlikely that biofilm developing on the membranes would contribute to isotope fractionation via either diffusion of chemicals through the biofilm or biotransformation.

In addition, similar microbial morphologies and community were present on the membranes from both copper and stainless-steel casings, suggesting that the copper did not prevent microbial attachment (Figure S4, Section 13), although copper mesh is often used for passive samplers to limit biofilm formation.[52,53] However, significant biofilm formation might be an issue for longer deployment time in other environmental settings. In such cases, composite extracts from multiple POCIS exposed in the water for a short time could be useful.

5. Environmental implications

Overall, our results demonstrated that POCIS are compatible with C-, N- and H-CSIA for most target compounds and can potentially be used for CSIA of other emerging polar compounds at trace levels. POCIS has been extensively used for hundreds of polar organic compounds at low

environmental concentrations[54–56] in wastewater, surface water, groundwater, marine water, wastewater treatment plants, constructed wetlands, and long-term remediation sites.[23,57] Thus, combining POCIS with CSIA has the potential to enable CSIA application to thousands of polar emerging contaminants present in the environment, such as pesticides, pharmaceuticals, and flame-retardants, possibly at low μg/L to ng/L levels. Therefore, POCIS could be a convenient sample preconcentration technique for C-, H-, and N-CSIA to understand *in situ* transformation of polar emerging contaminants in natural and engineering systems at extremely low concentrations.

The results presented here also demonstrate the importance to verify the appropriate sampling time to use POCIS for CSIA. POCIS field deployment time usually varies from one week to four weeks,[23] which aligns well with the sampling time requirement of most target compounds for CSIA. In addition, the measured isotope signature from POCIS will provide a pooled or time-integrated *in situ* information, although we did not observe any significant differences in isotope signatures obtained from POCIS and grab sampling after a 60-day deployment.

Lastly, achieving isotopic equilibrium in POCIS could depend on specific analyte-membrane-sorbent interactions and can potentially lead to significant but reproducible isotope fractionation. Thus, careful laboratory and *in situ* evaluation are recommended before applying POCIS-CSIA technique for new compounds. Predictive models could be developed to better understand the intermolecular interactions of PES and HLB with different compound groups. Such models could help evaluate the potential implications of the POCIS-CSIA technique for new compounds that might have special intermolecular interactions with HLB and PES phases.

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References

(1) Booth, G. Nitro Compounds, Aromatic. *Ullmann's Encyclopedia of Industrial Chemistry*; Wiley-VCH Verlag GmbH & Co. KGaA: Weinheim, Germany, 2000; pp 301–349.

(2) Ju, K. S.; Parales, R. E. Application of Nitroarene Dioxygenases in the Design of Novel Strains That Degrade Chloronitrobenzenes. *Microb. Biotechnol.* **2009**, *2* (2 SPEC. ISS.), 241–252.

(3) Elsner, M.; Schwarzenbach, R. P.; Haderlein, S. B. Reactivity of Fe(II)-Bearing Minerals toward Reductive Transformation of Organic Contaminants. *Environ. Sci. Technol.* **2004**, *38* (3), 799–807.

(4) Hussain, S.; Arshad, M.; Springael, D.; SøRensen, S. R.; Bending, G. D.; Devers-Lamrani, M.; Maqbool, Z.; Martin-Laurent, F. Abiotic and Biotic Processes Governing the Fate of Phenylurea Herbicides in Soils: A Review. *Crit. Rev. Environ. Sci. Technol.* **2015**, *45* (18), 1947–1998.

(5) Kaufman, D. D.; Plimmer, J. R.; Klingebiel, U. I. Microbial Oxidation of 4-Chloroahiline. *J. Agric. Food Chem.* **1973**, *21* (1), 127–132.

(6) United States Environmental Protection Agency (US EPA). Priority pollutant list https://www.epa.gov/sites/production/files/2015-09/documents/priority-pollutant-list-epa.pdf (accessed Jan 29, 2022).

(7) Spain, J. C.; Hughes, J. B.; Knackmuss, H.-J.; Hughes, J. B.; Knackmuss, H.-J. *Biodegradation of Nitroaromatic Compounds and Explosives*; Hughes, J., Spain, J., Knackmuss, H.-J., Eds.; CRC Press, 2000.

(8) Lekkas, T.; Kolokythas, G.; Nikolaou, A.; Kostopoulou, M.; Kotrikla, A.; Gatidou, G.; Thomaidis, N. S.; Golfinopoulos, S.; Makri, C.; Babos, D.; Vagi, M.; Stasinakis, A.; Petsas, A.; Lekkas, D. F. Evaluation of the Pollution of the Surface Waters of Greece from the Priority Compounds of List II, 76/464/EEC Directive, and Other Toxic Compounds. *Environ. Int.* **2004**, *30* (8), 995–1007.

(9) Schwarzbauer, J.; Ricking, M. Non-Target Screening Analysis of River Water as Compound-Related Base for Monitoring Measures. *Environ. Sci. Pollut. Res.* **2010**, *17* (4), 934–947.

(10) Hunkeler, D.; Meckenstock, R. U.; Sherwood Lollar, B.; Schmidt, T. C.; Wilson, J. T. A Guide for Assessing Biodegradation and Source Identification of Organic Ground Water Contaminants Using Compound Specific Isotope Analysis (CSIA). *USEPA Publ.* **2008**, *EPA 600/R-* (December), 1–82.

(11) Harrington, R. R.; Poulson, S. R.; Drever, J. I.; Colberg, P. J. S.; Kelly, E. F. Carbon Isotope Systematics of Monoaromatic Hydrocarbons: Vaporization and Adsorption Experiments. *Org. Geochem.* **1999**, *30* (8 A), 765–775.

(12) Slater, G. F.; Ahad, J. M. E.; Sherwood Lollar, B.; Allen-King, R.; Sleep, B. Carbon

Isotope Effects Resulting from Equilibrium Sorption of Dissolved VOCs. *Anal. Chem.* **2000**, *72* (22), 5669–5672.

(13) Xu, B. S.; Sherwood Lollar, B.; Passeport, E.; Sleep, B. E. Diffusion Related Isotopic Fractionation Effects with One-Dimensional Advective-Dispersive Transport. *Sci. Total Environ.* **2016**, *550*, 200–208.

(14) Melsbach, A.; Pittois, D.; Bayerle, M.; Daubmeier, M.; Meyer, A. H.; Hölzer, K.; Gallé, T.; Elsner, M. Isotope Fractionation of Micropollutants during Large-Volume Extraction: Heads-up from a Critical Method Evaluation for Atrazine, Desethylatrazine and 2,6-Dichlorobenzamide at Low Ng/L Concentrations in Groundwater. *Isotopes Environ. Health Stud.* **2021**, *57* (1), 35–52.

(15) Torrentó, C.; Bakkour, R.; Glauser, G.; Melsbach, A.; Ponsin, V.; Hofstetter, T. B.; Elsner, M.; Hunkeler, D. Solid-Phase Extraction Method for Stable Isotope Analysis of Pesticides from Large Volume Environmental Water Samples. *Analyst* **2019**, *144* (9), 2898–2908.

(16) Wang, Y.; Huang, Y.; Huckins, J. N.; Petty, J. D. Compound-Specific Carbon and Hydrogen Isotope Analysis of Sub-Parts per Billion Level Waterborne Petroleum Hydrocarbons. *Environ. Sci. Technol.* **2004**, *38* (13), 3689–3697.

(17) Goli, O.; Górecki, T.; Mugammar, H. T.; Marchesi, M.; Aravena, R. Evaluation of the Suitability of the Waterloo Membrane Sampler for Sample Preconcentration before Compound-Specific Isotope Analysis. *Environ. Technol. Innov.* **2017**, *7*, 141–151.

(18) BenIsrael, M.; Wanner, P.; Aravena, R.; Parker, B. L.; Haack, E. A.; Tsao, D. T.; Dunfield, K. E. Toluene Biodegradation in the Vadose Zone of a Poplar Phytoremediation

System Identified Using Metagenomics and Toluene-Specific Stable Carbon Isotope Analysis. *Int. J. Phytoremediation* **2019**, *21* (1), 60–69.

(19) Passeport, E.; Landis, R.; Mundle, S. O. C.; Chu, K.; Mack, E. E.; Lutz, E.; Sherwood Lollar, B. Diffusion Sampler for Compound Specific Carbon Isotope Analysis of Dissolved Hydrocarbon Contaminants. *Environ. Sci. Technol.* **2014**, *48* (16), 9582–9590.

(20) Passeport, E.; Landis, R.; Lacrampe-Couloume, G.; Lutz, E. J.; Erin Mack, E.; West, K.; Morgan, S.; Sherwood Lollar, B. Sediment Monitored Natural Recovery Evidenced by Compound Specific Isotope Analysis and High-Resolution Pore Water Sampling. *Environ. Sci. Technol.* **2016**, *50* (22), 12197–12204.

(21) Gilevska, T.; Passeport, E.; Shayan, M.; Seger, E.; Lutz, E. J.; West, K. A.; Morgan, S. A.; Mack, E. E.; Sherwood Lollar, B. Determination of in Situ Biodegradation Rates via a Novel High Resolution Isotopic Approach in Contaminated Sediments. *Water Res.* **2019**, *149*, 632–639.

(22) Alvarez, D. A.; Petty, J. D.; Huckins, J. N.; Jones-Lepp, T. L.; Getting, D. T.; Goddard, J. P.; Manahan, S. E. Development of a Passive, in Situ, Integrative Sampler for Hydrophilic Organic Contaminants in Aquatic Environments. *Environ. Toxicol. Chem.* **2004**, *23* (7), 1640–1648.

(23) Alvarez, D. A. Guidelines for the Use of the Semi Permeable Membrane Device (SPMD) and the Polar Organic Chemical Integrative Sampler (POCIS) in Environmental Monitoring. *Tech. Methods* **2010**, 1–D4, 28 p.

(24) Grass, G.; Rensing, C.; Solioz, M. Metallic Copper as an Antimicrobial Surface. *Appl. Environ. Microbiol.* **2011**, *77* (5), 1541–1547.

(25) Sherwood Lollar, B.; Hirschorn, S. K.; Chartrand, M. M. G.; Lacrampe-Couloume, G. An Approach for Assessing Total Instrumental Uncertainty in Compound-Specific Carbon Isotope Analysis: Implications for Environmental Remediation Studies. *Anal. Chem.* **2007**, *79* (9), 3469–3475.

(26) Brand, W. A.; Coplen, T. B. Stable Isotope Deltas: Tiny, yet Robust Signatures in Nature. *Isotopes Environ. Health Stud.* **2012**, *48* (3), 393–409.

(27) Vermeirssen, E. L. M.; Dietschweiler, C.; Escher, B. I.; Van Der Voet, J.; Hollender, J. Transfer Kinetics of Polar Organic Compounds over Polyethersulfone Membranes in the Passive Samplers Pocis and Chemcatcher. *Environ. Sci. Technol.* **2012**, *46* (12), 6759–6766.

(28) Belles, A.; Pardon, P.; Budzinski, H. Development of an Adapted Version of Polar Organic Chemical Integrative Samplers (POCIS-Nylon). *Anal. Bioanal. Chem.* **2014**, *406* (4), 1099–1110.

(29) Wanner, P.; Hunkeler, D. Carbon and Chlorine Isotopologue Fractionation of Chlorinated Hydrocarbons during Diffusion in Water and Low Permeability Sediments. *Geochim. Cosmochim. Acta* **2015**, *157*, 198–212.

(30) Bope, C. D.; Nalaparaju, A.; Ng, C. K.; Cheng, Y.; Lu, L. Molecular Simulation on the Interaction of Ethinylestradiol (EE2) with Polymer Membranes in Wastewater Purification. *Mol. Simul.* **2018**, *44* (8), 638–647.

(31) Endo, S.; Matsuura, Y. Characterizing Sorption and Permeation Properties of Membrane Filters Used for Aquatic Integrative Passive Samplers. *Environ. Sci. Technol.* **2018**, *52* (4), 2118–2125.

(32) Ng, C. K.; Bope, C. D.; Nalaparaju, A.; Cheng, Y.; Lu, L.; Wang, R.; Cao, B. Concentrating Synthetic Estrogen 17A-Ethinylestradiol Using Microporous Polyethersulfone Hollow Fiber Membranes: Experimental Exploration and Molecular Simulation. *Chem. Eng. J.* **2017**, *314*, 80–87.

(33) Schäfer, A. I.; Akanyeti, I.; Semião, A. J. C. Micropollutant Sorption to Membrane Polymers: A Review of Mechanisms for Estrogens. *Adv. Colloid Interface Sci.* **2011**, *164* (1–2), 100–117.

(34) Cockroft, S. L.; Hunter, C. A.; Lawson, K. R.; Perkins, J.; Urch, C. J. Electrostatic Control of Aromatic Stacking Interactions. *J. Am. Chem. Soc.* **2005**, *127* (24), 8594–8595.

(35) Cockroft, S. L.; Perkins, J.; Zonta, C.; Adams, H.; Spey, S. E.; Low, C. M. R.; Vinter, J. G.; Lawson, K. R.; Urch, C. J.; Hunter, C. A. Substituent Effects on Aromatic Stacking Interactions. *Org. Biomol. Chem.* **2007**, *5* (7), 1062–1080.

(36) Dias, N. C.; Poole, C. F. Mechanistic Study of the Sorption Properties of Oasis® HLB and Its Use in Solid-Phase Extraction. *Chromatographia* **2002**, *56* (5–6), 269–275.

(37) Bäuerlein, P. S.; Mansell, J. E.; Ter Laak, T. L.; De Voogt, P. Sorption Behavior of Charged and Neutral Polar Organic Compounds on Solid Phase Extraction Materials: Which Functional Group Governs Sorption? *Environ. Sci. Technol.* **2012**, *46* (2), 954–961.

(38) Kopinke, F. D.; Georgi, A.; Voskamp, M.; Richnow, H. H. Carbon Isotope Fractionation of Organic Contaminants Due to Retardation on Humic Substances: Implications for Natural Attenuation Studies in Aquifers. *Environ. Sci. Technol.* **2005**, *39* (16), 6052–6062.

(39) Imfeld, G.; Kopinke, F. D.; Fischer, A.; Richnow, H. H. Carbon and Hydrogen Isotope Fractionation of Benzene and Toluene during Hydrophobic Sorption in Multistep Batch Experiments. *Chemosphere* **2014**, *107*, 454–461.

(40) Wanner, P.; Parker, B. L.; Chapman, S. W.; Aravena, R.; Hunkeler, D. Does Sorption Influence Isotope Ratios of Chlorinated Hydrocarbons under Field Conditions? *Appl. Geochemistry* **2017**, *84*, 348–359.

(41) Świderek, K.; Paneth, P. Binding Isotope Effects. *Chem. Rev.* **2013**, *113* (10), 7851–7879.

(42) Rechavi, D.; Scarso, A.; Rebek, J. Isotopomer Encapsulation in a Cylindrical Molecular Capsule: A Probe for Understanding Noncovalent Isotope Effects on a Molecular Level. *J. Am. Chem. Soc.* **2004**, *126* (25), 7738–7739.

(43) Laughrey, Z. R.; Upton, T. G.; Gibb, B. C. A Deuterated Deep-Cavity Cavitand Confirms the Importance of C-H···X-R Hydrogen Bonds in Guest Binding. *Chem. Commun.* **2006**, No. 9, 970–972.

(44) Berg, M.; Bolotin, J.; Hofstetter, T. B. Compound-Specific Nitrogen and Carbon Isotope Analysis of Nitroaromatic Compounds in Aqueous Samples Using Solid-Phase Microextraction Coupled to GC/IRMS. *Anal. Chem.* **2007**, *79* (6), 2386–2393.

(45) Fauvelle, V.; Mazzella, N.; Belles, A.; Moreira, A.; Allan, I. J.; Budzinski, H. Optimization of the Polar Organic Chemical Integrative Sampler for the Sampling of Acidic and Polar Herbicides. *Anal. Bioanal. Chem.* **2014**, *406* (13), 3191–3199.

(46) Environmental Sampling Technologies. Environmental Sampling Technologies - The Leader in Polar Organic Chemical Integrative Samplers (POCIS) https://est-

lab.com/pocis.php (accessed Feb 2, 2022).

(47) Li, H.; Helm, P. A.; Paterson, G.; Metcalfe, C. D. The Effects of Dissolved Organic Matter and PH on Sampling Rates for Polar Organic Chemical Integrative Samplers (POCIS). *Chemosphere* **2011**, *83* (3), 271–280.

(48) Fauvelle, V.; Mazzella, N.; Delmas, F.; Madarassou, K.; Eon, M.; Budzinski, H. Use of Mixed-Mode Ion Exchange Sorbent for the Passive Sampling of Organic Acids by Polar Organic Chemical Integrative Sampler (POCIS). *Environ. Sci. Technol.* **2012**, *46* (24), 13344–13353.

(49) Kaserzon, S. L.; Kennedy, K.; Hawker, D. W.; Thompson, J.; Carter, S.; Roach, A. C.; Booij, K.; Mueller, J. F. Development and Calibration of a Passive Sampler for Perfluorinated Alkyl Carboxylates and Sulfonates in Water. *Environ. Sci. Technol.* **2012**, *46* (9), 4985–4993.

(50) Berho, C.; Claude, B.; Coisy, E.; Togola, A.; Bayoudh, S.; Morin, P.; Amalric, L. Laboratory Calibration of a POCIS-like Sampler Based on Molecularly Imprinted Polymers for Glyphosate and AMPA Sampling in Water. *Anal. Bioanal. Chem.* **2017**, *409* (8), 2029–2035.

(51) Berho, C.; Robert, S.; Coureau, C.; Coisy, E.; Berrehouc, A.; Amalric, L.; Bruchet, A. Estimating 42 Pesticide Sampling Rates by POCIS and POCIS-MIP Samplers for Groundwater Monitoring: A Pilot-Scale Calibration. *Environ. Sci. Pollut. Res.* **2020**, *27* (15), 18565–18576.

(52) Alvarez, D. A.; Maruya, K. A.; Dodder, N. G.; Lao, W.; Furlong, E. T.; Smalling, K. L. Occurrence of Contaminants of Emerging Concern along the California Coast (2009-10)

Using Passive Sampling Devices. *Mar. Pollut. Bull.* **2014**, *81* (2), 347–354.

(53) Vrana, B.; Allan, I. J.; Greenwood, R.; Mills, G. A.; Dominiak, E.; Svensson, K.; Knutsson, J.; Morrison, G. Passive Sampling Techniques for Monitoring Pollutants in Water. *TrAC - Trends Anal. Chem.* **2005**, *24* (10), 845–868.

(54) Harman, C.; Allan, I. J.; Vermeirssen, E. L. M. Calibration and Use of the Polar Organic Chemical Integrative Sampler-a Critical Review. *Environ. Toxicol. Chem.* **2012**, *31* (12), 2724–2738.

(55) Alvarez, D.; Perkins, S.; Nilsen, E.; Morace, J. Spatial and Temporal Trends in Occurrence of Emerging and Legacy Contaminants in the Lower Columbia River 2008-2010. *Sci. Total Environ.* **2014**, *484* (1), 322–330.

(56) Fent, K.; Zenker, A.; Rapp, M. Widespread Occurrence of Estrogenic UV-Filters in Aquatic Ecosystems in Switzerland. *Environ. Pollut.* **2010**, *158* (5), 1817–1824.

(57) Vallejo, A.; Prieto, A.; Moeder, M.; Usobiaga, A.; Zuloaga, O.; Etxebarria, N.; Paschke, A. Calibration and Field Test of the Polar Organic Chemical Integrative Samplers for the Determination of 15 Endocrine Disrupting Compounds in Wastewater and River Water with Special Focus on Performance Reference Compounds (PRC). *Water Res.* **2013**, *47* (8), 2851–2862.