New Strategies for Analysis of Particulate Loaded Water

by

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AUTHOR'S DECLARATION

I hereby declare that I am the sole author of this thesis. This is a true copy of the thesis, including any required final revisions, as accepted by my examiners.

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Abstract

This thesis describes the application of thin film solid phase microextraction (TF-SPME) for monitoring of time weighted average (TWA) concentrations and for investigating partitioning of UV filters and biocides in aquatic environments. The occurrence, distribution, fate, and transport of ingredients of personal care products in the aquatic environment are a subject of public concern, as their impact on human life and aquatic organisms has yet to be fully understood. As such, simple and reliable analytical techniques are necessary to monitor TWA concentrations and investigate the partitioning of UV filters and biocides on sediment and humic substances in aquatic environments. In the currently presented work, time weighted average (TWA) passive sampling with TF-SPME and liquid chromatography tandem mass spectrometry (LC-MS/MS) was used for the collection, identification, and quantification of UV filters and biocides in aquatic environment. First, a reliable flow-through aqueous standard generation (ASG) system was constructed to provide a steady state concentration, and also address issues related to in-vial preparation of aqueous standards of hydrophobic compounds, such as partial precipitation, loss due to analyte adhesion to surfaces, low solubility in water, and limited sample volumes. The developed ASG offers many advantages over traditional standard preparation methods, including convenient, inexpensive, solvent-free, and long term generation of analytes in the concentration ranges relevant to environmental values. The observed changes in the concentrations of target analytes were less than 20% within a 2 month span. The developed system was used subsequently for development of a TWA sampler, and for binding investigations using TF-SPME. Two types of TF-SPME passive samplers,

including a thin film-retracted device using a hydrophilic lipophilic balance coating (HLB) sorbent and an open bed configuration with octadecyl silica-based (C18) particles as extraction phases, were evaluated in the ASG. Laboratory calibration of the retracted device using the HLB coating showed a linear uptake of up to 70 days. In open bed configuration, the one-calibrant kinetic calibration technique was successfully applied by loading Benzophenone3-d5 as calibrant on the C18 coating to quantify all hydrophobic compounds. The experimental results showed that one-calibrant kinetic calibration can be used for determination of classes of compounds where deuterated counterparts are either not available or expensive. The developed passive samplers were deployed in wastewater-dominated reaches of the Grand River (Kitchener, ON) to verify their feasibility for determination of TWA concentrations in on-site applications. Field trials results indicated that these devices are suitable for long- and short-term monitoring of compounds varying in polarity, such as UV filters and biocide compounds in water. In addition, partitioning of UV filters and biocides on humic acid was investigated in the developed ASG. ASG enables the provision of large volumes of sample, an essential criteria when performing negligible depletion extraction by TF-SPME. Extraction time profiles of the analytes in humic acid-free and humic acid solutions were investigated, and matrix effects were observed for highly bonded analytes. The binding percentages and distribution coefficients (K_{DOC}) between the studied analytes and Sigma Aldrich humic acid were determined when the free and total concentrations of analytes as well as the concentration of dissolved organic carbon (DOC) were known. TF-SPME and liquid-liquid extraction were used respectively for measuring the free and total concentrations of analytes in the humic acid

solution. Furthermore, sediment-water partition coefficient K_d were also investigated in the batch mode for the analytes. The linear isotherm model was observed over a small concentration range and the analyte concentrations in sediment phase. TF-SPME using HLB and C18 sorbent was used for measuring $C_{W\text{-}SPME}$ instead of $C_{W\text{-}LLE}$. The data showed that the hydrophobic analytes have substantial association to the bed sediment and DOC in sediment porewater. This has several consequences on determination of fate, transport, and ecotoxicological effects by hydrophobic analytes. However, the low partitioning coefficients of hydrophilic analytes on sediment and DOC imply that these compounds are freely dissolved predominantly, which can have an adverse consequence on the quality of drinking water.

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Dedication

To my loving parents, my wife and Delsa

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List of Abbreviations

a- Time constant

A- Cross-sectional area of the needle opening

b- Thickness of the coating

BP- Binding percentage

C18-Octadecyl

C_f^{∞}- Equilibrium concentration of analyte on the

$C_{f\,max}$ - Maximum concentration of active sites on the coating

C_0- Initial analyte concentration

C_s -Analyte concentration in the bulk of the sample

CE- Collision energy

CID- Collision induced dissociation

CXP- Collision cell exit potential

d- Depth of the blade substrate

DP - Declusting potential

d - Days

Ds -Diffusion coefficient of analyte in the sample matrix

DI-Direct immersion

DOC- Dissolved organic carbon

EP- Entrance potential

DVB- Divinylbenzene

ESI- Electrospray ionization

ISTD- Internal standard Fibre- extraction phase

GC–Gas chromatography

HPLC- High performance liquid chromatography

HLB- Hydrophilic–lipophilic balance particles

HAB- Humic acid binding

J- Mass flux

K- Equilibrium constant of the analyte between the solid fiber surface and the sample matrix - Distribution coefficient of the analyte between the liquid fiber and the sample matrix

K_d- Sorption coefficient of the analyte between the sediment and the sample matrix

K_{DOC}- Distribution coefficient of the analyte between the dissolved organic carbon and the sample matrix

LC- Liquid chromatography

l- Length of the coating

LOD- Limit of detection

Log Kow- log of octanol/water partition coefficient

LOQ- Limit of quantitation

LLE- Liquid-liquid extraction

MRM- Multiple reaction monitoring mode

MDL- Method detection limit

MS- Mass spectrometer/spectrometry

MS/MS- Tandem mass spectrometry

MW- Molecular weight

MWCO- Molecular weight cut off

n- Amount of analyte extracted by SPME fibre at a specific time

n_e -Amount of analyte extracted by SPME fibre at equilibrium

NA- Not available

ND- Not detected

PAN- Polyacrylonitrile

Ppb- Parts per billion

Ppm- Parts per million

PS-DVB-WAX- Modified polystyrene-divinylbenzene copolymer with a weak anion exchanger

PPCPs- Pharmaceuticals and personal care products

POCIS- Polar organic chemical integrative sampler

PDMS- Polydimethylsiloxane

PSDs- Passive sampling devices

PTFE- Polytetrafluoroethylene

q_0- Amount of calibrant preloaded on fibre

Q- Amount of calibrant remaining on the fibre after extraction

QC -Quality control

r- Radius of the support wire

Rpm- Revolutions per minute

R^2- Linear regression coefficient

RSD- Relative standard deviation

s- Second

ASG- Aqueous standard generation

SPE- Solid phase extraction

SRM- Selected reaction monitoring mode

SPME- Solid-phase microextraction

S/N-Signal-to-noise ratio

SR- Sampling rate

TWA- Time weighted average

t- Sampling time

TF-SPME-Thin film solid phase microextraction

Z- Diffusion path length

t- Sampling time

V_f- Volume of fibre coating

Vs -Sample volume

w- Width of the coating substrate

δs- The thickness of the boundary layer

Chapter 1
Introduction

1.1 Emerging contaminants in aquatic environment

In industrial countries, the focus of interest of environmental scientists, government agencies, and public concern has gradually changed from conventional contamination, such as non-polar organic pollutants and heavy metals, to newly-emerging, unregulated contaminants.[1] According to the United States Environmental Protection Agency (USEPA), "emerging contaminants" can be classify as synthetic or naturally occurring chemicals or microorganisms that are not normally monitored in the environment, but have the potential to go into the environment and lead to known or suspected harmful effects on ecological or human health.[2] They consist of a large group of chemical compounds such as human and veterinary pharmaceuticals, personal care products, surfactants, surfactant residue, plasticizers, and a large number of industrial additives, all of which are used in large quantities in everyday life. [3] Of these contaminants, one of the most important classes is comprised of UV filters and biocides, which are mainly used for health and cosmetic purposes.[4] They enter the environment by two main input pathways, namely direct and indirect routes, through recreational release (e.g., swimming, showering) and wastewater treatment facilities, respectively.[5–8] Most operating wastewater-treatment plants (WWTP) are not designed to eliminate these contaminants completely, causing them to eventually enter the aquatic environment via sewage effluent, and reach detectable and potential harmful concentrations in receiving surface waters.[9–11] Great effort has been devoted to the introduction of sensitive analytical methods

for determination of trace and ultra-trace levels, and well as to develop protocols to quantify the incidence of occurrence, conduct risk assessments, and produce ecotoxicological data regarding this emerging contamination in aquatic environments. [3,12–20]

1.2 Sampling methods for water analysis

The main step for obtaining relevant environmental information, as well as the most important source of measurement uncertainty is sampling. Although, sample volume reduction is in the interest of the scientific community, the taken sample need to be representative sample. In this respect, obtained results from laboratory analyses may not be representative of the body under study due to inadequate or improper sample collection, and thus, not be validated.[21] Erroneous results can be generated if the sampling plan is poorly designed, or the wrong sampling equipment chosen, both which cannot be corrected post-sampling. Some of the main factors that need to be considered when developing a sampling plan include objectives, sample variability, analytical cost, and other non-technical factors. A realistic assessment of water pollution is conducted with rational preparation, careful decisions, proper equipment cleaning, and appropriate collection of water samples. Not only sample collection but also information related to various features of water quality should be taken into consideration and comprised in the information about the water being sampled. Parameters such as conductivity, temperature, and dissolved oxygen concentration, to name a few, need to be carefully taken into consideration for data analysis. Two types of water sampling strategies, in terms of timespan technology, can be applied towards sample collection: (1) discrete sampling and (2) composite sampling. A discrete sample, or grab sample, is a single sample collected in a

particular container. The sample is representative of the chemistry just at the time of sampling and only for the sampling location. The time frame for such sampling is normally less than 15 min. As such, discrete samples are mostly suitable for sample compositions that are not dependent on time. Conversely, a composite sample is composed of a number of small samples collected at a pre-set time or after a predetermined flow, and mixed together in the same bottle. Discrete and composite samplings can be achieved either through manual or automatic grab/spot sampling methods.[22,23] Traditional spot sampling methods may not provide a realistic picture of continuous exposure of aquatic-life to organic contamination for prolonged periods of time. When the level of water fluctuates (input of influence) within a water body, a large number of samples need to be collected over a long time period so as to obtain a time weighted average (TWA) water concentration, a process which is time consuming, laborious, and costly. [24-25] Passive samplers, as an alternative option, are widely used for monitoring of time weighted average (TWA) concentrations of environmental contaminants in water.[26,27] Passive samplers are human-made devices where sample collection and extraction happen at the same time in a fully passive way.[28] Principally, the sampler is placed in the body of water of interest, and left to accumulate target analytes, normally for a period of several weeks. As the passive sampling devices (PSDs) are placed in water for a long period of time, the issue of representative sampling is addressed in most circumstances. The passive sampling approach is based on the free movement of analytes from sample matrix to the extraction phase. The difference in the chemical potential of the analytes between the two media causes accumulation of the analytes into the extraction phase. [29-30] Passive sampling technology offers many substantial

advantages, such as simplicity, low cost, unattended operation, unlimited depth sampling, the ability to produce reliable data, in addition to not requiring expensive and complicated equipment or a power supply.[31] Moreover, passive sampling can integrate sampling collection, purification, and pre-concentration in a single step.[32] There are several passive sampling methods used for monitoring of contamination in water, including semi-permeable membrane devices (SPMDs),[33,34] polar organic chemical integrative sampler (POCIS),[35,36] chemcatcher,[24,37] ceramic dosimeter,[27,38] polymeric samplers,[39,40] diffusive gradient in thin film,[41,42] and membrane enclosed sorptive coating.[43]

1.3 Fate and transport in aquatic environment

Natural organic matter and sediment are ubiquitous matrix components of surface water. Natural organic matter is composed of a mixture of macromolecules originating from the biological and chemical degradation of plants or animals. In the aquatic environment, dissolved organic matter (DOM) is defined as organic macromolecules that can pass through the 1µm filter.[44] DOM contain of humic acid and fulvic acid. Fulvic acid is the soluble part in acidic and basic solutions, and humic acid is the non-soluble part at pH=2 which account for about two thirds and one third of DOM, respectively.[45] Humic substances are considered polyelectrolytes due to the presence of carboxylic and phenolic functional groups that also contain nonpolar regions. Spatial separation of these polar and nonpolar sites allows binding to organic molecules through ionic, hydrogen, and covalent bonding, charge transfer or electron donor–acceptor mechanisms, Van der Waals forces, ligand exchange, and hydrophobic bonding or partitioning.[46] Sorption of organic pollutants to sediment is dependent

on their molecular structure, the sediment properties, and the specific ionic composition of the aqueous phase.[47–49] The sorption behavior of organic pollutants is a relatively hydrophobicity-driven process, as demonstrated by the increase in sorption coefficient values with increasing hydrophobicity.[48] In addition, the relationship that exists between sorption affinity and the organic carbon content of natural sorbents displays that hydrophobic interactions are significant.[52,53]

The freely dissolved concentration of chemical compounds is very important in environmental chemistry, pharmacology, and toxicology. Free concentrations are the main driving force in the transport, distribution, and bioaccumulation of chemicals in the environment.[54] Consequently, free concentration is a basic factor in the interpretation of the bioavailability of a chemical. Accordingly, the binding or partitioning of chemicals to dissolved organic carbon (DOC) and sediment can decrease the free concentration and bioavailability or effectivity of a chemical.[44,55–58] In addition, sorption also affects the biological availability, therefore influencing bioaccumulation, biodegradation, and potential toxic effects in organisms.[59,60] Consequently, information about the degree of binding or partitioning of chemical compounds is needed in several scientific disciplines, as evidenced by the increase in studies directed towards the quantification of partition coefficients or binding affinities. These constants can be determined by quantifying either the bound or free concentrations of the chemical. Therefore, determination of free concentrations is necessary not only to study the bioavailability of a chemical, but also to measure the binding affinity or partition coefficient of a compound. Several methods have been used for determination of free concentrations,

including equilibrium dialysis, ultrafiltration, ultracentrifugation, reverse-phase chromatography, protein precipitation, fluorescence quenching, headspace analysis, and empore disk.[61–66] Although these methods are definitely useful, they are not suitable for determinations of all chemical compounds. In addition to their relative laboriousness, a main problem associated with the abovementioned methods is the loss of compounds that occurs due to filter binding or desorption of compounds from the binding agent, which results in a shift of equilibrium between bound and free compounds.[67,68]

1.4 Solid Phase Microextraction (SPME)

Solid-phase microextraction (SPME) is an equilibrium-based, solvent-free sample preparation technique that integrates sampling, extraction, and pre-concentration in a single step. [69] The SPME technique was established to address the need for fast sampling and sample preparation in the laboratory, and during on-site sampling. The first application of SPME was mostly focused on semi-volatile compound analyses coupled to gas chromatography (GC). Within the last two decades, however, its applications have expanded towards the determination of a wide range of substances, within a multitude of fields and disciplines. Coupled to liquid chromatography (LC), SPME has been applied towards the determination of pharmaceuticals in wastewater samples,[70] quaternary ammonium compounds in water samples, [71] phenolic compounds in wine, berry, and grape samples,[72] and prohibited substances in urine and plasma.[73,74] In this technique, a small amount of extractive phase immobilized on a solid substrate is exposed to the sample matrix for a specific period of time. The transport of analytes from the sample matrix to the fiber coating is initiated immediately after contact between the

two phases through the boundary layer until equilibrium is reached between the two phases (Figure 1-1)[75]. After extraction, the analytes concentrated in the coating are desorbed via thermal desorption in the injector of the instrument (i.e., GC application), or through solvent desorption (e.g., LC applications).

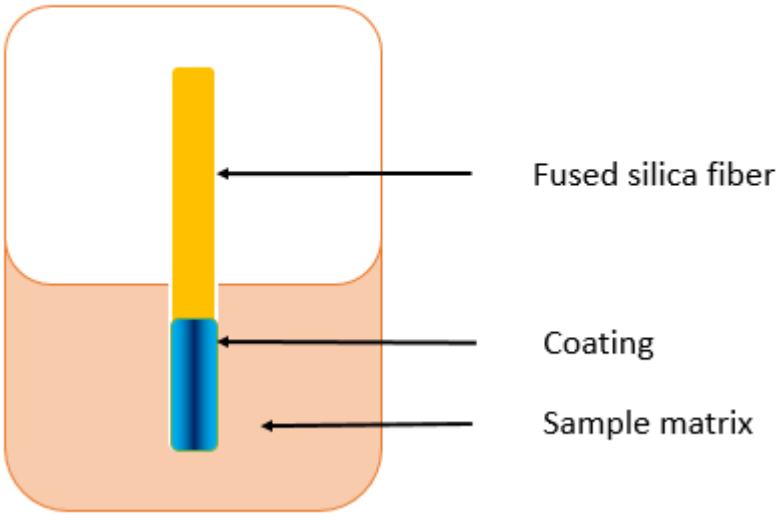


Figure 1.1 Schematic of equilibrium extraction of analyte with SPME in sample matrix

1.4.1 Introduction to the fundamentals of solid phase microextraction

Based on the fundamentals of SPME, the amount of extracted analytes is proportional to the volume of extraction phase, as shown in Equation 1.1:

$$n = \frac{K_{fs} V_f V_s C_0}{K_{fs} V_f + V_s}$$ Equation 1.1

where n is the amount of analyte extracted at equilibrium, K_{fs} is the partition coefficient between the extraction phase and the sample matrix, V_f is the volume of extraction phase, V_s is the volume of sample, and C_0 is the original concentration of the analyte. For large sample volumes, $K_{fs}V_f << V_s$, and equation 1.1 can be simplified to Equation 1.2:

$$n = K_{fs}V_f C_0 \qquad \text{Equation 1.2}$$

Here, K_{fs} is defined as the ratio of analyte concentration on the SPME coating to the analyte concentration in the sample matrix in equilibrium, as exemplified in Equation 1.3:

$$K_{fs} = \frac{C_f^{\infty}}{C_s^{\infty}} \qquad \text{Equation 1.3}$$

While the K_{fs} value depends on the nature of the analyte and selected coating, it can also be affected by experimental conditions such as pH, temperature, organic solvent, and ionic strength; as such, it is important that these parameters are kept constant throughout experiments. The principle of SPME is based on the interactions between analytes in the sample matrix and the fiber coating via absorption or adsorption and depends on either the solid or liquid coating that is used. In liquid coating, the extraction of analytes from the sample matrix into the liquid coating is based on the partition in which the analytes are absorbed by the coating. In this case if the coating is thin and a long extraction time is applied, the molecule can diffuse to the whole volume of the coating. On the other hand, solid coatings have a "glassy" or "crystalline" structure that allows for extraction of analytes through physical trapping/interactions that occur in the pores or high surface areas of the coating; consequently, analytes can only be adsorbed on the pores, and do not diffuse within the whole volume of the coating. The micropores (<20 Å) and mesopores (20–500 Å) of solid coatings can trap small

and medium size analytes, and are generally capable of retaining analytes until energy is applied, or a desorption solvent used to displace analytes. Conversely, macropores (> 500 Å), which are primarily found on the surface of coatings, can adsorb larger analytes via different interactions such as hydrogen bonding, pi-pi bonding, or van der Waals. One of the disadvantages associated with solid coatings is the limited site of adsorption on the coating surface, as there is always a competitive mechanism between analytes with different affinities toward the coating. When longer extraction times are used, low-affinity analytes are displaced either by analytes with high affinity, or those that are present in the sample solution in high concentrations.

The amount of analyte extracted by the solid coating at equilibrium, n_e, is obtained with Equation 1.4:

$$n_e = \frac{C_0 K_{Afs} V_s V_f \left(C_{fmax} - C_f^{\infty}\right)}{V_s + K_{Afs} V_f \left(C_{fmax} - C_f^{\infty}\right)} \qquad \text{Equation 1.4}$$

Where $C_{f\,max}$ is the maximum concentration of active sites on the coating, C_f^{∞} is the equilibrium concentration of analyte on the coating surface, and K_{Afs} is the analyte's adsorption equilibrium constant, which is defined as the ratio of surface concentration of analytes on the porous solid extractive phase to the concentration of analytes in the sample matrix at equilibrium. It can be concluded from the Equation 1.4 that if $C_f^{\infty} << C_{f\,max}$, then the number of occupied sites is low, and the amount of analyte extracted is linearly proportional to initial sample concentration. On the other hand, if analyte concentrations are sufficiently high, saturation of the coating surface can occur, causing adsorption isotherms to become nonlinear (Figure 1.2).[76]

1.4.1.1 Kinetics of solid phase microextraction

Equilibrium time is defined as the time when 95% of the extracted amount in equilibrium is extracted from a sample matrix. The time required to reach equilibrium is defined by Equation 1.5. The kinetics of extraction defines the speed of the extraction process in SPME. The kinetics theory recognises the extraction rate as the "bottleneck" of SPME, and designates some strategies for enhancing the rate of extraction.[73] All diffusion proccesses are assumed to be governed according to Fick's first law of diffusion. The extraction of analytes from a homogeneous sample to a liquid polymer extractive phase can be designed under three conditions: (1) perfect agitation (2) static conditions (3) practical agitation. Under the perfect agitation condition, the aqueous phase is moved rapidly in comparison to the SPME fiber, and all of the analytes have equal access to the coating. As the coating is continuously in contact with the fresh solution, the speed of extraction is determined by the diffusion of analytes into the polymeric coating. The time needed to reach equilibrium in perfect conditions is determined by Equation 1.5:

$$t_e = t_{95\%} = \frac{2\,(r_0 - r_i)^2}{D_f} \qquad \text{Equation 1.5}$$

Where r_i is the inner radius of the fiber coating, r_0 is the outer radius of the fiber coating, $(r_0 - r_i)$ is the fiber coating thickness, and D_f is the analyte diffusion coefficient in the fiber coating. In static conditions, the analytes must first diffuse through an extended boundary layer in the sample solution, and then through the fiber coating. In this situation, the mass transfer of analytes from the sample solution to the fiber coating is determined by the diffusion in the ever-broadening boundary layer; consequently, long extraction times are anticipated.

However, the extraction rate can be improved by applying agitation, as agitation allows the mass transport to increase. Under particle agitation, a boundary layer is always present around the coating. As the distance from the surface of the coating increases, the movement of fluid increases accordingly, until it is equal to the bulk flow in the sample. According to the kinetic theory of extraction, when extraction rate is controlled by diffusion of the analyte in the boundary layer, equilibration time can be estimated by the Equation 1.6:

$$t_e = t_{95\%} = \frac{3\ K_{fs}\,(b-a)\delta}{D_s} \qquad \text{Equation 1.6}$$

Where δ represents the thickness of the boundary layer, D_s represents the diffusion coefficient of the analyte in the sample matrix, (b - a) represents the coating thickness, and K_{fs} represents the distribution constant. The boundary layer thickness is determined by both the agitation conditions and the diffusion coefficient of the analyte in the sample matrix. According to Equation 1.6 equilibrium time depends on convection conditions, the distribution constant, the physicochemical properties of the analytes, and thickness of the SPME coating. So, efficient agitation and a thin coating are recommended to shorten the equilibrium time and sample throughput. It is worth to note that analytes with high K_{fs} values will have longer equilibrium times, even in cases where the boundary layer is very thin under high agitation conditions.

1.4.2 Thin-film microextraction

Thin film solid phase microextraction has been introduced to overcome the low sensitivity issue of traditional SPME fibers that result from the small amount of extraction phase on

(a)

(b)

(c)

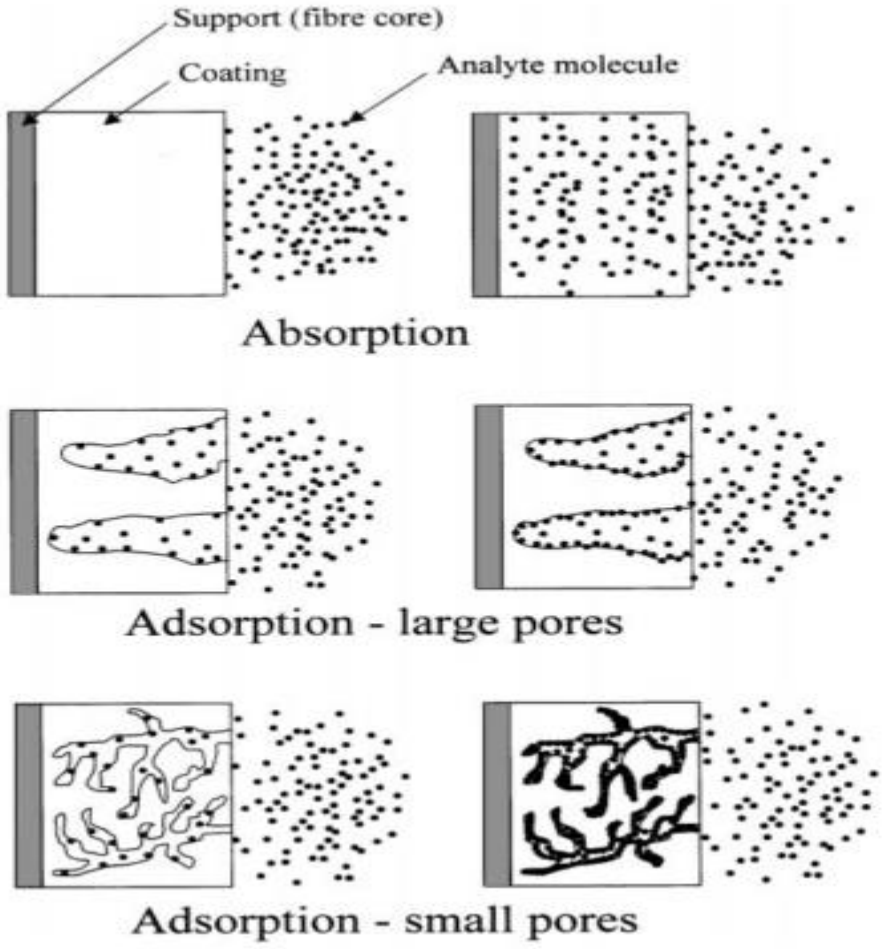


Figure 1.2 Sorption mechanisms for (a) liquid SPME coating, (b) solid SPME coating with large pores, and (c) solid SPME coatings with small pores. Figure reprinted from reference 76 with the permission of the publisher

the fiber. This drawback is more pronounced in cases where extractions are performed from complex matrices with a high degree of binding. Accordingly, efforts have been expanded to improve the sensitivity of the SPME method. According to the fundamental principle of SPME (Equation 1.2), the amount of extracted analytes is proportional to the

amount of extraction phase. As such, extraction amounts can be increased by both increasing the extraction phase volume and increasing the surface area of the extraction phase. [77-78] As previously stated, increasing the thickness of the extraction phase also results in an increase in extraction time; as such, the best way to increase the extraction phase volume, and thus the sensitivity of the method, without compromising extraction time, is to increase the surface area of the extraction phase. Thin film geometry has two main advantages over the traditional SPME fiber. First, a thinner SPME coating improves the convection conditions and the mass transfer of analytes, which leads to faster equilibrium extraction time. Secondly, the thin film geometry provides high initial mass uptake rates proportional to the surface areas, which are beneficial when performing pre-equilibrium extraction (Equation 1.7).

$$\frac{dn}{dt} = \frac{D_S A}{\delta} C_s \qquad \text{Equation 1.7}$$

In Equation 1.7 dn/dt is the rate of extraction, A is the surface area of the extraction phase, δ is the thickness of the boundary layer, D_s is the diffusion coefficient of the analyte in the sample matrix, C_s represents the initial concentration of analyte in the sample matrix, and t is the extraction time. Based on the equation above, given the same extraction time, thin film is able to extract more analyte in comparison to SPME fiber. Cudjoe et al. has demonstrated that extraction using C18 particles immobilized on the flattened end of a stainless steel support improved extraction efficiency by a factor of 2.[79]

1.4.3 Calibration in SPME

Calibration is an action that correlates the instrument signal to the concentration of the analytes in the sample matrix. As SPME is a non-exhaustive extraction method, selection of the

appropriate calibration method is very important in obtaining accurate quantitative results. Several calibration strategies have been developed for SPME, including traditional calibration (external, standard addition and internal calibration), equilibrium extraction, pre-equilibrium extraction, exhaustive extraction, and diffusion-based calibration. Diffusion-based calibration methods are based on Fick's first law of diffusion, interface and cross-flow models, and kinetic calibration methods (standard in-fiber and standard-free calibration).[80]

1.4.3.1 **Equilibrium extraction**

Equilibrium extraction is the most commonly used method for quantitation in SPME, particularly for field sampling. In this method, the SPME coating or sorbent is exposed to the sample matrix until equilibrium is reached. The amount of extracted analyte in equilibrium is directly proportional to the analyte concentration in the sample when the two phases are considered. When the volume of the sample is very large Equation 1.1 can be simplified to Equation 1.2. In such cases, the amount of analyte extracted is directly proportional to its concentration in the sample matrix. This feature of SPME is one of the many advantages of the method for on-site sampling when the volume of the matrix is unknown, and external calibration unfeasible. It is worth to mention that distribution coefficient between analytes and coating is necessary to be determined in advance. On-site sampling can eliminate errors associated with grab sampling, including errors derived from analyte loss via decomposition, or adsorption onto the surface of the sample container.

1.4.3.2 Pre-equilibrium Extraction

The time required to reach equilibrium is dependent on several factors, such as agitation, physicochemical properties of analyte, coating chemistry and coating thickness. Extraction in equilibrium provides high sensitivity, as the amount of extracted analyte onto the coating at equilibrium is maximum. If sensitivity is not a concern, extraction can be stopped before equilibrium is reached to increase sample throughput. Due to the displacement effect that occurs at high concentrations, pre-equilibrium extraction is recommended when solid coatings are used for extraction. The kinetic of extraction by the liquid coating is based on the diffusion-controlled mass transfer process proposed by Equation 1.8 [81,82]

$$C_0 = \frac{n(K_{fs}V_{f+V_s})}{K_{fs}V_fV_s(1-e^{-at})} \qquad \text{Equation 1.8}$$

Where "a" is the time constant, indicating how fast an equilibrium can be reached, which is depended on the sample volume, the extraction phase volume (liquid coatings) or the extraction phase surface area (solid coatings), the mass transfer coefficients, and the distribution coefficients between the analytes and the coating.[83] Equation 1.8 becomes Equation 1.1 when extraction time is sufficiently long to describe equilibrium extraction. Equation 1.8 shows a linearly proportional relationship between the amount of analyte extracted onto the fiber (n) at the time of pre-equilibrium, and the initial analyte concentration in the sample matrix (C_s^0). This relationship indicates that SPME quantification is possible by pre-equilibrium extraction if experimental conditions such as agitation conditions, sampling time, and temperature are kept constant.[80,84]

1.4.3.3 Diffusion-based Calibration Diffusion

Molecular diffusion is a special case of mass transfer in which solute molecules or particles move from a high concentration area to a low concentration area merely because of the kinetic energy of the solution molecules, while no external forces are applied to move fluid.[85] Several diffusion-based calibration methods have been developed for the quantification of SPME. These calibration methods are developed based on Fick's first law of diffusion, the interface model, the cross-flow model, and the kinetic processes of absorption and desorption.[80,86] They are mostly used for on-site sampling, including spot sampling and time-weighted average (TWA) sampling.

1.4.3.4 Fick's First Law of Diffusion

Fick's first law of diffusion was used for calibration of the SPME device. In this type of SPME device, the fiber or sorbent is retracted a certain distance into the needle housing during the sampling period.[58–60] Analyte molecules diffuse through the stagnant air/water gap between the opening and the fiber coating to access the fiber coating. The sampling rate is related to the geometry configuration of the sampler. It is directly proportional to the diffusion coefficient of the analyte, and the ratio of the cross sectional area of the opening (A) to the diffusion path length (Z). Due to the exceptionally small opening of the sampler, the performance of the device is independent of the convection conditions of the sample, particularly for on-site sampling, where the velocity of water is constantly varying, and difficult to measure and calibrate. Therefore, the amount of analyte collected during the sampling period can be calculated with the use of Fick's first law of diffusion (Figure 1.3, a). The concentration of

analytes can be calculated using Equation 1.9 if the sorbent behaves as a "zero sink" for the target analytes (Figure 1.3, b)

$$C = \frac{nZ}{AD_s t} \qquad \text{Equation 1.9}$$

Where C is the TWA concentration of the target analytes in air or water during the sampling time t, Z is the diffusion path length, A is the cross-sectional area of the needle opening, D is the diffusion coefficient of the target analytes in air or water, and n is the amount of analytes extracted by the fiber during sampling time. When used for water sampling, first, the air in the diffusion path should be completely replaced with ultrapure water, otherwise calibration with Equation 1.9 would be unsuitable. Additionally, hydrophobic analytes adsorb to the outside of the needle wall. To address this problem, the design of the originally developed sampler was modified by addition of a removable needle. The new redesigned sampler applied to measure TWA concentration pf PAHs compounds in Hamilton Harbour and Laurel Creek, Ontario, Canada.[87,90]

The disadvantage of this device is its very low sampling rate, which accordingly influences the need for long sampling times when analyte concentrations are low. However, it is worth to note that long sampling time periods are the final purpose of TWA sampling.

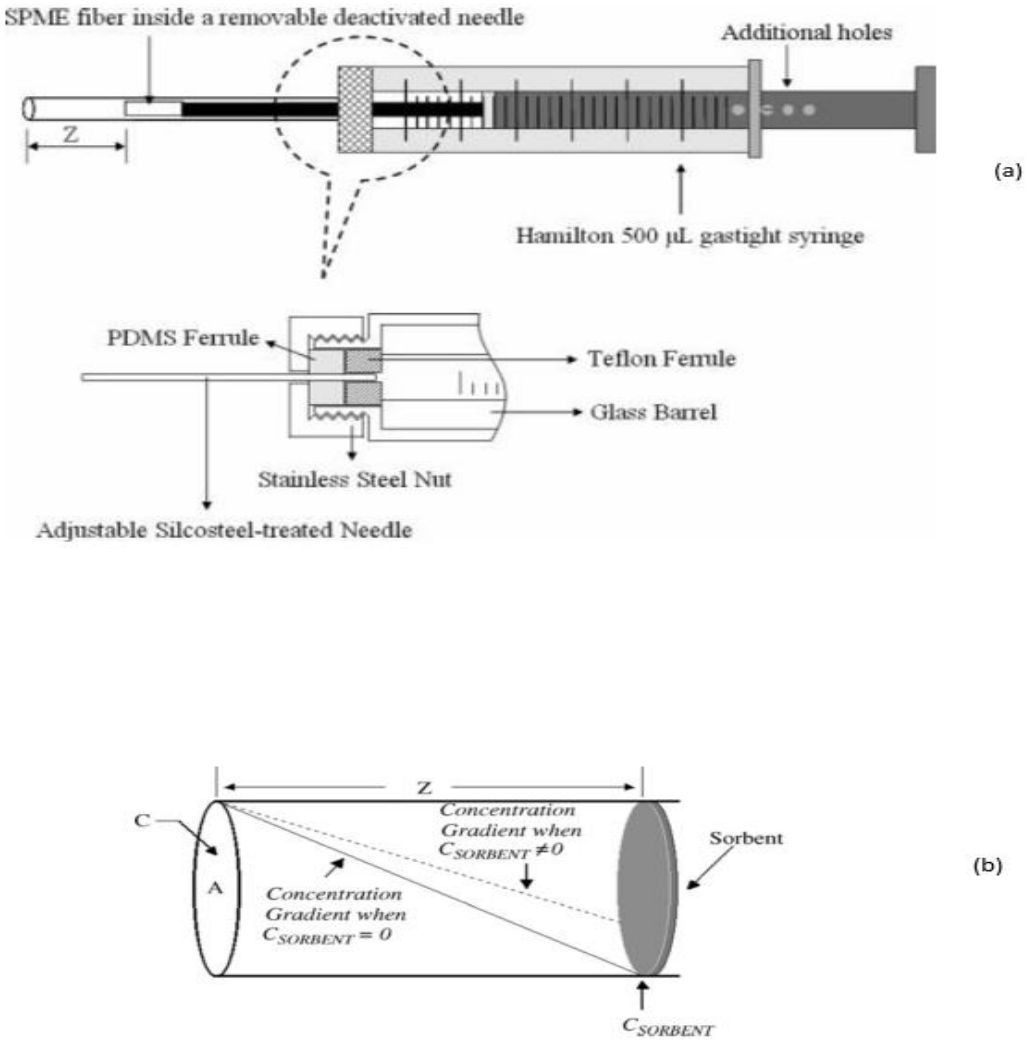


Figure 1.3 Schematic diagram of the (a) fiber-in-needle SPME device for TWA water sampling and the adjustable/removable needle; (b) schematic diagram of fiber-retracted SPME device and concentration gradient. Figure reprinted from reference 86 with the permission of the publisher

1.4.3.5 Kinetic Calibration

The absorption process of the analyte from the sample solution into an SPME liquid coating can be defined by Equation 1.10: [53, 54]

$$\frac{n}{n_e} = 1 - \exp(-at)$$ Equation 1.10

Where n is the extracted amount of analyte at sampling time t, and n_e is the extracted amount of analyte at equilibrium. Based on the above model, Chen et al. showed the isotropy between absorption of the analyte from the sampling matrix and desorption of the analyte from the coating, allowing for the introduction of the kinetic calibration approach for SPME applications.[91,92] The kinetic calibration technique uses desorption information of pre-loaded standards in the extraction phase of SPME to calibrate the extraction of the analytes. The method was validated in-vial and with the use of a flow-through system, and can be described by the Equation 1.11: [92,93]

$$\frac{Q}{q_0} = \exp(-a`t)$$ Equation 1.11

Where the relation between extraction and desorption is defined by Equation 1.12:

$$\frac{n}{n_e} + \frac{Q}{q_0} = 1$$ Equation 1.12

Where a is the adsorption rate constant, a' is the desorption rate constant, n is the amount of extracted analyte at extraction time, n_e is the amount of analyte extracted onto the coating at equilibrium, q_0 is the amount of loaded calibrant on the extraction phase, Q is amount of calibrant remaining in the extraction phase after exposure of the extraction phase to the sample matrix for a pre-determined sampling time. The concentration of the analytes in the matrix sample in the kinetic portion of the extraction time profile can be obtained by Equation 1.13:

$$C_s = \frac{n}{KV_f[1-\exp(-at)}$$ Equation 1.13

Kinetic calibration has been previously theoretically validated and practically applied to obtain TWA concentrations of PAHs compounds with the use of a flow-through system, and in pesticide sampling in jade plants.[90,94–96] The effect of environmental variables such as biofouling, temperature, and convection conditions was shown to be compensated with this approach.

1.4.4 Standard aqueous generation system

Preparation of hydrophobic compounds in water is challenging. Partial precipitation and losses on the surface, low solubility in water, and limited sample volume are the main problems encountered when spiking these compounds in water. The dynamic standard aqueous generation (ASG) system has been introduced to address the abovementioned issues, and has been extensively used for development and validation of the proposed passive sampler, as well as in obtaining partition coefficients between the PDMS coating and hydrophobic compounds such a PAHs in water. Two types of ASG were developed. Infusion of the standard solution prepared in organic solvent into the stream of water was the first configuration used for obtaining the partition coefficient between PAHs compounds and the PDMS coating.[97] A syringe pump was used for precise infusing of the standard organic solution into the water; however, the pump was a mechanical/electrical device that was susceptible to failure, and not convenient for long term preparation, as the syringe needed to be filled periodically. In addition, organic solvent had to be added to the system, which affected SPME method recovery.[98] Latter, another system was

developed based on the dialysis membrane for solid hydrophobic compounds.[99] It was consisted of a permeation chamber, mixing chamber and sampling chamber, sampling cylinder (Figure 1.4). The system was used for evaluation of SPME based passive samplers.[99] In addition, it was used for obtaining partitioning and accumulation artefacts into the PDMS thin film, and black worms to assess the bioaccumulation potential of hydrophobic organic chemicals. Bio-concentration, defined as the ratio of chemical concentration in the worm to chemical concentration in the flow-through system at equilibrium, was obtained for five PAHs compounds.[101]

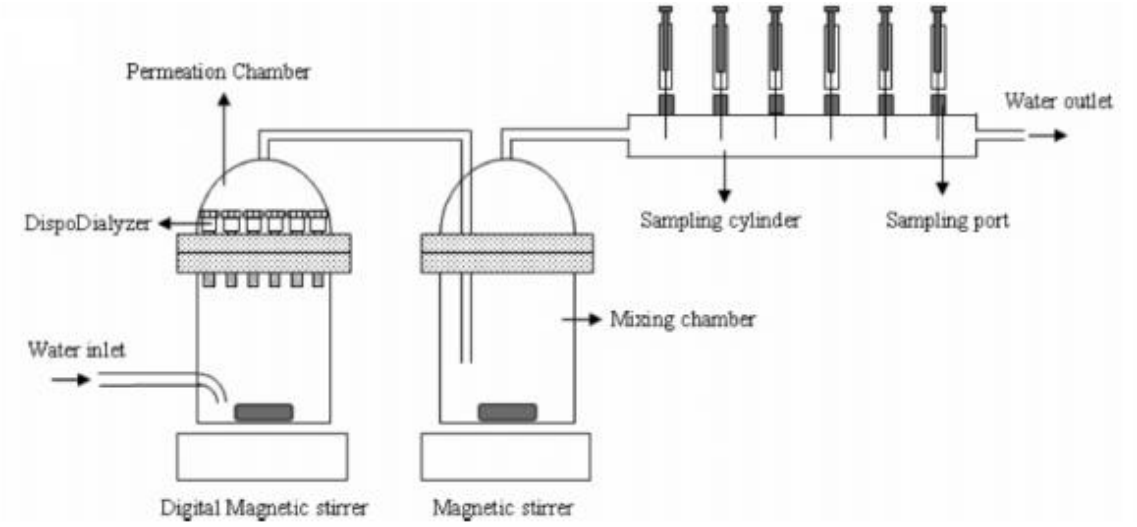


Figure 1.4 Schematic diagram of the flow-through system based on permeation. Figure reprinted from reference 140 with the permission of the publisher

1.4.5 Application of SPME for binding studies

SPME has been used for investigation of receptor-ligand interactions in pharmacology and toxicology applications (Figure 1.5).[57,68,102–114] SPME can measure the free concentration of analytes in complex matrices if the resulting extract is negligible in relation to the free concentration; this technique has been named negligible depletion-SPME (nd-SPME).[115] The basic conditions for application of nd-SPME for measuring a free dissolved concentration accurately in a complex matrix are: (1) establishment of equilibrium between the bound and free dissolved analytes (2) the depletion of the free fraction by extraction should be negligible to prevent disturbance of equilibrium between the bound and free chemical (3) the binding matrix should not affect the uptake of kinetic or adsorb to the coating. Of these three conditions, the second condition is the main criteria for performing nd-SPME accurately. Since the depletion cannot be 0%, a limit of 1-5% is set until the depletion is considered negligible.[57]

If the depletion limit is 1%, the volume of the sample should be $V_s \geq 100 K_{fs} V_f$. The free concentration of the analytes is calculated by preparation of a matrix-free calibration. It is generally prepared in matrix-free media such as ultrapure water that matches the pH and ionic strength of the surface water.[57] The free dissolved concentration of the analyte in the matrix

was proportional to the extracted amount of analyte from the sample according to Equation 1.14:

$$C_f = \frac{n}{K_{fs}V_f}$$ Equation 1.14

Where C_f is the free concentration of the analyte, n is the amount of the analyte extracted by SPME, K_{fs} is the distribution constant between the extraction phase (coating) and the sample, and V_f is the volume of the extraction phase. The fiber constant is the product of distribution coefficient and volume of the fiber in liquid coating or active surface area in the solid coating. In ultrapure water, the total spiked concentration and the free concentration were equal; henceforth, the fiber constant (Kfs×Vf) could be determined when the value of n was determined experimentally. The fiber constant can be determined by both single or multiple point calibrations.[112,114] The total concentration can also be determined with matrix -matched calibration, standard addition and spiking isotopic labled standard tosample.[114,116]

The total concentration is obtained with the same approach used for free concentration using of Equation 1.15:

$$C_{total} = \frac{n}{K_{fb}V_f}$$ Equation 1.15

Where $K_{fb}V_f$ is the fiber constant in matrix blank, and n is the amount of analyte extracted by the SPME fiber in equilibrium.

The total concentration can also be determined by spiking isotopic labeled standards in real samples when equilibrium is reached between the analytes and DOM. The total concentration of the non-labeled counterpart is calculated from the Equation 1.16:

$$C_{total} = C^* \frac{A_{SPME}}{A^*_{SPME}}$$ Equation 1.17

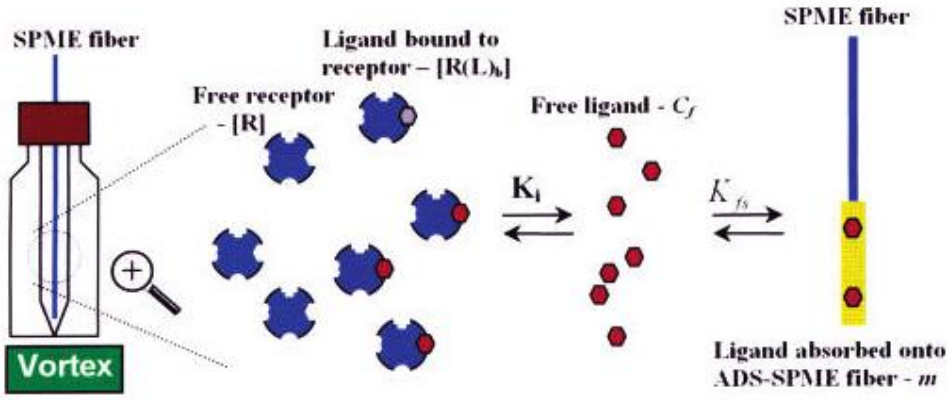


Figure 1.5 Schematic representation of experimental setup for binding studies. Figure reprinted from reference 113 with the permission of the publisher

Where A_{SPME} and A^*_{SPME} are the instrument responses (in peak area) of the non-labeled and labeled analytes measured by the SPME device, C* is the concentration of the spiked labeled standard, and C_{total} is the total concentration of the non-labeled counterpart. Ideally, the isotopic labeled standard should be the same as the analyte. However, if no labeled standards are available for all of the analytes under study, the total concentration can be obtained by considering the partition coefficient (K) of the analyte between the SPME fiber and analyte, and the response factor (RF) of the instrument (Equation 1.17).

$$C_{total} = C^* \frac{A_{SPME} \times K}{A^*_{SPME} \times RF}$$ Equation 1.18

With both free and total concentrations, the binding percentage of the analyte is calculated based on Equation 1.18.

$$PB\% = \frac{C_{total} - C_{free}}{C_{total}} \times 100 \qquad \text{Equation 1.19}$$

The distribution coefficient of the analyte (K_{DOC}) can be calculated if the C_{free}, C_{total} and DOC content are known by using Equation 1.19.[45,55,117]

$$K_{DOC} = \frac{C_{total} - C_{free}}{C_{free}} \times \frac{1}{[DOC]} \qquad \text{Equation 1.20}$$

1.5 Thesis objective

The presence of ingredients of personal care products in aquatic environment is a concern among the public and researchers as these compounds may have an impact on aquatic life. This has led to development various analytical methodologies not only for environmental monitoring but also for partitioning studies in the aquatic environment. The measurement of freely dissolved time weighted average concentration and partitioning coefficients between the chemicals and environmental compartments are two important key parameters for relevant toxicology studies. As a consequence, improvement of current analytical methods, particularly for on-site sampling and measuring freely dissolved concentration in complex matrices is paramount. There are some problems with the current methods which need to be addressed. First, preparation of aqueous standard solutions of hydrophobic compounds in vial for is challenging due to loss of analytes on vial surfaces, limitations of sample volumes, and low solubility in water. Second, there are no comprehensive method available that can be used for

monitoring TWA concertation of chemical compounds with wide range of physical-chemical properties. Third, the detection of highly bonded chemical in complex matrices in limited sample volume is difficult due to low free concertation available. With these in mind, the main objective of this thesis is to develop a dynamic generation of a standard aqueous solution containing UV filters and biocides for development and evaluation of a comprehensive passive sampling method based on thin film solid phase microextraction technique and binding investigations studies between target analytes, humic acid, and sediment.

This thesis is divided into three main chapters. First, preparation of a dynamic flow through aqueous standard generation of UV filters and biocides with a wide range of physical and chemical properties in liquid and solid states, ranging from highly hydrophobic to highly hydrophilic compounds (chapter 2). Secondly, time weighted average concentration monitoring based on thin film solid phase microextraction is described in chapter 3. Thirdly, partitioning of UV filters and biocides to different environmental compartments such as humic acid and sediment is described in chapter 4. Conclusions of the research are summarized in Chapter 5, in addition to suggestion for future directions.

Chapter 2

Dynamic flow through system to calibrate thin film solid phase micro-extraction devices for environmental monitoring of personal care ingredients

2.1 Introduction

UV filters and biocides are two classes of compounds widely used as ingredients in an extensive variety of personal care products (PCPs), and comprised of a wide range of physical-chemical properties, ranging from high water solubility to extreme insolubility.[13,118] Large quantities of UV filters and biocides enter the environment through both direct and indirect pathways such as recreational release (e.g., swimming) as well as effluent from wastewater treatment plants (WWTP)[119]. Accordingly, public concern has been increasing over the widespread consumption of PCPs, as their impact on the environment and human health is not entirely known.[120] As such, there is a current need for the development of sampling and monitoring tools that would enable accurate collection of data to ensure the safety of the environment. Typically, sampling involves automated or manual grab-sampling from a specific field site. Samples should be stored and transferred in a manner that ensures that sample integrity is maintained until extraction and analysis can be performed in the laboratory. The entire process is time-consuming and costly, requiring a significant amount of method development, and to incorporate quality assurance and quality control steps specific to

each analyte included in the analysis.[16,121,122] Passive sampling devices could greatly improve the assessment of levels of chemicals used in PCPs. As the concentrations of these compounds in aquatic environments are typically found in trace and ultra-trace levels, suitable sample preparation steps and sensitive analytical methods are required. In natural aquatic environments such as lakes and rivers, the concentrations of aquatic contaminants may change daily or even hourly; accordingly, long term monitoring is required in order to obtain a realistic picture of the time-weighted average (TWA) concentrations of contaminants.[16] A passive sampling approach was found to be a more promising technique for measuring the concentrations of a wide range of pollutants in environmental samples. However, in order to validate long-term passive sampling applications to monitor organic pollutants in water, the methods or equipment should first be tested and evaluated in the laboratory, and then applied to the field.[123] However, preparation of aqueous standard solutions of hydrophobic compounds in vial for evaluation of passive sampler devices is problematic due to loss of analytes on vial surfaces, limitations of sample volumes, and low solubility in water.[87,124,125] To address these problems, dynamic preparation of aqueous standard solutions was introduced. Two configurations based on syringe infusion and permeation methods have been developed to date.[97,99] Syringe infusion has been replaced by the permeation method as the method of choice due to the former method need for an expensive syringe pump that required periodic filling, as well as subsequent addition of organic solvents to standards. The dialysis membrane, which is supplied with a certain molecular weight cut-off, is the key component of the permeation-based

system. It is simple, cheap, solvent-free, has a long lifetime, and allows for preparation of high concentrations of target analytes.[99]

ASG has been used for finding partitioning coefficients of PAHs between water and PDMS coating, calibration of SPME devices and other sampling devices for on-site or in vivo sampling, as well as for obtaining apparent distribution coefficients between SPME fibers and blood matrix/buffer sample matrices. [97,99,116,126] Data obtained from dynamic systems is more accurate than data gathered with static systems, as the former avoids common limitations of the latter such as analyte loss due to surface adhesion; in addition, dynamic systems allow fiber uptake to be corrected. [127]

Thin film solid phase microextraction has been introduced in thin film sheet and blade configuration to overcome the low capacity limitation and also acceleration of the extraction rate to shorten extraction time by increasing surface area-to-volume ratio [77-78]. In blade configuration, different types of particles including C18, PS-DVB, HLB are immobilized on a thin stainless steel mount by spray methods for extraction wide range of analytes in different matrixes.[78] It has been widely used for determination of pharmaceuticals in wastewater samples[70] , quaternary ammonium compounds in water samples [71] phenolic compounds in wine, berry and grape samples [72], and prohibited substances in urine.[73]

In this study, dynamic generation of aqueous standard containing UV filters and biocides with a wide range of physical and chemical properties in liquid and solid states, ranging from highly hydrophobic to highly hydrophilic compounds, was prepared and used for development of thin

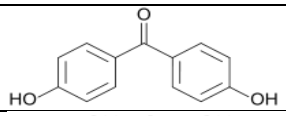
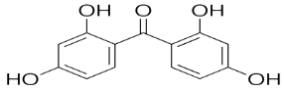
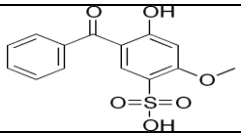
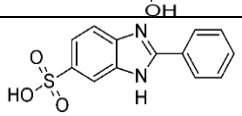
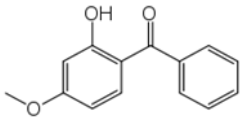
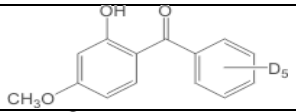
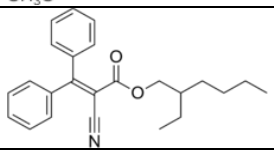
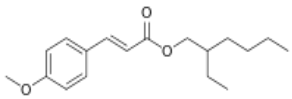
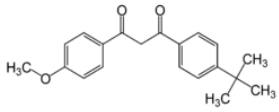
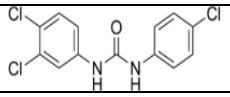
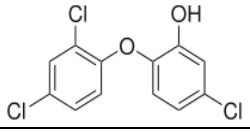
film solid phase microextraction method for the analytes. In Table 2.1 the physicochemical properties of the UV fitters and biocides studied in this thesis are demonstrated.

2.2 Materials and methods

2.2.1 Chemical and materials

All chemicals and reagents utilized in this study were obtained at the highest available purity (>99%) and used without further purification. The standards octyl methoxycinnamate (OMC), benzophenone-1 (Ben-1), benzophenone-2 (Ben-2), benzophenone-3 (Ben-3), benzophenone-4 (Ben-4), 2-phenylbenzimidazole-5-sulphonic acid (PBSA), octocrylene (OCR), butylmethoxydibenzoylmethane (BM-DBM), triclosan (TCS), and triclocarban (TCC), as well as HPLC-grade solvents acetonitrile (ACN), methanol, isopropyl alcohol (IPA), ethyl acetate, formic acid (LC-MS grade), formic acid (reagent grade) sodium carbonate, sodium hydrogen carbonate, and ammonium formate (HPLC grade) were obtained from Sigma (Oakville, ON, Canada). Nanopure water was obtained from a Barnstead nanopure water purification system with 18.2 MΩcm resistivity. Oasis HLB (hydrophilic lipophilic balanced) polymeric reversed-phase particles (30 µm particle size) were obtained from Waters (ON, Canada), C18 (5 µm particle size) particles were supplied by Supelco. Easy modified polystyrene-divinylbenzene-weak anion exchange (PS-DVB-WAX)) (Macherey-Nagel) particles were obtained from VWR International (Mississauga, Canada). Polyacrylonitrile (PAN), obtained from Aldrich (Oakville, ON, Canada), was dissolved in N, N-dimethylformamide (Sigma-Aldrich, Oakville, ON, Canada) and used as glue for the immobilisation of functional particles to the SPME blades. HLB and C18, PS-DVB-WAX particle coated blades were prepared as reported by

Table 2.1 Physical-chemical properties of UV filters and biocide compounds

Compound	Chemical structure	Log P	pka	Water solubility (mg/L)
Ben 1		2.9[b]	7.53[b]	236[b]
Ben 2		2.1[b]	6.98[b]	399[b]
Ben 4		0.37[a]	-0.7[b]	250000[a]
PBSA		1.03[b]	-0.87[b]	23600[d]
Ben 3		3.8[b]	7.56[b]	68.6[a]
Ben 3-d5		-	-	-
OCR		6.4[a]	-	0.001[c]
OMC		5.8[a]	-	0.003[c]
BM-DBM		4.5[a]	9.74[b]	0.008[c]
TCC		4.9[b]	12.77[b]	0.0237[b]
TCS		4.76[a]	8.14[b]	10[a]

nd; no data

na; not applicable

[a] Experimental values, from database of physicochemical properties. Syracuse Research Corporation: http://www.syrres.com/esc/physdemo.htm, accessed on Oct, 2015

[b] Software-calculated value, from SciFinder Scholar Database 2006: ttp://www.cas.org/products/sfacad/

[c]Unilever internal report (2013). UV – filters Partition coefficient (n-octanol/water): slow stirring method and water solubility (column generator method)

[d]Estimated value, from database of Royal Society of Chemistry's databases; Chemspider: http://www.chemspider.com, accessed on Oct,2015

Mirnaghi et al.[78] Dialysis membrane units of 100-500 Da and 500-1000 Da molecular weight cut off (MWCO) were obtained from Spectrum Laboratories (Rancho Dominguez, CA). Porous frit (0.5 µm) were obtained from Swagelok (Ontario, Canada). PTFE permeation tubes were obtained from Zeus (New Jersey, USA). The digital conductivity meter used for measuring conductivity and total dissolved solid was obtained from VWR (ON, Canada). Benchtop Orion 3 Star pH meter from Thermo Scientific ((NJ, U.S.) was used for measuring the pH of the water samples and aqueous standard generation system. Individual stock solutions were prepared either in methanol (Ben-1, Ben-2, Ben-3, Ben-4, TCS, TCC, OCR, OMC and BM-DBM), or in pure water with the addition of a few drops of 2 M sodium hydrogen carbonate (in the case of PBSA) at a 2 $mgmL^{-1}$ concentration. Mixed standard

solutions were prepared at a 100 µg mL^{-1} concentration and stored at 4 °C. Instrument calibration standards were prepared daily in MeOH/H2O 50/50 (v/v).

2.2.2 Instrumental analysis method (LC/MS/MS)

A Shimadzu (LC-10 AD) high-performance liquid chromatograph (HPLC) and an Applied Biosystems API 4000 triple quadrupole mass spectrometer (equipped with TurboIonSpray source) were used for separation and quantitative analysis of analytes. The chromatographic column used was a Waters Symmetry Shield RP18 with dimensions of 2.1mm × 50 mm and 3.5 µm particle diameter. Sample volumes of 20 µL of both standards and extracted analytes were injected into the LC-MS/MS system using a HTC PAL autosampler from Leap Technologies (HTC Analytics, NC). Two different chromatography methods were used for negative and positive mode, respectively. In positive mode, mobile phase A consisted of ACN/H2O (50/50, v/v), with a 10 mM ammonium formate buffer with the pH adjusted to 3.2 with formic acid, and mobile phase B of isopropanol with 0.1% formic acid (v/v). The applied chromatographic gradient was started at 10% of B and kept at this composition for 2.2 min, then linearly increased to 50% of B within the next 2 min, where it was held for an additional 2 min. Finally, within the next 1 min, the gradient was returned to 10% of B and held for an additional 1 min at the same composition (total cycle was 8.2 min). This method prevented carry-over for hydrophobic compounds.[128] In negative mode, the mobile phase consisted of solvent A (water) and solvent B (acetonitrile, ACN), both containing 1 mM acetic

acid (HOAc). The applied chromatographic gradient was started at 10% of B and kept at this composition for 2.2 min, then linearly increased to 100% of B within the next 2 min, where it was held for an additional 2 min. Finally, within the next 1 min, the gradient was returned to 10% of B and kept for an additional 1 min at the same composition (total cycle was 8.2 min). The ionization efficiency was improved by using acetic acid in negative mode, as acetate has a high basicity function in the gas phase.[129] After each injection, the autosampler system was cleaned by washing the syringe and injector port with two separate washing solutions. Washing solvent A was composed of ACN/IPA 50/50 (v/v) and washing solvent B was methanol. MS/MS analysis was performed in positive and negative modes in a separate run under multiple reaction monitoring (MRM) conditions. A summary of the MS/MS parameters is given in Table 2.2.

2.3 Aqueous standards generation (ASG)

To prepare a robust and reliable standard aqueous generation of UV filters and biocides, a careful investigation was conducted in regards to the physicochemical properties of each analyte, including solubility in water, physical state (liquid or solid), polarity, and molecular weight. Figure 2.1. Shows a schematic diagram of the standard aqueous solution generation system based on permeation. The system consisted of a permeation chamber, mixing chamber and sampling chamber. An 18 L polypropylene reservoir was filled with water delivered by a Series 200 Perkin Elmer pump (Shelton, Connecticut, USA) at a speed of 3 $mLmin^{-1}$.

Table 2.2 Mass spectrometry conditions for the analytes: optimized ionization source values

Negative mode: ion source gas 1 (GS1) = +40, ion source gas 2 (GS2) = +40, curtain gas = +50, collision gas = 10, spray ionization voltage = -4500 V, and temperature = 500◦C.

Positive mode: Negative mode: ion source gas 1 (GS1) = +40, ion source gas 2 (GS2) = +60, curtain gas = +50, collision gas = 10, spray ionization voltage = 5500 V, and temperature = 500◦C.

*DP=Declustering potential, EP= entrance potential, CE=Collision energy, and CXP=Collision cell exit potential

Compound	Q1 mass (amu)	Q3 mass (amu)	*CE (V)	*CXP (V)	*DP (V)	*FP (V)	Ionization mode in ESI-MS/MS
TCS	287	35	-58	-58	-32	-32	Negative
Ben 2	245	109	-72	-72	-31	-31	Negative
TCC	315	162	-69	-69	-22	-22	Negative
Ben 4	307	227	-96	-96	-32	-32	Negative
PBSA	273	193	-84	-84	-36	-36	Negative
Ben 1	213	169	-85	-85	-29	-29	Negative
OCR	362	232	39	5	5	16	Positive
BM-DBM	311	135	55	8	8	24	Positive
BM-DBM	311	177	55	8	8	15	Positive
Ben 3	229	151	70	6	6	11	Positive
Ben 3-d5	234	151	73	10	10	11	Positive
Ben 3-d5	234	110	40	10	10	15	Positive
OMC	291	161	37	4	4	11	Positive

to the permeation chamber (capacity is about 1000mL). The key part of the ASG is the permeation chamber, which consists of dialysis membranes, a permeation tube, and a porous frit coated with epoxy glue. The permeation chamber had an inlet for introduction of fresh water close to the bottom of the chamber, and an outlet (close to the top) connected to the mixing chamber. The mixing chamber was used to ensure that the resulting solution was homogeneous, and that the sampling chamber can be used for SPME optimization, passive sampler evaluation, and binding studies. The entirety of the system was covered with aluminum foil to prevent any photodegradation of analytes.

2.3.1 Preparation of aqueous standards generation system

A permeation tube (PTFE) with 0.25 mm thickness was used for standard generation of OCR and OMC (liquid state). Permeation tubes for OCR and OMC were made by placing pure chemicals inside a 50 mm long (1/4 inch) tube capped with 20 mm long solid PTFE plugs, and 6 mm (1/4 in.) Swagelok caps. Cellulose acetate dialysis membranes with 500-1000 Da MWCO or MWCO 100-500Da were used for TCS, TCC, BM-DBM, and Ben-1, Ben-2 and Ben-3, respectively. A stainless steel porous frit (0.5µm) coated with epoxy glue was used for Ben-4 and PBSA. Dialysis membranes and the porous frit were partially filled with 100 mg of each solid and 1 mL water, then tightly sealed, placed into a beaker (containing 300 mL of water), and sonicated for 30 min. The prepared membranes and porous filters were then placed in the permeation chamber. The partially dissolved analytes (or

saturated solution, in the case of solid compounds) were further diffused through the membrane walls and transported via flowing water, first to the mixing chamber, and then to the sampling chamber.

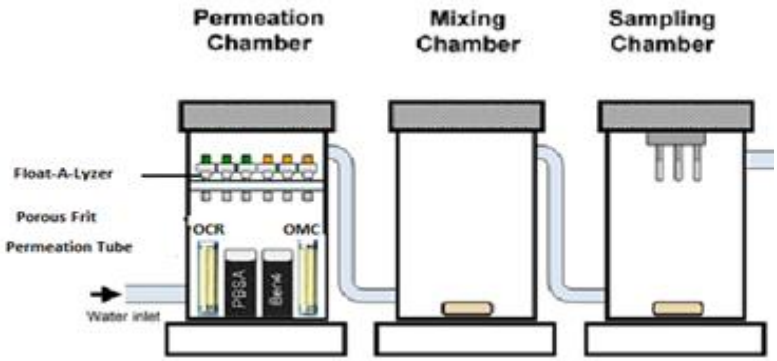


Figure 2.1 Schematic diagram of the aqueous standard generation (AGS) system

2.3.2 Determination of the analytes in ASG

The concentrations of the analytes in the aqueous standard generation system were measured in two different ways. For low concentration and/or hydrophobic compounds, 250 mL of water sample was collected in an amber bottle from downstream of the sampling chamber, and the analytes were extracted using 50 mL of ethyl acetate. Second and third sequential extractions were performed with fresh ethyl acetate, and combined together. The collected extract was then evaporated to dryness and reconstituted in 10.0 mL of MeOH/H2O (50/50, v/v). For high concentration and/or hydrophilic analytes, 0.7 mL of the solution was taken from the ASG, mixed with the same volume of methanol, and then injected directly to the LC-MS-MS instrument.

2.3.3 Quality assurance and quality control

The limits of detection and quantification of the instrument were defined as the concentrations of working standards which provide signal-to-noise ratios of 3 and 10, respectively. In the calibration experiments, these were determined to be in the range of 0.1-5.0 $ngmL^{-1}$ and 0.5-15.0 $ngmL^{-1}$, for detection and quantification, respectively. The extraction method detection limit and quantification limit were determined by considering the extraction recovery and instrumental limit of detection, which were in the range of 0.004-0.1 $ngmL^{-1}$ and 0.02-0.2 $ngmL^{-1}$ respectively. Extraction recoveries were determined in ultrapure water spiked with 1$ngml^{-1}$ concentration of the analytes and they were found to be in the ranges of 85±5% and 110±6%, with the exception of Ben-4 and PBSA. A laboratory blank, consisted of ultrapure water, was analyzed, and Ben-1 and Ben-2 were found in concentrations of 0.3 and 0.6 $ngmL^{-1}$, respectively.

2.4 Thin film solid phase microextraction procedure

TF-SPME method development comprised of selection of a suitable coating, desorption solvent, desorption time, and preconditioning time. For selection of the best extraction phase three different coating types were evaluated, namely, C18, HLB and WAX-PS-DVB coated blades. Prior to extraction, the coatings were preconditioned in MeOH/H2O (50/50, v/v) for 30 min. Conditioned coatings were placed in the sampling chamber of the ASG, for an extraction time of 180 min. Following extraction, to find the best solvent for desorption of the analytes, four

desorption solvents were tested simultaneously. This step was performed in a 2 mL amber vial containing 1.8 mL of desorption solvent using vortex agitation at 1500 rpm. The desorption step was followed with a second desorption to evaluate the carryover in each extraction phase/desorption solvent pair. The amounts of extracted analytes were determined in the LC-MS/MS method described above using instrumental calibration solutions prepared in MeOH/H2O (50/50, v/v) in a range of 0.1 to 100 $ngmL^{-1}$. A summary of the experimental conditions is given in Table 2.3. Gloves were worn during sample preparation and separate solvents and glassware were used. All glassware was sequentially rinsed three times each with acetone, methanol, and ultrapure water before use.

Table 2.3 Summary of experimental conditions used throughout the evaluation of coatings and desorption solvents

1) Preconditioning conditions		**2) Rinsing conditions**	
Time	30 min	Time	5 sec
Agitation	1500 rpm	Agitation	1500 rpm
Solvent	50/50 ($MeOH/H_2O$)	Solvent	H_2O
Volume	1.8 mL	Volume	1.8 mL
3) Desorption conditions			
Time	15-30 min		
Agitation	1500 rpm		
Volume	1.8 mL		
Desorption solvent	50:25:25 (MeOH/ACN/IPA)		

2.5 Results and discussion

Before commencing preparation of the ASG, the chemistries of the chemical compounds under study as well as of the membrane being used were examined. The most important property of the membrane is its ability to control the rate of permeation of the chemicals. The model that describes the mass transfer of chemical species through the membrane is solution-diffusion, in which the chemical species dissolve in the membrane, and then diffuse through due to the concentration gradient. The permeation rate depends on the solubility of the material in the membrane as well as the rate of diffusion through the membrane. The solution-diffusion model can be described by first Fick's law of diffusion, which states:

$$J_i = -\frac{D_i}{RT}\ \frac{\partial \mu_i}{\partial x} \qquad \text{Equation 2.1}$$

Where Ji is the rate transfer of species i or flux ($gcm^{-2}.s$), dci/dx is the concentration gradient of species i, and D_i is the diffusion coefficient (cm^2s^{-1}). Fundamentally, diffusion is a slow process. In solution-diffusion, the pore (free volume) consists of small spaces between polymeric chains, which are caused by the thermal motion of the polymer molecule. The free volume of the polymer appears and disappears at the same timescale as the species traverses the membrane.[130] The driving force that produces the movement of permeate is the gradient in its chemical potential. The flux of a chemical can be described by Equation 2.2:

$$J_{i=} = -\frac{L_i}{RT}\ \frac{\partial \mu_i}{\partial x} \qquad \text{Equation 2.2}$$

Where dμi/dx is the chemical potential gradient of component i and L_i is a coefficient of proportionality relating the chemical potential to the flux. Driving forces such as gradients in

concentration, temperature, pressure, and electrical potential can be expressed as chemical potential gradients.[131] Competing against this gradient, however, are the resistance to permeation across the membrane and the resistance in the interface adjacent to the interior and exterior of the membrane. The overall mass resistance is the combination of the boundary layer and membrane mass resistance. The flux in the boundary layer interface can be written as:

$J_i=K_{bl}\ (C_{ib}-C_{i0})$ Equation 2.3

Where k_{bl} is the mass transfer coefficient, and C_{io} is the concentration of component i in the fluid at the feed/membrane interface. The flux across the membrane can be written as:

$J_i=K_m\ (C_{i0}-C_{ip})$ Equation 2.4

Where K_m is the mass transfer coefficient in the membrane. The total concentration drop is the sum of concentration drop across the boundary layer and membrane. The mass resistance is described as follow:

$$\frac{1}{Kov}=\frac{1}{Km}+\frac{1}{Kbl}$$ Equation 2.5

When the boundary layer mass transfer is high, the mass resistance is small and the rate limiting step will be the mass resistance in the membrane. When the mass transfer in the boundary layer is low, the mass resistance becomes high, forms a large fraction of overall mass resistance. Boundary layer thickness is dependent on the thickness of the membrane, fluid's linear velocity, kinematic viscosity, temperature, and the diffusion coefficient of the analyte in the liquid. Increasing fluid's linear velocity past the membrane surface reduces the boundary layer thickness, which increases the mass transfer and depletes permeates in the bulk side if the boundary layer on the outer interface is the rate-limiting step.

Although, increasing linear velocity will simultaneously dilute the sample and decrease the concentration in the ASG. Permeate accumulates at the membrane surface until a sufficient gradient has formed to allow it to diffuse to the bulk solution; a steady state is then reached. In this study, three different devices, namely dialysis membrane, permeation tube, and a porous frit coated with epoxy glue, were used to produce a steady state concentration of a wide range of chemicals. Other parameters may also contribute to the diffusion of analytes from the membrane to the water system could be the diffusion coefficient in water, the diffusion coefficient in the membrane, temperature, and time, concentration of species, membrane surface area, membrane thickness, molecular charge, molecular size, and agitation.

2.5.1 Dialysis membrane

The simplest application of the solution-diffusion model is dialysis, as only concentration gradients are involved. The solutes diffuse from a high concentration region to a low concentration region across a semipermeable membrane. The application illustrates which molecular weight cut off (MWCO) yields constant molecular permeation. As the dialysis membrane is a spongy matrix of cross-linked polymers, the pore rating referred to as MWCO, is a measure of the retention performance. The membrane MWCO is determined as the solute size that is retained by the membrane at least 90%. The permeability of a solute is dependent on molecular shape, degree of hydration, ionic charge, and polarity. When selecting an MWCO, one should consider a feature that provides less fluctuation in analyte concentration. In dialysis membranes, for instance, if the MWCO of the dialysis membrane is too small, this may result in a low concentration of analytes. On the other hand, if the MWCO is too large,

this may cause fluctuations in the concentration of the analytes. In this study, two dialysis membranes with MWCO 100-500 and 500-1000 were used for hydrophilic and hydrophobic compounds, respectively. Since the water solubility of hydrophilic analytes is high, the concentration gradient on both sides of the membrane is high. The lowest MWCO membrane was used to keep the permeability of the membrane low to create a low concentration in the ASG. However, for hydrophobic analytes, a high MWCO membrane was used to improve the permeability of these analytes. The concentration of the analytes in ASG can be increased by adding more dialysis membranes to the permeation chamber. Special care was also taken to maintain a stable room temperature (24±1 ◦C). Any change in temperature might change the diffusion rate of the analytes and affect analyte concentrations inside the membrane.

2.5.2 Permeation tube

OCR and OMC are liquid hydrophobic compounds, and as such, their aqueous standards need to be prepared in a different way. PTFE tubing was used, as described in the experimental section. Fluorocarbon resin are chemically inert; since no chemical reaction and solubility occurs in Teflon. Liquid analyte permeates by random molecular motion between plastic molecules due to concentration gradient across the polymer. Permeation depends on temperature, surface contact, plastic density, and thickness. An increase in temperature will increase molecule activity of the material, and hence, permeation rate.

2.5.3 Porous frit coated epoxy glue

As with membranes for hydrophilic compounds, dialysis membranes with a molecular weight cut off 100-500 were selected and tested in the permeation

chamber. Initially, 200 mg of Ben-4 and PBSA (100 mg of each) were loaded in the dialysis membrane and placed in the permeation chamber. Visual inspection showed that the membrane had been disrupted after 24 hrs. The high water solubility of PBSA and Ben-4 decreases the chemical potential inside the membrane, allowing water molecules to migrate across the membrane in order to reach equilibrium. The dialysis membrane was replaced by a stainless steel porous frit cup with a 0.5 µm porosity, selected to withstand against osmotic pressure. After this change, the permeation rate was noted to significantly increase due to the large microcavities in the porous frit. To address this, the porous frit was covered with epoxy glue with approximately 0.2 mm thickness in order to restrict the permeation rate. The transfer mechanism changed to solution-diffusion, since epoxy has a hydrophilic structure that allows analytes to dissolve in and diffuse through the glue. After reaching a steady state, the average concentrations of individual analytes were 379, 138, 28, 459, 3.5, 118, 0.25, 0.08, 1.2, and 0.12 $ngmL^{-1}$ for PBSA, Ben 1, Ben 3, Ben 4, TCS, Ben 2, OCR, OMC, TCC, and BM-DBM, respectively. As the solids (UV filters and biocides) and liquid (water) co-existed inside of the membranes, the concentrations of the analytes remained constant if the temperature and flow-rate of water were maintained constant. The observed change in the concentrations of the eight UV filters and biocide compounds was less than 20% over a 3 month period (Figure 2.2).

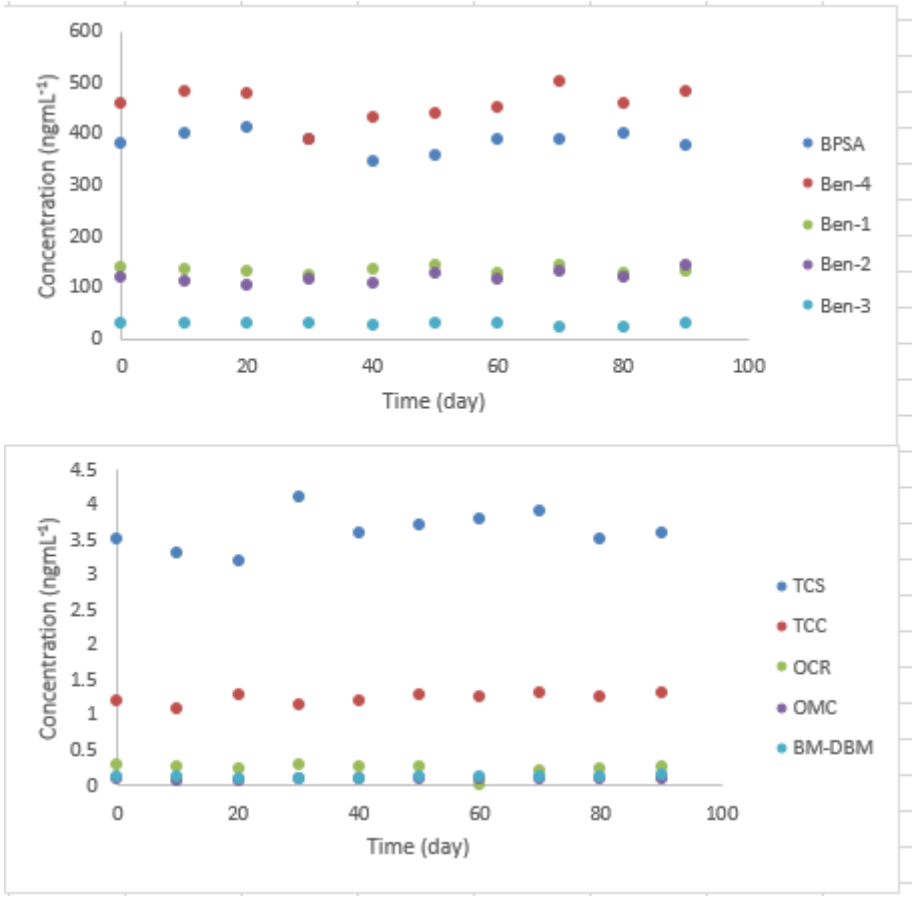


Figure 2.2 Variation of concentration-time for flow-through system

2.5.4 Thin-film solid phase microextraction

2.5.4.1 Selection of the coating and desorption solvent

In order to find the optimum SPME coating for the extraction of the analytes of interest, three different extractive phases (i.e., WAX-PS-DVB, C18, and HLB) were prepared in blade format and evaluated. Evaluations of coating and desorption solvent were performed simultaneously by considering the extraction capability and observed carryover in each coating. After

extractions for 180 min in the ASG, first and second desorptions were performed with a range of desorption solvents for 60 min. In this work, MeOH/ACN/IPA (50/25/25, v/v/v), MeOH/ACN/H2O (40/40/20, v/v/v), ACN/H2O (50/50, v/v), and MeOH/H2O (80/20, v/v), were studied to find the most appropriate desorption solvent. The results indicated that the MeOH/ACN/IPA (50/25/25, v/v/v) gave the best recovery and the lowest carryover (less than 4%) in comparison with other desorption solvents. HLB and C18 extractive phases showed enhanced extraction abilities for hydrophilic and hydrophobic compounds, respectively. The WAX-PS-DVB coating was not considered for further studies, as the extraction efficacies for most of the analytes were low and carryover was high in comparison with C18 and HLB coatings. The obtained results for the coating and desorption solvent optimization experiments are shown in Table 2.4-Table 2.13 Evaluation of various blades in terms of extraction amount and carryover of Ben-3 After selection of the extraction phases and desorption solution the necessity of preconditioning step of the extraction phases was evaluated in two experiments. In the first experiment, prior to extraction the extraction phases were conditioned in MeOH/H2O (50/50, v/v) for 30 min. In the second experiment, extraction was performed directly with dry coating without preconditioning. The obtained results for extraction with HLB coated blades revealed no significant difference in extraction efficiencies between conditioned and non-conditioned coatings. Conversely, the extraction efficiencies of C18 coated blades were affected from presence or absence of conditioning step revealing higher recoveries when conditioning step is used. (Data not shown). Therefore, the C18 coating was preconditioned for 30 min in MeOH/H2O (50/50, v/v) before extractions. Desorption time was also optimized, where 15

and 30 min were found as optimum desorption times for HLB and C18 coatings, respectively (Figure 2.3).

2.6 Conclusion and future directions

The flow-through aqueous standard generation described in this study was able to generate constant concentrations of UV filters and biocides in water. The concentration variation was less than 20% for three months. A porous frit coated by epoxy glue (0.5µm), dialysis membranes with 100-500 and 500-1000 Da MWCO and PTFE permeation tube were used as a basic element for this purpose. The ASG overcame problems with loss of analytes on the system surfaces, limited sample volume and poor dissolution of highly hydrophobic analytes. The developed system described in this study offers many advantages, including convenient, inexpensive, solvent-free and long lifetime. This system can be used for SPME method development, passive sampler evaluation and binding study with sediment and humic substances.

Table 2.4 Evaluation of various blades in terms of extraction amount and carryover of TCS

Coating	Solvent type	1st desorption ng	RSD %	2nd desorption ng	Carryover %
HLB	A	195.1	4.3	30.7	13.6
	B	243.1	9.3	42.6	14.9
	C	54.0	1.6	37.7	41.1
	D	98.5	12.5	60.9	38.2
C18	A	211.2	11.8	7.5	3.4
	B	133.7	12.3	1.6	1.2
	C	147.4	13.3	5.7	3.8
	D	122.2	12.9	5.4	4.2
PS-DVB	A	30.3	3.4	14.1	31.7
	B	23.9	16.8	8.8	26.8
	C	24.7	10.3	13.0	34.5
	D	45.6	10.0	17.6	27.9

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

Table 2.5 Evaluation of various blades in terms of extraction amount and carryover of Ben-2

Coating	Solvent type	1^{st} desorption ng	RSD %	2^{nd} desorption ng	Carryover%
HLB	A	954.0	6.7	14.8	1.3
	B	897.6	4.4	136.6	13.2
	C	385.3	1.5	104.0	21.3
	D	553.8	5.8	170.3	23.5
C18	A	97.4	4.9	2.2	2.2
	B	77.4	9.2	3.1	3.9
	C	64.0	8.8	1.0	1.6
	D	44.5	2.8	1.2	2.7
PS-DVB	A	124.2	8.5	9.0	6.7
	B	119.9	11.3	12.3	9.3
	C	146.1	11.5	20.0	12.1
	D	72.0	7.7	5.4	6.9

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

Table 2.6 Evaluation of various blades in terms of extraction amount and carryover of TCC

Coating	Solvent type	1st desorption ng	RSD %	2nd desorption ng	Carryover %
HLB	A	44.4	4.5	20.7	31.8
	B	46.0	5.8	20.9	31.3
	C	6.8	12.7	4.7	40.9
	D	10.1	11.7	13.9	57.8
C18	A	40.4	13.4	0.4	1.0
	B	19.8	12.3	0.2	1.5
	C	19.8	11.9	0.3	1.5
	D	18.6	6.0	0.2	1.2
PS-DVB	A	5.4	12.6	2.6	32.2
	B	2.5	11.3	0.7	21.6
	C	2.1	12.7	0.8	27.0
	D	2.7	13.3	1.3	32.3

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

Table 2.7 Evaluation of various blades in terms of extraction amount and carryover of Ben-4

Coating	Solvent type	1st desorption ng	RSD %	2nd desorption ng	Carryover %
HLB	A	57.9	9.5	1.5	2.5
	B	53.2	12.8	0.6	1.2
	C	23.2	6.3	0.9	3.7
	D	31.8	8.9	1.9	5.7
C18	A	8.3	0.9	1.0	10.7
	B	2.2	0.4	1.0	32.8
	C	2.2	8.1	0.6	21.9
	D	3.0	12.8	2.0	39.3
PS-DVB	A	19.2	8.5	3.3	14.6
	B	28.7	11.4	2.3	7.4
	C	22.9	6.8	1.9	7.8
	D	27.2	10.8	1.1	4.0

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

Table 2.8 Evaluation of various blades in terms of extraction amount and carryover of PBSA

Coating	Solvent type	1st desorption ng	RSD %	2nd desorption ng	Carryover %
HLB	A	12.6	8.1	0.5	3.8
	B	11.8	9.2	0.6	4.8
	C	6.7	5.1	0.9	11.8
	D	7.8	7.2	1.9	19.6
C18	A	6.8	5.2	0.9	11.7
	B	5.3	3.7	1.3	19.7
	C	5.3	6.1	0.6	10.2
	D	4.8	8.1	1.5	23.8
PS-DVB	A	4.6	5.9	1.8	28.1
	B	4.8	9.1	1.7	26.2
	C	4.9	8.1	1.9	27.9
	D	5.1	9.1	1.1	17.7

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

Table 2.9 Evaluation of various blades in terms of extraction amount and carryover of Ben-1

Coating	Solvent type	1st desorption ng	RSD %	2nd desorption ng	Carryover %
HLB	A	857.4	10.4	30.2	3.4
	B	750.6	1.9	104.7	12.2
	C	216.0	7.6	84.0	28.0
	D	565.2	9.4	169.0	23.0
C18	A	198.3	8.6	5.8	2.8
	B	19.3	4.1	0.5	2.5
	C	110.3	9.3	2.2	2.0
	D	124.2	8.9	1.5	1.2
PS-DVB	A	131.2	7.7	12.8	8.9
	B	111.9	11.6	15.7	12.3
	C	102.5	10.2	22.3	17.9
	D	86.8	4.5	9.3	9.7

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

Table 2.10 Evaluation of various blades in terms of extraction amount and carryover of OCR

Coating	Solvent type	1st desorption ng	RSD %	2nd desorption ng	Carryover %
HLB	A	12.0	7.4	4.9	28.9
	B	7.1	16.1	15.3	68.3
	C	2.2	9.3	1.3	36.7
	D	3.7	11.5	1.8	33.0
C18	A	11.1	11.2	0.3	2.6
	B	5.7	12.0	0.3	4.4
	C	6.2	10.3	1.1	15.0
	D	3.4	13.2	1.3	27.3
PS-DVB	A	4.6	12.9	1.7	26.7
	B	1.7	0.7	0.6	27.0
	C	0.9	3.0	0.6	40.8
	D	2.0	3.5	0.0	1.8

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

Table 2.11 Evaluation of various blades in terms of extraction amount and carryover of BM-DBM

Coating	Solvent type	1^{st} desorption ng	RSD %	2^{nd} desorption ng	Carryover %
HLB	A	2.8	10.0	0.1	4.0
	B	2.0	10.1	2.1	52.0
	C	0.6	7.7	0.8	57.9
	D	0.8	5.5	1.7	68.4
C18	A	7.9	4.7	0.3	3.6
	B	5.9	7.9	0.7	10.1
	C	1.6	7.0	0.6	28.1
	D	2.3	5.0	0.9	27.6
PS-DVB	A	1.9	3.3	1.9	50.6
	B	0.7	8.2	0.6	44.4
	C	0.6	10.0	0.7	53.3
	D	0.9	12.5	0.4	30.7

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

Table 2.12 Evaluation of various blades in terms of extraction amount and carryover of OMC

Coating	Solvent type	1^{st} desorption ng	RSD %	2^{nd} desorption ng	Carryover %
HLB	A	5.8	13.9	1.9	25.0
	B	2.8	10.4	1.6	36.2
	C	8.6	6.9	3.7	30.1
	D	1.5	6.0	0.8	35.1
C18	A	12.1	7.0	0.4	3.2
	B	1.9	9.8	0.6	23.2
	C	7.6	1.7	4.5	5.9
	D	2.5	7.9	0.2	8.2
PS-DVB	A	10.1	8.3	1.9	16.0
	B	7.6	7.1	0.8	9.2
	C	0.6	2.0	0.3	31.2
	D	1.5	7.3	0.1	3.4

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

Table 2.13 Evaluation of various blades in terms of extraction amount and carryover of Ben-3

Coating	Solvent type	1st desorption ng	RSD %	2nd desorption ng	Carryover %
HLB	A	652.1	1.8	23.0	3.4
	B	748.2	6.9	98.5	11.6
	C	189.8	6.7	94.5	33.2
	D	607.1	8.0	197.8	24.6
C18	A	540.0	7.5	23.3	4.1
	B	106.7	7.4	2.6	2.4
	C	311.4	2.1	7.2	2.3
	D	279.6	5.5	10.6	3.6
PS-DVB	A	61.4	7.6	16.8	21.5
	B	57.9	4.2	10.2	15.0
	C	32.7	7.3	16.5	33.5
	D	52.3	7.5	11.4	17.9

A: (50/25/25) MeOH/IPA/ACN

B: (40/40/20) MeOH/ACN/H_2O

C: (50/50) ACN/H_2O

D: (80/20) MeOH/H_2O

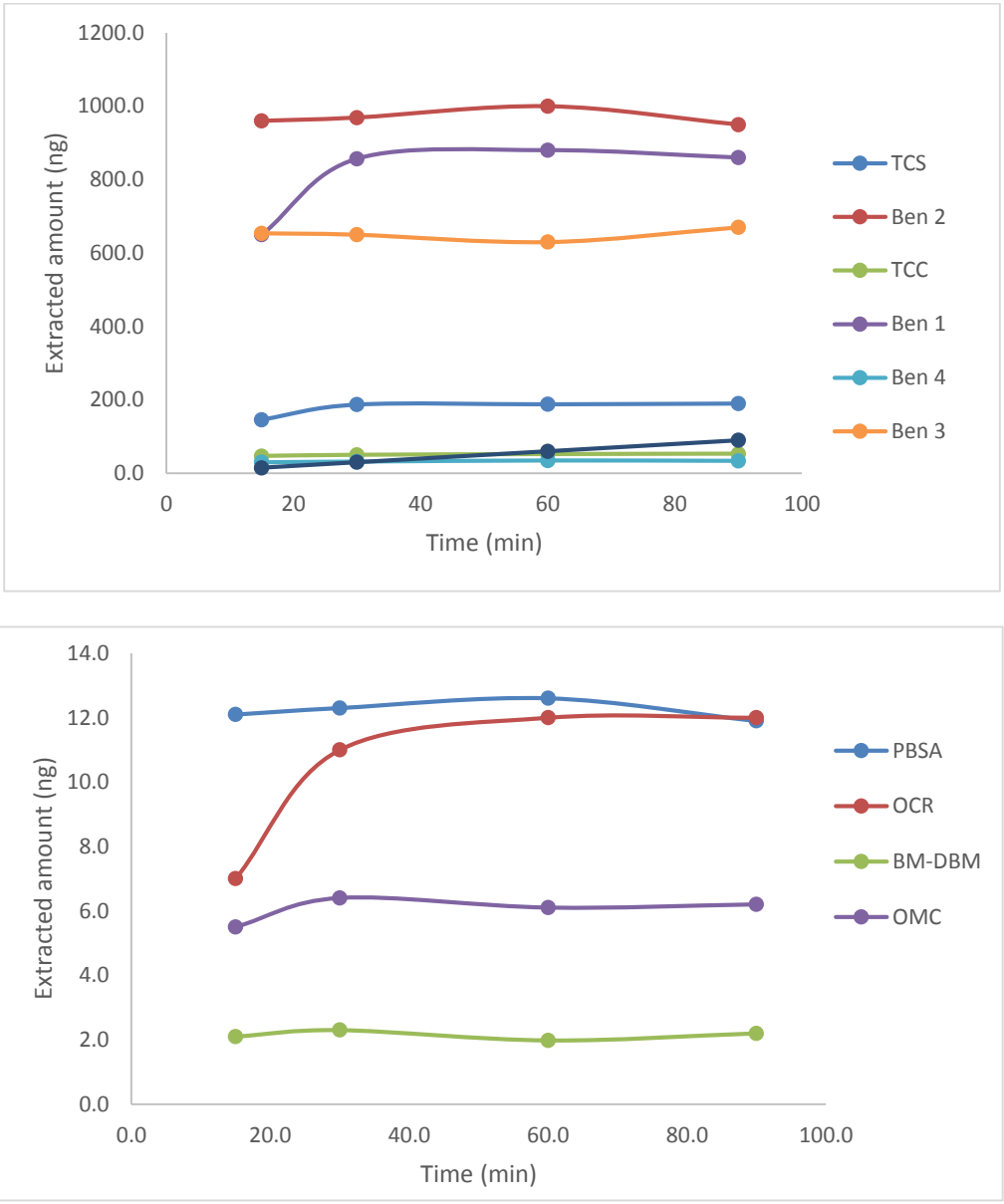


Figure 2.3 Desorption time profile for analytes under study, extraction from ASG

Chapter 3
Time weighted average concentration monitoring based on thin film solid phase microextraction

3.1 Introduction

The environmental impact of biocides and UV filters has gained increasing interest as these ingredients can be found in various products used in everyday life, such as personal care products (PCPs), cleaning agents, paints, and coatings. [132-133] They are used in large quantities on a daily basis, and are continuously discharged into municipal wastewater treatment plants. Eventually, these compounds are dispersed into the aquatic environment through wastewater effluent discharges,[9-134] where they can reach detectable and potentially harmful concentrations.[135] Monitoring their presence at trace level concentrations is an ongoing challenge for analytical chemists. Sampling is the first and most time-consuming step in the entire analysis procedure, and can be achieved either through grab/spot sampling, or passive sampling methods. However, the spot sampling method can only provide information regarding the system at the time and point of sampling. Consequently, spot sampling may not provide a realistic picture of aquatic-life continuous exposure to PCPs.[29] When levels of contaminants fluctuate within a body of water, traditionally collection of large numbers of samples over a long period of time is needed to obtain a time weighted average (TWA) water concentration, a process which is time consuming, laborious, and costly. [24-25] Conversely, passive sampling methods, as an alternative option to the process described above, are now

widely used for monitoring of TWA concentrations of environmental contaminants.[26,27] The passive sampling approach is based on the free movement of analytes from sample matrix to the extraction phase. Differences in the chemical potential of the analytes between two media cause accumulation of the analytes under study into the extraction phase.[29-30] Passive sampling eliminates power requirements, reduces analysis cost, and also prevents decomposition of the analyte during storage and transportation.[31] Several passive sampling methods are used for monitoring of organic contaminants in water, including semi-permeable membrane devices (SPMDs),[33,136] polar organic chemical integrative samplers (POCIS),[35,36] Chemcatcher,[24,37] ceramic dosimeters,[27,38] polymeric samplers,[39,40] diffusive gradients in thin film,[41,42] and membrane enclosed sorptive coatings.[137] Solid phase microextraction (SPME) was developed as a solvent-free sample preparation technique that integrates sampling, extraction, and pre-concentration in a single step.[69] Due to these advantages, it has been widely used for on-site sampling of a broad range of target analytes in different matrices.[80-138]A SPME retracted device, in which the SPME fiber is retracted a certain distance into the needle housing during the sampling, has been used for TWA sampling in air[139-140] and water[90-87] for volatile and semi volatile compounds. However, there are some limitations associated with this sampler as hydrophobic compounds are lost along the diffusion path.[141] Additionally, the low surface-to-volume ratio of the SPME fiber may constrain the sampling time, as well as the sensitivity of the method.[142] Moreover, the chemistry of the coating is mostly limited to PDMS, which is not an appropriate coating for hydrophilic analytes, and does not act as a zero sink for volatile compounds.[139, 141] Thin film solid phase microextraction (TF-SPME) provides an increase in

the surface area-to-volume ratio of the sampling device and has been introduced to overcome the low-capacity limitations and low extraction rates of traditional SPME fibers.[77-78] TF-SPME was applied for TWA sampling of polycyclic aromatic hydrocarbons (PAHs) in water, and the kinetic calibration technique using an isotopically labelled standard was used for quantitation.[90,95] However, there are factors that limit the application of traditional kinetic calibration for multiple compound analysis, since preloading of a number of calibrants is a complex process, and isotopically labelled counterparts may not always be available or affordable for all target analytes. To address these limitations, Ouyang introduced a one-calibrant kinetic calibration technique, where one isotopically labelled standard was used in order to quantify multiple analytes of interest. [143]

In this study, we propose two types of TF-SPME passive sampler, namely a retracted TF-SPME device and an open bed configuration TF-SPME device. This proposal offers an integrated approach for TWA determination of analytes, with a wide range of physical-chemical properties, for on-site application. The proposed methods were investigated in a flow-through aqueous standard generation system (ASG) which provides accurate calibration data of the devices over a wide polarity range of PCP chemicals. The use of a flow-through system allowed for the provision of an environmentally realistic calibration of hydrophobic compounds without the samplers depleting the system. The samplers were then used in the Grand River (ON, Canada) for on-site application.

3.2 Experimental

3.2.1 Chemical and materials

All chemicals and reagents utilized in this study were described in 2.2.1. The internal standard (IS) 2-hydroxy-4-methoxybenzophenone-2',3',4',5',6'-d5 (Ben-3-d5), was obtained from CDN isotopes (Pointe-Claire, QC, Canada).

3.2.2 Instrumental analysis method (LC/MS/MS

The LC-MS/MS method was described in 2.2.2.

3.2.3 Aqueous standards generation system

The ASG system was described in 2.3. The system reached a constant value after an initial induction period of 1 week for all compounds, and showed variations in concentrations of less than 20% within three months period. Average concentrations of individual analytes were 377, 138, 28, 459, 118, 3.26, 0.35, 0.08, 1.26 and 0.12 $ngmL^{-1}$ for PBSA, Ben 1, Ben 3, Ben 4, Ben 2, TCS, OCR, OMC, TCC and BM-DBM, respectively.[99]

3.2.4 Retracted thin film solid phase microextraction

The design of the retracted TF-SPME TWA sampler is illustrated in Figure 3.1. The sampler consisted of a copper tube, caps, a Teflon rod assembly, a Teflon spacer (all made at the Machine Shop at the University of Waterloo), and a thin film (HLB) coated blade made in the laboratory. The copper tube was made from a copper rod that was drilled, creating a hole with a 0.76 mm inner diameter and 10.0 mm length (diffusion path). To avoid the trapping of air bubbles in the sampler, all parts of the sampler were assembled under ultrapure water. Assembly of the sampler was conducted by first locating the Teflon spacer inside the copper

tube, followed by insertion of the thin film on the Teflon holder inside the copper tube, and finally, tightly screwing the cap to fix the blade in place. The sampler was then removed from the ultrapure water basin and transferred to the sampling chamber.

3.2.5 Open bed thin film solid phase microextraction

Open bed TF-SPME coated with C18 (Figure 3.1) was evaluated for monitoring of hydrophobic compounds with the one-calibrant kinetic calibration approach. The calibrant was first loaded on the coating, then subsequently transferred to the sampling chamber of the ASG to investigate isotropism between absorption and desorption. The ASG provides a steady state free concentration of the analytes during the experiment. Extraction times ranged from 1 h to 250 h. Benzophenone-3 (Ben-3) and Benzophenone-3-d5 (Ben-3-d5) were used as analyte and calibrant, respectively.

3.2.5.1 Loading of the calibrant on open bed TF-SPME

C18 was used as a coating in the TWA passive sampler in open bed configuration. The initial loading of the calibrant (Ben-3-d5) was optimized and subsequently used for evaluation and on-site sampling. The amount of calibrant (150ng) on the coating should be sufficient to be detected by the instrument after sampling. Loading was performed by extraction from an aqueous solution composed of 100 ng mL^{-1} of the calibrant in a 2 mL amber vial for 60 min at 800 rpm agitation. The amount of loaded calibrant was calculated after desorption and analysis by the instrument, and quantified by external calibration. The relative standard deviation of the loading procedure was less than 7%.

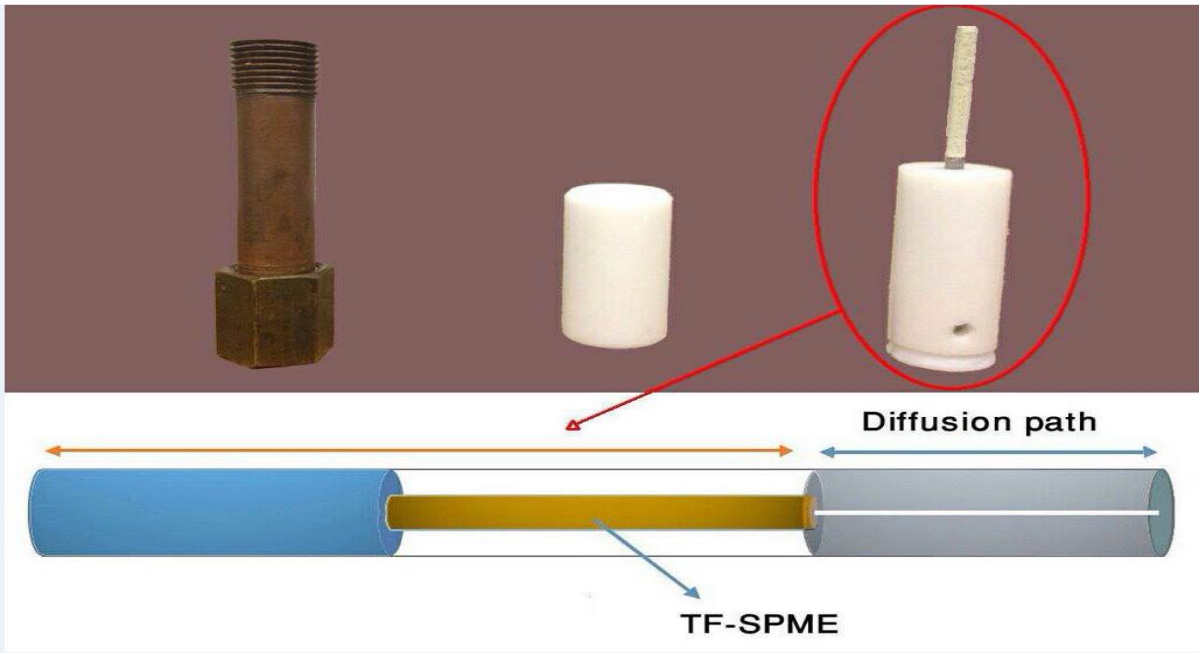


Figure 3.1 Retracted TF-SPME TWA sampler

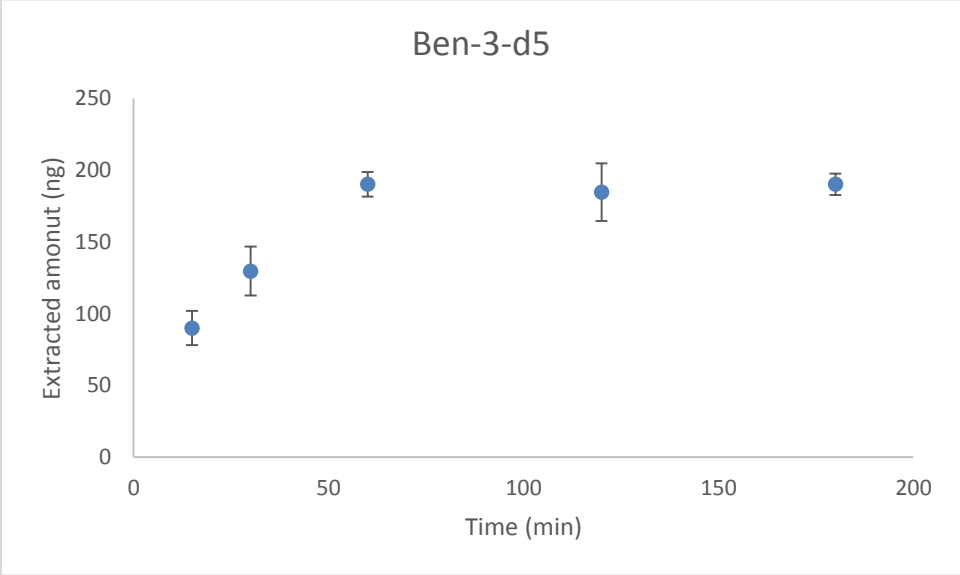


Figure 3.2 Extraction time profile of Ben3-d5 (n=3)

3.2.6 Preparation of field samplers and on-site sampling procedures

On site sampling was performed with a retracted TF-SPME sampler and an open bed C18 TF-SPME device. Three retracted devices with a 10 mm diffusion path were assembled under ultrapure water in the laboratory, and the opening of the device was covered by copper mesh in order to prevent biofouling from the sampling environment. The samplers were transported to the sampling location in appropriate containers (Figure 3.3 a) to ensure that individual passive sampling devices remained isolated from the environment, potential sources of contamination and each other during storage, transport to the deployment site and return to the laboratory following retrieval. Three open bed C18 TF-SPME devices were loaded with calibrant and wrapped with aluminium foil, then transported in a cold box filled with dry ice to the sampling location. Upon arrival, SPME devices were inserted individually into copper bags (Figure 3.3 b) to secure them in the sampling environment. All samplers were then placed in the plastic cage (Figure 3.3 c) and deployed at the sampling site. The sampling time selected was based on the convection conditions of the river water and was 90 days for the retracted devices, while the sampling time for the open bed configuration was set to 5 days. Once the sampling time had elapsed, the samplers were retrieved and transferred into a desorption solution, then transported to the laboratory where they were analyzed by LC-MS/MS. For comparison, spot samples were collected concurrently and from the same location as the passive sampling experiments.

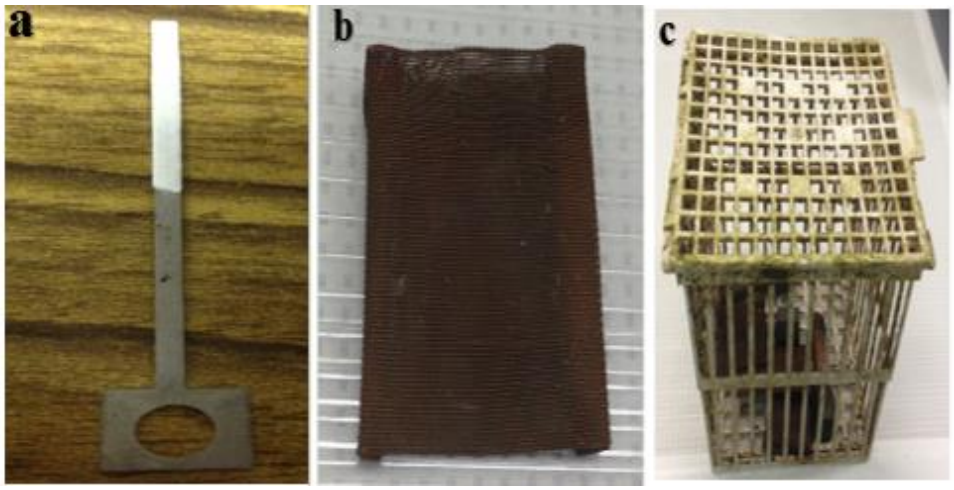


Figure 3.3 Thin-film passive samplers: (a) C18 thin-film sampler, (b) copper mesh, (c) samplers' cage

Two different types of spot sampling approaches were used, namely grab sampling followed by laboratory analysis and equilibrium on-site sampling with an open bed TF-SPME. Different spot sampling methods were selected in order to provide adequate sensitivities necessary for the analyses. Grab sampling in bottle was selected for determination of the non-polar analytes with C18 TF-SPME and external calibration method. However On–site sampling using open bed HLB TF-SPME with equilibrium extraction was used for polar analytes as better sensitivity for determination can be obtained due to large sample volume. Two spot samples were retrieved at the sampling location at the time of deployment and retrieval of the TWA sampler. Samples were stored in amber glass bottles previously washed with acetone, methanol, and ultrapure water, then transferred to the laboratory and stored at 4◦C until extraction. Spot samples were analyzed in triplicate in a 250 mL amber bottle by external SPME calibration technique, using HLB TF-SPME as the extraction phase. It should be noted

that loss of analyte onto the glassware was shown to be negligible. On-site equilibrium spot sampling with open bed TF-SPME was performed every 10 days in parallel to retracted TF-SPME TWA sampling. Three open bed TF-SPME devices were exposed directly into the river at the sampling point for 10 days. After extraction, the devices were desorbed in MeOH/ ACN/IPA (50/25/25, v/v/v) and desorption solvents were evaporated to dryness under N_2. The residues were reconstituted in 0.30 mL MeOH/H2O (50/50, v/v), which was equivalent to the initial solvent composition in the chromatographic method, and subsequently analyzed by LC-MS/MS. The quantification of analytes in the samples was performed using the equilibrium method. Equilibrium sampling not only provides high sensitivity for detection of analytes, it also avoids the need for transportation of samples to the laboratory.

3.2.7 Blank samples

Two blank samples were prepared and considered in all steps, including preparation, assembling, transportation, storage, deployment, and retrieval (Standard ISO-5667). The procedural blank sampler was used to evaluate if any possible contamination occurred during preparation, assembling, loading of the calibrant, storage, transportation, processing, and analysis. Field blank was exposed to ambient air during deployment and retrieval of samplers. Both blank sample were transferred to the laboratory and stored at -20 °C until processing.

3.3 Results and discussion

3.3.1 Thin film SPME passive samplers

The main objective of this study was to identify and evaluate TF-SPME approaches for the TWA determination of PCP compounds which possess a wide range of polarity. In preliminary

evaluations, it has been found that relatively polar analytes such as Ben-1, Ben-2, Ben-3, Ben-4 and PBSA can reach equilibrium in a short time if they are exposed directly to the sampling location (open bed configuration). Thus the retracted device, in which the coating is placed in a tube with a well-defined path length with analytes diffusing through the narrow opening to the coating, was selected. This geometry allows longer equilibration time in extraction which means longer TWA sampling is possible and use of a simple diffusion based calibration approach (see the next section for details). According to the results in Table 2.4 to Table 2.13, it can be concluded that HLB coating has higher affinity for the polar compounds compared to the C18 extraction phase. Thus taking into account the necessity of high sensitivity of the final method and considering the fact that the high affinity coatings can provide a longer time to reach equilibrium, HLB was selected as the coating for retracted TF-SPME samplers.

For non-polar compounds retracted devices may not provide reasonable TWA concentrations as the measurements may suffer from non-specific secondary interactions of the analytes within the diffusion path of the sampler. Thus for the non-polar compounds in this study (OCR, OMC, TCC, TCS and BM-DBM) an open bed geometry was selected which can be easily calibrated with a kinetic calibration approach. It was found that the C18 coated TF-SPME provided better desorption of the calibrant from the sampler to the solution than the HLB coated TF-SPME sampler which makes C18 coating a more suitable extraction phase for kinetic calibration and therefore was selected for the open bed TF-SPME device. Consequently, to cover a wide range of compounds two different TF-SPME passive samplers have been selected. Retracted HLB coated TF-SPME with diffusion based calibration approach

was used for relatively polar compounds (log P < 4) and open bed C18 coated TF-SPME with kinetic calibration was employed for relatively non-polar compounds log P > 4. The key characteristics of the two approaches are described in the following sections.

3.3.2 Retracted TF-SPME TWA sampler

To perform TWA sampling with retracted TF-SPME, three basic prerequisites have to be met.[142,144] First, the coating of the TWA sampling device should act as a zero sink for all of the analytes - the mass uptake rate should not be influenced by the amount of analyte already sorbed. A zero sink test was performed by continuous and intermittent exposure of the thin film in the ASG for the same extraction time.[139] A t-test was conducted to compare the results obtained for the different sampling regimes, indicating that there was no statistically significant difference between mass amounts adsorbed for intermittent and continuous exposure. Thus, it could be concluded that the HLB coating behaved as a zero sink for all of the target analytes due to its strong affinity towards the analytes under study, and the large capacity of the coating. The second requirement is that the passive sampler should respond proportionally to changes of analyte concentrations at the face of the device. The capability to integrate high peak concentrations is an important function of any passive sampler. This function is directly related to the response time of the sampler, where the response time is the mean dwelling time of an analyte in the diffusion zone of the sampler.[142] For the thin film retracted device with a diffusion path length of 10.0 mm, the response time for the analytes is 4-10 h. Nevertheless, response time is negligible in comparison to sampling time, which could be as long as 90 days. The response time can be changed by designing a passive sampler with a shorter or longer

diffusion path. The third condition is that the bulk concentration (C_{bulk}) of analytes must be equal to the concentration at the face of the device ($C_{bulk}=C_{face}$). The overall mass-transfer resistance for transportation of analyte from the bulk of the samples to the collecting medium of passive samplers should be limited to the diffusion path of the sampler. To confirm the third condition, a face velocity effect test was carried out in a well-agitated sampling chamber (800 rpm, linear flow rate was ~50 cm sec^{-1}) and in a mixing chamber where the linear velocity was low (0.15 cm sec^{-1}). The sampling time was 30 days, and each measurement was performed in triplicate. The obtained results showed that there was no significant difference between accumulated masses in the samplers for both conditions. This is a desired feature of the sampler for on-site applications, where convection conditions are difficult to measure and calibrate.

3.3.2.1. Calibration of retracted TF-SPME passive sampler

Laboratory calibration of the TWA sampler was performed by exposing nine samplers in the ASG simultaneously for different time intervals. Since the diffusion of analytes in stagnant water between the thin film and the opening of the sampler is controlled by mass-transfer, the diffusion is assumed to follow Fick's first law under a steady state condition, and the mass uptake can be calibrated by use of Fick's first law of diffusion (Equation 3.1).

$$C = \frac{nZ}{ADt} \qquad \text{Equation 3.1}$$

Where C is the TWA concentration, n is the amount of analyte extracted, Z is the diffusion path length, A is the cross-sectional area of the opening, D is the diffusion coefficient, and t is sampling time. The diffusion coefficients of the uncharged organic molecules and charged organic molecules in water can be calculated with the empirical Equation 3.8 and Equation

3.9. The ratio of theoretical sampling rate (R) to analyte diffusion coefficient (D) depends on the geometric configuration of the sampler only, i.e. (R/D) = A/Z. The ratio of theoretical sampling rate to the experimental sampling rate (SR) should be equal to 1 which verifies that the sampler can be calibrated by diffusion based calibration. SR values of each compound can be determined experimentally with a sampler with known diffusion path. For this purpose, samplers with three different diffusion paths were exposed in the sampling chamber with known concentrations of analytes for a defined time. The sampling rate could then be calculated with Equation 3.2:

$$SR = \frac{n}{Ct} \qquad \text{Equation 3.2}$$

Table 3.1 summarizes the experimental results for (RZ)/(AD) at 5.0, 10.0, and 15.0 mm diffusion path lengths. The obtained results show that the values of (RZ)/ (AD) for all analytes are close to 1. This indicates the zero sink behavior of the HLB coating for the target analytes. Figure 3.4-Figure 3.8 demonstrate good correlations between the theoretical and experimental data, as well as good linear relationships between mass uptake and sampling times of up to 70 days. According to the trends observed for the amount of analytes extracted, sampling time can potentially be further extended, because the thin film coating has a sufficiently large capacity. The experimental results indicated that retracted TF-SPME passive sampling by HLB coating works very well for TWA sampling of hydrophilic compounds in water. The retracted TF-SPME device was shown to meet all the required criteria for successful performance as a TWA sampler, i.e. zero sink behaviour, fast response, and independence from face velocity. The disadvantage of this sampler is its low sampling rate; accordingly, it should be ensured

that sampling time is sufficiently long to enable measurement of TWA concentrations in real environmental application.

Table 3.1 Comparison of experimental values of (RZ)/ (AD) (n=3)

	(RZ)/ (AD)		
	Z=0.5 cm	Z=1 cm	Z=1.5 cm
Ben 2	0.98±0.11	0.92±0.13	1.1±0.12
Ben 4	0.96±0.16	0.88±0.18	0.98±0.15
PBSA	1.1±0.15	0.96±0.15	0.89±0.16
Ben 1	0.89±0.15	0.91±0.18	0.93±0.20
Ben3	0.99±0.19	0.92±0.14	0.97±0.21

3.3.3 Open bed TF-SPME passive sampler

In the retracted device described in the previous section the boundary layer thickness is constant (diffusion path) and it is not affected by convection condition; therefore, the calibration is straightforward and follows Fick's law. In open bed configuration, however, since the sampler is directly exposed into the sampling location, the boundary layer is strongly dependent on the convection conditions, thus a calibrant is necessary for adequate calibration. The application of a kinetic calibration process was assessed in the ASG. This included the evaluation of the symmetry of adsorption and desorption of the analytes as well as an

investigation into the possibility to use a single calibrant for all tested compounds. The zero sink test was performed for C18 coating and analytes (Ben-3, OCR, OMC, TCC, TCS and BM-DBM). The results showed that C18 coating behaved as a zero sink for all of the analytes.

3.3.3.1 Symmetry of desorption and adsorption verification

To ensure the applicability of the method, the existence of isotropic behaviour of adsorption of the analyte and desorption of the calibrant should be verified. This experiment involved the simultaneous determination of the desorption time profile of Ben-3-d5 as a calibrant, and the extraction time profile of Ben-3. Pre-loaded C18 thin film blades were exposed in the sampling chamber of the ASG for different sampling times in triplicate at a linear velocity of 50 cm sec^{-1}.

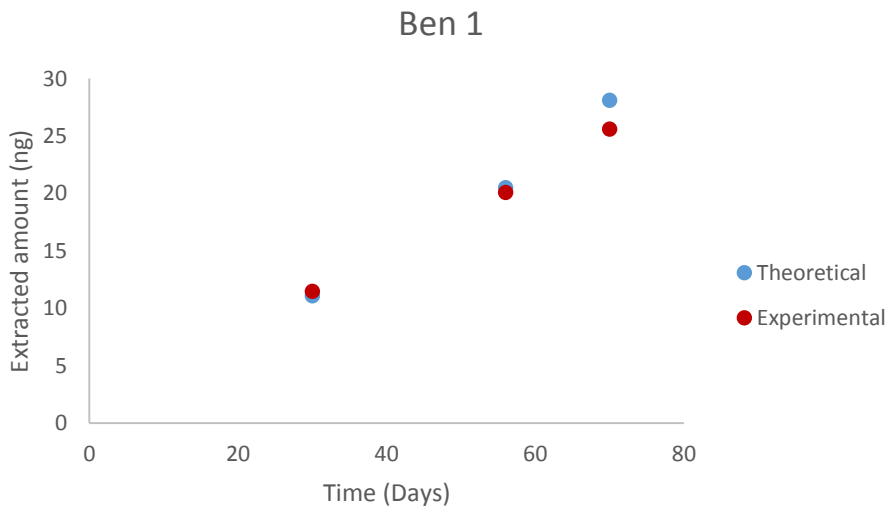


Figure 3.4 TWA concentration of Ben-1 using the retracted TF SPME sampler (diffusion path: 10 mm and analyte concentration in the ASG: 138 ng mL^{-1})

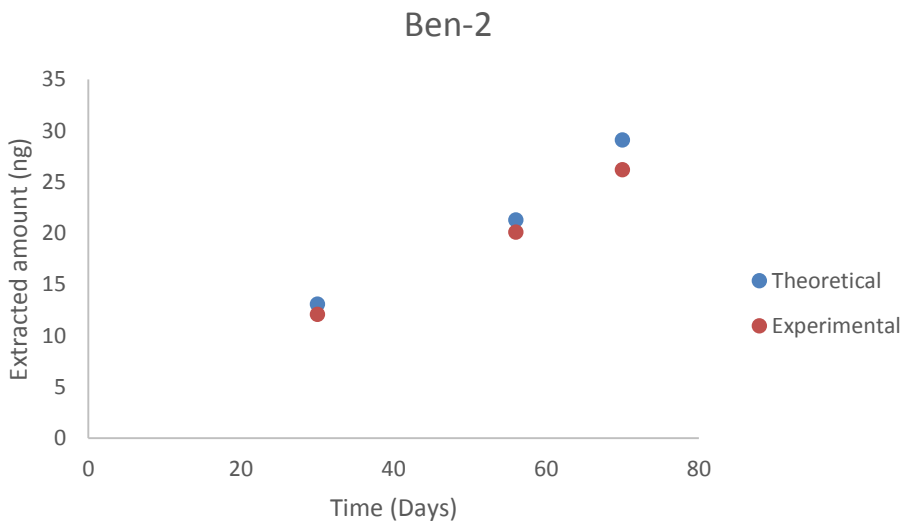


Figure 3.5 TWA concentration of Ben-2 using retracted TF-SPME (diffusion path: 10 mm and analyte concentration in the ASG:118 ng mL^{-1})

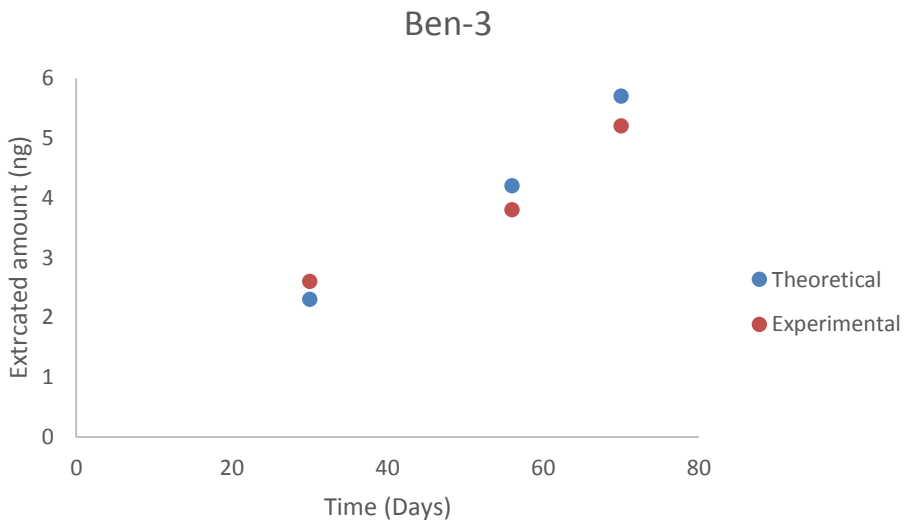


Figure 3.6 TWA concentration of Ben-3 using retracted TF-SPME (diffusion path: 10 mm and analyte concentration in the ASG: 28 ng mL^{-1})

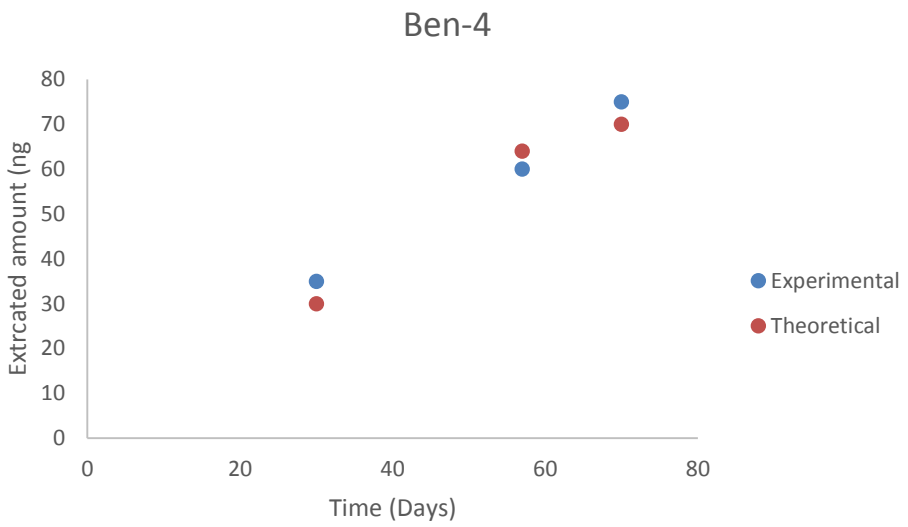


Figure 3.7 TWA concentration of Ben-4 using retracted TF-SPME (diffusion path: 10 mm and analyte concentration in the ASG: 459 ng mL-1)

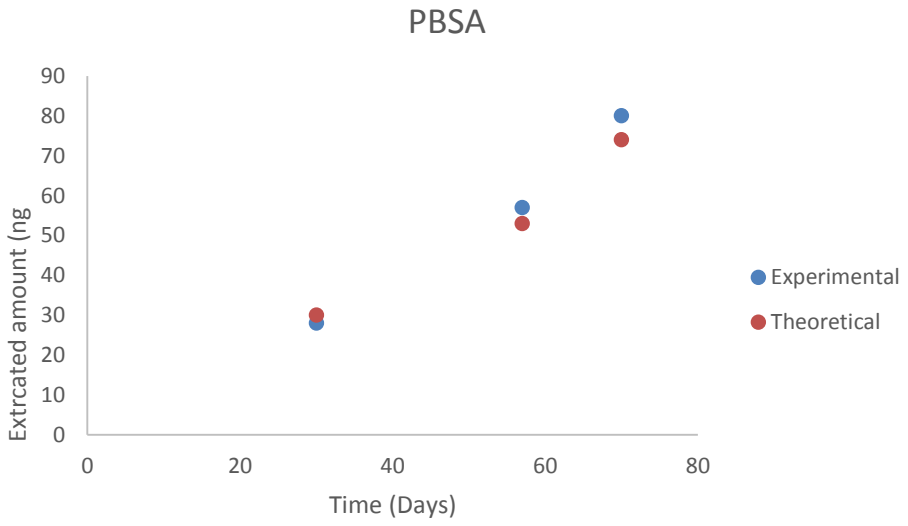


Figure 3.8 TWA concentration of PBSA using retracted TF-SPME (diffusion path: 10 mm and analyte concentration in the ASG: 377 ng mL-1)

After each specified extraction time, the thin film blades were removed from the solution and desorbed in the selected desorption solvent. Figure 3.9 (A) presents the isotropy of the adsorption and desorption processes. The desorption process of the pre-loaded C18 thin film blades can be described by the Equation 3.3:

$$\frac{Q}{q_0} = \exp(-a't) \qquad \text{Equation 3.3}$$

Where q_0 is the amount of loaded calibrant on the extraction phase, Q is the amount of calibrant remaining in the extraction phase after exposure to the sample matrix, a′ is the desorption rate constant of the preloaded calibrant and t is sampling time. The kinetics of the absorption process for the TF-SPME can be defined using the Equation 3.4:

$$\frac{n}{n_e} = 1 - \exp(-at) \qquad \text{Equation 3.4}$$

Where n is the amount of extracted analyte at sampling time t, ne is the amount of analyte extracted onto the coating at equilibrium and a is the absorption rate constant of the analytes. The sums of Q/q_0 and n/ne at any time were close to 1, demonstrating the isotropy of the method (Equation 3.5).

$$\frac{n}{n_e} + \frac{Q}{q_0} = 1 \qquad \text{Equation 3.5}$$

The linearized absorption and desorption time constant profiles can be obtained with ln (1 - n/n_e) or ln (Q/q_0) as the y-axis, where the regression slope is –a, based on Equation 3.3 and Equation 3.4. The obtained data exhibited a good linear relationship between ln (1 - n/ne) or ln (Q/q_0) and time (R >0.99), demonstrating that Equation 3.3 and Equation 3.4

accurately describe the kinetics of desorption and absorption of the C18 coating towards UV filters and biocides (Figure 3.9 B, C). Where a is the adsorption rate constant, and a' is the desorption rate constant. The rate constant a is described by Equation 3.6:

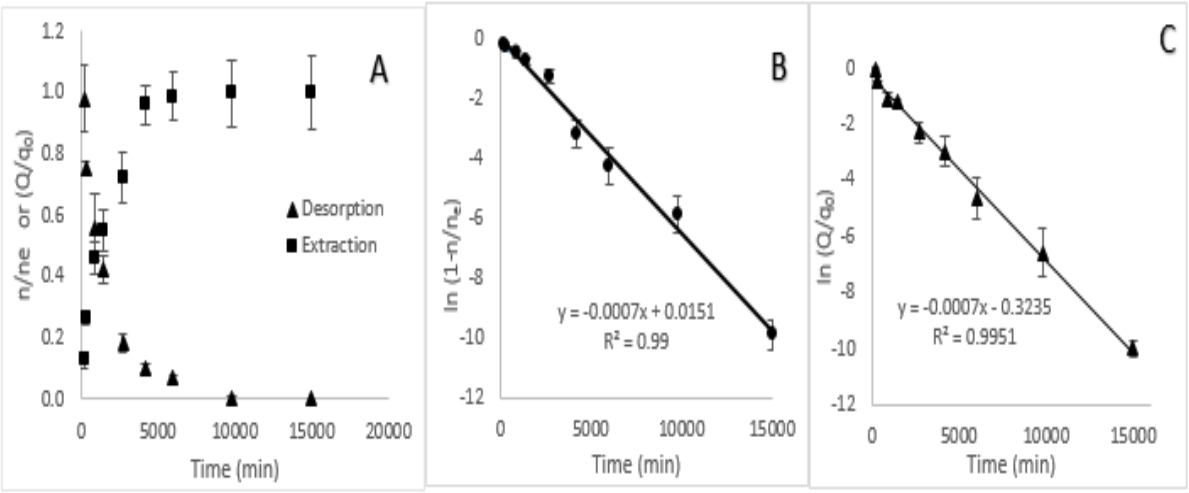


Figure 3.9 The isotropy of absorption and desorption of Ben-3 and Ben-3-d5 (A) Extraction time profile of Ben-3 (B) and desorption time profile of Ben-3-d5 (C)

$$a = \frac{D_s A}{V_f \delta_s K}$$ Equation 3.6

Where K is the distribution coefficient of the analyte between the C18 coating and the sample matrix, A is the surface area, Vf is the volume of the coating, Ds is the diffusion coefficient of the analyte in the sample matrix; and δs is the thickness of the boundary layer, which is mainly affected by the agitation. Desorption of the calibrant follows Equation 3.7.

$$a' = \frac{D'_s A}{V_f \delta'_s K'}$$ Equation 3.7

Where D′s is the diffusion coefficient of the calibrant in water, and K′ is the distribution coefficient of the calibrant between the C18 coating and sample matrix. Since the thickness of the diffusion layer, the surface area, and the volume of the coating are the same for both the extraction of the target analytes (δs) and desorption of the calibrant (δ′s), then the relationship between the rates constants a and a' can be shown by Equation 3.10. The distribution coefficients of the analytes and calibrant between the C18 coating and water can be determined experimentally. The diffusion coefficients for uncharged and charged organic molecules are calculated with the Equation 3.8 and Equation 3.9:

$$D_{w=}\frac{1326\times10^{-4}}{\eta w^{1.14}V^{0.589}} \qquad \text{Equation 3.8}$$

$$D=\frac{RT}{F^2}\left(\frac{\frac{1}{n^+}+\frac{1}{n^-}}{\frac{1}{\lambda_+^\circ}+\frac{1}{\lambda_-^\circ}}\right) \qquad \text{Equation 3.9}$$

Where D is the liquid-phase diffusion coefficient at infinite dilution, cm^2s^{-1}; R is the universal gas constant, 8.314 J $(mol\cdot K)^{-1}$; T is the absolute temperature, K (273 + ◦C); n+ is cation valence and n− anion valence; F is Faraday's constant, 96,500 Ceq-1; λ◦+ = limiting positive ionic conductance, $cm^2\cdot seq^{-1}$ and λ◦− limiting negative ionic conductance, $cm^2\cdot seq^{-1}$. Limiting ionic conductance of Ben-4 and PBSA were obtained by the conductivity method, and measured at 30 and 25 $cm^2\cdot seq^{-1}$, respectively.[145]

3.3.3.2 One-calibrant kinetic calibration

Verification of the one calibrant kinetic calibration approach for measuring the TWA concentration of the hydrophobic analytes (OCR, OMC, BM-DBM, TCC, and TCS) was

performed in the ASG. Ben-3-d5 was selected as the calibrant and absorption rates related to the analytes were calculated with the use of Equation 3.10:

$$\frac{a}{a'} = \frac{DsK}{D'sK'} \qquad \text{Equation 3.10}$$

Where D_S' is the diffusion coefficient of the calibrant in pure water, and K' is the distribution coefficient of the calibrant between the C18 coating and sample matrix, K is the distribution coefficient of the analyte between the C18 coating and the sample matrix and D_S is the diffusion coefficient of the analyte in the sample matrix. Equation 3.10 can be used to specify the relationships that exist between the desorption rate constant of the calibrant and the absorption rate constants of different compounds; accordingly, all extracted analytes can be quantified with a single pre-loaded calibrant. The partition coefficients of the analytes (K) and the calibrant (K') were determined by equilibrium extraction of TF-SPME in the ASG (Table 3.2). The partition coefficient of Ben-3 was used for the calibrant Ben-3-d5, as both compounds display the same retention time in the chromatographic separation on the C18 column, which indicates the similar physical-chemical properties between Ben-3-d5 and Ben-3. To investigate the validity of Equation 3.10 for determination of TWA concentrations of other analytes, three C18 TF-SPME devices were loaded with the calibrant and exposed in the sampling chamber at 800 rpm for 15 hrs, where the kinetics of uptake is still in the linear portion of the extraction time profile, and the obtained concentrations can be assumed to be TWA concentrations. The concentration of the analytes in the matrix sample in the kinetic portion of the extraction time profile can be obtained with the Equation 3.11:

$$C_s = \frac{n}{KV_f[1-\exp(-at)]}$$ Equation 3.11

Since a=a', then Equation 3.11can be rewritten as the Equation 3.12**:**

$$C_s = \frac{n}{KV_f\left[1-\exp\left(\frac{Q}{q_0}\right)t\right]}$$ Equation 3.12

Where C_s, K and V_f are the concentration of the analyte in the sample matrix, distribution coefficient between the coating and analyte and volume of the coating respectively. Equation 3.12 demonstrates that the open bed samplers can be deployed with a pre-loaded calibrant into the sample medium, and retrieved after the sampling period has elapsed. The concentration or TWA concentration of the target analyte can then be calculated, since n and Q can be determined, and K, V_f, and q_0 are already known. Liquid-liquid extraction with ethyl acetate was used to measure the concentrations of the analytes in the ASG as a confirmatory approach. The quantitative results are shown in Table 3.2. It is clear that one-calibrant kinetic calibration is able to quantify TWA concentrations of other analytes in cases where their deuterated counterparts are either not available or too expensive.

3.3.4 Determination of distribution coefficient

The extraction time profiles of the hydrophobic analytes (TCS, TCC, OMS, OCR and BM-DBM) using TF-SPME C18 coating were investigated in the ASG. TF-SPME coated samplers were placed in the sampling chamber at 800 rpm from 30-7230 min in triplicate to determine the equilibrium time. Equilibrium time was defined as the time when the extracted amount is statistically constant. The distribution coefficient was obtained based on $n = K_{fs}V_fC_0$ Equation 3.13,

$n = K_{fs}V_fC_0$ Equation 3.13

When the amount of extracted analyte in equilibrium and initial concentration are known. The volume of the thin film blade coatings was calculated based on the length (l = 20 mm) and thickness (b = 200 μm) of the coating, and the width (w = 2.5 mm) and depth (d = 0.7 mm) of the blades, using the .[146]

$$V_f = 2[lb(w + 2b)] + 2[lb(d + 2b)] + [b(d+2b)(w+2b)]$$ Equation 3.14

3.3.5 Limit of detection

3.3.5.1 Limit of detection of the TWA samplers

KfsVfC0VsKfsVf+Vs

Equation 3.15 and can be simplified to $n=K_{fs}V_fC_0$ Equation 3.13 when Vs>>KfsVf in ASG

$$n = \frac{K_{fs}V_fC_0V_s}{K_{fs}V_f+V_s}$$ Equation 3.15

Where n, V_s,$K_{fS}V_f$, and C_0 are the amount of extracted analyte, volume of sample, fiber constant and initial concentration. Equation 3.15 was used for calculation of the LOD of the passive sampler when n (absolute detection limit of the instrument) and fiber constant were known. The absolute instrumental detection limit was obtained by multiplying the known instrumental detection limit and desorption solvent volume. For the retracted device, equation 1 was used for calculating the limit of detection when sampling time, diffusion path, and absolute instrumental detection limit were known. The sampling time and diffusion path were 90 days and 10.0 mm, respectively.

Table 3.2 Values of distribution coefficient (K), diffusion coefficient (Ds, cm^2s^{-1}), absorption rate (a) and TWA concentrations of analytes in ASG (ng L^{-1}) (Mean± Standard Deviation; n =3)

Analytes	$D_s \times 10^{5a}$	K^b	$a \times 10^{4b}$	C_{TWA}	C_{ASG}
OCR	0.408	68000±4760	0.434±0.039	250±30	240±20
OMC	0.458	11800±590	2.81±0.14	80±10	80±6
BM-DBM	0.460	118000±9440	0.244±0.030	120±20	120±10
TCS	0.358	20000±1000	1.25±0.11	3200±400	3300±300
TCC	0.536	14000±840	2.75±0.31	1200±150	1300±100
Ben-3-d5	0.508	5600±168	6.55±0.52	28000±350	28000±220

a: Calculated

b: Determined

3.3.5.2 Limit of detection of the grab samplers

Two different grab sampling techniques were used for both passive sampling techniques. Spot sampling by direct exposure of TF-SPME for 10 days at sampling site with equilibrium method was selected for hydrophilic analytes, while bottle sampling with external calibration was used for hydrophobic analytes. The limit of detection of the on-site sampling was calculated when the absolute limit of detection and known fiber constant using Equation 3.13.

Table 3.3 Limits of detection of the TWA sampler, grab samplers, and instrument

	LOD of TWA samplers and grab samplers (ng L^{-1})					
	Ben-1	Ben-4	PBSA		Ben-2	Ben-3
Thin film retracted device	630	799	160		131	499
On-site equilibrium sampling	1.1	247	106		0.53	1.1
Instrument	500	2500	500		100	1000
	LOD of TWA samplers and grab samplers (ngL^{-1})					
	OCR	Ben-3	OMC	TCS	TCC	BM-DBM
Open bed TWA sampler	0.14	0.56	0.04	0.23	0.07	0.01
External calibration	20	7.5	20	10	1.0	3.0
Instrument	1000	1000	500	100	1	160

3.3.6 On-site water sampling with TWA samplers

The two TWA samplers were developed and validated in the laboratory, and subsequently used for an on-site investigation. The downstream of the Doon (Kitchener) municipal wastewater treatment plant was selected for deployment of the devices. The sampling time for retracted TF-SPME TWA samplers was set as 90 days. Sampling time was selected based on levels of concentration found for these analytes in surface water in primarily investigation, as well as the detection limit of the LC-MS/MS instrument. In addition to TWA sampling, spot sampling was performed every month using open bed TF-SPME and nine samplers were deployed from 5 to 15 days, with results showing the coating reached equilibrium after 10 days. Spot sampling

was carried out by direct exposure of three HLB coated TF-SPME samplers for 10 days sampling time, and equilibrium concentrations were calculated with Equation 3.16:

$$C_0 = \frac{n}{f_c}$$ Equation 3.16

Where C_0 is the equilibrium concentration, fc is the fiber constant, which is the product of the partition coefficient of the analytes and the active surface area of the solid coating, and n is the amount of extracted analyte. Average concentrations and relative standard deviations (RSD) of spot sampling and TWA sampling results are shown in Table 3.4. The TWA concentrations of the analytes were calculated with the use of Equation 3.1. The results obtained by the two methods are similar. Ben-1, Ben-3, Ben-4, and PBSA were detected in spot sampling, while only PBSA and Ben-4 were detected in TWA sampling. This is due to the low sampling rate of the device and the low concentrations of Ben-1 and Ben-3. In addition, procedural and field blank samples related to these samples were analyzed, and none of the analytes under the study were detected. For the in-field trial for the open bed TF-SPME TWA samplers, nine thin film samplers were loaded with the calibrant and transported to the sampling location on dry ice. Upon arrival, samplers were placed into individual copper cages to prevent biofouling, then subsequently placed in plastic cages before deployment. Three of the samplers were retrieved after 2, 5, and 10 days. Analysis showed that 50% of the calibrant was lost within 5 days of sampling, which was determined to be the optimum time for measuring TWA concentrations. TCS, OCR, and Ben-3 were detected and quantified by Equation 3.12, with results shown in Table 3.4. In addition to TWA sampling with open bed TF-SPME, spot sampling using a grab sampling approach was investigated for determination of the concentration of analytes over

the TWA sampling time. Three spot samples were collected in 1 L amber bottles at different days throughout the sampling period, and transported to the laboratory for analysis. The samples were analyzed in triplicate by external calibration TF-SPME. Moreover, procedural and field blank samples were analyzed, and none of the analytes under study were detected. The data shows good agreement between the TWA passive sampling methods and relevant spot sampling checks made. The field study showed that one-calibrant kinetic calibration was perfectly suitable for quantitation of hydrophobic compounds in river waters. The concentrations that were found by the developed passive samplers are consistent with typical concentrations reported in the literature.[119,147–152] The field study showed that the one-calibrant kinetic calibration is perfectly suitable for quantitation of hydrophobic compounds in river waters. The concentrations that were found by the developed passive samplers are consistent with typical concentrations reported in the literature.[119,147–152]

3.4 Conclusions and future directions

This study demonstrated the feasibility of TWA concentration measurement with two different passive sampling methods, namely, open bed TF-SPME and retracted TF-SPME. The combination of both developed methods offers an integrated approach for TWA determination of analytes with wide range of physical-chemical properties for on-site applications. Retracted TF-SPME devices using HLB sorbent offers a promising passive sampling method to monitor hydrophilic analytes in water without calibrating or controlling the convection condition. The implementation of this sampler is easy compared to other passive sampling methods such as

Table 3.4 Field sampling results of retracted TF-SPME and open bed TF-SPME TWA samplers in Grand River, ON

Open bed TF-SPME (Spot sampling)	Concentration (ngL^{-1})					
	Ben-1 (RSD, %)	Ben-3 (RSD, %)	Ben-4 (RSD, %)	PBSA (RSD, %)	Ben- 2	
June (n=3) 10 Days	5.2 (7)	23 (12)	4529.0 (12)	2379 (20)	<LOD[b]	
July (n=3) 10 Days	5.7 (13)	17 (11)	6058.1 (11)	4060 (12)	<LOD[b]	
August (n=3) 10 Days	6.4 (8)	19 (6)	6604.8 (15)	4982 (11)	<LOD[b]	
Ave	5.6	20.0	5730.6	3807.8	<LOD[b]	
Retracted TF-SPME TWA Sampler (n=3)	<LOD[a]	<LOD[a]	5420.4 (15)	4009.7(12)	<LOD[a]	
Procedural blank	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]	
Field blank	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]	
One-calibrant kinetic calibration	Concentration (ngL^{-1})					
	OCR (RSD, %)	TCS (RSD, %)	Ben-3 (RSD, %)	OMC	TCC	BM-DBM
Open bed TF-SPME TWA sampler (n=3)	90.5 (9)	36.5 (5)	27.4 (19)	<LOD[d]	<LOD[d]	<LOD[d]
Grab sampling (n=6) (spot sampling)	129.5 (15)	50.5 (15)	28.4 (10)	<LOD[e]	<LOD[e]	<LOD[e]
Procedural blank	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]
Field blank	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]	<LOD[c]

LOD[a] is the limit of detection of the retracted TF-SPME device

LOD[b] is the limit of detection of equilibrium on-site sampling

LOD[c] is the limit of detection of the instrument

LOD[d] is the limit of detection of the open bed TWA sampler

LOD[e] is the limit of detection of the external calibration

polar organic chemical integrative sampler (POCIS), does not required calibration in the laboratory or field. However the sampling rate in the sampler was very low and required a long sampling time which could be increased by selecting a shorter diffusion path. Open bed configuration using one-calibrant with C18 sorbent showed its capability for measuring the TWA concentration of hydrophobic compounds, where their isotopic labelled standards are either not available or affordable. The limitation of this sampler is its dependency on the convection conditions which effects the desorption rate of the calibrant and it cannot be used for long term monitoring where the convection conditions are high. In addition to that, a predetermined K value was necessary for this approach. This method potentially could be applied to TWA measurement of other personal care ingredients by using different calibrants to cover a wide range of compounds. The proposed method focused on measurement of the free concentration of analytes.

Chapter 4

Partitioning of UV filters and biocides to different environmental compartments

4.1 Introduction

UV filters and biocides are used as ingredients in an extensive variety of personal care products with a wide range of physical-chemical properties.[149,153–155] Due to their widespread and extensive use, their concentrations in aquatic environments are increasing to detectable levels.[5,132,135] Previous studies have shown that hydrophobic organic chemicals (HOCs) interact with natural sorbents and dissolved organic matter (DOM) as environmental counterparts. In environmental matrices such as surface water, runoff effluents, and soil or sediment porewater, HOCSs are distributed between the sorbent phases and water. These interactions decrease the freely dissolved concentration (C_{free}) and affects the ecotoxicological effects, biodegradation and bioavailability of contaminant.[156–159] Therefore, accurate determination of distribution coefficients between HOCSs and dissolved organic matter and sediments are necessary to quantify the strength of affinity and bioavailability of HOCSs. This will help to determine the potential impact of HOCSs concentrations on the environment.[124,160,161] The determination of distribution coefficient of dissolved organic carbon (DOC) is complicated, because the sorbent is dissolved in the aqueous phase, and a classical separation of the two phases without disturbing the equilibrium is difficult to achieve.[162] Therefore, sorption behavior of hydrophobic substances to DOC is often studied by measuring

the free concentration without active separation of the two phases. Several methods have been proposed for measuring free concentrations, including equilibrium dialysis,[163,164]water solubility enhancement,[165,166] complexation–flocculation,[59] liquid–liquid partitioning,[167] reversed-phase separation,[168] liquid-liquid extraction[169] and fluorescence quenching.[170] Negligible solid phase microextractionc(nd-SPME) has been used for measuring free concentrations of organic contaminants in humic acid and sediment.[44,55,58,106,117,171–176] Equilibrium and pre-equilibrium extraction techniques were used for measuring C_{free} concentration[55,125,175,177]. Thin film solid phase microextraction (TF-SPME), while maintaining the major advantages of SPME also shorten the extraction time without scarifying method's sensitivity, as it can provide larger surface-to-volume ratio.[73,100,178] The objective of the currently presented work was binding investigations of UV filters and biocides to humic acid and sediment using thin film solid phase microextraction. Equilibrium extraction method was used to determine C_{free} concentration of the analytes in both matrixes. The sorbed concertation was obtained by traditional liquid extraction method. Eventually, distribution coefficients of DOC (K_{DOC}) and sediment (K_d) of the target analytes were calculated.

4.2 Experimental

4.2.1 Chemical and material

All chemicals and reagents utilized in this study were described in 2.2.1. Humic Acid sodium salt was obtained from Sigma Aldrich (Oakville, ON, Canada).

4.2.2 Instrumental analysis method (LC/MS/MS

The LC-MS/MS method was described in 2.2.2.

4.2.3 Preparation of humic acid solution

500 mg of HA sodium salt was dissolved in 1000 mL of ultrapure water and left to stir overnight. The solution was first filtered by 5 µm cellulose nitrate, then further filtered with the use of a 0.25 µm cellulose nitrate filter. The concentration of total organic carbon (TOC) was 150 mgL^{-1} measured with a Shimadzu CHNO Analyzer.

4.2.4 Matrix free calibration media

It is very important to match the conditions of matrix-free media close to those of environmental water samples in terms of pH and ionic strength. In order to have an idea about the average pH, total dissolved solids (TDS), and ionic strengths of typical surface water, various samples were collected from different locations. The obtained results are shown in Table 4.1 in accordance with the values obtained for the collected environmental samples, the pH of the water samples was set at pH= 7.5 with 5 mM sodium carbonate-bicarbonate buffer (pH was adjusted by formic acid), producing a conductivity of 850 µS/cm, and TDS of 550 ppm. These values are in the range of typical surface waters. For further studies, the pH and ionic strength of water in the ASG were kept constant at these values.

4.2.5 Aqueous standards generation system

The ASG system was described in 2.3. Average concentrations of individual analytes were 296, 15, 12, 288, 30, 4.1, 0.21, 0.085, 1.1 and 0.95 $ngmL^{-1}$ for PBSA, Ben 1, Ben 3, Ben 4, Ben 2, TCS, OCR, OMC, TCC and BM-DBM, respectively For binding investigation in presence of humic acid, the humic acid stock solution was continuously added to the mixing chamber with the flow rate of 0.2$mLmin^{-1}$ to adjust the DOC equal to 10mgL^{-1}.

Table 4.1 Typical pH, TDS and conductivity of surface waters sample

Sample	pH	Conductivity (μS/cm)	TDS (ppm)
Laurel Creek	8.1	774	480
GR1	7.4	470	270
RW1	7.5	410	310
RW2	7.8	391	280
RW3	7.9	387	300
Tap water	8.0	740	490
carbonate buffer 5 mM	7.5	850	550

4.2.6 Sorption kinetic on humic acid

Measurement of the sorption kinetic of UV filters and biocides on HA was performed in a 1000 mL amber bottle containing 1.0 $ngmL^{-1}$ of the target analytes and 10 mgL^{-1} of TOC. pre-equilibrium SPME was used for extraction of analytes so as to meet negligible extraction conditions,[58,117] while ensuring that the extraction was still in the dynamic range. However, the LC-signal intensities reflected the free concentration of the analytes as a function of the DOC sorption time. First, 1 $ngmL^{-1}$ of mixture standard was prepared in 930 mL ultrapure water in the amber bottle and stirred for 10 min, and then 70 mL of HA solution was added to adjust the TOC concentration to 10 mgL^{-1}. Vigorous mixing of HA with the standard solution was applied for 5, 10, 15, 20, 30 and 60 min. Extraction times were set as 2 min for all cases.

4.2.7 Extraction time profiles in humic acid free and humic acid solution

Extraction time profiles were investigated by C18 and HLB TF-SPME in humic acid free and humic acid solutions, with extraction times ranging from 30 to 7230 min in the sampling

chamber, agitation set at 800 rpm, and all extractions conducted in triplicate. A stock humic acid solution was introduced to the mixing chamber of the system to adjust the total organic compounds (TOC) to 10mgL^{-1}. After each extraction, desorption was performed in a 2 mL amber vial containing 1.8 mL of desorption solvent using (MeOH/ACN/IPA 50/25/25, v/v/v)) vortex agitation at 1500 rpm. The collected extract was evaporated to dryness and reconstituted in 0.3 mL of MeOH/H2O (50/50 v/v). The general experimental conditions are given in Table 2.3. A calibration curve was constructed from 1-100 ngmL^{-1} of analytes prepared in MeOH/H2O (50/50) to calculate the amount of extracted analytes. C18 was used for extraction of OCR, OMC, BM-DBM, TCC, TCS and Ben-3, and HLB was used for extraction of Ben-1, Ben-2, Ben-4, and PBSA.

4.2.8 Blade constant and C_{free} measurement

The sorbent to water partition coefficients is an essential parameter for measuring the C_{free} concentration of chemicals in complex matrix such as sediment and humic acid. After equilibrium extraction in matrix free media in ASG with selected coatings desorption was performed in a 2 mL amber vial containing 1.8 mL of desorption solvent using (MeOH/ACN/IPA 50/25/25, v/v/v)) vortex agitation at 1500 rpm. The amounts of extracted analytes were determined in the LC-MS/MS method described above using instrumental calibration solutions prepared in MeOH/H2O (50/50, v/v) in a range of 0.1 to 100 ngmL^{-1}.

4.2.9 Sediment collection

The sediment sample was collected from Staverton (Northamptonshire, UK) from the surface sediment (10 cm). The sediment was brown, and the texture was loamy. The sample was

collected by grab sampler, passed through a 2 mm sieve, and transferred to a plastic container for transport to the laboratory. The total organic matter was 1.6% (w/w %), and the cation exchange capacity at sodium saturation at pH=7 was 6.2 meq/100g. Sediment particle size distribution was 87% (w/w %) from 2-0.05mm, 7% (w/w %) for 0.05-0.002mm, and 6% (w/w %) for less than 0.002 mm. Sediment was extracted through application of the Soxhlet method, using a mixture of methanol and ethyl acetate, then analyzed; no target analytes were detected.

4.2.10 Sorption kinetics on sediment

The sorption kinetic was investigated in the batch mode in which the sampling chamber (500 mL) was equilibrated for 24 h with the standard solution in flow through system to eliminated losses of the analytes on the surface. Then 5.0 grams (dry weight equivalent) sediment were added to the sampling chamber while the inlet and outlet were blocked initially with the septum and it was transferred to the mechanical shaker. The sorption kinetics was investigated in mechanical shaker at 250 rpm for 0.5, 1, 5, 10, and 24 hrs. The supernatant was separated from sediment through the 0.45µm cellulose acetate filter and transferred to a 250 mL amber bottle according to procedure described in 2.3.2.

4.2.11 TF-SPME extraction time profile in sediment

Extraction time profiles were investigated in aqueous standard solution containing sediment in sampling chamber bottle (500mL) with C18 and HLB coating. The glass bottle was pre-equilibrated for 24 hrs in ASG to eliminate surface adsorption. 5.0 gram (dry weight equivalent) of sediment was added to the aqueous standard solution. Further, C18 and HLB coated blade was added to the three separate glass bottles. The bottles were agitated in 250 rpm

on mechanical shaker and blades were retrieved after 1, 3, 5, 10, 23, 49, 63 hrs. After extraction, the devices were desorbed in MeOH/ACN/IPA (50/25/25, v/v/v) and desorption solvents were evaporated to dryness under N_2. The residues were reconstituted in 0.30 mL MeOH/H2O (50/50, v/v), which was equivalent to the initial solvent composition in the chromatographic method, and subsequently analyzed by LC-MS/MS. The amounts of extracted analytes were determined by the LC-MS/MS method.

4.2.12 Adsorption isotherm experiment

The concentration of the analytes in the ASG was changed by adding intermittent flow of water (3, 7, 17, 37 $mLmin^{-1}$) to the mixing chamber to create different concentrations. In each concentration 5.0 grams (dry weight equivalent) of sediment was placed in the sampling chamber (500 mL) and agitated at 250 rpm on the mechanical shaker for 5 hrs to reach an equilibrium. Upon equilibrium, the sediment sample was separated from the supernatant through the 0.45µm cellulose acetate filter and transferred to an amber bottle, then 50 mL of MeOH/EtOAc (50/50 v/v) solvent was added, followed by 60 min of ultrasonic irradiation at 60°C in a water bath. The sediment was extracted with the same solvent two more times. The extracts were combined and evaporated to dryness under a nitrogen stream. A 10 ml volume of methanol was added to the bottle and sonicated for 2 min to re-dissolve the extract, then transferred to a 20 ml amber vial and evaporated to dryness and reconstituted in 1.5 mL MeOH/H2O (50/50, v/v). After filtration, the total concentrations of the analytes in supernatant was measured by LLE using EtOAc. The concentration of the Ben-4 and PBSA was measured from the subtraction of initial concentration in the aqueous solution and concentration on the

sediment. An aliquot of the supernatant was removed for measuring DOC level by combustion at 720 °C on Shimadzu CHNO analyzer after acid digestion which was 5 mgL^{-1}.

4.3 Results and discussion

4.3.1 Kinetics of sorption in humic acid

As described above, the kinetics of sorption between humic substances and target analytes was investigated with application of the short term SPME procedure. The results (Figure 4.1-Figure 4.2) indicated that sorption equilibrium times for Ben-1, Ben-2, Ben-3, Ben-4, PBSA, and TCS were all less than 5 min. The main interactions occurring between the analytes and HA were hydrogen bonding and van der Waals interactions. Total sorption equilibrium occurs within 10 min for the OCR, TCC, OMC, and BM-DBM. The kinetics of sorption of hydrophobic compounds with HA is very low, because the hydrophobic part of HA is in its interior part, and rearrangement is necessary for interaction.[179]

4.3.2 Thin film solid phase microextraction

4.3.2.1 Extraction time profile

Extraction time profiles were investigated in humic acid free and humic acid solutions in ASG. TF-SPME were placed in a sampling chamber at 800 rpm from 30-9660 min in triplicate. Equilibrium time was defined as the time when the extracted amount is statistically constant. Normalized extraction time profiles for all analytes are shown in Figure 4.1-Figure 4.10. For hydrophobic compounds, TCS, TCC, OMC, OCR, BD-BDM, the kinetic of extraction in the presence of HA showed an enhancement effect, resulting in shorter equilibrium times, which

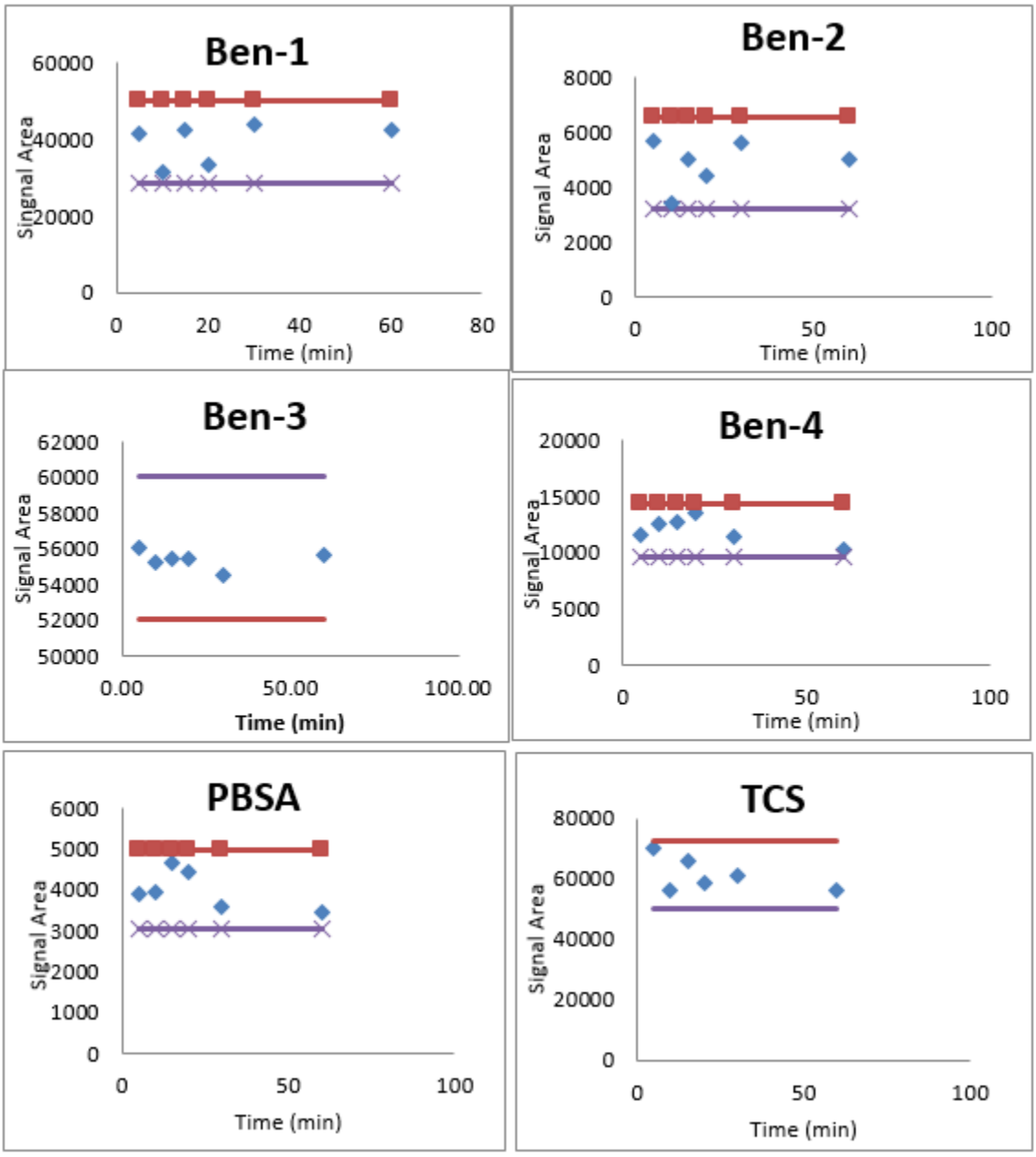


Figure 4.1 Sorption kinetics of Ben-1, Ben-2, Ben-3, Ben-4, PBSA and TCS on a dissolved humic acid, red line and violet line are upper limit and lower limit respectively (±SD)

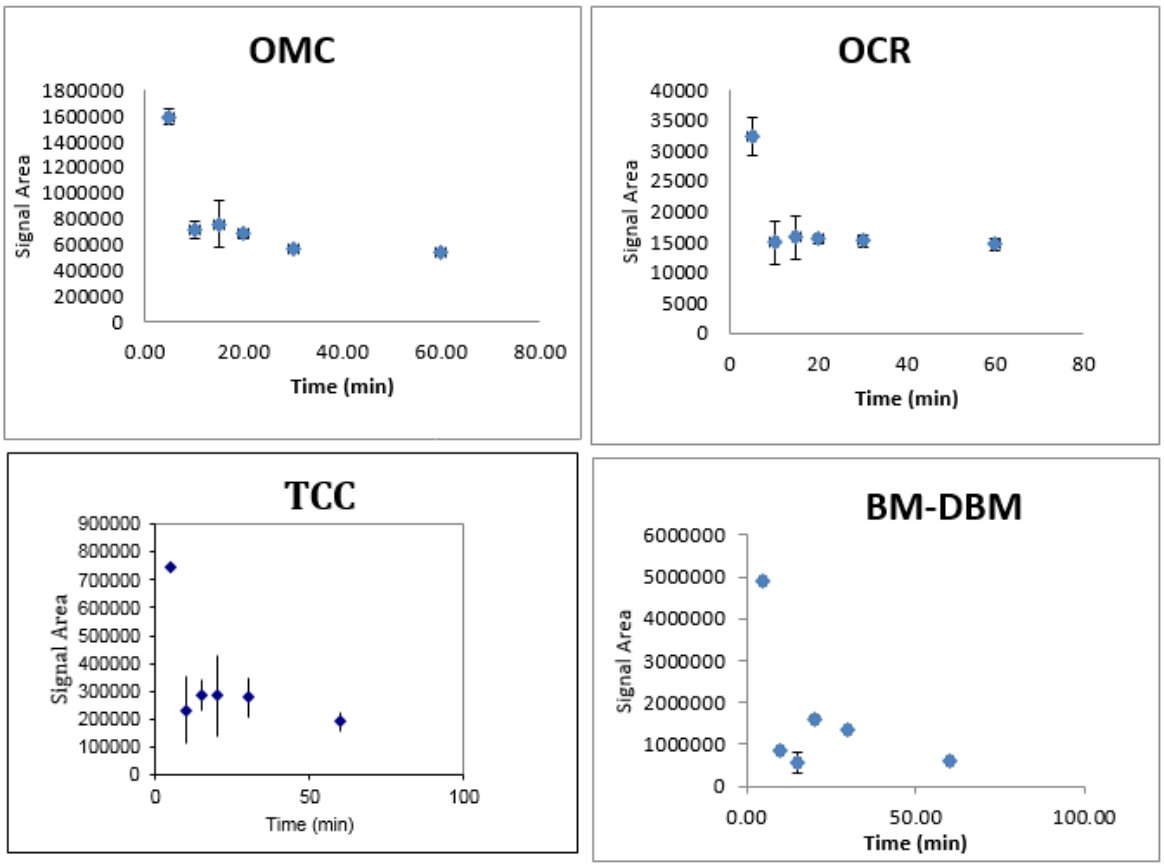


Figure 4.2 Sorption kinetics of OMC, OCR, TCC and BM-DBM on dissolved humic acid

is in accordance to previous research in the literature.[57,177] This phenomenon can be explained by local depletion of the analytes from the matrix in the boundary layer due to adsorption by the coating. The diffusion of bound chemicals is faster than free chemicals within the boundary layer, [44,57,162,177,180,181] In contrast, Alam et al., have shown using mathematical modelling that the increase in uptake of extraction in the presence of matrix is dependent on the equilibrium

dissociation constant (K_D) between the matrix component and the analytes [182]. The reaction between the matrix component and analytes can be expressed by Equation 4.1:

$$A + B \rightleftarrows AB \qquad \text{Equation 4.1}$$

Where A is the freely dissolved analyte, B is the binding matrix component, and AB is the bound species. The rate of dissociation constant (K_D) determines the strength of interaction between the matrix and analyte. This affects the rate of analyte release from the matrix, as defined by the Equation 4.2:

$$K_D = \frac{k_r}{k_f} = \frac{C_A C_B}{C_{A\,B}} \qquad \text{Equation 4.2}$$

Where k_r and k_f are the rate of dissociation and the rate of association, respectively, and C_A, C_B, and C_{AB} are the molar concentrations of the freely dissolved analyte, matrix component, and bound matrix, respectively. It was shown that significant enhancement was observed by increasing the K_D value from 10^{-3} to 10^{-6}. As explained previously, when the affinity between the matrix and analytes is very high, the amount of free analytes is decreased, and the coating requires less analytes to reach equilibrium. In addition, the concentration gradient is at a shorter distance from the coating, and therefore, the equilibrium time is shorter. Our results strongly support the extraction process is controlled by diffusion within the boundary layer and the extraction should be performed under equilibrium conditions to prevent overestimation of free concentrations in non-equilibrium extraction conditions.

4.3.2.2 Determination of partition coefficients

The amount of extracted analytes by TF-SPME in equilibrium is directly proportional to their free concentrations in the sample. The relationship between the free concentration and amount of analyte extracted can be expressed by using Equation 4.3:

$$C_f = \frac{n}{fc} \qquad \text{Equation 4.3}$$

Where C_f is the free concentration of the analyte in the sample, n is the amount of extracted analyte by extraction phase, and f_C is the blade constant, which is a product of K_{fs} (partition coefficient between the extraction phase and the analytes) and the volume of the extraction phase for liquid coatings or active surface for solid coatings. TF-SPME can extract negligible amounts of analytes in equilibrium if $V_S \geq 100 \times f_C$, which means that the equilibrium between the matrix and analytes remains undisturbed for biding investigation studies. The ASG system can provide large volumes of samples in order to meet the main requirements of the binding study by TF-SPME. Single-point calibration was used to obtain blade constants by using C18 and HLB TF-SPME for related analytes in matrix free media. The experiment was performed in triplicate, with the obtained results indicating low inter-thin film variability.[183]

4.3.3 Determination of free concentration

When SPME is used for binding studies between the matrix component and analytes, three basic conditions need to be met: (i) equilibrium between the matrix and analyte must be established; (ii) SPME should extract a negligible amount of the analyte to prevent equilibrium disturbances between matrix and analyte; (iii) the matrix should not have an effect on the kinetic of uptake if the extraction is supposed to be performed in the kinetic regime. Based on

the sorption kinetic study of HA and target analytes, the minimum contact time for establishing equilibrium between the analytes and HA was are 10 min. So, HA was added to the mixing chamber before the analytes reached the sampling chamber for extraction. Based on the flow rate of the system and the geometry of the vessel, the hydraulic retention time of HA was approximately 392 min when the analytes reached the sampling chamber. The depletion of free concentration of the analytes in the presence of the matrix was less than 4% by considering the humic acid binding (HAB). The uptake of the kinetic for hydrophobic analytes was affected in the presence of HA. Consequently, SPME extraction for determination of free concentration was performed in equilibrium to prevent from any overestimation in free concentration. The free concentration of the anlaytes in matrix were obtained by Eq-SPME using Equation 4.3, where the blade constant between the analytes and coating in water and amount of extracted analytes in matrix were known.

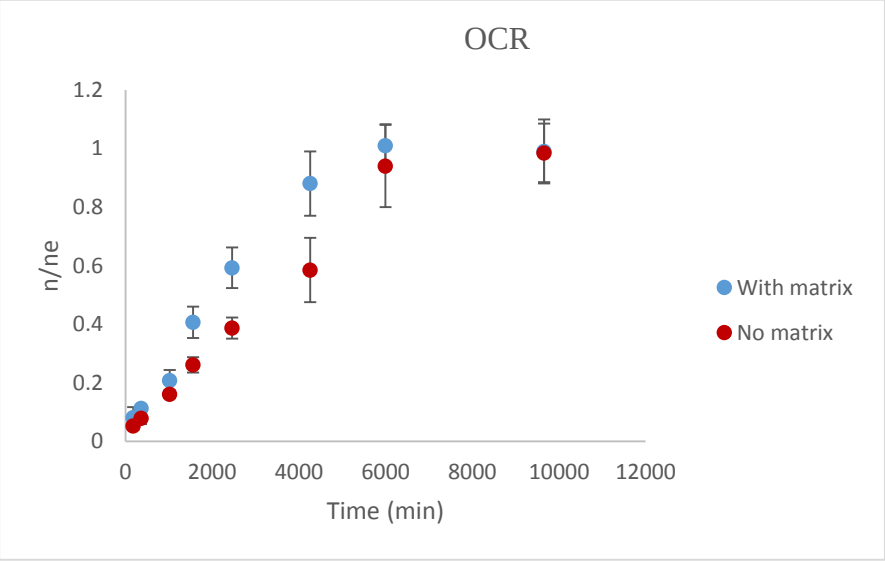


Figure 4.3 Normalized extraction time profiles for OCR in the ultrapure water and HA solution.

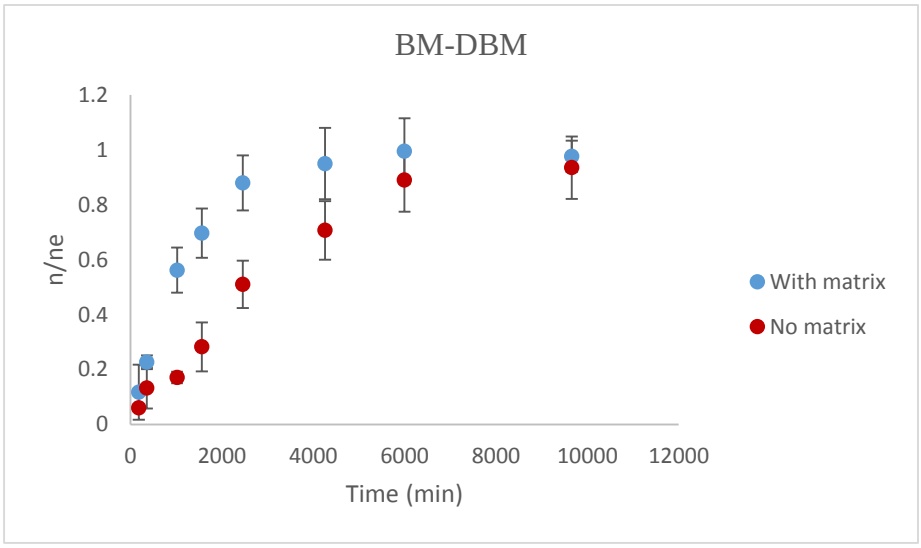


Figure 4.4 Normalized extraction time profiles for BM-DBM in the ultrapure water and HA solution.

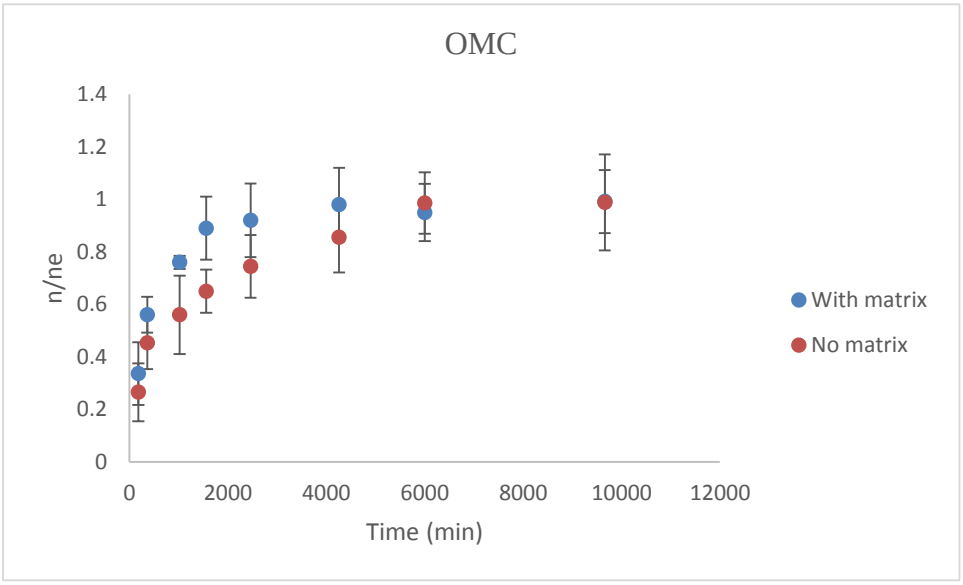


Figure 4.5 Normalized extraction time profiles for OMC in the ultrapure water and HA solution

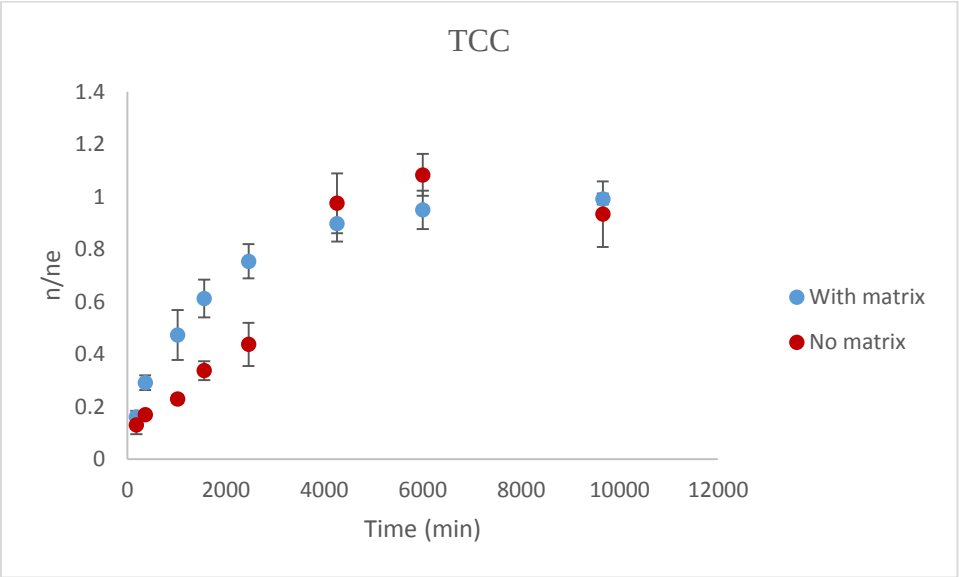


Figure 4.6 Normalized extraction time profiles for TCC in the ultrapure water and HA solution

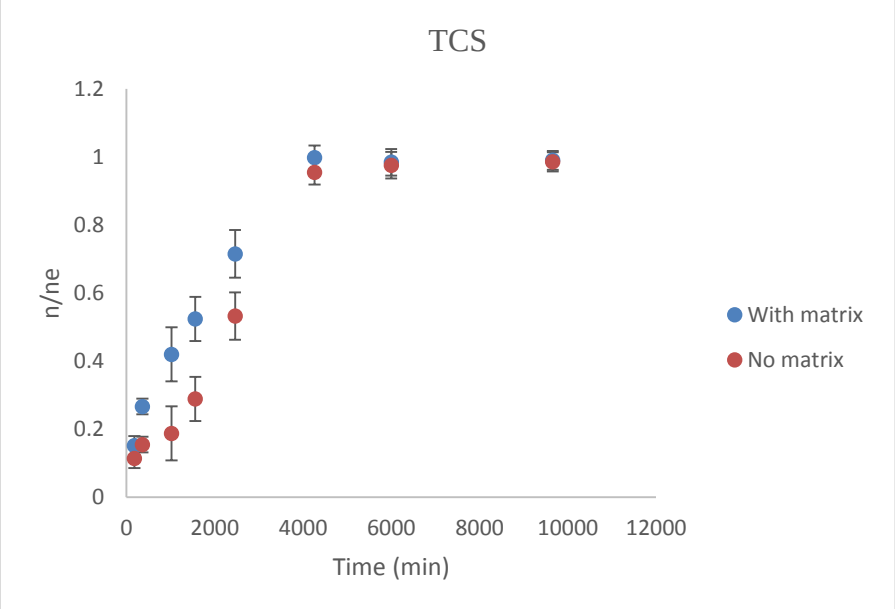


Figure 4.7 Normalized extraction time profiles for TCS in the ultrapure water and HA solution

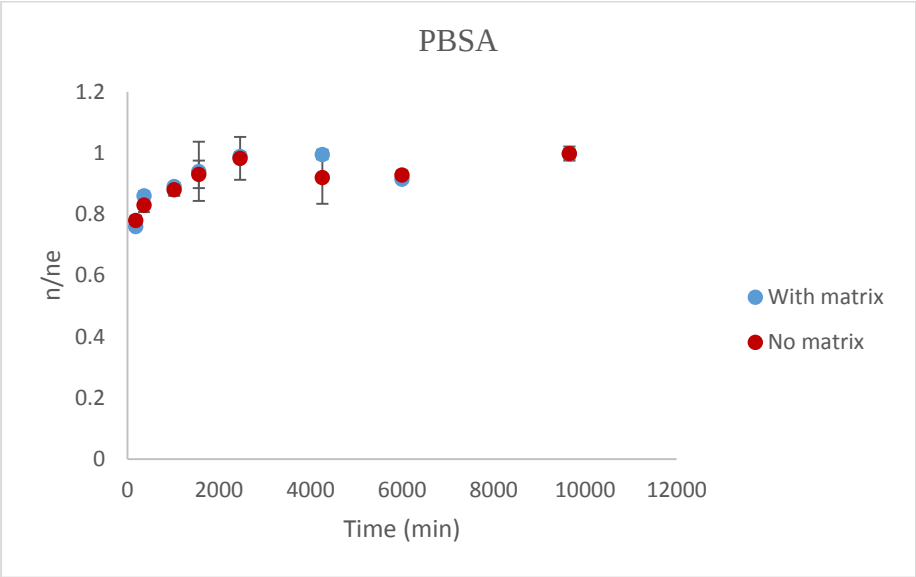


Figure 4.8 Normalized extraction time profiles for PBSA in the ultrapure water and HA solution

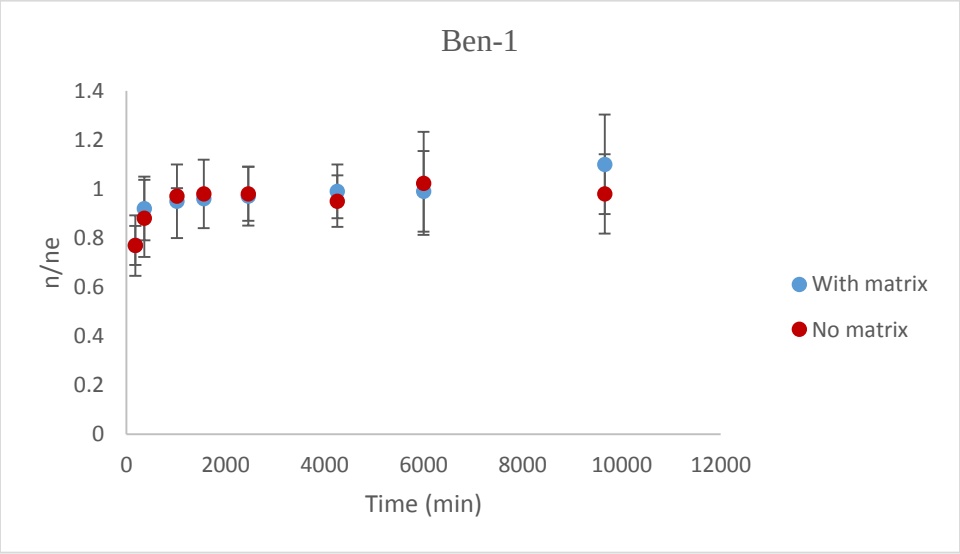


Figure 4.9 Normalized extraction time profiles for Ben-1 in the ultrapure water and HA solution

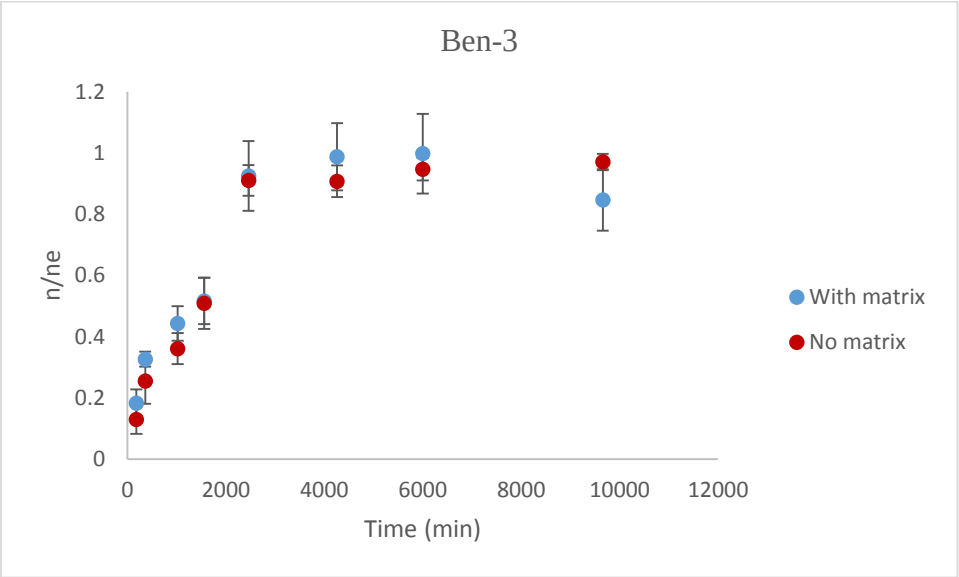


Figure 4.10 Normalized extraction time profiles for Ben-3 in the ultrapure water and HA solution

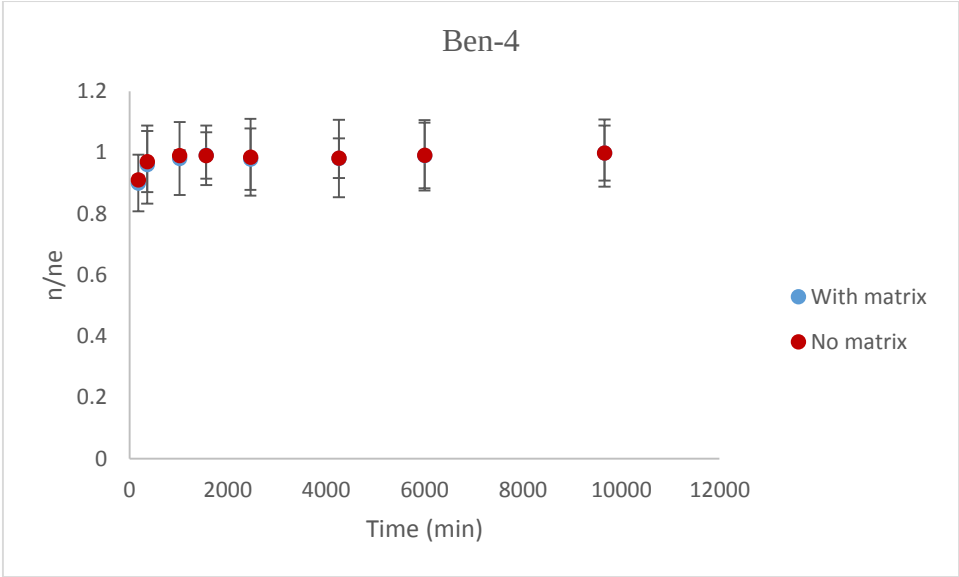


Figure 4.11 Normalized extraction time profiles for Ben-4 in the ultrapure water and HA solution

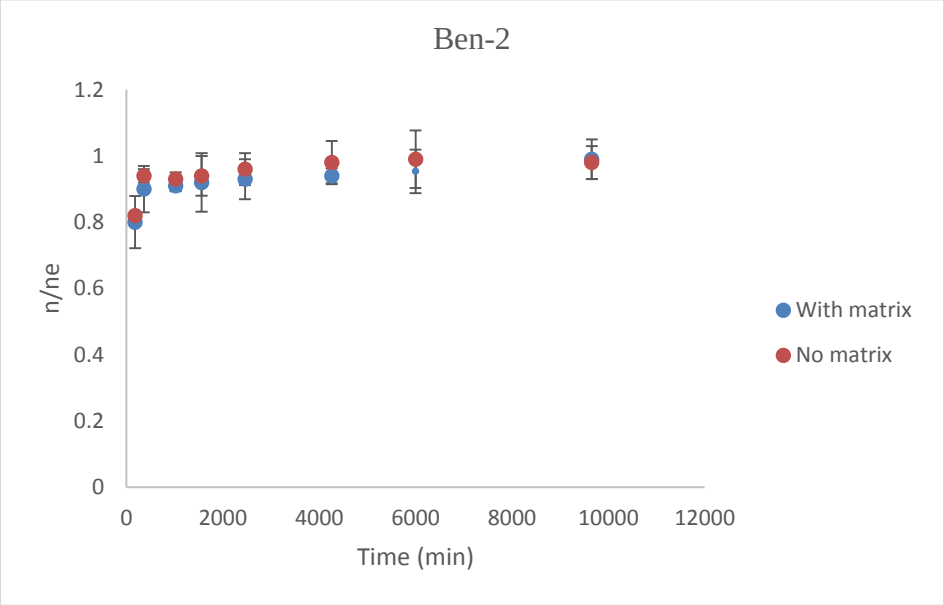


Figure 4.12 Normalized extraction time profiles for Ben-2 in the ultrapure water and HA solution

4.3.4 Determination of total concentration

In DOC containing aqueous sample, the total concentration of the analytes were obtained by single point matrix matched calibration in humic acid solution and traditional liquid-liquid extraction method. The total concentration of the analytes in humic acid solution was obtained based on the Equation 4.4:

$$C_{total} = \frac{ne}{K_{fb}V_{f\,(Sa)}} \qquad \text{Equation 4.4}$$

Where n_e is the amount of extracted in equilibrium in humic acid solution and K_{fb} is the distribution coefficient between the analytes coatings in humic acid solution and V_f and S_a are the volume and surface active of the coating. Two-tailed paired t tests (α=0.05) showed that for the all analytes, there was no significant difference between the two methods. (Table 4.2).

Table 4.2 Concentration (ngL^{-1}) of UV filters and biocides in HA solution measured by SPME and LLE, (Mean, Standard Deviation; n =3)

Compounds	C_{free}	$C_{total\text{-}LLE}$	$C_{total\text{-}SPME}$
TCS	1440±60	4100±120	4200±176
TCC	251±5	1100±40	1200±60
OCR	35±2	210±20	230±7
OMC	13±0.4	85±2	90±5
BM-DBM	1.6±-0.1	95±3	88±5
Ben-1	10350±500	15000±500	14000±1000
Ben-2	24000±1000	30000±1000	32000±2000
Ben-3	6500±300	13000±600	12000±720
Ben-4	260000±22000	288000±15000	260000±25000
PBSA	266000±13000	296000±9000	265000±11000

4.3.5 Estimation of distribution coefficient (K_{DOC})

A new guideline of the U.S. Environmental protection Agency recommends that the partitioning coefficient K_{DOC} is considered so as to account for bioavailability when the toxicity effect is estimated from total concentrations.[55] K_{DOC} can be obtained according to the using Equation 4.5:

$$K_{DOC} = \frac{(C_{total} - C_{free})}{C_{free}\,[DOC]} \qquad \text{Equation 4.5}$$

Free and total concentrations, as well as DOC concentration, are necessary to estimate the partition coefficients of analytes on dissolved organic carbon. The binding of analytes to HA can be calculated from the using Equation 4.6:

$$HAB\% = \frac{C_{total} - C_{free}}{C_{total}} \times 100 \qquad \text{Equation 4.6}$$

Since some of the under studied analytes have a phenolic and sulfonic functional groupneutral compounds and anions are present simultaneously, under experimental conditions (Table 2.1). In such cases, the acid dissociation constant (Ka) should be considered for calculation of partition coefficients of natural molecules. The fraction of non-dissociated analytes was calculated using Equation 4.7 in pH=7.5 and the obtained partition coefficients corrected by considering the fraction of neutral species that interact with DOC.

$$\alpha_{HA} = 1 - \frac{1}{1+10^{(Pka-pH)}} \qquad \text{Equation 4.7}$$

The free concentration of the analyte in presence of HA was measured using Eq-SPME technique. The reduction of free fraction of the analytes was observed for all of the analytes, however the reduction was different for all of them. The partition coefficients (K_{DOC}) were calculated by using the Equation 4.5 when the free concentration, total concentration and DOC concentration were known. The results, which are summarized in Table 4.4 indicated that higher binding values are belong to hydrophobic compounds; TCC, TCC, OCR, OMC, and BM-DBM. The main mechanisms for OMC, TCS, TCC, OCR and BM-DBM would be non-specific hydrophobic bonding with aliphatic and aromatic moieties in HA and hydrogen bonding with the phenolic moieties of HA.[165,184,185] There are two kinds of hydrophobic sites for HA. First

is interior hydrophobic sites which have a strong affinities at pH below 5 for hydrophobic compounds. At pH>5 the interior hydrophobic sites of HA would be destroyed due to conformational HA changes. Second is surface hydrophobic site which are where hydrophobic moieties such as the poly (methylene) groups are exposed to water but keep their capacity to bind nonpolar chemical compounds.[186] On the other hand, association of hydrophilic analytes; Ben-1, Ben-2, Ben-3, Ben-4 and PBSA, with HA were low. These observations imply that these low interactions with HA were due to electrostatic repulsion between the deprotonated analytes and the negatively charged of HA in the working condition (pH=7.5). A greater portion of acidic functionalities in HA dissociate and ionize and thereby making their surfaces more negatively charged and decreasing the H-bonding with the analytes. Our results show that hydrophobicity is not the only property that can describe distribution coefficients of analytes under study; other factors, such as polarity, aromaticity, electrostatic charge, may play a significant role.[187] Data obtained from this binding studies strongly supports the association of hydrophobic analytes with HA, which in turn affects the bioavailability, fate, and transport of these compounds in aquatic environments. In this case, the distribution and total mass of a pollutant in an environments depends on humic material-hydrophobic pollutant binding. However, it is important to note that the Aldrich HA might behave differently from natural humic substances.

4.3.6 Sorption on sediment

4.3.6.1 Kinetic of sorption

The kinetic of sorption was investigated in mechanical shaker at 250 rpm by measuring the analytes concentration in aqueous solution and the results are presented in Table 4.3. The equilibrium time for hydrophilic analytes (Ben-1, Ben-2, Ben-3, Ben-4, and PBSA) was quick. The rapid equilibrium times for these compounds can be interpreted to be due to physical adsorption mechanisms via hydrogen bonding and van der Waals interactions with the organic carbon content (OC) in the sediment. Ben-4 and PBSA were totally anionic, and the electrostatic repulsion between Ben-4 and PBSA with the OC or sediment surface, which carries a total negative charge (62meq/kg),[188] was predominant. Ben-1, Ben-2, and Ben-3 were in natural and ionic forms, and also reached equilibrium within 1.5 hrs. The main interactions taking place were hydrogen bonding and van der Waals interactions with DOC and mineral surfaces of the sediment. When sediment is fully hydrated, adsorption of the organic molecule by sediment becomes comparatively insignificant compared to the uptake by partitioning into dissolved organic matter; water is preferentially adsorbed by oxygen in the surface of sediment, a hydrogen bonding phenomenon known as the "solvent effect". In such cases, the mechanism of sorption is likely hydrogen-bond formations between the sorbate phenolic hydroxyl groups and the hydrogen bonding sites on the sediment organic matter.[189,190] Conversely, equilibrium times for hydrophobic compounds (TCS, TCC, OCR, OMC and BM-DBM) were 5 hrs. Compounds with high octanol-water coefficients were found to show slower sorption. Hydrophobic compounds do not have a tendency to to interact with sediment minerals (e.g.,

clays and metal oxides) due to polar and charged nature of these minerals. Based on many studies that have revealed that HOCSs sorption is strongly connected to the organic carbon content of the sediment, their association can be concluded to occur primarily with sediment organic matter via hydrophobic partitioning.[191,192]

Table 4.3 Concentrations of UV filters and biocides in the aqueous phase (ngL^{-1}) at different time intervals from the onset of equilibration for the sediment

Time (h) / Compound	1.5	5	10	24
TCS	351	645	655	660
TCC	61	162	165	170
OCR	30	57	61	55
OMC	16	28	30	26
BM-DBM	12	26	28	30
Ben-1	14000	13000	15000	13000
Ben-2	19000	18000	17000	20000
Ben-3	10000	11000	12000	13000
Ben-4	569000	560000	555000	570000
PBSA	455000	440000	460000	438000

4.3.6.2 Uptake kinetic in sediment-water

The uptake kinetics of analytes in water containing sediment samples under agitated conditions are shown in Figure 4.13 and Figure 4.14. In general, an apparent equilibrium was reached within 24 hr for all analytes with selected coatings. Eq-SPME was used in order to measure the free

concentration of the analytes in the suspension solution by knowing pre-determined blade constant in the same volume in nondepletive SPME condition.

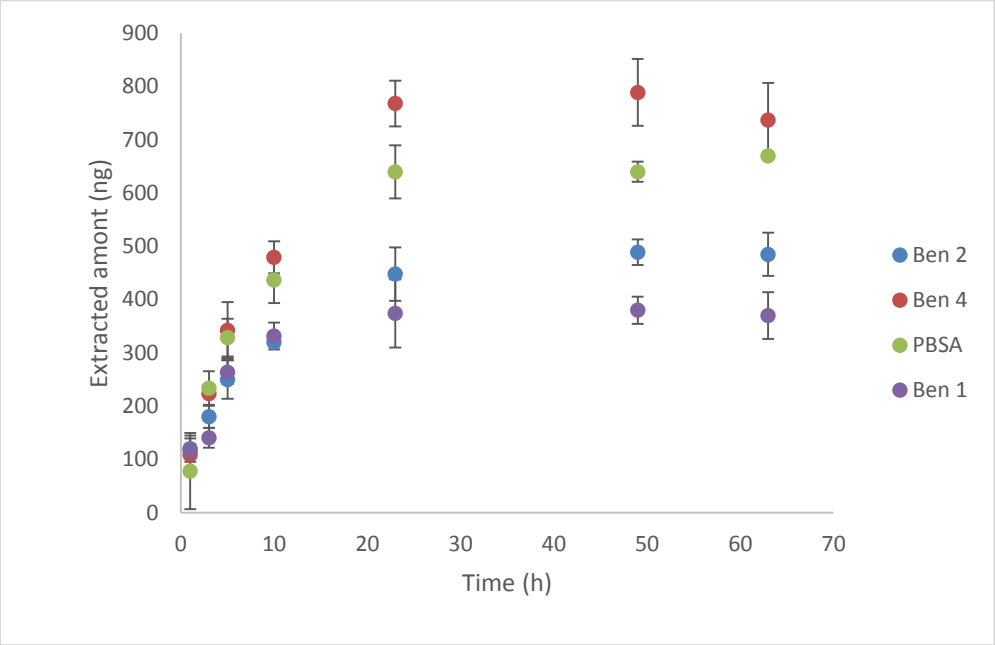


Figure 4.13 Uptake kinetics of Ben-1, Ben-2, Ben-4 and PBSA from sediment

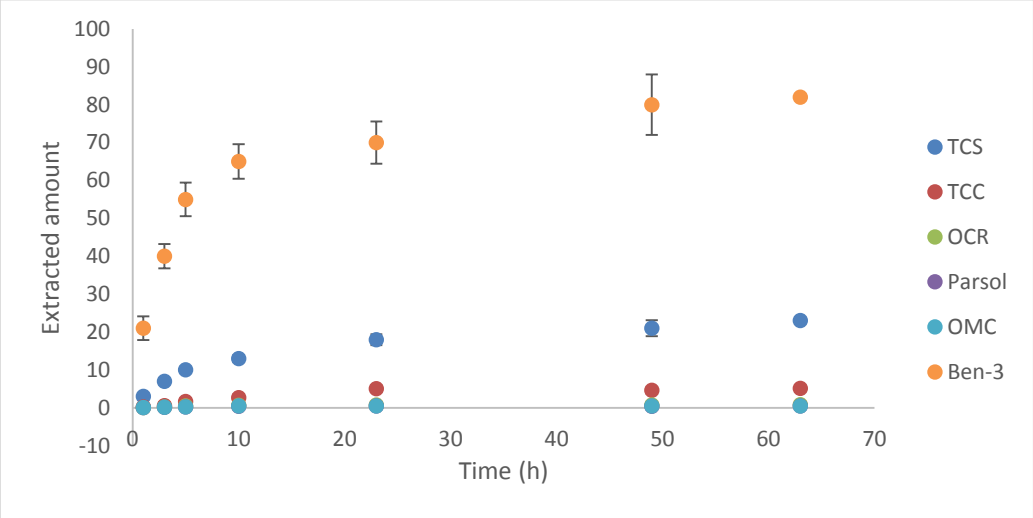


Figure 4.14 Uptake kinetics of OCR, OMC, TCC, TCS, BM-DBM and Ben-3 from sediment

4.3.6.3 Estimation of K_d, K_{OC} and K_{DOC} in sediment

The adsorption isotherms were investigated in a relatively small concentration range and they were fitted to a linear relationship to estimated K_d using TF-SPME. The good linear relationship is due to use of small concentration range.

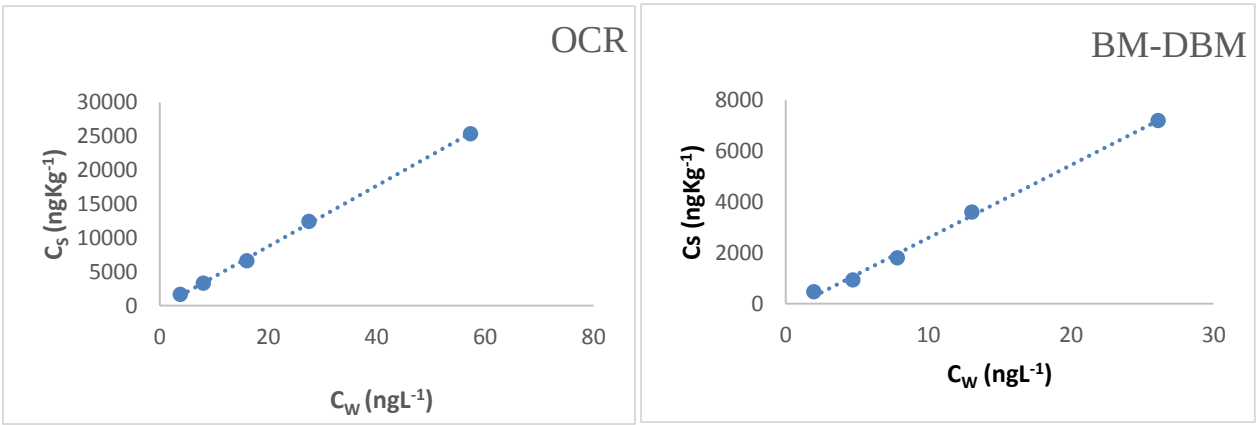


Figure 4.15 Sorption isotherms of OCR and BM-DBM in sediments measured using liquid-liquid extraction (LLE)

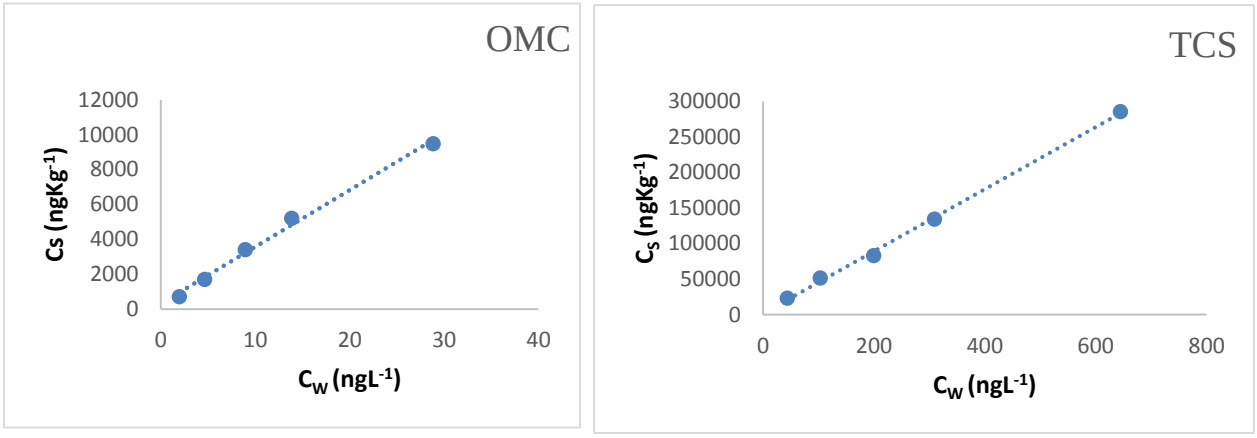


Figure 4.16 Sorption isotherms of OMC and TCS in sediments measured using liquid-liquid extraction (LLE)

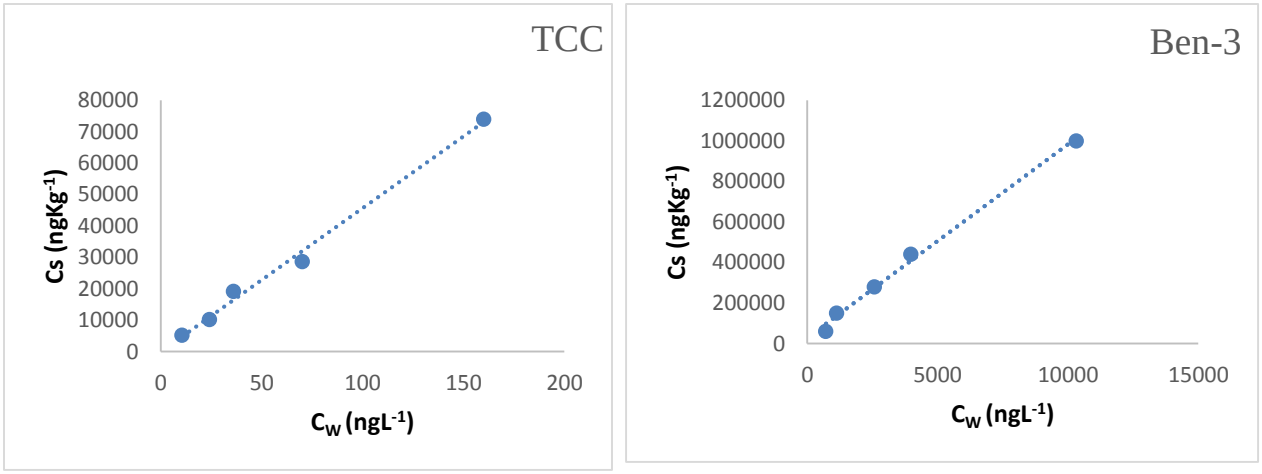


Figure 4.17 Sorption isotherms of TCC and Ben-3 in sediments measured using liquid-liquid extraction (LLE)

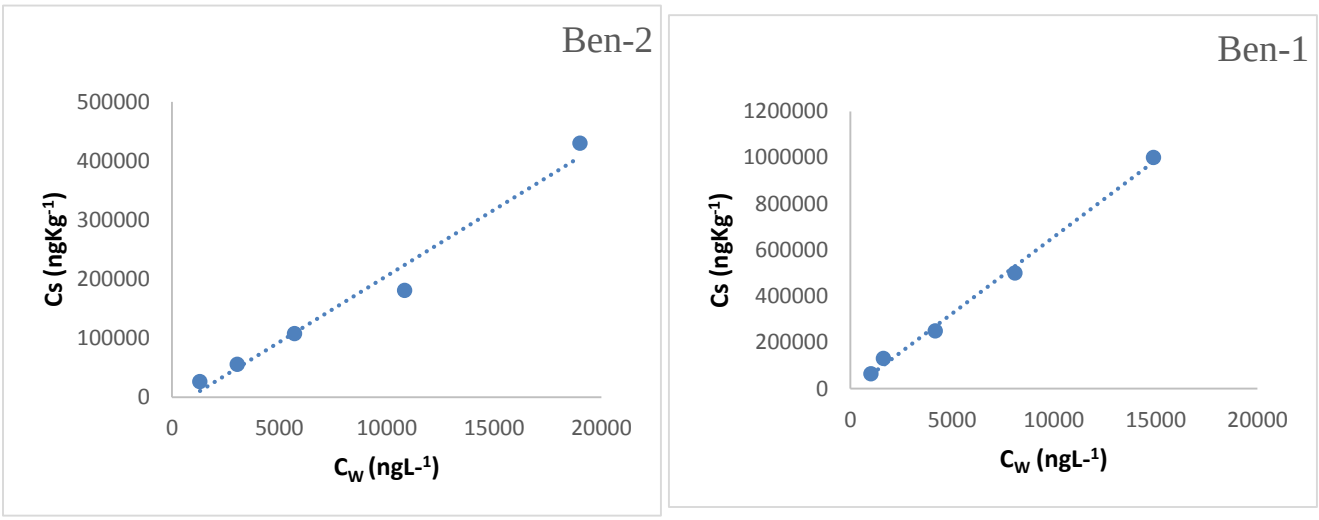


Figure 4.18 Sorption isotherms of Ben-2 and Ben-1 in sediments measured using liquid-liquid extraction (LLE)

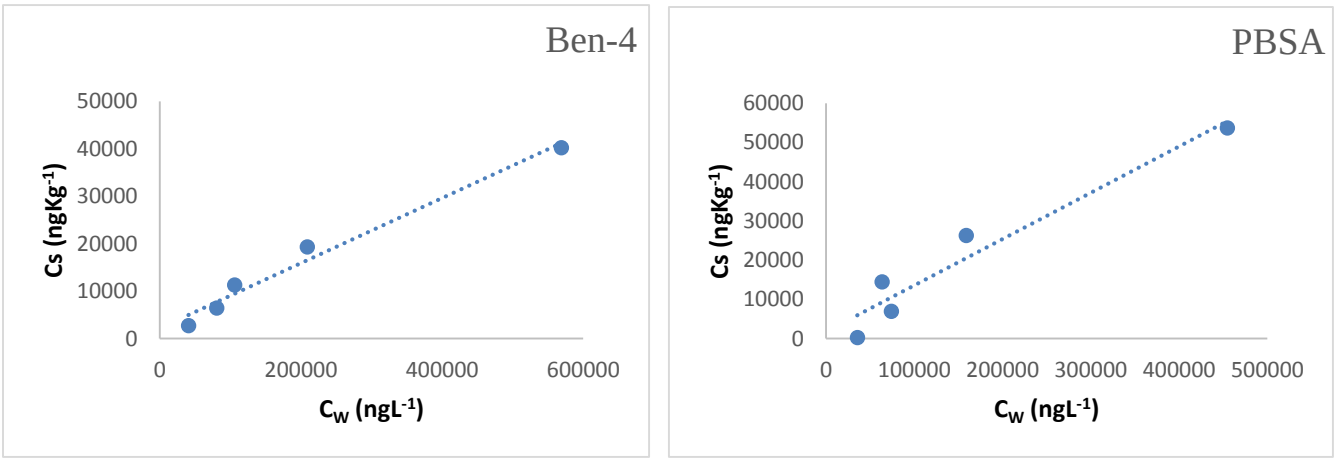


Figure 4.19 Sorption isotherms of Ben-4 and PBSA in sediments measured using liquid-liquid extraction (LLE)

Sorption coefficient (K_d) of the analytes in the sediment at equilibrium was determined by measuring aqueous concentration ($C_{w\text{-}SPME}$) and sediment concentration (C_s) according to using Equation 4.8:

$$K_d = \frac{C_s}{C_{w-SPME}} \qquad \text{Equation 4.8}$$

$C_{w\text{-}SPME}$ and C_s are obtained by SPME and liquid extraction method on aqueous and sediment phases. The K_d values of the analytes are illustrated in Table 4.4 were in the range of 0.06-1600 Lkg^{-1}. The data implied that the sorption coefficients of analytes on the sediment increased as the hydrophobicity of the analytes increased.[58] Hydrophobic analytes sorbed primarily due to hydrophobic interactions with insoluble (humin) and soluble (humic and fulvic acids) fractions of sorbent organic matter through hydrophobic bonding on the surface or in internal voids of a molecular sieve-type structural arrangement.[193] This types of bonding is attributed to van der Waals forces and thermodynamic gradient which drives hydrophobic compounds with low solubility out of solution, because the interaction of these analytes with organic matter in sediment is favourable energetically. However, K_d values for hydrophilic analytes (Ben-1, Ben-2, Ben-3, Ben-4 and PBSA) are low as they are present in both anionic and/or molecular forms in the experimental pH=7.5, hence electrostatic interactions occurring between silt/clay and the ionic forms of the analytes. A similar phenomenon was observed for diclofenac and ketoprofen in other research.[194,195] Sorption mechanisms of less hydrophobic and ionisable analytes containing polar functional groups may in addition to hydrophobic partitioning into sorbent organic matter, include hydrogen bond formations and charge transfers.[188,196,197]

The partition coefficient (K_d) values was normalized on organic carbon content (OC) of the sediment according to using Equation 4.9:

$$K_{OC} = \frac{K_d}{f_c} \qquad \text{Equation 4.9}$$

Where KOC was the sediment OC normalized partition coefficient and fc is the organic carbon fraction in the sediment. The KOC were plotted against the octanol water partitioning coefficient (log KOW) values, and good correlation was obtained, as shown in Figure 4.20.

It was observed that the DOC or fine particles are present in the supernatant phase after centrifuging which caused overestimation of the partitioning coefficient by conventional extraction method on aqueous phase.[198] However, the SPME was used to measure Cfree of the analytes in aqueous solution and improved the estimation of Kd and further KOC.[176] The KDOC was calculated further by measuring the CW-LLE and CW-SPME using Equation 4.10:

$$K_{DOC} = \frac{C_{W-LLE} - C_{W-SPME}}{C_{W-SPME}\,[DOC]} \qquad \text{Equation 4.10}$$

Where [DOC] is the level of DOC. The partition coefficient between analytes and dissolved organic carbon (K_{DOC}) is a valuable factor for predicting toxicity and bioaccumulation abilities of hydrophobic organic compounds in sediments or soils. U.S. EPA recommended to use K_{DOC} data in relating chemical concentrations to toxicity to benthic organisms in its sediment quality guidelines.

Table 4.4 Sediment-water partition coefficient (Kd), organic-carbon normalized partition coefficient (K_{OC}) and dissolved organic carbon partition coefficient (K_{DOC}) and Aldrich humic acid partition coefficient (K_{DOC}) and binding percent

	Sediment			Aldrich Humic acid	
	K_d (LKg^{-1})	Log K_{OC}	Log K_{DOC}	K_{DOC}	HAB%
TCS	700±35	4.64±0.23	5.21±0.31	5.21±0.15	65±2
TCC	1000±60	4.79±0.28	5.38±0.43	5.56±0.11	78±2
OCR	1500±60	4.97±0.19	5.78±0.52	5.61±0.67	83±10
OMC	1000±80	4.79±0.38	5.62±0.33	5.72±57	84±8
BM-DBM	1600±144	4.99±0.45	6.02±0.48	6.72±0.41	98±6
Ben-1	60±6	3.57±0.35	4.9±0.34	4.55±0.40	33±3
Ben-2	24±1	3.17±0.21	4.35±0.35	3.71±0.33	20±2
Ben-3	100±8	3.79±0.31	3.92±0.16	4.83±0.15	50±2
Ben-4	0.06±0.005	0.57±0.05	3.72±0.18	3.77±0.31	5±0.4
PBSA	0.12±0.009	0.87±0.06	4.31±0.32	4.12±0.45	10±1

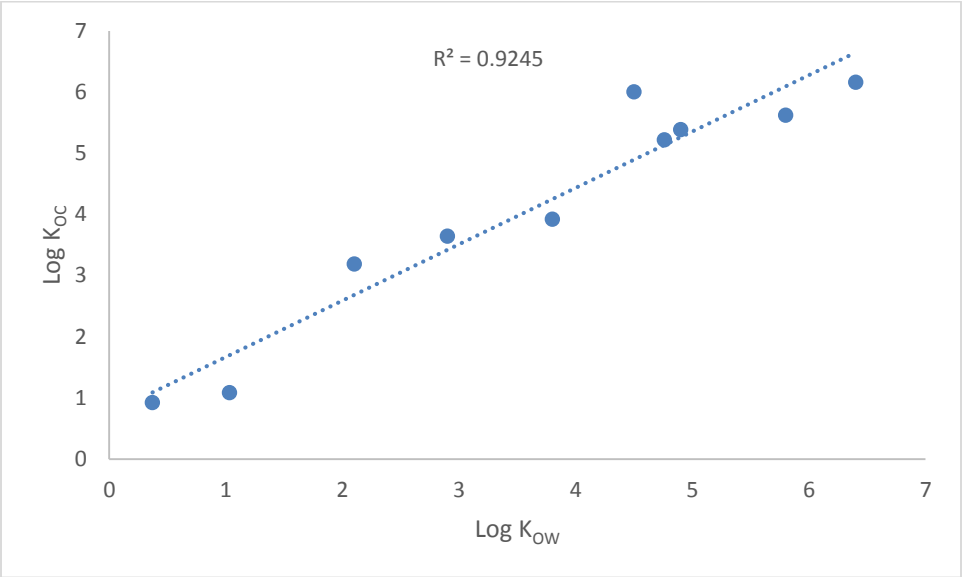


Figure 4.20 Partitioning coefficients normalized on organic carbon content (log K_{OC}) versus octanol water partitioning coefficients (log K_{OW})

4.4 Conclusions and future direction

Laboratory sorption experiments were conducted for UV filters and biocides, of relatively high potential ecological risk and high consumption, dissolved humic acid and sediments as environmental counterpart using negligible depletion thin film solid phase microextraction to measure free concentration in solution and suspension. The results of partitioning coefficient suggest that hydrophobic compounds exhibit relatively higher partition on humic acid and sediment, whereas hydrophilic analytes did not bond strongly with solid phase and liquid phase. The high partitioning coefficients of hydrophobic analytes on sediment and dissolved

humic acid are attributed of their high octanol-water coefficients and organic carbon content in both cases which happen via hydrophobic partitioning. This high partitioning has several implications on the fate, transport, and ecotoxicological effects by these compounds. The fact that sorbed compounds to DOC implies that in natural surface streams DOC may act as an effective carrier in surface runoff or stream- flow for them and thus enable to move over relatively long distances. Low partitioning of hydrophilic analytes implies that these analyte don't interact significantly due to electrostatic repulsion between the deprotonated analytes and the negatively charged of HA and sediment and organic carbon and hindrance of forming hydrogen bonds in the working condition (pH=7.5). The presented aspects of low partition of selected influence the concentration of micropollutants flowing into the reservoir, which has a negative impact on drinking water quality.

Chapter 5
Summary and future prospective

5.1 Summary

Time weighted average (TWA) concentrations of organic contaminants provide a more realistic picture of the exposure of organisms to contaminants than concentrations determined by one or few grab samples. For this reason, the development of on-site sampling tools that enable the collection of precise and accurate data are an important step towards ensuring the safety of the environment and human health. Solid phase microextraction (SPME) was developed as a solvent-free sample preparation technique that integrates sampling, extraction, and pre-concentration in a single step. SPME has been broadly applied in on-site sampling for determination of TWA concentrations of hydrophobic organic contaminants in surface water. In addition, SPME has been used to measure free concentrations of chemical substances in complex matrices such as blood, plasma, and humic acid solution. Thin film solid phase microextraction (TF-SPME) has been introduced to overcome the relative low capacity, sensitivity, and extraction rate of traditional SPME fibers by increasing surface area-to-volume ratio. In this thesis, a dynamic gaseous standard generation (ASG) system was created to provide accurate calibration data of passive sampling devices over a wide polarity range of PCP chemicals. Use of the flow-through system meant it was possible to provide an environmentally realistic calibration of hydrophobic compounds without depleting the system, and to investigate the partitioning of PCP chemicals onto humic acid and sediment. ASG also

addresses issues related to preparation of aqueous standards of hydrophobic compounds in-vial, such as partial precipitation, analyte loss due to adhesion to surfaces, low solubility in water and limited sample volume. The introduction of the thin film retracted device using HLB as a sorbent showed the potential application of TF-SPME for monitoring of hydrophilic compounds in aquatic environments. Moreover, the thin film open bed configuration using the one-calibrant kinetic calibration technique was successfully applied for TWA measurement of multiple compounds in water when isotopic counterparts are either not available or affordable. The obtained results show that the combination of the TF-SPME technique with diffusion-based and kinetic calibrations is a successful quantitative technique for on-site sampling of wide range of chemicals substances in surface water. This is helpful in cases where the composition of the sample matrix is complex, and/or the convection conditions of the sample matrix are variable or uncontrolled. Additionally, TF-SPME was used for a binding investigation of UV filters and biocides in environmental counterparts. The association of the chemicals with the dissolved organic matter decrease the free concentrations of analytes, consequently affecting transport, and the overall fate of the chemical in aquatic environment. Free concentrations of analytes were measured in the presence of Aldrich humic acid by negligible depletion solid phase microextraction (nd-SPME) in the ASG with the use of equilibrium extraction, while total concentrations were measured with the use of liquid-liquid extraction and single point matrix match calibration. Extraction time profile comparisons between the ultrapure water and humic acid solutions show that the uptake kinetics is increased in the presence of matrix for highly bonded analytes. This observed effect can be explained by

desorption of analytes from the matrix into the boundary layer, where the transport of analytes to the fiber coating occurs only by diffusion. Another explanation would be that the concentration of free analytes is decreased in the presence of the matrix; as such, the coating require less analyte to reach equilibrium when the affinity between matrix and analytes is very high. Therefore, the concentration gradient is in a shorter distance to the coating, and equilibrium time would be shorter. Binding and distribution coefficients (K_{DOC}) between analytes and the dissolved organic carbon (DOC) were calculated. The quantitative determination of analytes within the humic acid solution, as shown in Chapter 4, clearly demonstrates the potential of application of nd-SPME using thin film technology coupled to LC-MS/MS when free concentrations decrease significantly due to humic acid binding. Partition coefficients (K_d) of the UV filters and biocides were also investigated on the sediment as another environmental counterpart. Batch sorption experiment using TF-SPME and conventional solvent extraction method were used for measuring the concentration of the analytes in aqueous phase and sediment phase and consequently the partition coefficient were calculated. The results of partitioning coefficient indicated that hydrophobic compounds show relatively higher sorption, however the hydrophilic analytes show an opposite trend. The results of this part suggest that sorption mechanism of the analytes to the sediment is happen via hydrophobic partitioning into sediment organic matter.

5.2 Future perspective

5.2.1 Aqueous standard generation system

Although the developed ASG was mainly focus on UV filter and biocides in this thesis, it can be used for different class of personal care products ingredients. Future step of this thesis, currently in progress in our laboratory, involves the creation of ASG for long chain cationic surfactants. The goal of these applications is to address the surface adsorption issue and generating steady state concentration in water. Cationic surfactants were loaded on the HLB particle by extraction from an aqueous standard (50 ppm) and then the particles were transferred to the permeation chamber. The primarily results so far show that the system is able to provide steady state concentration for 2 months with less than 20% variation.

5.2.2 One-calibrant kinetic calibration

Measuring free concentration of hydrophobic compounds in sediment, soil and humic acid is important because freely dissolved concentration is bioavailable which controls diffusive mass transfer phenomena such as evaporation, sorption, and uptake into macro- and microorganism. Solid phase microextraction is an equilibrium based extraction technique which can measure the free concentration of hydrophobic compounds in such a complex matrix by performing extraction in equilibrium time and knowing fiber constant in matrix free media. However, the equilibrium time may takes weeks or months for highly hydrophobic compounds. In order to overcome this problem, kinetic calibration can be used to find the free concentration by performing extraction in a shorter time. One-calibrant kinetic calibration techniques which was used in this thesis showed its capability for measuring the TWA concentration of multiple

compounds in water sample. This idea can be used for measuring the free concentration of hydrophobic compounds in sediment-water and humic acid-water. Kinetic calibration may extract lower amount of analytes in comparing to equilibrium based extraction, but using thin film solid phase microextraction and high sensitive mass spectrometer will overcome these limitations.

5.2.3 Binding investigation

Evaluation fate, transport, bioavailability and toxicity of organic contaminants in natural systems needs knowledge of their distribution between the solid and aqueous phases. The developed ASG and TF-SPME methodology in this thesis have opened up new possibilities for SPME application in combination with very powerful mass spectrometers in binding investigation of other new emerging contaminants such as pharmaceuticals and other ingredients of personal care products in soil, sediment and dissolved organic matter. In addition, this technique can be used for analysis of field-contaminated sediment.

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Title: Theory of analyte extraction by selected porous polymer SPME fibres†

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References

(1) Petrovic, M. *TrAC Trends Anal. Chem.* **2003**, *22*, 685–696.

(2) USGS Emerging contaminants in the environment http://toxics.usgs.gov/regional/emc/index.html (accessed Oct. 2015)

(3) Agüera, A.; Martínez Bueno, M. J.; Fernández-Alba, A. R. *Environ. Sci. Pollut. Res.* **2013**, *20*, 3496–3515.

(4) USEPA Pharmaceuticals and personal care products (PPCPs) 2008 http://www.epa.gov/ppcp/ (accessed Jan 2015).

(5) Balmer, M. E.; Buser, H.-R.; Müller, M. D.; Poiger, T. *Environ. Sci. Technol.* **2005**, *39*, 953–962.

(6) MacManus-Spencer, L. a; Tse, M. L.; Klein, J. L.; Kracunas, A. E. *Environ. Sci. Technol.* **2011**, *45*, 3931–3937.

(7) Poiger, T.; Buser, H.-R.; Balmer, M. E.; Bergqvist, P.-A.; Müller, M. D. *Chemosphere* **2004**, *55*, 951–963.

(8) Silvia Díaz-Cruz, M.; Llorca, M.; Barceló, D. *TrAC Trends Anal. Chem.* **2008**, *27*, 873–887.

(9) Ying, G. G.; Kookana, R. S. *Environ. Int.* **2007**, *33*, 199–205.

(10) Jacquet, R.; Miège, C.; Bados, P.; Schiavone, S.; Coquery, M. *Environ. Toxicol. Chem.* **2012**, *31*, 279–288.

(11) Kupper, T.; Plagellat, C.; Brändli, R. C.; de Alencastro, L. F.; Grandjean, D.; Tarradellas, J. *Water Res.* **2006**, *40*, 2603–2612.

(12) Richardson, S. D. *Anal. Chem.* **2012**, *84*, 747–778.

(13) Richardson, S.; Ternes, T. a. *Anal. Chem.* **2014**, *81*, 4645–4677.

(14) Zhang, Z.; Hibberd, A.; Zhou, J. L. *Anal. Chim. Acta* **2008**, *607*, 37–44.

(15) Yan, S.; Subramanian, S. B.; Tyagi, R. D.; Surampalli, R. Y.; Zhang, T. C. Pract. Period. Hazard. Toxic Radioact. Waste Manage., **2010**, 2–20.

(16) Gautam, P.; Carsella, J. S.; Kinney, C. a. *Water Res.* **2014**, *48*, 247–256.

(17) Singer, H.; Müller, S.; Tixier, C.; Pillonel, L. *Environ. Sci. Technol.* **2002**, *36*, 4998–5004.

(18) Kameda, Y.; Kimura, K.; Miyazaki, M. *Environ. Pollut.* **2011**, *159*, 1570–1576.

(19) Traverso-soto, J. M.; González-mazo, E.; Lara-martín, P. a. *Chromatogr. - Most versatile method Chem. Anal.* **2012**, 187–216.

(20) Kot-Wasik, A.; Dębska, J.; Namieśnik, J. *TrAC Trends Anal. Chem.* **2007**, *26*, 557–568.

(21) Wardencki, W.; Namiesnik, J. Comprenhensive Analytical Chemistry; Pawliszyn, J., Ed.; Elsevier: Amsterdam (The Netherlands), 2002. pp 33–60.

(22) Nielsen, D. M. The Essential Handbook of Ground-Water Sampling; Nielsen, D. M.; Nielsen, G. L., Eds.; CRC Press: Boca Raton, FL, 2006.

(23) Dick, E. M. Principles of Environmental Sampling; Keith, L. H., Ed.; American Chemical Society, 1996.

(24) Aguilar-Martínez, R.; Palacios-Corvillo, M. a; Greenwood, R.; Mills, G. a; Vrana, B.; Gómez-Gómez, M. M. *Anal. Chim. Acta* **2008**, *618*, 157–167.

(25) Thomatou, A. A.; Zacharias, I.; Hela, D.; Konstantinou, I. *Environ. Sci. Pollut. Res.* **2011**, *18*, 1222–1233.

(26) Alvarez, D. a; Stackelberg, P. E.; Petty, J. D.; Huckins, J. N.; Furlong, E. T.; Zaugg, S. D.; Meyer, M. T. *Chemosphere* **2005**, *61*, 610–622.

(27) Cristale, J.; Katsoyiannis, A.; Chen, C.; Jones, K. C.; Lacorte, S. *Environ. Pollut.* **2013**, *172*, 163–169.

(28) Huckins, N.J.; Booij, K.; Jimmie, D. P.; Monitors of Organic Chemicals in the Environment, Semipermeable Membrane Devices, Spribger: New York, 2006.

(29) Vrana, B.; Allan, I. J.; Greenwood, R.; Mills, G. a.; Dominiak, E.; Svensson, K.; Knutsson, J.; Morrison, G. *TrAC Trends Anal. Chem.* **2005**, *24*, 845–868.

(30) Petty, J. D.; Orazio, C. E.; Huckins, J. N.; Gale, R. W.; Lebo, J. a; Meadows, J. C.; Echols, K. R.; Cranor, W. L. *J. Chromatogr. A* **2000**, *879*, 83–95.

(31) Seethapathy, S.; Górecki, T.; Li, X. *J. Chromatogr. A* **2008**, *1184*, 234–253.

(32) Górecki, T.; Namienik, J. *TrAC - Trends Anal. Chem.* **2002**, *21*, 276–291.

(33) Gourlay-Francé, C.; Lorgeoux, C.; Tusseau-Vuillemin, M.-H. *Chemosphere* **2008**, *73*, 1194–1200.

(34) Esteve-Turrillas, F. a; Yusà, V.; Pastor, A.; de la Guardia, M. *Talanta* **2008**, *74*, 443–457.

(35) Harman, C.; Allan, I. J.; Vermeirssen, E. L. M. *Environ. Toxicol. Chem.* **2012**, *31*, 2724–2738.

(36) Bayen, S.; Segovia, E.; Loh, L. L.; Burger, D. F.; Eikaas, H. S.; Kelly, B. C. *Sci. Total Environ.* **2014**, *482-483*, 15–22.

(37) Vermeirssen, E. L. M.; Dietschweiler, C.; Escher, B. I.; van der Voet, J.; Hollender, J. *Anal. Bioanal. Chem.* **2013**, *405*, 5225–5236.

(38) Bopp, S.; Weiß, H.; Schirmer, K. *J. Chromatogr. A* **2005**, *1072*, 137–147.

(39) Jacquet, R.; Miège, C.; Smedes, F.; Tixier, C.; Tronczynski, J.; Togola, A.; Berho, C.; Valor, I.; Llorca, J.; Barillon, B.; Marchand, P.; Coquery, M. *Chemosphere* **2014**, *98*, 18–27.

(40) Allan, I. J.; Harman, C.; Ranneklev, S. B.; Thomas, K. V; Grung, M. *Environ. Toxicol. Chem.* **2013**, *32*, 1718–1726.

(41) Sun, Q.; Zhang, L. P.; Ding, S.; Li, C.; Yang, J. Y.; Chen, J.; Wang, P. *Environ. Sci. Process. Impacts* **2015**, *17*, 570–577.

(42) Dočekalová, H.; Škarpa, P.; Dočekal, B. *Talanta* **2015**, *134*, 153–157.

(43) Allan, I. J.; Booij, K.; Paschke, A.; Vrana, B.; Mills, G. a; Greenwood, R. *Environ. Sci. Technol.* **2009**, *43*, 5383–5390.

(44) de Perre, C.; Le Ménach, K.; Ibalot, F.; Parlanti, E.; Budzinski, H. *Anal. Chim. Acta* **2014**, *807*, 51–60.

(45) Pörschmann, J.; Kopinke, F.-D.; Pawliszyn, J. *J. Chromatogr. A* **1998**, *816*, 159–167.

(46) Höllrigl-Rosta, A.; Vinken, R.; Lenz, M.; Schäffer, A. *Environ. Toxicol. Chem.* **2003**, *22*, 746–752.

(47) Jones-Hughes, T.; Turner, A. *Environ. Sci. Technol.* **2005**, *39*, 1688–1697.

(48) Schellenberg, K.; Leuenberger, C.; Schwarzenbach, R. P. *Environ. Sci. Technol.* **1984**, *18*, 652–657.

(49) Jafvert, C. T.; Westall, J. C.; Grieder, E.; Schwarzenbach, R. P. *Environ. Sci. Technol.* **1990**, *24*, 1795–1803.

(50) Acta, J. F. B. B.; Enuirorz, J. M. A. Environ. Sci. Techno **1987**, 370–373.

(51) Westall, J. C.; Chen, H.; Zhang, W.; Brownawell, B. J. *Environ. Sci. Technol.* **1999**, *33*, 3110–3118.

(52) Rico-Rico, Á.; Temara, A.; Behrends, T.; Hermens, J. L. M. *Environ. Pollut.* **2009**, *157*, 377–383.

(53) Westall, J. C.; Chen, H.; Zhang, W.; Brownawell, B. J. *Environ. Sci. Technol.* **1999**, *33*, 3110–3118.

(54) Mackay, D.; Paterson, S. *Environ. Sci. Technol.* **1991**, *25*, 427–436.

(55) Bondarenko, S.; Gan, J. *Environ. Sci. Technol.* **2009**, *43*, 3772–3777.

(56) *Lützhøft, H.-C.H.; Vaes W.H.J.; Freidig A.P.; Halling- SØrensen B.; Hermens, J.L.M.; Environ. Sci. Technol. 2000, 34, 4989–4994.*

(57) Heringa, M. B.; Hermens, J. L. M. *TrAC Trends Anal. Chem.* **2003**, *22*, 575–587.

(58) Poerschmann, J.; Kopinke, F. D.; Pawliszyn, J. *Environ. Sci. Technol.* **1997**, *31*, 3629–3636.

(59) Laor, Y.; Rebhun, M. *Environ. Sci. Technol.* **1997**, *31*, 3558–3564.

(60) Kumke, M. U. Analyst, **1994**, *1*, 997–1001.

(61) Oravcova´, J.; Bo¨hs, B.; Lindner, W. *J. Chromatogr. B Biomed. Sci. Appl.* **1996**, *677*, 1–28.

(62) Landrum, P. F.; Nihart, S. R.; Eadie, B. J.; Gardner, W. S. *Environ. Sci. Technol.* **1984**, *18*, 187–192.

(63) Rosner, W.; Toppel, S.; Smith, R. N. Biochem. Biophy. Acta, 1974, 351, 92–98.

(64) Kopinke, F.-D.; Georgi, A.; Mackenzie, K. *Acta Hydrochim. hydrobiol.* **2001**, *28*, 385–399.

(65) Horstmann, M.; McLachlan, M. S. *Environ. Sci. Technol.* **1992**, *26*, 1643–1649.

(66) Freidig, P. A; Garicano, E. L.; Busser, F. J. M; Hermens, J.L.M; Environ. Toxicol. Chem. 1998, 17, 998–1004.

(67) Vuignier, K.; Schappler, J.; Veuthey, J.-L.; Carrupt, P.-A.; Martel, S. *Anal. Bioanal. Chem.* **2010**, *398*, 53–66.

(68) Heringa, M. B.; Pastor, D.; Algra, J.; Vaes, W. H. J.; Hermens, J. L. M. *Anal. Chem.* **2002**, *74*, 5993–5997.

(69) Arthur, C.; Pawlyszin, J. *Anal. Chem.* **1990**, *62*, 2145–2148.

(70) Togunde, O. P.; Cudjoe, E.; Oakes, K. D.; Mirnaghi, F. S.; Servos, M. R.; Pawliszyn, J. *J. Chromatogr. A* **2012**, *1262*, 34–42.

(71) Boyacı, E.; Sparham, C.; Pawliszyn, J. *Anal. Bioanal. Chem.* **2014**, *406*, 409–420.

(72) Mirnaghi, F. S.; Mousavi, F.; Rocha, S. M.; Pawliszyn, J. *J. Chromatogr. A* **2013**, *1276*, 12–19.

(73) Jiang, R.; Pawliszyn, J. *TrAC Trends Anal. Chem.* **2012**, *39*, 245–253.

(74) Reyes-Garcés, N.; Bojko, B.; Pawliszyn, J. *J. Chromatogr. A* **2014**, *1374*, 40–49.

(75) Pawliszyn, J. *Handbook of Solid Phase Microextraction*; Elsevier, 2012.

(76) Górecki, T.; Yu, X.; Pawliszyn, J. Analyst, **1999**, 643–649.

(77) Boyaci, E.; Gorynski, K.; Rodriguez-Lafuente, A.; Bojko, B.; Pawliszyn, J. *Anal. Chim. Acta* **2014**, *809*, 69–81.

(78) Mirnaghi, F. S.; Chen, Y.; Sidisky, L. M.; Pawliszyn, J. *Anal. Chem.* **2011**, *83*, 6018–6025.

(79) Cudjoe, E.; Vuckovic, D.; Hein, D.; Pawliszyn, J. **2009**, *81*, 4226–4232.

(80) Ouyang, G.; Pawliszyn, J. *Anal. Chim. Acta* **2008**, *627*, 184–197.

(81) Ai, J. *Anal. Chem.* **1997**, *69*, 3260–3266.

(82) Ai, J. *Anal. Chem.* **1997**, *69*, 1230–1236.

(83) Zhang, X.; Es-Haghi, A.; Cai, J.; Pawliszyn, J. *J. Chromatogr. A* **2009**, *1216*, 7664–7669.

(84) Ouyang, G.; Pawliszyn, J. *Anal. Bioanal. Chem.* **2006**, *386*, 1059–1073.

(85) Crittenden, J. C.; Trussell, R. R.; Hand, D. W.; Howe, K. J.; Tchobanoglous, G.; Borchardt, J. H. MWH's Water Treatment Principles and Design, 3rd ed. Wiley: New Jersey, **2012**.

(86) Ouyang, G.; Pawliszyn, J. *J. Chromatogr. A* **2007**, *1168*, 226–235

(87) Ouyang, G.; Zhao, W.; Alaee, M.; Pawliszyn, J. *J. Chromatogr. A* **2007**, *1138*, 42–46.

(88) Woolcock, P. J.; Koziel, J. a.; Cai, L.; Johnston, P. a.; Brown, R. C. *J. Chromatogr. A* **2013**, *1281*, 1–8.

(89) Chen, Y.; Pawliszyn, J. *Anal. Chem.* **2003**, *75*, 2004–2010.

(90) Ouyang, G.; Zhao, W.; Bragg, L.; Qin, Z.; Alaee, M.; Pawliszyn, J. *Environ. Sci. Technol.* **2007**, *41*, 4026–4031.

(91) Chen, Y.; O'Reilly, J.; Wang, Y.; Pawliszyn, J. *Analyst* **2004**, *129*, 702–703.

(92) Wang, Y.; O'Reilly, J.; Chen, Y.; Pawliszyn, J. *J. Chromatogr. A* **2005**, *1072*, 13–17.

(93) Chen, Y.; Pawliszyn, J. *Anal. Chem.* **2004**, *76*, 5807–5815.

(94) Zhao, W.; Ouyang, G.; Alaee, M.; Pawliszyn, J. *J. Chromatogr. A* **2006**, *1124*, 112–120.

(95) Bragg, L.; Qin, Z.; Alaee, M.; Pawliszyn, J. *J. Chromatogr. Sci.* **2006**, *44*, 317–323.

(96) Zhou, S. N.; Zhao, W.; Pawliszyn, J. *Anal. Chem.* **2008**, *80*, 481–490.

(97) Shurmer, B.; Pawliszyn, J. *Anal. Chem.* **2000**, *72*, 3660–3664.

(98) Lord, H. L.; Pawliszyn, J. *Anal. Chem.* **1997**, *69*, 3899–3906.

(99) Ouyang, G.; Chen, Y.; Pawliszyn, J. *J. Chromatogr. A* **2006**, *1105*, 176–179.

(100) Qin, Z.; Bragg, L.; Ouyang, G.; Pawliszyn, J. *J. Chromatogr. A* **2008**, *1196-1197*, 89–95.

(101) Qin, Z.; Mok, S.; Ouyang, G.; Dixon, D. G.; Pawliszyn, J. *Anal. Chim. Acta* **2010**, *667*, 71–76.

(102) Yuan, H.; Ranatunga, R.; Carr, P. W.; Pawliszyn, J. *Analyst* **1999**, *124*, 1443–1448.

(103) Hu, X.; Peng, J.; Liu, J.; Jiang, G.; Jönsson, J. A. *Chemosphere* **2006**, *65*, 1935–1941.

(104) Artola-Garicano, E.; Vaes, W. H.; Hermens, J. L. *Toxicol. Appl. Pharmacol.* **2000**, *166*, 138–144.

(105) Broeders, J. J. W.; Blaauboer, B. J.; Hermens, J. L. M. *J. Chromatogr. A* **2011**, *1218*, 8529–8535.

(106) Lützhøft, H. C. H.; Vaes, W. H. J.; Freidig, A. P.; Halling-SØrensen, B.; Hermens, J. L. M. *Environ. Sci. Technol.* **2000**, *34*, 4989–4994.

(107) Heringa, M. B.; Hogevonder, C.; Busser, F.; Hermens, J. L. M. *J. Chromatogr. B. Analyt. Technol. Biomed. Life Sci.* **2006**, *834*, 35–41.

(108) Vaes, W. H.; Ramos, E. U.; Hamwijk, C.; van Holsteijn, I.; Blaauboer, B. J.; Seinen, W.; Verhaar, H. J.; Hermens, J. L. *Chem. Res. Toxicol.* **1997**, *10*, 1067–1072.

(109) Yuan, H.; Pawliszyn, J. *Anal. Chem.* **2001**, *73*, 4410–4416.

(110) Theodoridis, G. *J. Chromatogr. B. Analyt. Technol. Biomed. Life Sci.* **2006**, *830*, 238–244.

(111) Musteata, F. M.; Pawliszyn, J.; Qian, M. G.; Wu, J.; Miwa, G. T. **2006**, *95*, 1712–1722.

(112) Vuckovic, D.; Pawliszyn, J. *J. Pharm. Biomed. Anal.* **2009**, *50*, 550–555.

(113) Musteata, F. M.; Pawliszyn, J Proteome Res; **2005**, 789–800.

(114) Bojko, B.; Vuckovic, D.; Cudjoe, E.; Hoque, M. E.; Mirnaghi, F.; Wąsowicz, M.; Jerath, A.; Pawliszyn, J. *J. Chromatogr. B. Analyt. Technol. Biomed. Life Sci.* **2011**, *879*, 3781–3787.

(115) Vaes, W. H. J.; Urrestarazu Ramos, E.; Verhaar, H. J. M.; Seinen, W.; Hermens, J. L. M. *Anal. Chem.* **1996**, *68*, 4463–4467.

(116) Zhang, X.; Es-haghi, A.; Musteata, F. M.; Ouyang, G.; Pawliszyn, J. *Anal. Chem.* **2007**, *79*, 4507–4513.

(117) Poerschmann, J.; Zhang, Z.; Kopinke, F.-D.; Pawliszyn, J. *Anal. Chem.* **1997**, *69*, 597–600.

(118) Plagellat, C.; Kupper, T.; Furrer, R.; de Alencastro, L. F.; Grandjean, D.; Tarradellas, J. *Chemosphere* **2006**, *62*, 915–925.

(119) Rodil, R.; Moeder, M. *J. Chromatogr. A* **2008**, *1179*, 81–88.

(120) Tolls, J.; Berger, H. *Environ. Toxicol. Chem.* **2009**, *28*, 2485–2489.

(121) Greenwood, R.; Mills, G. a.; Vrana, B. *J. Chromatogr. A* **2009**, *1216*, 631–639.

(122) Söderström, H.; Lindberg, R. H.; Fick, J. *J. Chromatogr. A* **2009**, *1216*, 623–630.

(123) Vallejo, a; Prieto, a; Moeder, M.; Usobiaga, a; Zuloaga, O.; Etxebarria, N.; Paschke, a. *Water Res.* **2013**, *47*, 2851–2862.

(124) Ter Laak, T. L.; Durjava, M.; Struijs, J.; Hermens, J. L. M. *Environ. Sci. Technol.* **2005**, *39*, 3736–3742.

(125) Xie, M.; Yang, Z.-Y.; Bao, L.-J.; Zeng, E. Y. *J. Chromatogr. A* **2009**, *1216*, 4553–4559.

(126) Koziel, J. a; Martos, P. a; Pawliszyn, J. *J. Chromatogr. A* **2004**, *1025*, 3–9.

(127) Poerschmann, J.; Gorecki, T.; Kopinke, F. *Environ. Sci. Technol.* **2000**, *34*, 3824–3830.

(128) Boyacı, E.; Sparham, C.; Pawliszyn, J. Anal Bioanal Chem, **2014,** 406, 409–420.

(129) Zhang, X.; Clausen, M. R.; Zhao, X.; Zheng, H.; Bertram, H. C. *Anal. Chem.* **2012**, *84*, 7785–7792.

(130) Alexander Stern, S. *J. Memb. Sci.* **1994**, *94*, 1–65.

(131) Lonsdale, H. K. *Membrane Technology and Applications*, 1st ed; Weily: London, 1985

(132) Liu, H.; Liu, L.; Xiong, Y.; Yang, X.; Luan, T. *J. Chromatogr. A* **2010**, *1217*, 6747–6753.

(133) Richardson, S.; Ternes, T. a. *Anal. Chem.* **2014**, *81*, 4645–4677.

(134) Lindström, A.; Buerge, I. J.; Poiger, T.; Bergqvist, P. A.; Müller, M. D.; Buser, H. R. *Environ. Sci. Technol.* **2002**, *36*, 2322–2329.

(135) Cuderman, P.; Heath, E. *Anal. Bioanal. Chem.* **2007**, *387*, 1343–1350.

(136) *Chelan, L.; Ellis, S. G.; Booij, K.; Kaputa,* M. Chemosphere, *2006, 72, 1112–1117.*

(137) Allan, I. J.; Booij, K.; Paschke, A.; Vrana, B.; Mills, G. a.; Greenwood, R. *Environ. Sci. Technol.* **2009**, *43*, 5383–5390.

(138) Duan, C.; Shen, Z.; Wu, D.; Guan, Y. *TrAC Trends Anal. Chem.* **2011**, *30*, 1568–1574.

(139) Martos, P. a; Pawliszyn, J. *Anal. Chem.* **1999**, *71*, 1513–1520.

(140) Chen, Y.; Pawliszyn, J. *Anal. Chem.* **2004**, *76*, 6823–6828.

(141) Ouyang, G.; Chen, Y.; Pawliszyn, J. *Anal. Chem.* **2005**, *77*, 7319–7325.

(142) Chen, Y.; Pawliszyn, J. *Anal. Chem.* **2003**, *75*, 2004–2010.

(143) Ouyang, G.; Cui, S.; Qin, Z.; Pawliszyn, J. *Anal. Chem.* **2009**, *81*, 5629–5636.

(144) Ouyang, G.; Chen, Y.; Pawliszyn, J. **2010**, *77*, 7319–7325.

(145) Apelblat, A. *J. Mol. Liq.* **2002**, *95*, 99–145.

(146) Mirnaghi, F. S.; Monton, M. R. N.; Pawliszyn, J. *J. Chromatogr. A* **2011**.

(147) Rodil, R.; Quintana, J. B.; López-Mahía, P.; Muniategui-Lorenzo, S.; Prada-Rodríguez, D. *Anal. Chem.* **2008**, *80*, 1307–1315.

(148) Wick, A.; Fink, G.; Ternes, T. a. *J. Chromatogr. A* **2010**, *1217*, 2088–2103.

(149) Bratkovics, S.; Sapozhnikova, Y. *Anal. Methods* **2011**, *3*, 2943.

(150) Wu, J.-L.; Lam, N. P.; Martens, D.; Kettrup, A.; Cai, Z. *Talanta* **2007**, *72*, 1650–1654.

(151) Cheng, C.-Y.; Wang, Y.-C.; Ding, W.-H. *Anal. Sci.* **2011**, *27*, 197–202.

(152) Hua, W.; Bennett, E. R.; Letcher, R. J. *Environ. Int.* **2005**, *31*, 621–630.

(153) Fent, K.; Zenker, A.; Rapp, M. *Environ. Pollut.* **2010**, *158*, 1817–1824.

(154) Kawaguchi, M.; Ito, R.; Honda, H.; Endo, N.; Okanouchi, N.; Saito, K.; Seto, Y.; Nakazawa, H. *J. Chromatogr. A* **2008**, *1200*, 260–263.

(155) Zhang, Z.; Ren, N.; Li, Y.-F.; Kunisue, T.; Gao, D.; Kannan, K. *Environ. Sci. Technol.* **2011**, *45*, 3909–3916.

(156) Alexander, M. *Environ. Sci. Technol.* **2000**, *34*, 4259–4265.

(157) Reid, B. J.; Jones, K. C.; Semple, K. T. *Environ. Pollut.* **2000**, *108*, 103–112.

(158) Semple, K. T.; Doick, K. J.; Jones, K. C.; Burauel, P.; Craven, A.; Harms, H. *Environ. Sci. Technol.* **2004**, *38*, 228A – 231A.

(159) Schwarzenbach, R. P.; Escher, B. I.; Fenner, K.; Hofstetter, T. B.; Johnson, C. A.; von Gunten, U.; Wehrli, B. *Science* **2006**, *313*, 1072–1077.

(160) Burkhard, L. P. *Environ. Sci. Technol.* **2000**, *34*, 4663–4668.

(161) Oucette, W. I. J. D. *Environ. Toxicol.* **2003**, *22*, 1771–1788.

(162) Urrestarazu Ramos, E.; Meijer, S. N.; Vaes, W. H. J.; Verhaar, H. J. M.; Hermens, J. L. M. *Environ. Sci. Technol.* **1998**, *32*, 3430–3435.

(163) Carter, C. W.; Suffet, I. H. *Environ. Sci. Technol.* **1982**, *16*, 735–740.

(164) De Paolis, F.; Kukkonen, J. *Chemosphere* **1997**, *34*, 1693–1704.

(165) Chin, Y. P.; Aiken, G. R.; Danielsen, K. M. *Environ. Sci. Technol.* **1997**, *31*, 1630–1635.

(166) Chiou, C. T.; Kile, D. E.; Brinton, T. I.; Malcolm, R. L.; Leenheer, J. a. *Environ. Sci. Technol.* **1987**, *21*, 1231–1234.

(167) Lee, S.; Gan, J.; Liu, W. P.; Anderson, M. A. Environ. Sci. Technol. **2003**, 37, 5597−5602.

(168) Prosen, H.; Zupančič-Kralj, L. *Chromatographia* **2000**, *51*, S155–S164.

(169) Delgado-Moreno, L.; Wu, L.; Gan, J. *Environ. Sci. Technol.* **2010**, *44*, 8473–8478.

(170) Doll, T. E.; Frimmel, F. H.; Kumke, M. U.; Ohlenbusch, G. *Fresenius. J. Anal. Chem.* **1999**, *364*, 313–319.

(171) Ohlenbusch, G.; Kumke, M. U.; Frimmel, F. H. *Sci. Total Environ.* **2000**, *253*, 63–74.

(172) Kopinke, F. D.; Georgi, A.; Mackenzie, K. *Environ. Sci. Technol.* **2001**, *35*, 2536–2542.

(173) Mackenzie, K.; Geobgi, A.; Kumke, M.; Kopinke, F. D. *Environ. Sci. Technol.* **2002**, *36*, 4403–4409.

(174) Prosen, H.; Fingler, S.; Zupancic-Kralj, L.; Drevenkar, V. *Chemosphere* **2007**, *66*, 1580–1589.

(175) Cui, X.; Bao, L.; Gan, J. *Environ. Sci. Technol.* **2013**, *47*, 9833–9840.

(176) Jia, F.; Cui, X.; Wang, W.; Delgado-Moreno, L.; Gan, J. *Environ. Pollut.* **2012**, *167*, 34–40.

(177) Oomen, a G.; Mayer, P.; Tolls, J. *Anal. Chem.* **2000**, *72*, 2802–2808.

(178) Qin, Z.; Bragg, L.; Ouyang, G.; Niri, V. H.; Pawliszyn, J. *J. Chromatogr. A* **2009**, *1216*, 6979–6985.

(179) Avena, M. J.; Koopal, L. K. *Environ. Sci. Technol.* **1999**, *33*, 2739–2744.

(180) Mayer, P.; Karlson, U.; Christensen, P. S.; Johnsen, A. R.; Trapp, S. *Environ. Sci. Technol.* **2005**, *39*, 6123–6129.

(181) Haftka, J. J. H.; Parsons, J. R.; Govers, H. a J.; Ortega-Calvo, J.-J. *Environ. Toxicol. Chem.* **2008**, *27*, 1526–1532.

(182) Alam, M. N.; Ricardez-Sandoval, L.; Pawliszyn, J. *Anal. Chem.* **2015**, 150907063856006.

(183) Vuckovic, D.; Pawliszyn, J. **2009**, *50*, 550–555.

(184) Niederer, C.; Schwarzenbach, R. P.; Goss, K. U. *Environ. Sci. Technol.* **2007**, *41*, 6711–6717.

(185) Akkanen, J.; Vogt, R. D.; Kukkonen, J. V. K. **2004**, *66*, 171–177.

(186) Ferreira, J. a; Nascimento, O. R.; Martin-Neto, L. *Environ. Sci. Technol.* **2001**, *35*, 761–765.

(187) Wei-Haas, M. L.; Hageman, K. J.; Chin, Y. P. *Environ. Sci. Technol.* **2014**, *48*, 4852–4859.

(188) DiVincenzo, J. P.; Sparks, D. L. *Arch. Environ. Contam. Toxicol.* **2001**, *40*, 445–450.

(189) Delle Site, A. *J. Phys. Chem. Ref. Data* **2001**, *30*, 187–439.

(190) Kragulj, M.; Tričković, J.; Kukovecz, Á.; Jović, B.; Molnar, J.; Rončević, S.; Kónya, Z.; Dalmacija, B. *RSC Adv.* **2015**, *5*, 24920–24929.

(191) Luthy, R. G.; Aiken, G. R.; Brusseau, M. L.; Cunningham, S. D.; Gschwend, P. M.; Pignatello, J. J.; Reinhard, M.; Traina, S. J.; Weber, W. J.; Westall, J. C. *Environ. Sci. Technol.* **1997**, *31*, 3341–3347.

(192) Means, J. C.; Wood, S. G.; Hassett, J. J.; Banwart, W. L. *Environ. Sci. Technol.* **1980**, *14*, 1524–1528.

(193) Accardi-Dey, A.; Gschwend, P. M. *Environ. Sci. Technol.* **2002**, *36*, 21–29.

(194) Styszko, K. J Soils Sediments, 2015, **16, 677-686**.

(195) Xu, J.; Wu, L.; Chen, W.; Chang, A. C. *J. Environ. Qual.* **2009**, *38*, 1177–1182.

(196) Stipicević, S.; Fingler, S.; Drevenkar, V. *Arh. Hig. Rada Toksikol.* **2009**, *60*, 43–52.

(197) Huang, X.; Wu, C.; Hu, H.; Yu, Y.; Liu, J. *Ecotoxicol. Environ. Saf.* **2015**, *116*, 76–83.

(198) Wang, W.; Delgado-Moreno, L.; Ye, Q.; Gan, J. *Environ. Sci. Technol.* **2011**, *45*, 1521–1527.