ABSTRACT

DEVELOPMENT OF MICRO-SCALE AND AUTOMATED MEMBRANE EXTRACTION SYSTEMS FOR WATER ANALYSIS

by

Xiaoyan Wang

Monitoring of trace level contaminants in water involves the extraction and enrichment of analytes. Over the years several techniques have been developed for this purpose. Currently there is an urgent need for the development of micro-scale extraction techniques that can lead to simple, fast trace analysis. Automated instruments that can be used for continuous, on-line analysis are also important. Membrane extraction can address both these needs, because it can be carried out in a continuous fashion and can also be miniaturized. This research addresses these two aspects of membrane extraction for the next generation of sample preparation.

Micro-Scale Membrane Extraction:

Micro-scale hollow fiber membrane extraction was studied for extraction and preconcentration of non-polar analytes as well as haoacetic acids in water. They represent two important classes of pollutants. The goal was to optimize solvent systems to provide high enrichment and reproducible results. The concept of using a barrier film as a means of providing a diffusional barrier on the membrane fiber was developed. It was found to play an important role by protecting the organic acceptor from migration, resulting in much less acceptor loss. The analytes were extracted from water sample without any pretreatment into an organic acceptor via an organic layer on the membrane (referred to as barrier film). The concentrated extract was analyzed by HPLC and the separation was achieved within 12 minutes. Enrichment factors as high as 4555 were obtained in a 60-

minute extraction. This represented a 27 times enhancement over that has been preiously published in the literature. Detection limits were at low to ppt level with RSDs between 1.60 and 7.65%. Large linear dynamic ranges with good linearity (R^2 between 0.9870 and 0.9997) were achieved. The implementation of a barrier film represents a major development in the area of membrane based microextraction because it allows a wide variety of solvents to be used and also the use of rigorous extraction conditions.

Micro-fluidic devices were designed and fabricated for analyte enrichment. The analytical approach is based on supported liquid membrane extraction (SLME) followed by direct HPLC-UV detection without any derivatization. Channel dimensions and the flow rates affected enrichment factors and extraction efficiencies. Enrichment factors (EF) as high as 54 were obtained on a 2×2 cm extraction module. Large linearity ranges with good linearity (R^2 between 0.9895 and 0.9996), high precisions (RSD between 3.56 and 8.54%) and detection limit as low as 2 ng/mL were obtained.

Automated, On-line Membrane Extraction:

On-line extraction involves the continuous introduction of water on the feed side of the membrane and continuous enrichment of analytes on the permeate side. In fact membranes represent the only extraction media where such a continuous process is possible. Two different extraction approaches were investigated. The first was for real-time monitoring of haloacetic acids in water, and the second was for on-line liquid-liquid membrane extraction followed by membrane concentration.

Hollow fiber liquid-liquid membrane extraction (LLME) and SLME followed by on-line HPLC-UV detection were developed for continuous monitoring of the nine HAAs. With continuous LLME, seven halo-acetic acids could be analyzed and EF was

around 50. All the nine acids could be extracted and quantified by continuous SLME. Experiments with laboratory standards demonstrated that EF and extraction efficiency could be as high as 500 and 54%, respectively. Relative standard deviations based on seven replicates were between 3.3 and 10.3 %, and the MDLs were at sub-ppb levels.

A total analytical system (TAS) was developed by interfacing continuous membrane extraction, pervaporation and on-line HPLC-UV detection. Organics from a water sample were extracted into an organic solvent, and then concentrated via pervaporation prior to HPLC-UV detection. Factors affecting the system performance were studied. With optimized experimental parameters enrichment factors as high as 192 were obtained, the method detection limits were at low ng/mL levels, and the precisions were better than 5%.

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CHAPTER 1

INTRODUCTION

1.1 Water Analysis and Sample Preparation

1.1.1 Water Analysis

Water quality is threatened by industrial discharge, urban runoff and agricultural activities, such as pesticide usage. The US Congress passed the Clean Water Act (CWA) to protect surface water, and the Safe Drinking Water Act (SDWA) for ground and drinking water. The US Environmental Protection Agency (USEPA) has published a list of the priority contaminants and regulated their Maximum Contamination Levels (MCLs). Today, water contamination appears to be a serious problem and the US EPA has stated that over 40% of the assessed water does not meet the water quality standard. Thus, significant efforts are required to monitor and clean up polluted waters.

Typical water analysis involves four steps:

- Sampling at the site;
- Transporting/storage of samples;
- Sample preparation;
- Instrumental analysis.

The whole process takes place at different times and places. Errors are introduced via evaporation, degradation and contamination, which result in decreases in accuracy and precision. Moreover, it is time-consuming labor-intensive, and consumes large amount of reagents, resulting in high cost per sample. Real-time, continuous, on-line monitoring methods need to be developed and implemented to avoid these limitations.

1.1.2 Sample Preparation

In the overall water analysis process, sample preparation plays an important role and is the bottleneck in many analytical methodologies [1]. A survey in 1991 claimed that sample preparation accounted for about 61% of the effort of typical analytical chemists [2]. Poor sample treatment may destroy the whole assay even with the most powerful detection method.

The aims of sample preparation are: removal of the undesired gradients from the sample to achieve higher selectivity, concentration of the analyte for higher sensitivity or to bring the analyte concentration within the instrumental linear dynamic range. In some other cases, the analyte may not be in an analyzable form, thus further derivatization is needed. For example, acids need to be converted to esters to be detected by gas chromatography (GC). In many cases, analytes are separated from the sample and transferred to another matrix, which is termed "extraction". Interferences may need to be removed from the matrix, which is referred to as "clean-up". Since environmental pollutants are present in trace concentrations (ppm to ppt level) and in a complex matrix, sample preparation for preconcentration or enrichment is very important as it increases the analyte concentration to a level suitable for instrumental detection.

1.1.3 Conventional Sample Preparation Methods

Many pollutants in water are organic compounds, which can be classified into two categories: volatile organic compounds (VOCs) and semi-volatile organic compounds (SVOCs). The conventional sample preparation methods for VOCs are headspace analysis, purge and trap (PT). While liquid-liquid extraction (LLE) and solid phase extraction (SPE) are the conventional methods for SVOCs.

In headspace analysis, the water sample is placed in a vial and sealed with a cap. VOCs are evaporated into the headspace of the vial due to their high volatility. The equilibrium of evaporation can be enhanced by heating or stirring. The headspace air (VOCs in air) is drawn and transferred for instrumental analysis after equilibrium is reached. In PT, the VOCs are expelled from a solution by flushing with an inert gas, and then trapped into a cryogenic sorbent trap [3]. PT is used widely in US EPA standard methods for VOC analysis. However, these methods are not suitable for compounds with high water solubility.

LLE has been widely used for a few centuries and is still in use by many US EPA standard procedures. In LLE, the analytes are extracted from an aqueous sample into an organic solvent. Good selectivity and high recovery is achieved by selecting suitable solvents for different analytes. However, LLE has several disadvantages. It uses a large amount of organic solvents and the extract often needs further concentration via solvent evaporation. It is labor-intensive and time-consuming. Emulsion formation is another issue in LLE, making the separation of the organic and aqueous phases difficult. Classical LLE is hard to couple directly to analytical instruments for continuous on-line monitoring.

SPE was first reported about 50 years ago and developed significantly during the last 20 years [4]. In SPE, the organic analytes are extracted from an aqueous sample onto a solid sorbent made of bonded silica or different polymers, and then eluted with a suitable solvent by altering the polarity or pH. The selectivity of SPE can be achieved by choosing a variety of sorbents and solvents. Compared to LLE, SPE uses less organic solvent and can be coupled to high performance liquid chromatography (HPLC), GC or other instruments for on-line analysis. However, it involves multiple batch processes such as

conditioning, washing, and elution. The SPE cartridges are disposable, which represents a high consumable cost. Moreover, it is quite challenging when the analytes are very polar as they are difficult to extract from water.

1.1.4. Emerging Sample Preparation Approaches

Although many conventional sample methods are still in use, there exists the need for the development of alternative higher performance methods. It is important to develop techniques for [2]:

- Smaller sample sizes
- Higher selectivity in extraction
- Automation for on-line analysis, reducing manual operations
- Less reagent and organic solvent consumption, and less waste generation

Alternative sample preparation methods, such as, flow injection analysis (FIA), solid phase microextraction (SPME), stir-bar extraction (SBE), liquid phase microextraction (LPME), and membrane technology have been evolved recently based on the goals.

FIA has been used intensively since its introduction in 1975 [5], mainly in clinical and environmental analysis. It offers several advantages: capable of on-line analysis, reduced human intervention in time-consuming procedures, shorter analysis time, decreased reagent consumption and waste generation. Another attractive characteristic is the possibility of using unstable reagents or analyzing unstable compounds that decompose when using conventional procedures [6].

SPME was invented by Pawliszyn and coworkers in 1990, and has emerged as a promising technique [7]. In SPME, the analytes are adsorbed onto a fused-silica or polymer coated fiber and then thermally desorbed in a GC injection oven, or extracted directly into

the eluent of HPLC. For VOC analysis headspace SPME is usually performed. SPME is a solvent-free technique and the instrumentation is simpler. However, it is only suitable for high concentration analysis due to its low sensitivity. The fiber is fragile and the replacement is costly.

The development of SBE was prompted by the small volume of the stationary phase of the SPME fiber, which may result in incomplete extraction. In SBE, a magnetic stirrer bar is coated with a bonded sorbent layer. It is simply stirred in the sample, removed after a certain stirring time, and finally desorbed thermally (with a thermal desorption unit) for GC analysis, or extracted into a solvent for HPLC analysis [8-9]. The disadvantage of SBE is that it is difficult to remove the stir-bar out of the sample automatically.

LPME is a miniaturized LLE that was first reported in 1996 [10]. It is based on hanging a droplet of organic solvent at the end of a micro syringe and placing it into an aqueous sample, the analytes are extracted into the organic droplet, and finally the droplet is withdrawn back to the syringe and injected into analytical instruments for detection. LPME reduces the usage of organic solvents and offers high selectivity through solvent selection and pH variation. However, it requires careful human attention, as the droplet can mix or dissolve in the sample.

1.2 Membrane Technology

1.2.1 Membrane and Membrane Technology

Membrane technology has been evolved as one of the most promising sample preparation alternatives to conventional methods, not only in analytical laboratories, but also in large-scale industrial applications [11]. The concept of membrane separation is shown in

Figure 1.1. Generally speaking, the membrane is a selective barrier between two phases [12]. The sample (referred to as the donor) contacts one side of the membrane, while another phase (referred to as the acceptor) collects the species that permeate through. Mass transfer occurs by one or more driving forces, such as, concentration, pressure, pH, and electrical potential gradient.

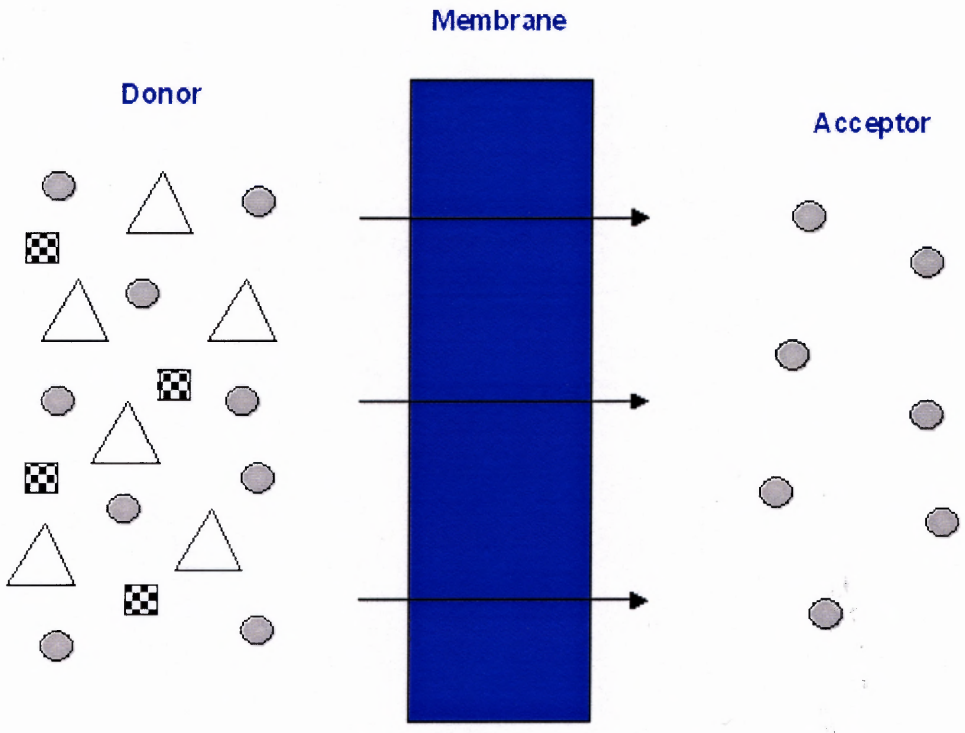


Figure 1.1 Concept of membrane separation [35].

A wide variety of membrane materials can be used. In many cases a membrane is a porous network of polymers, such as polypropylene, polysulfone or cellulose derivatives [13]. With these membranes, separation takes place based on size-exclusion: small molecules can permeate through the membrane, while larger ones do not. However

membrane separation is not only based on size difference, but also on the ionic properties of the compound. In ion-exchange membranes, ionic compounds with the same charge as the membrane ions are excluded. Some other membranes, for example, nonporous membrane that consist of a layer of liquid or polymer is different. For compounds of the same size, the separation is based on the different physicochemical properties of the compounds. Geometrically speaking, membrane can be classified into flat sheet or hollow fiber membranes. Hollow fiber membranes have larger surface to volume ratios and higher packing density, as a large number of fibers can be packed into a small volume.

1.2.2 Applications of Membrane Technology

Membrane technology has been used widely because it is simple, inexpensive, consumes less solvent, and offers high enrichment and clean-up efficiency. Moreover, membrane separation has been coupled to various instruments for continuous on-line analysis.

Gas permeation, pervaporation, filtration, dialysis, electrodialysis, and membrane extraction are the most frequently used membrane applications for sample preparation. Gas permeation takes place when some components in the donor pass through the membrane more rapidly than others. Microporous and nonporous membranes are applicable for gas permeation. Large molecules are excluded from the pores of microporous membranes. For a nonporous membrane, gas dissolves in the membrane material and then diffuse through it. The driving forces for gas permeation are concentration, pressure, and temperature differences. The main application of gas permeation on sample preparation is for the detection and removal of volatile organic compounds from air samples.

Pervaporation is characterized by the existence of a membrane between a liquid and a gaseous phase, selective mass transfer occurred across the membrane to the gas side.

Since different components pass through the membrane at different speeds, a compound in the donor side can be highly enriched compared to the permeate side. In the meanwhile, the species on the donor side can be concentrated by losing the components that have higher permeating speeds. The pervaporation process can be speeded up by applying a vacuum at the permeate side which removes the permeated species quickly. The driving forces for pervaporation could be chemical potential gradient, pressure and temperature differences. The main applications of pervaporation include water or solvent removal from organic or aqueous samples, organic/organic separation and vapor permeation [11].

Membrane filtration can be classified into ultrafiltration and microfiltration. Ultrafiltration is primarily a size-exclusion separation process. The driving force is pressure gradient. The average pore sizes of ultrafiltration membranes range from 10 to 1000 angstroms and the molecular weight of the retaining species range from 300 to 500,000 Daltons, including sugars, polymers, biomolecules, and colloids [11]. Microfiltration is used for the separation of micro-sized particles from fluids. Separation occurs when a pressure-driven flow (1 to 50 psi) is applied across a membrane. Microfiltration has seen many successful applications, such as syringe filters and filter systems for HPLC mobile phase.

Dialysis is a membrane process in which microsolutes pass through while the macrosolutes are blocked. On the permeate side, an aqueous solution is used to receive the permeated microsolutes. The selectivity is achieved by sieving in the membrane pores and the driving force is the concentration gradient between the two sides of membrane. In electrodialysis, a pair of electrodes is placed on both sides of the membrane, which creates

an electric potential gradient. Another driving force of electrodialysis is the concentration difference across the membrane.

Membrane extraction has evolved as one of most promising sample preparation methods and has been used widely in analytical processes. In membrane extraction, the solutes first dissolve into the membrane and then diffuse through it. At the other side of membrane the acceptor receives the solutes. The mechanism is described as solution-diffusion theory. The driving force of membrane extraction is the concentration gradient between the two sides of the membrane. Membrane extraction has been used for the separation of a variety of volatile, semi-volatile and non-volatile compounds from different types of samples, with high enrichment and selectivity. The details of membrane extraction will be discussed in the following section.

1.3 Membrane Extraction

Membrane extraction is a family of techniques that can be applied to many extraction problems [14]. It requires very little organic solvent, and offers high enrichment and clean-up efficiency. Systems based on membrane extraction can be automated and connected with various analytical instruments for continuous on-line analysis [15-27]. Both inorganic and organic analytes can be extracted with different kinds of membrane extraction modules. [15-30]. There are three main types of membrane extraction, supported liquid membrane extraction, liquid-liquid membrane extraction and polymeric membrane extraction. The phase conditions of the three types of membrane extraction are listed in Table 1.1.

Table 1.1 Phases of the Three Types of Membrane Extraction

Name	Abbreviation	Phases (Donor-membrane-acceptor)
Supported liquid membrane extraction	SLME	Aqueous-organic-aqueous
Liquid-liquid membrane extraction	LLME	Aqueous-organic-organic or organic-organic-aqueous
Polymeric membrane extraction	PME	Aqueous-polymer-aqueous or organic-polymer-aqueous or aqueous-polymer-organic

1.3.1 Supported Liquid Membrane Extraction (SLME)

The application of SLME to sample preparation in analytical chemistry was first introduced by Audunsson in 1986 [31]. Since then it has been developed quickly as a useful tool for preconcentration of complex samples, such as blood plasma, urine and wastewater, etc [32]. The main applications of SLME are for biological and environmental analysis. SLME has also been used for industrial separations, such as the extraction of metal ions and organic acids in wastewater.

In SLME, on one side of the membrane the donor is adjusted to a certain pH so that the analytes are in their molecular forms. On the other side another aqueous solution, which is called acceptor, is adjusted at a different pH to receive the analytes via an

acid-base reaction. The supported liquid membrane is made by soaking a piece of hydrophobic microporous membrane (support) in an organic solvent (liquid) for a while, so that the solvent is held in the membrane pores by capillary forces. The organic solvents for SLME are long-chain hydrocarbons, which should be non-volatile and insoluble in water, to achieve long stability in the membrane. Typical solvents used are n-undecane, kerosene and polar compounds like dihexyl ether and dioctyl phosphate [33] for moderate polar to nonpolar compounds. For very polar compounds, hydrogen-bonding reagents, such as tri-n-octylphosphine oxide (TOPO) can be added to the membrane liquid to increase the efficiency of such extraction [28-29, 34].

Figure 1.2 shows the basic principle of SLME of an acid [35]. The acid molecules (HA) move from the bulk donor solution to the surface of the membrane, dissolve into the organic solvent in the membrane or the membrane pores, and are back extracted into the acceptor solution via deprotonation. Extraction and back-extraction take place simultaneously resulting in efficient mass transfer. Neutral compounds can pass through the membrane but without enrichment. The base ions are excluded from extraction because they are charged. Similarly, the acid ions cannot be back-extracted into the donor side, resulting in the enrichment of acid ions in the acceptor solution.

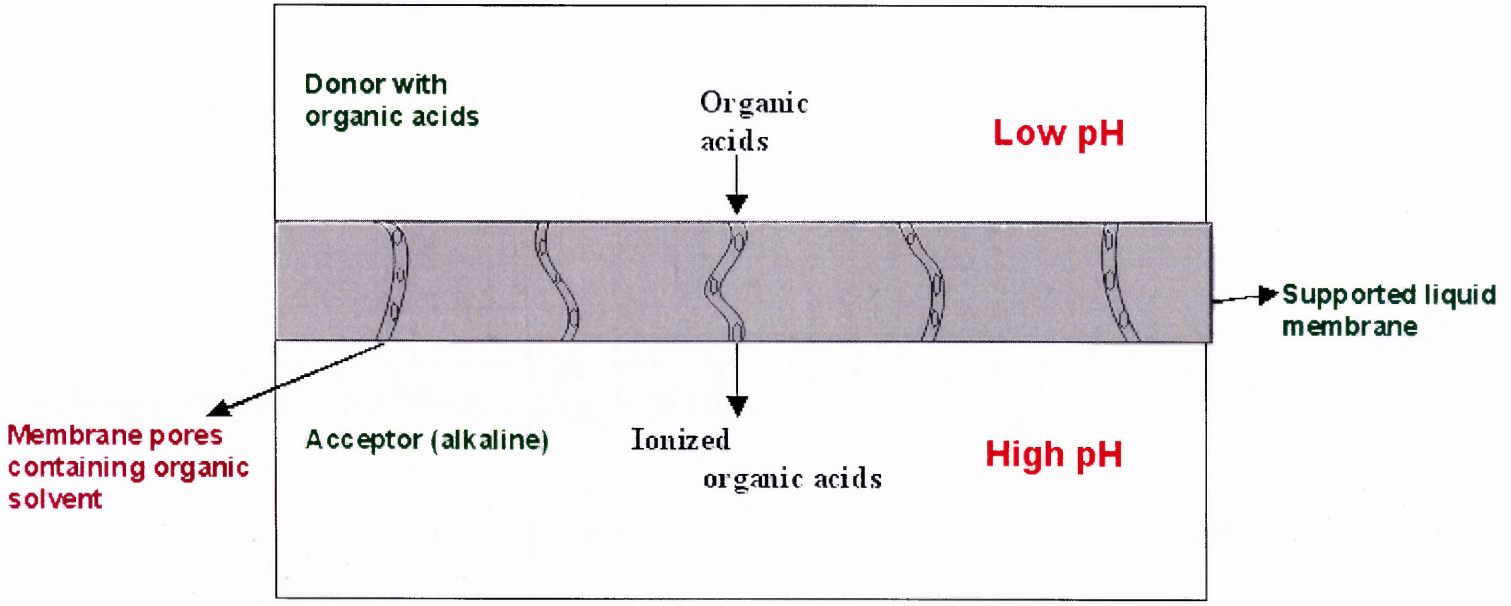


Figure 1.2 The concept of SLME of organic acids.

SLME offers high enrichment and selectivity, and has been applied to polar compounds, such as organic acids or bases, charged compounds, and metal ions [36]. The selectivity is achieved by tuning the chemistry of the three phases for different classes of compounds. Simply, basic analytes can be extracted in an analogous manner by reversing the pH conditions. In addition to pH, the organic solvent, membrane, and ion strength of the donor are the factors that tune selectivity in SLME. In summary, a neutral, extractable species should be formed in the donor phase by adjusting pH. The species should be transported through the membrane and into the acceptor phase, in another non-extractable form. In short, SLME is a combination of extraction into an organic solvent followed by a back-extraction into another aqueous phase [37]. The mass transfer from the donor to acceptor is proportional to the concentration difference, ΔC, over the membrane, which can be expressed as:

$$\Delta C = \alpha_D C_D - \alpha_A C_A \tag{1.1}$$

Where C_D and C_A are the concentration in the donor and acceptor respectively, and α_D and α_A are the fractions of the analytes that are the extractable (molecular) form in the donor and acceptor phase. Normally the extraction conditions are set so that α_D is close to unity and α_A is very small, for instance, to an alkaline donor and an acidic acceptor if bases are to be extracted, the C_A is zero from the beginning of the extraction and increases successively, usually to a value well above C_D at the end. The maximum enrichment factor (EF_{max}) is obtained when ΔC reaches zero:

$$EF_{max} = (C_A / C_D)_{max} = \alpha_D / \alpha_A \tag{1.2}$$

The extraction efficiency (EE) is usually expressed as:

$$EE = n_A / n_{DI} \tag{1.3}$$

Where n_A and n_{Di} are the number of moles of analyte in the acceptor and the initial donor, respectively. Extraction efficiency is a function of many parameters, including the partition coefficient (K) of the analyte between the aqueous phase and the organic liquid in the membrane, the acceptor conditions, flow rates of the donor and acceptor, and the characteristics and dimensions of the membrane and channels [33]. The influence of K is still not straightforward. For relatively hydrophilic compounds with a low K, the analytes are not sufficiently extracted into the organic solvent and the overall mass transfer is limited by the diffusion through the membrane, which leads to a low EE. For intermediate K, the mass transfer is limited by the analyte transport in the donor phase. For very hydrophobic compounds (high K values), the limiting factor is the transport of analyte into the acceptor phase. It was found that the most efficient extraction occurred when the octanol-water partition coefficient was around 10^3 [38]. The acceptor conditions are also important for the extraction efficiency, which decreases if the trapping is not complete and subsequently limits the enrichment factor that can be achieved [39].

1.3.2 Liquid-liquid Membrane Extraction (LLME)

LLME is another approach to membrane extraction. It is a two-phase extraction system, where the analytes are extracted from an aqueous phase into an organic phase or from an organic phase into an aqueous phase. Both hydrophobic and hydrophilic membranes are used in LLME. In a hydrophobic membrane, the membrane pores are filled with an organic solvent, the analytes in the aqueous donor phase move toward the membrane, dissolve in the membrane, and then reach into the organic acceptor. For a hydrophilic membrane, the aqueous phase wets and fills the membrane pores. The analytes are extracted from an

organic donor into an aqueous acceptor phase. LLME with hydrophilic membranes are seldom used for analytical purposes.

LLME is more suitable than SLME for hydrophobic compounds, such as, hydrocarbons. These compounds are more easily extracted from an aqueous phase into an organic solvent. The schematic diagram of a continuous LLME is shown in Figure 1.3.

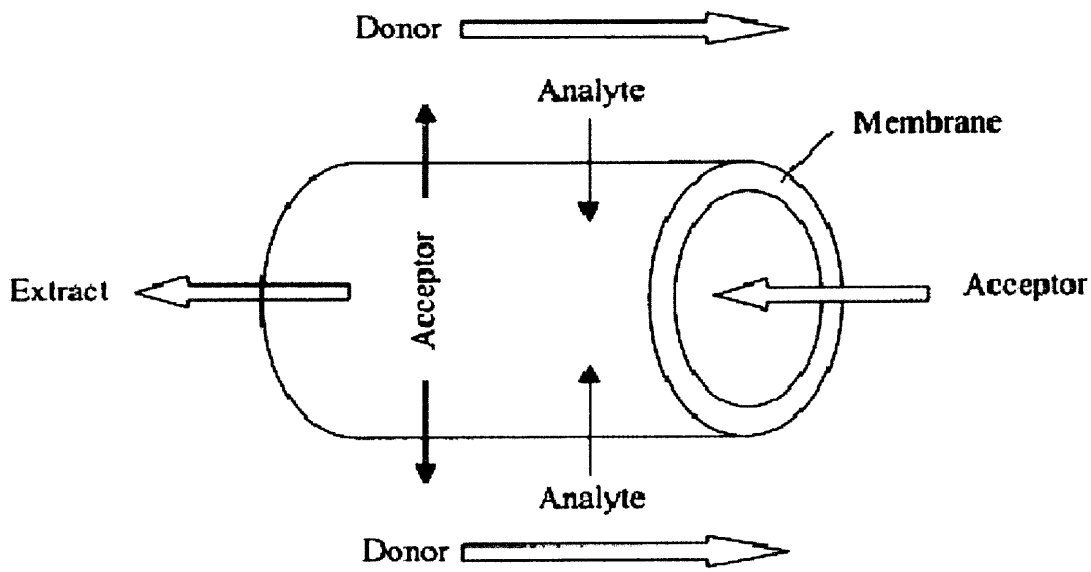


Figure 1.3 Schematic diagram of a continuous LLME [61].

The principle of LLME is equivalent to a conventional LLE, but may be performed in a flow system that allows easy automation by interfacing to analytical instruments. This technique is easily interfaced with GC and HPLC. Due to the presence of a membrane in LLME, there is no emulsion formation, which is a common phenomenon when the two phases are directly contacted, such as in LLE. In LLME, the aqueous phase and organic

phases are never mixed, and mass transfer between the phases takes place at the surface of the membrane.

The extraction efficiency of LLME is also limited by the partition coefficient. It is possible to obtain a considerable enrichment into a relatively small acceptor volume. When the partition coefficient is small, flow of the acceptor phase moves very slowly to accept and remove the extracted analytes, and maintain the diffusion through the membrane. However, this may lead to a smaller degree of enrichment.

Polymeric membrane extraction (PME) is another application of membrane extraction. The most commonly used membrane for PME is silicon rubber, which leads to a longer lifetime of the membrane. There are possibilities for both aqueous-polymer-aqueous extraction, which is similar to SLME, and aqueous-polymer-organic extraction (similar to LLME). Melcher et al first described both principles [40-41]. However, PME is not used as frequently as SLME and LLME due to the slow mass transfer rate and less possibilities for chemical tuning.

1.3.3 Applications

Membrane extraction has been applied widely to various analytes and different matrices [33]. In environmental analysis many kinds of analytes are extracted and enriched. These include VOCs, which can be extracted from aqueous samples into a gaseous acceptor through membrane pervaporation, which can be coupled to GC or mass spectrometry (MS) for the on-line monitoring of VOCs. This application has been studied in our laboratory [16, 27, 42-43]. Kou et el reported gas injection membrane extraction (GIME) followed by GC detection for fast analysis of VOCs in water. In GIME aqueous sample was introduced into the membrane extraction system by gas injection, which reduced the formation of

boundary layer, and increased the overall diffusion coefficient [16]. VOCs in air samples can also be extracted via membrane extraction [15]. Compounds such as benzene, toluene, ethylbenzene, xylenes and similar compounds were analyzed [15-16, 27, 42-43]. Acidic and basic compounds, mainly from surface water samples can be extracted using SLME. Compounds such as pesticides [44], phenoxy acids [45], sulfonylurea herbicides [46-47], and phenolic compounds [48] in water have been reported. Carboxylic acids in air samples [49] and in soil samples [50-51] were also analyzed by SLME. Basic analytes, such as aliphatic amines [52], triazine herbicides [53-55], and aniline derivatives [56] also have been extracted and enriched. Other compounds, such as metal ions [57-59], and anionic surfactants [60] in environmental samples also were extracted by SLME successfully. Nonionizable compounds or those with low polarity can be extracted with LLME, in which the analystes are extracted into an organic extract. Analytes, such as naphthalene [24, 30], pentachlorophenol [26, 61], triazine herbicides [62], cationic surfactants [63], and organotin compounds [64] have been analyzed by LLME.

Membrane extraction has also been applied widely in bioanalysis to determine drugs and other compounds as well in biological fluids, such as blood, plasma and urine [33]. The sample volume is limited in bioanalysis, thus the enrichment factor is generally smaller than in environmental analysis. SLME has been used for the analysis of ropivacaine metabolites in urine [18], amperozide in blood plasma [65], and some other drug compounds [23, 66] with a high degree of selectivity achieved. SLME with other compounds, such as heavy metals [67-68] and aliphatic amines [69] from urine were reported. LLME has also been applied to bioanalysis, such as to anaesthetics in blood plasma [17].

For food analysis, membrane extraction has been used in the determination of vitamin E in butter [70], pesticide residues in lipid matrices [71], and phenoxy herbicides in milk [72]. Some interesting applications of SLME were to solid or semi-solid food samples. Nicotine in snuff [73], vanillin in chocolate [74], and caffeine in coffee and tea [75] have been extracted.

1.4 Membrane Modules

The membrane modules can be made from flat sheet and hollow fiber membranes. The flat sheet membrane modules are usually constructed of two blocks of inert material with grooved channels. The blocks are clamped or screwed together with a piece of flat sheet membrane sandwiched in between. Figure 1.4 shows the schematic diagram of such modules.

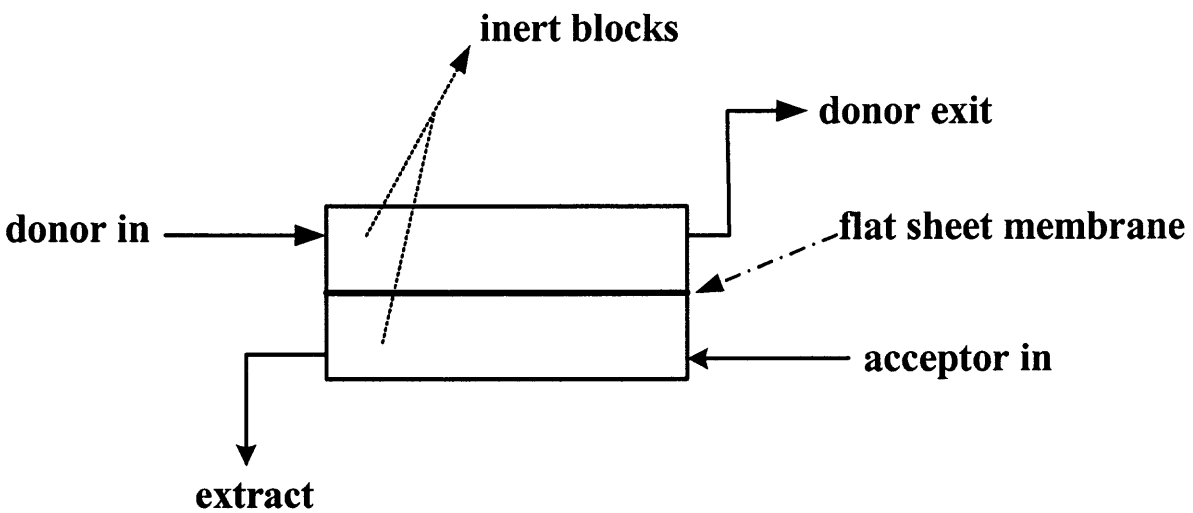


Figure 1.4 Schematic diagram of flat sheet membrane module.

Hollow fiber membrane modules are made by packing a number of hollow fibers inside a stainless steel or PTFE tubing. Two T-unions are connected at each end of the tubing to connect the donor and acceptor line. Epoxy glue is used to seal the ends to prevent the mixing of the two phases. Figure 1.5 shows an example of this module.

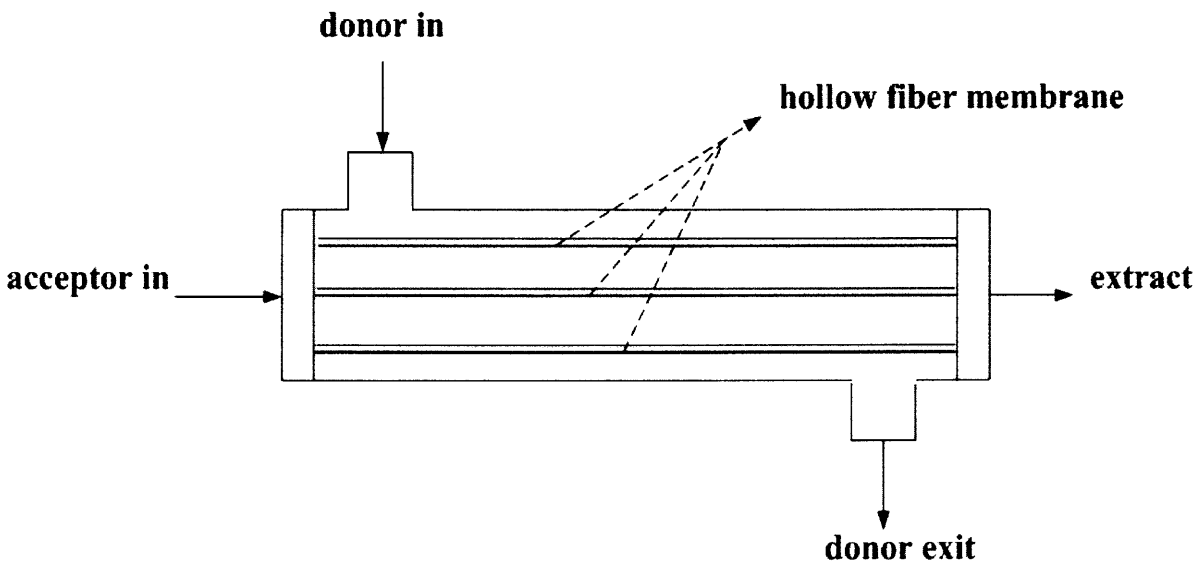


Figure 1.5 Hollow fiber membrane module.

These two modules can be used for continuous on-line analysis. For off-line extraction, a simple setup is shown in Figure 1.6. Two syringes taped together are used for introducing and withdrawing acceptor into the hollow fiber membrane, and holding the membrane in place at the same time. A magnetic stirrer is utilized to speed up the mass transfer in the donor phase.

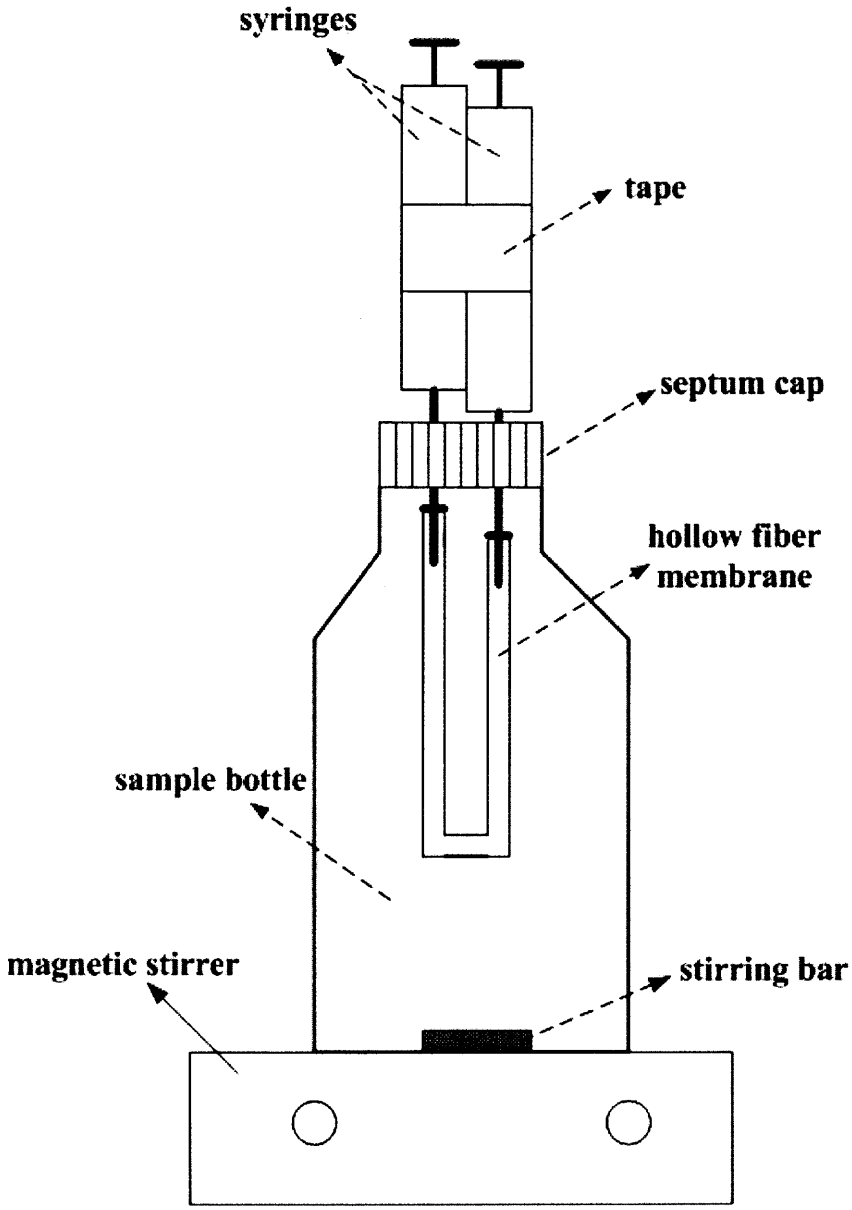


Figure 1.6 Off-line membrane extraction setup.

CHAPTER 2

RESEARCH OBJECTIVS

As mentioned in Chapter 1, the important needs in the analytical instrumentation arena are miniaturization and automation. Membrane separation is a promising sample preparation method that can facilitate both. The overall objectives of this study were to develop the miniaturized membrane modules for sample enrichment and to develop automated membrane systems for continuous on-line monitoring of both polar and non-polar compounds in water.

Hollow fiber based microextraction techniques have shown much promise in the extraction and preconcentration of analytes from aqueous matrices. Typically, a solvent held in the membrane is used as an extractant (or an acceptor). However, a problem of the micro-scale extraction is the outflow of the extract, especially during long extractions, and with vigorous stirring. To enhance the performance, the stabilization of the acceptor is important. A barrier film that can decrease the extract loss will be implemented and its effect will be studied. Hollow fiber microextraction will be carried out in two different approaches: liquid-liquid membrane microextraction and supported liquid membrane microextraction for the determination non-polar and acidic analytes in water.

A great advantage of membrane extraction is that it can be miniaturized onto a chip. It involves the flow of sample on one side of the membrane, while an extractant flows on the other side, and the analytes selectively permeate across the membrane. Miniature, on-chip devices will be designed and fabricated on polycarbonate wafers. The dimensions of the chips, and the flow rates of the sample and extractant will be investigated.

Automated online measurements are less expensive, provide real-time information and have better accuracy and precision. Since there is less manual sample handling, these techniques tend to be less prone to contamination. Membrane extraction can be coupled with analytical instruments for continuous, on-line monitoring. Haloacetic acids (HAAs) are the major non-volatile disinfect byproducts generated during the chlorination of drinking water. Currently there is no continuous method for the determination of all the nine HAAs. Here an automated, online membrane extraction system will be developed for the continuous monitoring of HAAs.

There has been much interest in integrating different analytical functions onto a single platform referred to as total analytical system (TAS). Trace level analysis involves the extraction of the analytes from the matrix, concentration of the extract, and instrumental detection. Therefore an automated TAS that can perform on-line extraction, concentration, and detection is desirable. It can be implemented by interfacing continuous membrane extraction, pervaporation and on-line HPLC-UV detection.

In summary, the specific objectives of this research were:

I. Developments in micro-scale membrane modules:

- To develop hollow fiber membrane microextraction based on liquid-liquid membrane extraction and supported liquid membrane extraction for the determination of non-polar and polar compounds in water.
- To design and fabricate a miniaturized on-chip device for sample enrichment.

II. Developments in automated, on-line membrane systems:

- To develop a continuous membrane extraction system for on-line monitoring of trace level haloacetic acids in water.
- To develop a total analytical system by interfacing membrane extraction, pervaporation, and HPLC-UV detection for continuous, on-line monitoring of non-polar organics in water.

SECTION A:

MICRO-SCALE MEMBRANE EXTRACTION

CHAPTER 3

SOLVENT SYSTEMS FOR MICRO-SCALE MEMBRANE EXTRACTION

3.1 Implementation of Barrier Film in Hollow Fiber Microextraction

3.1.1 Introduction

The challenge in trace analysis lies in the separation of analytes from complex matrices and the achievement of low detection limits. Separation and preconcentration are usually necessary prior to analysis and are often accomplished by procedures, such as liquid-liquid extraction (LLE), solid phase extraction (SPE), and solid phase micro-extraction (SPME). Recently, there has been significant interest in microextraction techniques because of their simplicity and miniature scale.

An interesting microextraction approach has been referred to as liquid-phase microextraction (LPME). It is carried out by exposing a single droplet of an organic solvent, hung from the end of a micro syringe needle and into the aqueous sample [77-85]. Passive sampling of analytes into the organic droplet occurs via diffusion. Stirring can be utilized to enhance the mass transfer and extraction. This technique is inexpensive and uses small amounts of organic solvent. However, it is not rugged because the droplet may be lost during the extraction, especially at high stirring rate or during a long extraction [86]. Moreover, samples such as plasma may emulsify the organic solvent and affect the stability of the droplet [87].

Enclosing the organic solvent into a piece of hollow fiber membrane with an open sample end is a modification of LPME [88-89]. Compared to the single drop LPME, it is more robust and the loss of extract is reduced significantly. However, the solvent and the

extract can still be lost by dissolving into the sample matrix or be extruded due to vigorous stirring. Low reproducibility in extract volume results in variable enrichment factors and extraction efficiency. Moreover, as the solvent is open to the sample, which reduces selectivity. Emulsion formation is also possible during the direct contact between the aqueous sample and organic solvent.

Pedersen-Bjergaard et al. reported a hollow fiber based LPME approach, where the extractant was completely enclosed in a membrane and isolated from the aqueous sample [87, 90]. Two syringes were used for injecting the acceptor into the hollow fiber and withdrawing the extract. The membrane served as a selective barrier for the analytes to permeate in while preventing solvent outflow and extract loss. Thus higher stirring rate could be applied to enhance extraction.

Hollow fiber based LPME can be carried out by two approaches: supported liquid membrane microextraction (SLMME) and liquid-liquid membrane microextraction (LLMME). SLMME is a three-phase system in which the analytes are extracted from an aqueous sample into an aqueous acceptor via an organic extractant held in the pores of the membrane by capillary force. It has shown great promise for the determination of highly polar and ionizable compounds [87, 28-29]. LLMME is a two-phase system where the analytes are extracted from an aqueous sample to an organic acceptor. It is the equivalent of liquid-liquid extraction across a membrane and can be used in any application as long as the analytes can be enriched into an organic solvent [24, 61]. While the presence of membrane prevents direct interaction with the extractant, the later is still lost via diffusion through the membrane and by dissolving in the aqueous phase [61]. These are more pronounced for certain solvents that have high diffusion coefficient in the membrane

material and high solubility in water. Vigorous conditions (e.g. high stirring rate) and long extraction time that are needed for high enrichment only enhance the loss of the acceptor. The reduction/elimination of solvent loss is important for enhancing the performance of membrane based microextraction.

The objective of this study is to investigate solvent systems that can be implemented to reduce the loss of solvent (and the extract) enclosed in the membrane. Two approaches are investigated. The first involves the coating of the membrane fiber with a barrier film of a solvent that has lower miscibility/solubility with water. This layer serves as a diffusion barrier that prevents the outflow of the acceptor through membrane. Now LLMME becomes a three-phase system, where the analyte has to first partition into the barrier film before being extracted by the solvent. This is shown in Figure 1. The latter way may be simply select a solvent that is less miscible/soluble with water.

3.1.2 Experimental Section

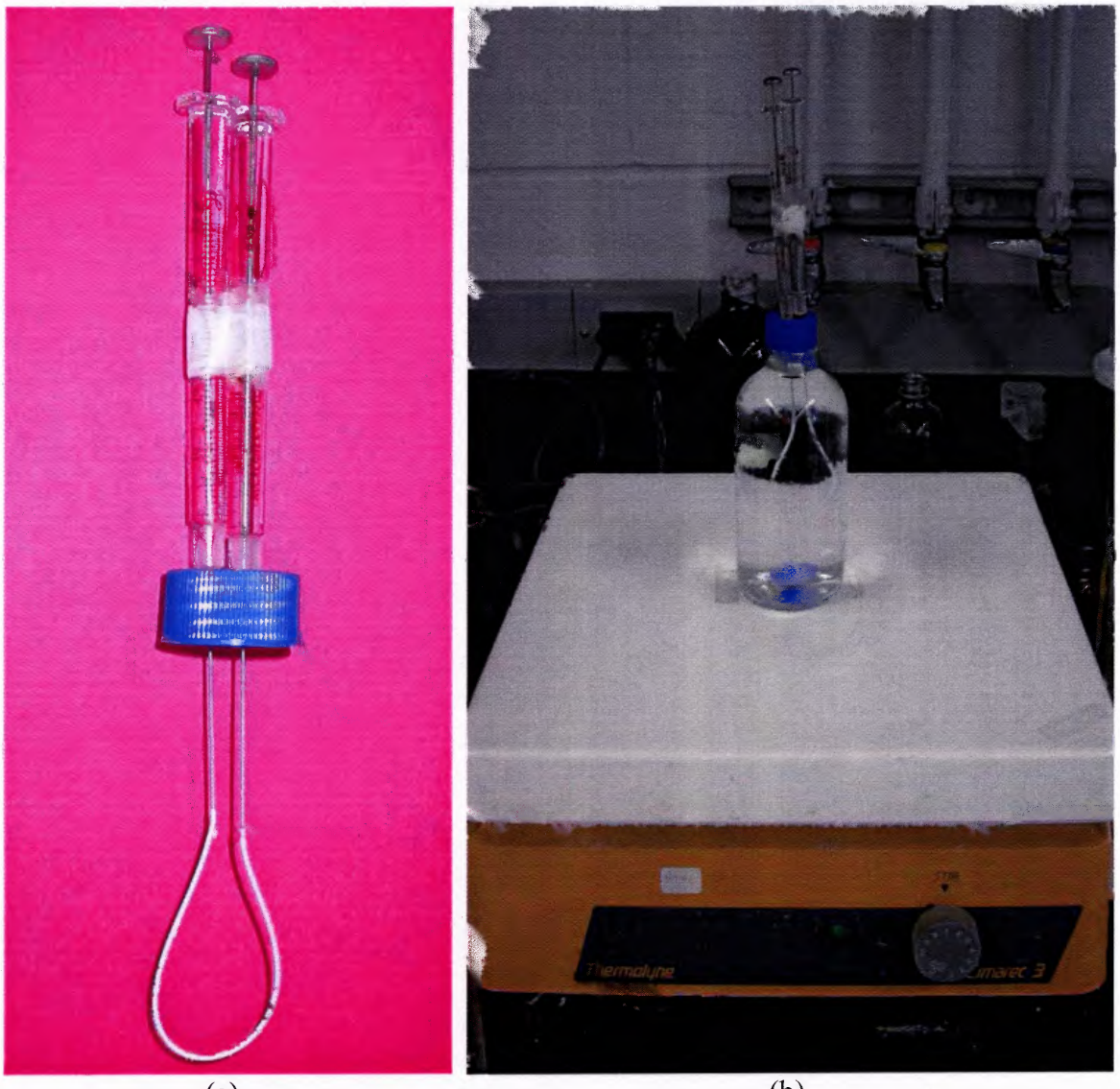
3.1.2.1 Reagents

Six PAHs (naphthalene, fluorene, acenaphthene, phenanthrene, anthracene and pyrene) purchased from Supelco (Supelco Park, PA, US) were used as the model compounds in this study. HPLC grade acetonitrile (Fisher Scientific, NJ, US) was used as the mobile phase in HPLC analysis and for preparing stock solution. All other chemicals used in this study were ACS reagent grade (Sigma Chemical Co., St. Louis, MO, US). Deionized (DI) water was obtained from a Millipore Gradient A10 water purification system (Millipore Co., Bedford, MA, US). The filter membrane of the system has an average pore size of 0.22 μm.

3.1.2.2 Apparatus and Procedures

Photographs of the hollow fiber based microextraction system is shown in Figure 3.1. It is quite simple and inexpensive. Two 50 μL HPLC manual injection syringes (Hamilton, Reno, NV, USA) with flat needles were used to hold the membrane in place (Figure 3.1a). The two syringes pierced through a septum cap and were fixed together, which made it easily screwed to the donor bottle. One syringe was used to inject the acceptor into the membrane, while the other was used to withdraw the extract. The liquid membrane was placed into a donor bottle that contained the water sample. A magnetic stir plate (Cimarec 3, Barnstead/Thermolyne, Dubuque, Iowa, USA) was used to speed up the extraction. The whole setup is shown in Figure 3.1b. The schematic diagram of such setup has been already described in Figure 1.6.

The hollow fiber membrane used in this study was Accurel PP Q3/2 (Membrana GmbH, Wuppertal, Germany). It had an I.D. of 600 μm and an O.D. of 1000 μm with an average pore size of 0.2 μm with 75% of the surface being porous. A 10.8 cm long membrane was soaked in the barrier fluid, and then attached to one of the syringes filled with about 50 μL of organic acceptor. The syringe plunger was then released; 30 μL of the acceptor remained in the membrane lumen while the rest went out through the other end of the membrane. The other syringe needle was attached immediately on the other end. The membrane assembly was then suspended into the sample bottle. Extraction was carried out with stirring at relatively high speed. After the extraction, the acceptor was withdrawn and transferred into a vial insert (Fisher Scientific, Springfield, NJ, USA) for HPLC analysis. A new membrane fiber was used for each extraction, thus analyte carry over was not a concern.



(a) (b)

Figure 3.1 Photographs of the hollow fiber microextraction.

3.1.2.3 Chromatographic Separation

The chromatographic separation was carried out by a Hewlett-Packard 1050 HPLC equipped with a Waters 486 Tunable Absorbance UV detector. The HPLC column was a 150 mm × 6.0 mm I.D. YMC ODS-A C_{18} column with 5 μm packing. The HPLC mobile phase was 65:35 (v/v) acetonitrile: DI water with a flow rate of 2.0 mL/min. The

wavelength was set at 254 nm, and the injection volume was 20 μL. Minichrom V 1.62 software (VG Data System) was used for data acquisition and analysis. The complete separation of the six PAHs was achieved within 12 minutes.

3.1.3 Results and Discussion

3.1.3.1 Mass Transfer

Figure 3.2 shows the mass transfer in hollow fiber based microextraction. The analytes (A) in the sample (donor) partition in barrier film of the membrane, and then permeate through the membrane, finally dissolve in the organic solvent (acceptor) in the fiber lumen: $A_{donor} \rightarrow A_{barrier\ fluid} \rightarrow A_{acceptor}$. The whole process is driven by the partition of the analytes between the aqueous sample, barrier fluid and the organic acceptor.

3.1.3.2 Barrier Film

Toluene has been used as the acceptor for the PAHs extraction [89]. However, it was found to be easily lost by diffusion through the membrane and the pores. The extract loss is often accompanied by the loss of analyte leading to a low enrichment factor. The recovery of a variable amount of extract leads to increased uncertainty in quantitative analysis. When extraction was carried out for a long period, or the stirring rate was high all the toluene was lost. Consequently, the prevention of solvent loss is important. This can be done by choosing a solvent with low water immiscibility or coating the membrane with a thin barrier film of an immiscible solvent, thus preventing the outflow of extract. In some cases, a completely water-immiscible solvent may lead to very low extraction efficiency. So, the

barrier film should allow the diffusion of the analyte through it, to obtain the high extraction efficiency and enrichment factor.

The barrier film was implemented by soaking the membrane in the solvent prior to extraction. Four different organic solvents, dihexyl ether, n-undecane, 1-octanol and n-decane, were tested as the barrier film. Extraction without any barrier film was also performed for comparison. The donor was 250 mL, 0.1 μg/mL of the six PAHs in DI water. The acceptor was 30 μL toluene, the stirring level was set at 5 (arbitrary units) and extraction time was 10 minutes. The EF, the percentage of the extractant lost during the extraction is presented in Table 3.1. N-decane provided the best results, all the six PAHs were extracted and the EF was large. When no barrier film was used, the high water solubility of toluene resulted in high acceptor loss. Figure 3.3 (a) shows the chromatograms from the extraction with only toluene and with n-decane as the barrier film. Only four of the six PAHs were detected without the barrier film. When n-decane was applied, all the six of the PAHs were detected and the extraction efficiency and EF were higher.

Acceptor loss as a function of extraction time is shown in Figure 3.4. The donor (volume and composition) and the stirring rate were same as for the data in Table 3.1. Significant acceptor loss was observed without the barrier film, it increased with the increase in extraction time and was completely lost after 20 minutes. When either n-decane or dihexyl ether was applied as the barrier film, the acceptor loss was much lower although it increased with the extraction time. The extractant loss decreased further when n-decane was used as both the barrier film and the acceptor. Solvent loss was less than 3.3% for a 30-minute extraction.

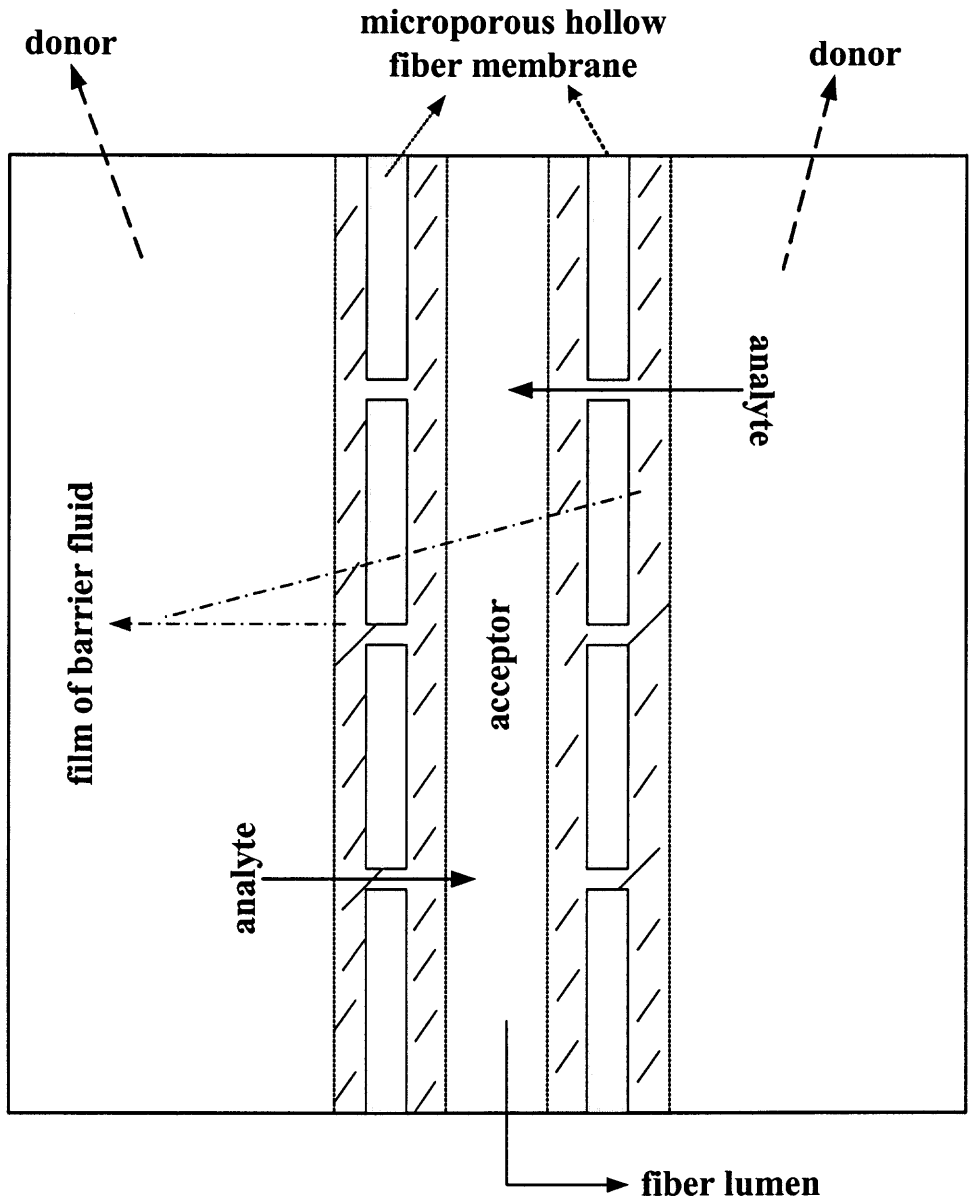
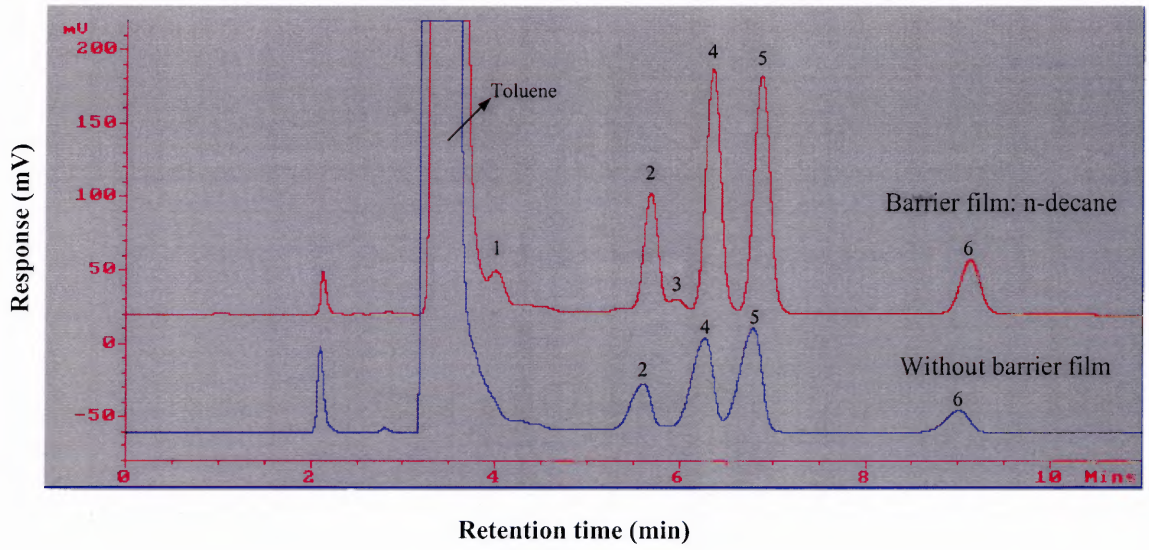


Figure 3.2 Mass transfer through barrier film in hollow fiber based microextraction.

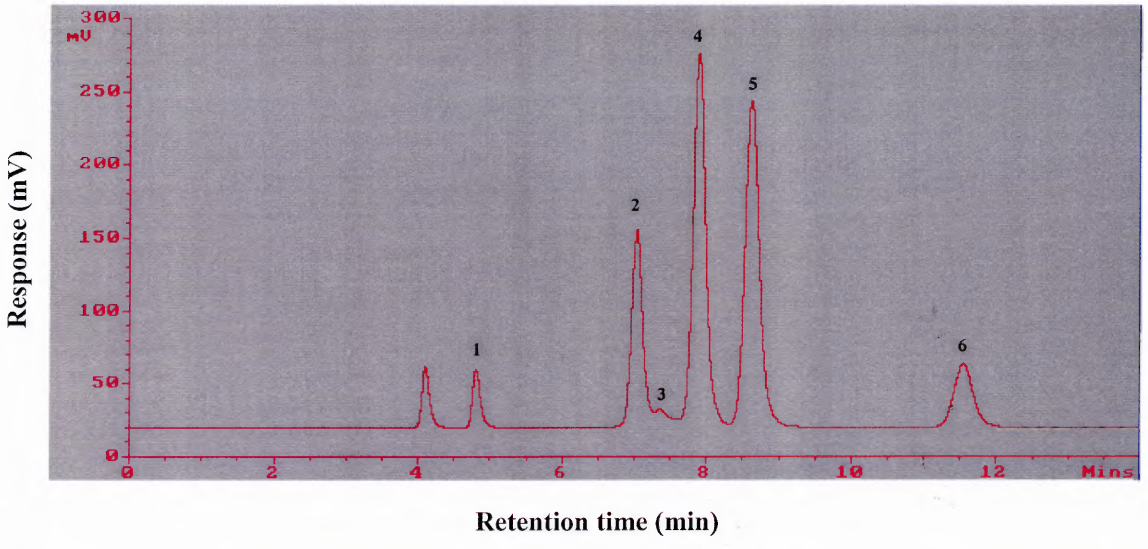
Table 3.1 The Effect of Barrier Film on Enrichment Factor and Acceptor Loss

Barrier film	Water solubility (20 °C)	Boiling point (°C)	Enrichment factor						Acceptor loss (%)
			naphthalene	fluorene	acenaphthene	phenanthrene	anthracene	pyrene	
Without barrier film	0.515 g/L[a]	110.6[b]	nd[c]	67.5	nd	68.0	54.6	76.6	73.3
dihexyl ether	insoluble	226.6	nd	164.1	nd	150.3	129.3	175.4	33.3
1-octanol	insoluble	194.5	nd	117.9	nd	117.9	77.3	117.6	26.7
n-undecane	insoluble	196.0	248.7	120.6	95.6	121.2	87.2	125.9	10
n-decane	0.009 ppm	174.0	485.6	146.2	185.7	144.0	94.6	158.2	10

[a] toluene's water solubility; [b] toluene's boiling point; [c] Not detected.



(a)



(b)

Figure 3.3 (a) Chromatograms from LLMME with and without n-decane as the barrier film, toluene was used as the acceptor. (b) Chromatogram from LLMME with n-decane as the acceptor and the barrier film. The peaks are: 1. naphthalene, 2. fluorene, 3. acenaphthene, 4. phenanthrene, 5. anthracene, and 6. pyrene.

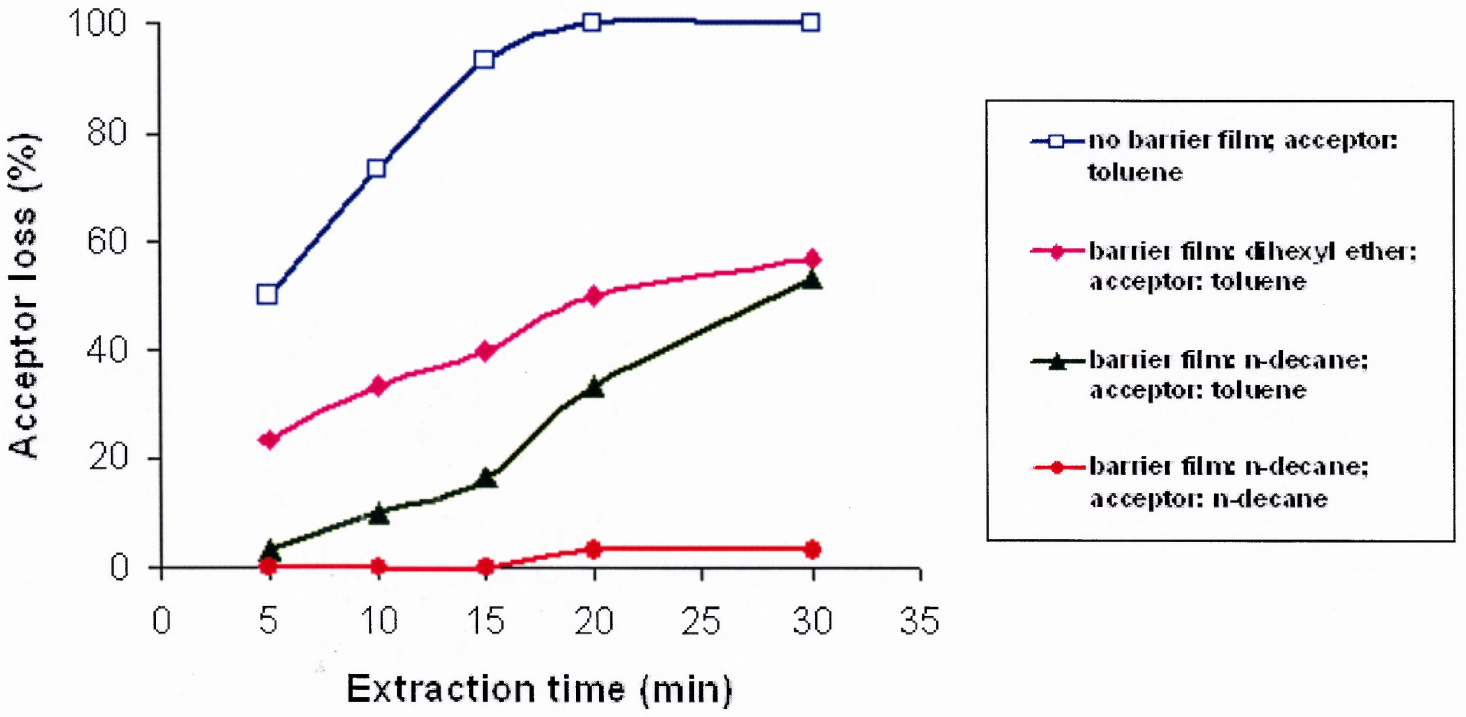


Figure 3.4 Acceptor loss as a function of extraction time. The donor was 250 mL, 0.1 μg/mL of the six PAHs in DI water. The acceptor was 30 μL toluene, and the stirring level was set at 5 (arbitrary units).

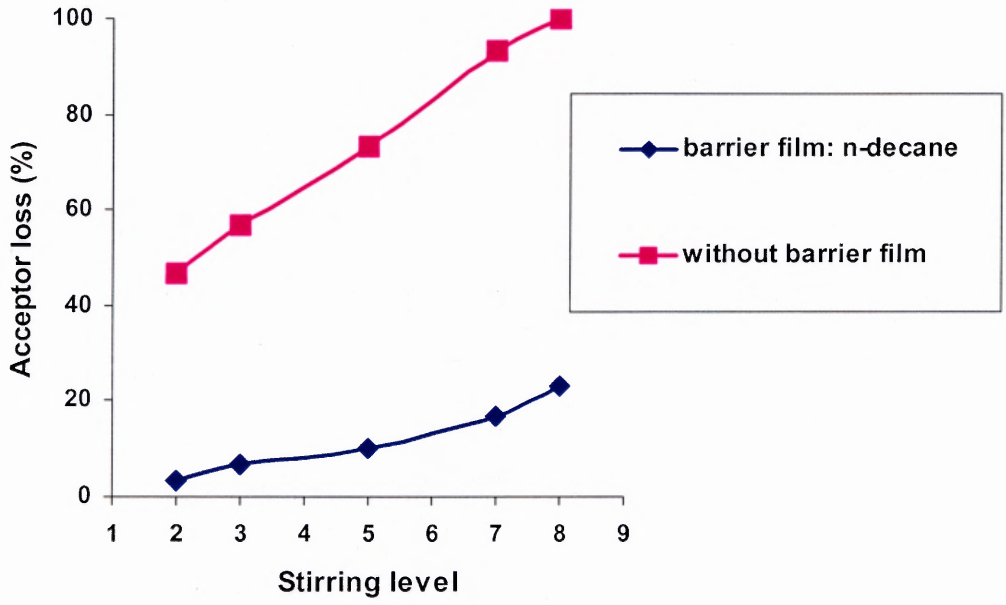
3.1.3.3 Stirring Effect

Stirring is known to enhance extraction because it increases the mass transfer [29]. However, higher stirring rate also increases solvent loss. Figure 3.5 shows the effect of stirring rate on acceptor loss (a) and on the peak areas of the PAHs in the extract (b). The donor was 0.1 μg/mL of the six PAHs in 250 mL DI water, the acceptor was 30 μL toluene and n-decane was used as the barrier film. The extraction time was 10 minutes. Stirrer setting was varied from 2 to 8 (arbitrary units). At higher stirring rates the permeation of toluene to the water phase increased, resulting in higher acceptor loss. With an n-decane barrier film, the solvent loss was less than 20% even when stirring rate was increased to 8, whereas the toluene was completely lost when there was no barrier protection. As expected, the peak area increased significantly when the stirring level increased from 2 to 7 (Figure 3.5b). However, it decreased when the stirring level was increased beyond 8, because the extract began to be lost under the high level of agitation. Thus level 7 was selected as the optimum stirring rate.

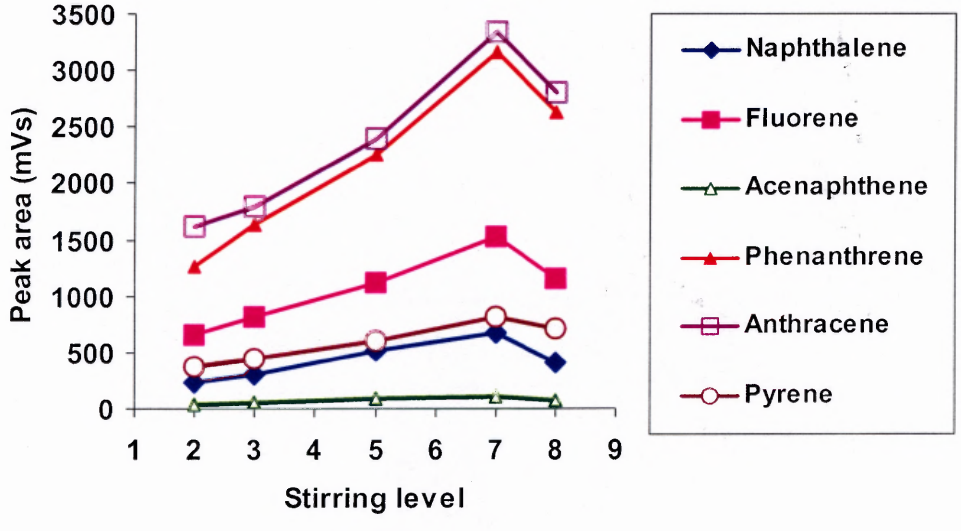
3.1.3.4 Acceptor Selection

Since the barrier film can be used to prevent acceptor loss, a variety of solvents can be used as the acceptor. The data presented so far used toluene as the extractant. Dihexyl ether and n-decane were tested as the acceptor and also the barrier film. Both solvents worked well for the extraction of the six PAHs and the acceptor loss was negligible. The baseline of the chromatogram obtained using n-decane was better than that with dihexyl ether, and it was studied in more details. A chromatogram after a 10-minute extraction of a sample

containing 0.1 μg/mL of the six PAHs using n-decane as the acceptor and barrier film is shown in Figure 3.3 (b). The peaks were sharper and no solvent interference was observed.



(a)



(b)

Figure 3.5 Effect of stirring level on (a) acceptor loss, and (b) on peak area of PAHs (toluene as the acceptor and n-decane as the barrier film).

Figure 3.6 compares the peak areas obtained with n-decane as the barrier film as well as the acceptor (n-decane/n-decane), to that with n-decane as the barrier film along with toluene as the acceptor (n-decane /toluene). Naphthalene had a higher extraction efficiency using n-decane/toluene combination, while the rest of the five PAHs were extracted better with the n-decane/n-decane system.

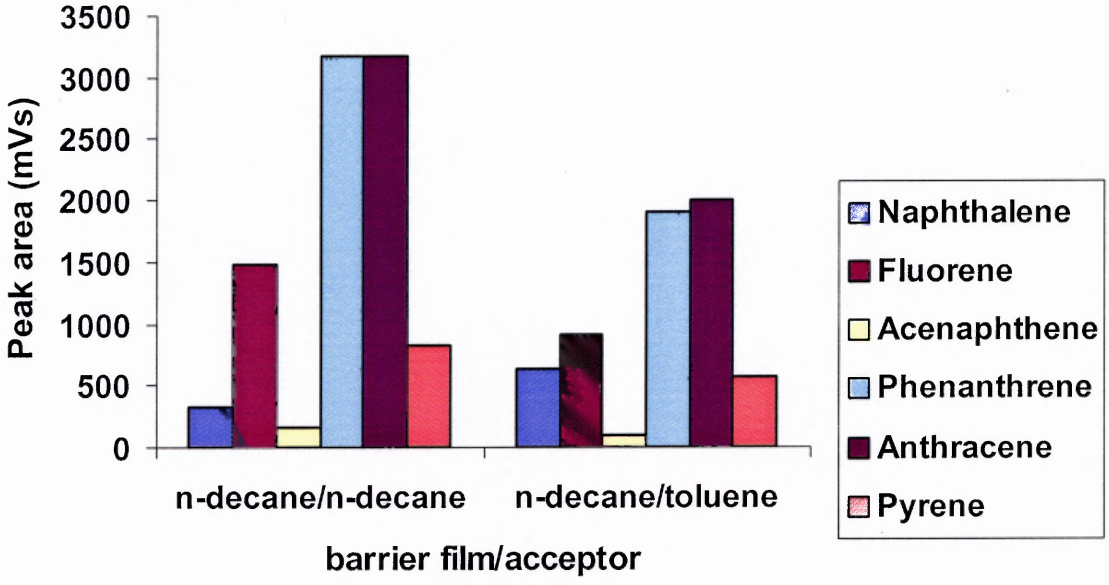


Figure 3.6 Peak area of PAHs using different acceptor (barrier film: n-decane).

3.1.3.5 Extraction Time

Different extraction times were investigated and the results are shown in Figure 3.7. The sample was 250 mL of 0.1 μg/mL six PAHs in DI water and the acceptor was 30 μL n-decane, which also served as the barrier film. Now that we had a solvent system that had significantly lower acceptor loss, high stirring speed and long extraction times were possible. The stirring level was set at 7 and the extraction time as long as 60 minutes were

studied. As can see from Figure 3.7, an increase of extraction time resulted in higher enrichment, and consequently higher sensitivity.

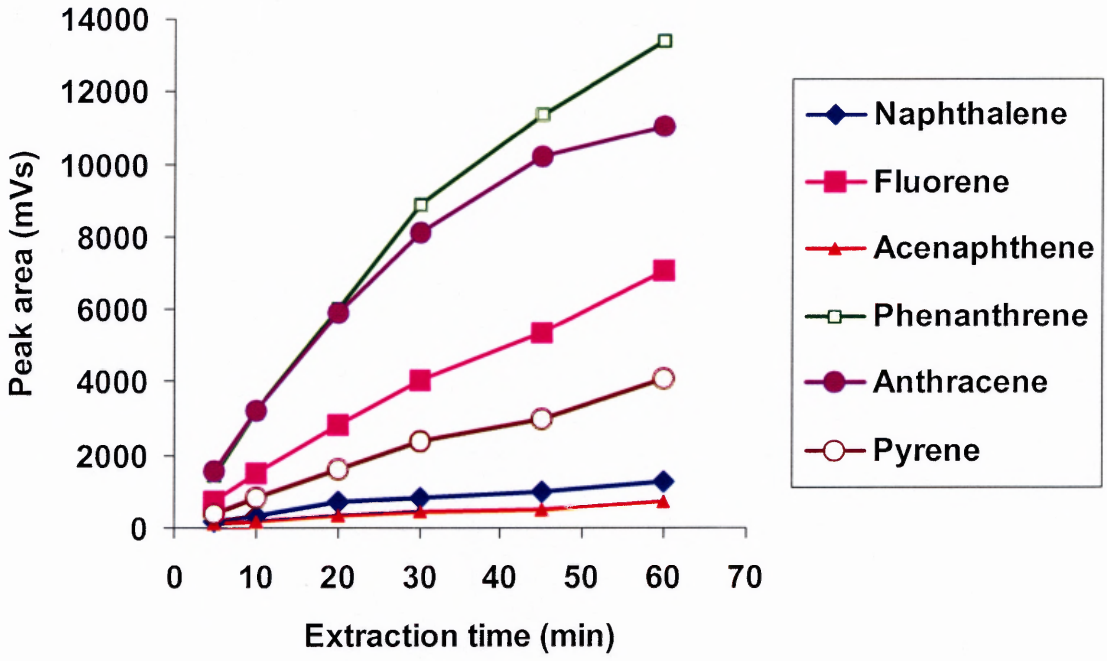


Figure 3.7 Effect of extraction time on peak area.

3.1.3.6 Analytical Performance

Based on the above experiments, the optimum conditions appeared to be use of n-decane as the barrier film and acceptor, with a 60-minute extraction at a stirring level of 7. DI water samples spiked with different PAHs concentrations ranging between 0.01 to 100 μg/L were extracted under these conditions. The linear dynamic ranges, regression coefficients (R^2), relative standard deviations (RSDs), method detection limits (MDLs), EF and EE% are listed in Table 3.2.

Table 3.2 Analytical Performance

	Water solubility (mg/L) at 25°C	Log K_{ow} [30]	Linear Dynamic Range (µg/L)	Regression Coefficient (R^2)	RSD (%) n=7	MDL[a] (ng/L)	Enrichment factor	Extraction efficiency (%)	Drinking water fountain (µg/L)	Bottle water (µg/L)
naphthalene	32	3.36	0.50-100	0.9997	7.65	120	494	5.93	nd[b]	nd
fluorene	1.9	4.18	0.10-100	0.9985	1.60	5	2037	24.44	0.083	0.015
acenaphthene	3.4	4.32	1.00-100	0.9933	2.75	86	1329	15.95	4.043	nd
phenanthrene	1.0-1.3	4.46	0.05-100	0.9870	3.24	5	1317	15.80	0.078	nd
anthracene	0.05-0.07	4.45	0.01-40	0.9997	6.51	2	4555	54.66	0.341	nd
pyrene	0.14	5.30	0.10-100	0.9891	3.55	11	2531	30.37	0.154	nd

* MDLs were obtained with a water sample containing 0.50 µg/L naphthalene, 0.10 µg/L fluorene and pyrene, 1 µg/L acenaphthene, 0.05 µg/L phenanthrene and 0.01 µg/L anthracene.

** Not detected.

The detection limits determined by the standard EPA procedure [113] were at the ng/L (ppt) level. The response was linear with a fairly large linear dynamic range, the regression coefficients (R^2) were between 0.9870 and 0.9997 indicating high degree of linearity in a large dynamic range. RSDs based on seven replicates measured over three different days, were between 1.60 and 7.65 %. The EF ranged from 494 to 4555, which was significantly higher than what has been reported previously, using toluene as the acceptor (between 46-167) [89]. EE% was between 6 and 55%.

3.1.3.7 Analysis of Real World Drinking Water

The optimized method was applied to the determination of PAHs in real world drinking water samples. Two drinking water samples were analyzed. The first was from a drinking water fountain located at New Jersey Institute of Technology (Newark, NJ, USA), and the other was bottled natural spring water (Acadia) bought from STOP & SHOP grocery store (Linderhurst, NJ, USA). The results were also listed in Table 3.2. Five of the six PAHs studied were detected in the water from the water fountain, with concentrations ranging between 0.078 and 4.043 μg/L. For the bottled water, only fluorene was found, at a concentration of 0.015 μg/L. The results showed that this technique could be employed to monitor trace level water samples.

3.1.4 Conclusion

A barrier film of a second solvent was coated on a hollow fiber membrane to stabilize the acceptor solvent within. The coating was implemented easily by dipping the membrane in the barrier solvent. Toluene, which has been typically used as an extractant for PAH

analysis could be stabilized by a barrier film of decane (or dihexyl ether), and its loss during extraction could be significantly reduced. Extraction time as long as 60 min was possible with a barrier film, and high stirring rate which would not be possible with pure toluene were now possible. The overall effect was significantly enhanced performance in terms of higher enrichment factor, lower detections limits and higher reproducibility. Running a large number of samples simultaneously can increase the sample throughput. The extraction device is simple, inexpensive and uses little organic solvent. It can also be used with GC or GC/MS detection to achieve even lower MDLs.

3.2 Supported Liquid Membrane Microextraction of Haloacetic Acids

3.2.1 Haloacetic Acids

Chlorination is one of the most common methods for disinfecting drinking water [91]. Chlorine reacts with naturally occurring acids to form halogenated disinfection byproducts (DBPs), some of which are known carcinogens. Trihalomethanes (THMs) and haloacetic acids (HAAs) are the major volatile and nonvolatile DBPs [92]. The names, abbreviations, pKa values and octanol-water partition coefficients (log P) of the nine HAAs are included in Table 3.3. USEPA has classified DCAA as a probable human carcinogen and TCAA as a possible human carcinogen. Furthermore, decarboxylation of HAAs results in the formation of THMs, which are also carcinogens. USEPA has regulated the total Maximum Contaminant Level (MCL) in drinking water of the five HAAs: MCAA, MBAA, DCAA, BCAA, and DBAA to be less than 60 μg/L [93].

Currently there are several USEPA approved methods for the determination of HAAs in drinking water (EPA method 552, 552.1, 552.2) [94-96]. All these methods

involve cumbersome liquid-liquid extraction or ion exchange and derivatization, followed by GC-ECD detection. They have several limitations, for example, EPA method 552.1 uses ion exchange and derivatization followed by GC-ECD detection. It consumes less solvent, however the interference from anions increases the detection limits [95], and it can only determine six of the HAAs. Typical analysis time for the above methods varies between three to four hours. Alternative methods that do not need the derivatization prior to analysis have been developed based on, liquid chromatography (LC) [97-98], ion chromatography (IC) [99-101], capillary electrophoresis (CE) [102], and electrospray ionization high- field asymmetric waveform ion mobility spectrometry and mass spectrometry (ESI-FAIMS-MS) [103]. ESI-FAIMS-MS provides low detection limit, has excellent sensitivity and selectivity, but the high cost limits its availability. The detection limits of the LC, IC and CE methods are higher than the GC methods. Many of the alternative methods have been used for five or six HAAs, and only a few are applicable for all the nine HAAs.

Table 3.3 Names, Abbreviation and Properties of Nine HAAs

Names	Abbreviation	LogP*	pKa*
Monochloroacetic acid	MCAA	0.22	2.87
Dichloroacetic acid	DCAA	0.92	1.26
Monobromoacetic acid	MBAA	0.41	2.89
Bromochloroacetic acid	BCAA	1.14	1.39
Dibromoacetic acid	DBAA	1.69	1.47
Trichloroacetic acid	TCAA	1.33	0.51
Bromodichloroacetic acid	BDCAA	2.31	1.09
Chlorodibromoacetic acid	CDBAA	2.91	1.09
Tribromoacetic acid	TBAA	3.46	3.13

*logP and pKa values are from Ref. [29].

3.2.2 Supported Liquid Membrane Microextraction

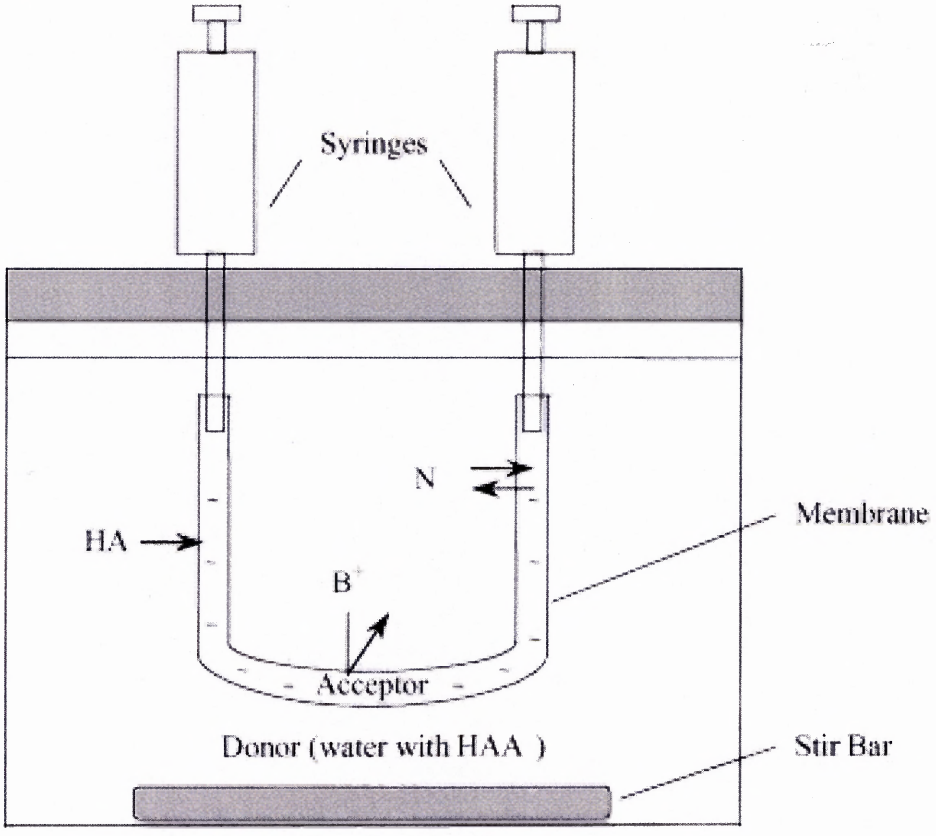


Figure 3.8 Schematic diagram of SLMME setup [29].

SLME has been used for the extraction of charged or ionized analytes [14]. Supported liquid membrane micro-extraction (SLMME) followed by ion-pair chromatography was developed for the analysis of nine HAAs in water [35]. The SLMME system is shown in Figure 3.8. The supported liquid membrane (SLM) used is made by impregnating a short segment (few cm) of microporous hollow fiber with a membrane liquid (organic solvent) for a few seconds, and the membrane pores are automatically filled with the liquid. The excess liquid is replaced by injecting micro liters of basic acceptor (NaOH solution) into

the membrane lumen. Two syringes are used to hold the membrane in place. One is used to inject (or withdraw) the acceptor into (or from) the membrane. The other serves as a support. The liquid membrane is placed in the acidified water sample (the donor). A magnetic stirrer is used to agitate the sample during extraction. After extraction, the acceptor solution is drawn into the syringe, and analyzed by ion-pair chromatography. This technique provides high enrichment in a relatively short analysis time.

When 0.05 M tris buffer was used as the acceptor instead of the NaOH solution, much less interference was observed at trace levels and the linear dynamic range was broader. Second, instead of IC with flow programming and mobile phase with high salt concentration (400 mM ammonium sulfate), an isocratic HPLC method was developed. The mobile phase was 15 mM KH_2PO_4/H_3PO_4 buffer at pH 2.24:acetonitrile (95:5, v/v) at a flow rate of 1.0 mL/min. This new method was found that the humic acid interference peak was separated well from the first analyte peak (MCAA). Moreover this HPLC method could avoid the frequent column washing caused by the salt crystallization in the column. The chromatograms before and after modifications are shown in Figure 3.9 and 3.10, respectively. With the optimized method, detection limits decreased about 1 to 8 times. The comparison of the linear dynamic ranges and method detection limits before and after optimization is showed in Table 3.4.

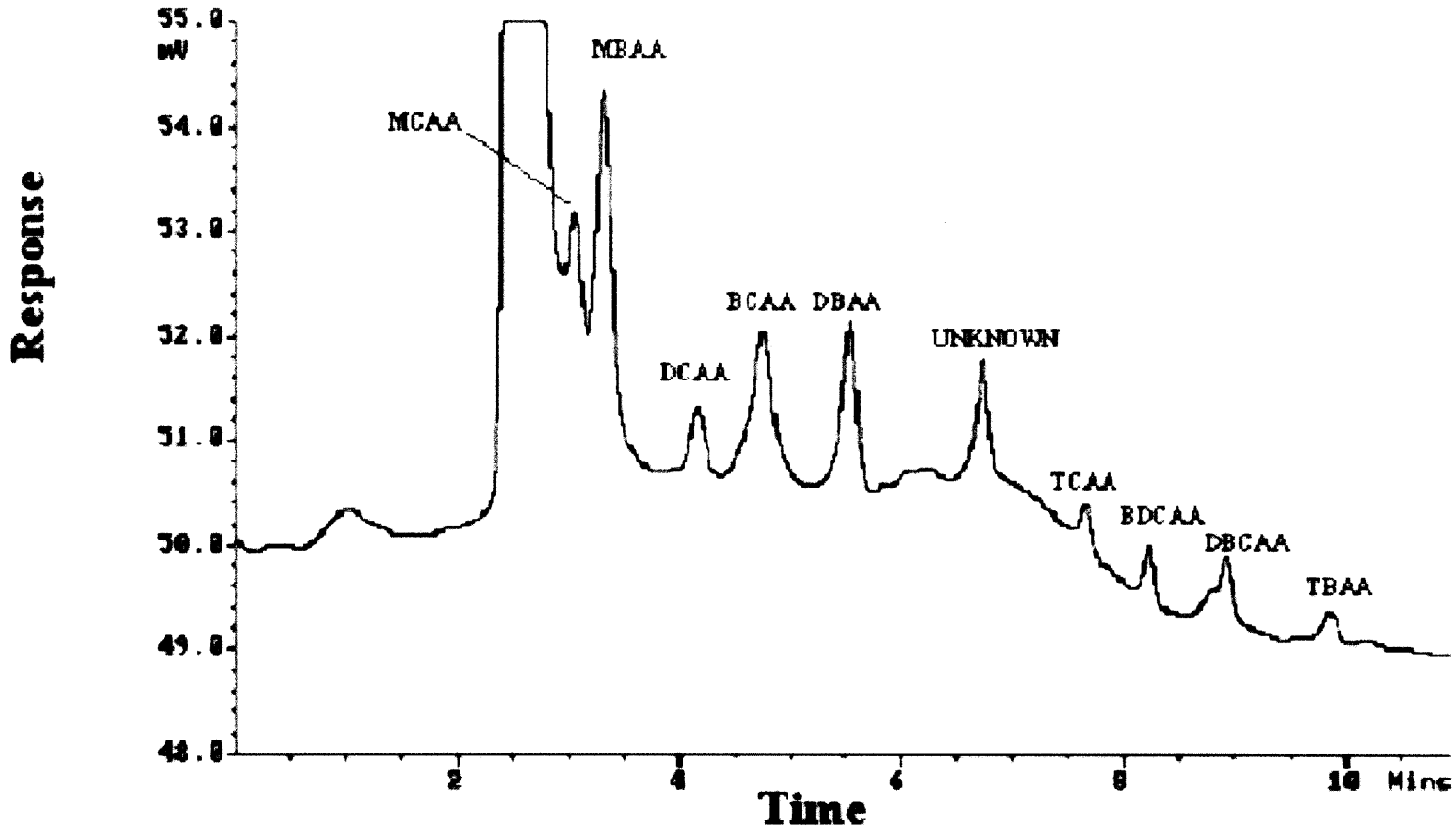


Figure 3.9 Chromatogram of nine HAAs in reagent water after SLMME (before method modification). The concentrations were: MCAA at 40 ppb, MBAA at 10 ppb, DCAA at 0.8 ppb, and the other six HAAs at 0.4 ppb.

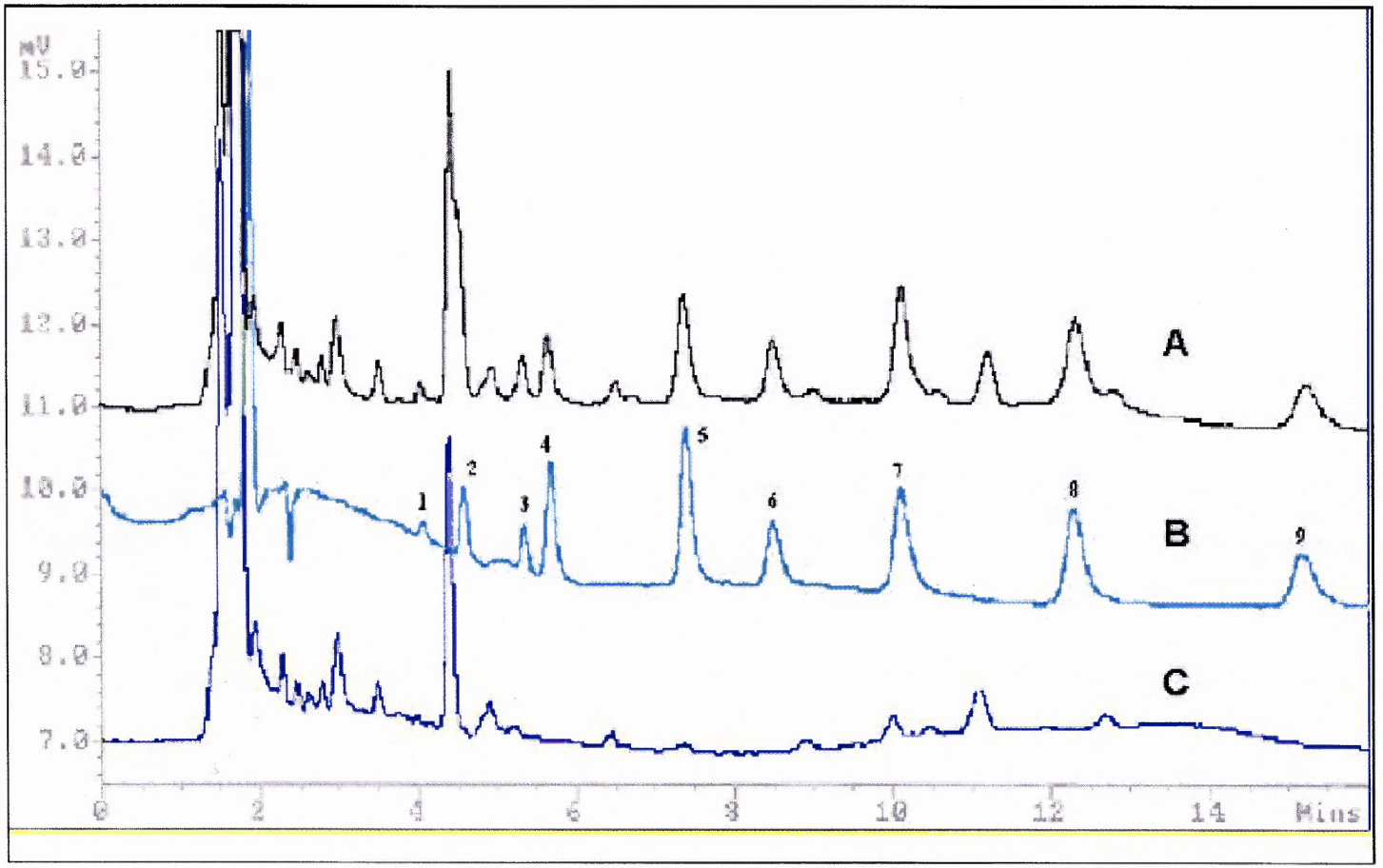


Figure 3.10 Overlay of chromatograms (after modification). A: SLMME of spiked water sample containing 8.4 ppb MCAA, 2.4 ppb DCAA, 4.4ppb MBAA, and the other 6 HAAs at 0.4 ppb each; B: direct injection of a standard solution containing nine HAAs at 1ppm each; and C: SLMME of a blank (reagent water). The numbered peaks in the chromatograms are: 1. MCAA, 2. DCAA, 3. MBAA, 4. BCAA, 5. DBAA, 6. TCAA, 7. BDCAA, 8. CDBAA, and 9. TBAA.

Table 3.4 Comparison of LDR and MDL Before and After Modification

HAAs	LDR (μg/L)		MDL (μg/L)	
	Before	After	Before	After
MCAA	20-160	8.4-160	7.69	2.69
DCAA	10-80	2.4-160	2.00	0.25
MBAA	0.8-20	4.4-160	0.21	0.23
BCAA	0.4-20	0.4-160	0.09	0.04
DBAA	0.4-20	0.4-160	0.10	0.06
TCAA	0.4-20	0.4-160	0.05	0.05
BDCAA	0.4-20	0.4-160	0.12	0.02
CDBAA	0.4-20	0.4-160	0.12	0.02
TBAA	0.4-20	0.4-160	0.08	0.03

SLMME followed by HPLC-UV analysis was developed for the extraction, concentration, and determination of nine HAAs in water. It was simple, sensitive, fast, used only a few microliters of organic solvent per sample, and did not require any derivatization. Enrichment factors in the range of 300- 3000 were obtained in a 60-minute extraction. The extract was directly analyzed by HPLC within 15 minutes. A large number of samples can be extracted simultaneously to increase the sample throughput. This method showed high precision, and the detection limits were lower than or comparable to those by the standard EPA methods. It is possible to use a new membrane for each extraction, so that the extraction is free of memory effects, and the membrane life is not a concern. This approach offers an attractive alternative to the current US EPA standard methods for HAA analysis, which require complex sample preparation and derivatization prior to GC-ECD detection.

CHAPTER 4

MICROFLUIDIC SUPPORTED LIQUID MEMBRANE EXTRACTION

4.1 Introduction

There has been much interest in the miniaturization of analytical instrumentation because it offers advantages, such as, inexpensive mass production and low reagent consumption. The possibility of parallel processing on a single substrate is also attractive, as it increases analytical throughput. A typical lab-on-a-chip application would involve the integration of multiple sample preparation and analytical functionalities, such as, mixing, reactions, extraction, separation and detection [104]. Microfluidic devices, which have one or more micron scale (less than 1000 μm) channels have been used in such applications; an advantage of such devices is their large surface to volume ratios.

Membrane extraction has been used in a variety of analytical applications in recent years [12-75, 105-106]. Typical membrane extraction involves the flow of sample on one side (referred to as the donor) of the membrane, while an extractant (gas or liquid) flows on the other side (referred to as the acceptor), and the analytes selectively permeate across the membrane. Since the donor and the acceptor can flow continuously, this approach offers the unique advantage of selective extraction on a continuous basis. Membranes have been interfaced with GC, HPLC, MS, CE, and other analytical instruments for continuous on-line monitoring [19-25].

Supported liquid membrane (SLM) extraction is an excellent technique for selective removal of ionizable and polar compounds from aqueous samples [107]. It is a

three-phase system, where the analytes are extracted from a water sample into an aqueous acceptor through an organic extractant held in the pores of the membrane by capillary action. The analytes on one side of the membrane are maintained at a pH where they are uncharged, and can be extracted into the organic phase in the membrane pores. On the other side of the membrane is a solution at a different pH, into which the analytes are back-extracted in their charged forms [37]. Typical SLM extraction modules have been made using flat membranes in a flow cell [18-20], or using multiple parallel hollow fibers in a shell-and-tube format [15-16, 24-27, 42, 61, 105-106, 108-112].

The ionic nature of environmental acids makes them a suitable candidate for the analysis by SLM. Haloacetic acids (HAAs) are an important regulated group of the disinfection by-products formed during the chlorination of drinking water [93]. Current USEPA approved methods for HAAs analysis involve cumbersome liquid-liquid extraction and derivatization followed by GC-ECD detection [94-96]. These methods consume significant amounts of reagents, and the overall analysis time is approximately four hours per sample. In a previous study, we reported the extraction of HAAs by supported liquid membrane micro-extraction [29]. Analysis of this type would benefit significantly from the development of miniature devices that can reduce solvent consumption and the extraction time.

The objective of this study is to develop a microfluidic extraction system for SLM. A device such as this can find other lab-on-a-chip applications as well. Although it is tested with HAAs, it can be used for any ionizable analyte including metals.

4.2 Experimental Section

4.2.1 Reagents

Nine standard solutions were purchased from Supelco (Supelco Park, PA, US), each containing an individual HAA. Acetonitrile used in HPLC analysis was HPLC grade (Fisher Scientific, NJ, USA). All other chemicals used in this study were ACS reagent grade (Sigma Chemical Co., St. Louis, MO, US). Deionized water was obtained from a Milli-Q® water purification system (Millipore Co., Bedford, MA, US).

4.2.2 Experimental System and Microfluidic SLM Modules

The schematic diagram and a photograph of the microfluidic SLM is shown in Figure 4.1 and 4.2, respectively. It includes an extraction module and two micro syringe pumps (Cole Parmer 74900) for liquid delivery. The first pump was used to deliver the water sample, and the other for the acceptor solution. The HAAs were extracted through the membrane liquid, and enriched into the acceptor. The extract was collected into a vial insert and then injected into the HPLC directly without any derivatization. Alternately, the extract could be also collected in the sample loop of a HPLC injector for direct on-line analysis.

The components of the microfluidic SLM module are shown in Figure 4.3 a, b and c. The acceptor and the donor flowed in the micro-channels fabricated on two separate polycarbonate wafers. The wafers were sandwiched with the membrane in between to form the SLM module (Figure 4.3a). The rectangular channels were micro-machined using a vertical milling machine (Bridgeport Machine Company, Bridgeport, CT, USA). The machining was carried out at 2750 rpm at the approximate rate of 1/4" per minute. A serpentine shape allowed long channels to be fabricated in a small area. Alternate

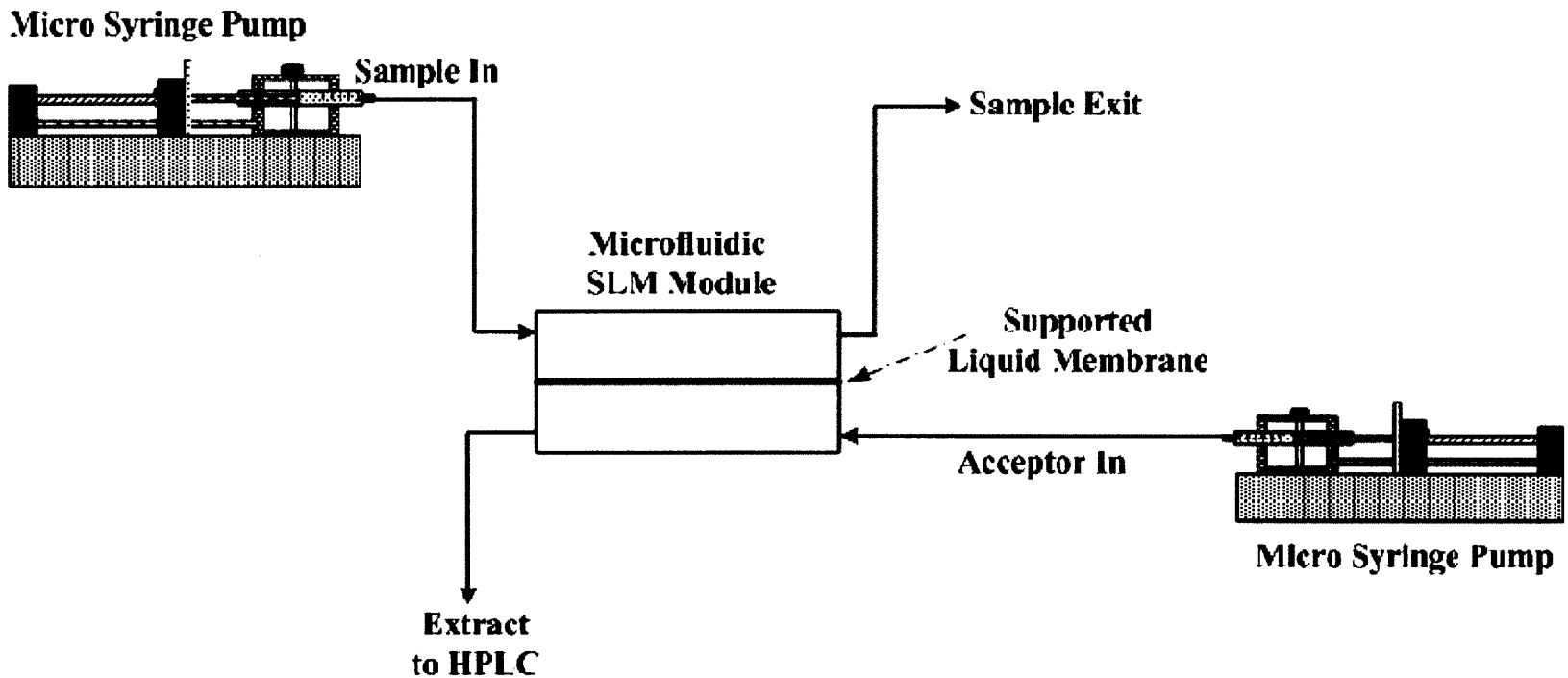


Figure 4.1 Schematic diagram of microfluidic SLME.

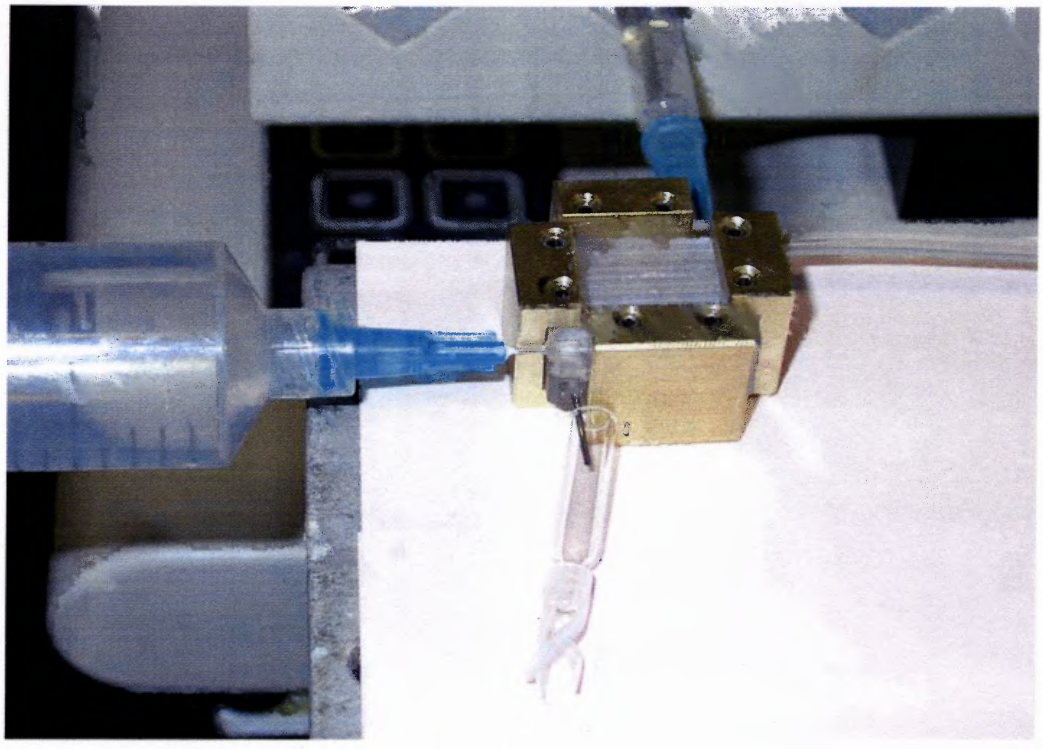


Figure 4.2 Photograph of microfluidic SLME (Module C).

fabrication techniquse such as lithography, or laser ablation could also be used to produce this type of micro-channels. The channels for the flow of the two phases mirrored each other so that they could be face to face. Steel capillaries with a 250 μm I.D. were glued into the channels to serve as the inlets and outlets. Three microfluidic SLM modules (Module A, B and C) with different dimensions were designed and fabricated, Module A being the largest and C the smallest. The details of these SLM modules are listed in Table 4.1. The width and depth were between 100 to 1000 μm. The width of the donor and the acceptor channels were always the same, but the depth varied as shown in Figure 4.3b. The ratio of the donor and the acceptor volume was close to 8 for modules B and C. A size comparison of the micro channels and a U.S. quarter is shown in Figure 4.3c.

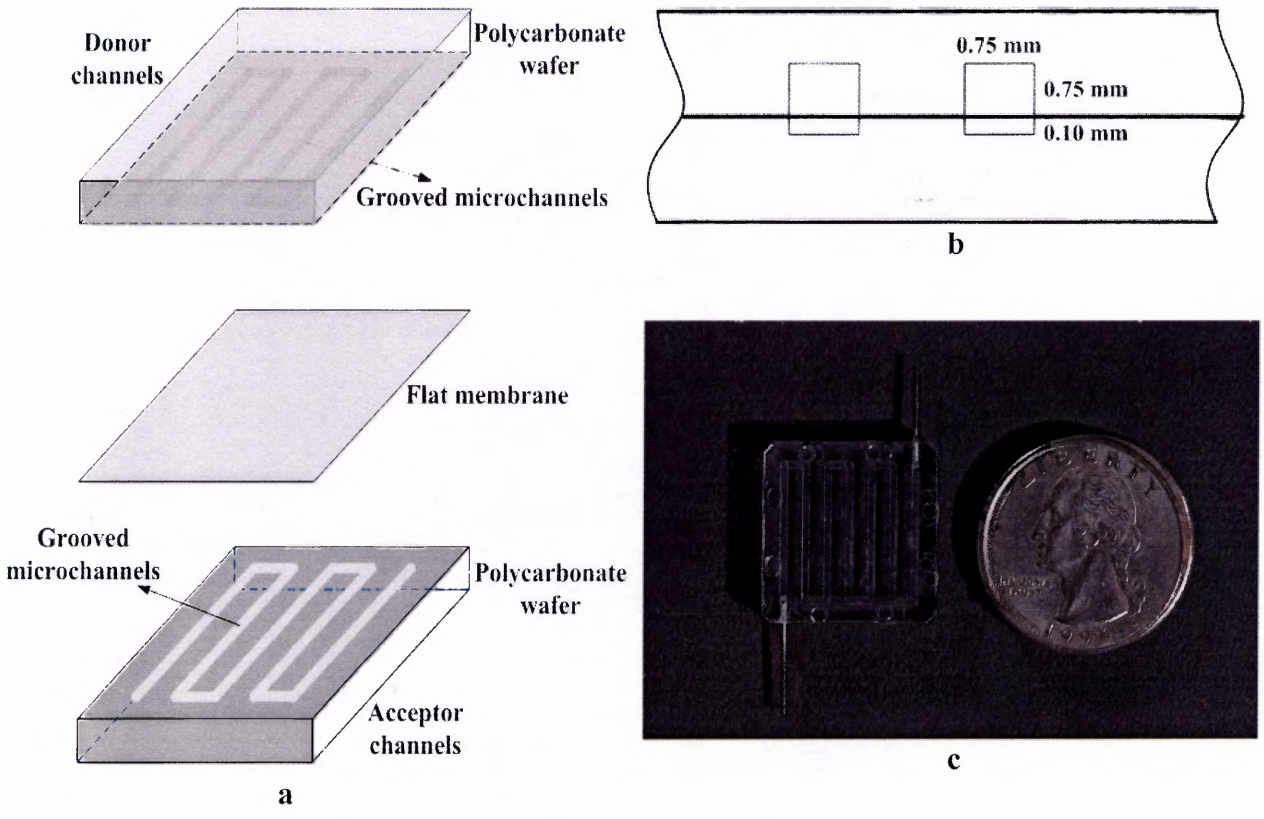


Figure 4.3 Details of the microfluidic SLM modules:
a. Microfluidic SLM module showing the acceptor and the donor channels with the membrane in between.
b. Cross-sectional view of the two channels (Module C)
c. Photograph of the micro channels (Module C), a U.S. quarter is presented for size comparison.

Table 4.1 Dimensions of the Three Microfluidic SLM Modules

	Module A	Module B	Module C
Length of microfluidic channel (cm)	22.8	40.2	11.1
Width of micro channel (μm)	1000	750	750
Surface area of micro channels (cm^2)	2.28	3.02	0.83
Depth of donor channel (μm)	1000	750	750
Depth of acceptor channel (μm)	1000	100	100
Volume of donor channel (μL)	228	226	62
Volume of acceptor channel (μL)	228	30	8
Membrane surface area (cm^2)	9	6	2.25
Maximum EF	10	65	54
Maximum EE (%)	16	34	50

The flat sheet membrane used in this study was Celgard® 2400 microporous polypropylene membrane (Hoechst Celanese, Charlotte, NC, US). It had an average thickness of 25 micron, the pore size was 0.05 μm, and 38% of the surface was porous.

4.2.3 Procedures

The supported liquid membrane was made by soaking a small piece of membrane in 5% trioctylphosphine oxide (TOPO) in di-hexyl ether (DHE) for 10 minutes. A few microliters of membrane liquid was consumed for each soakage. The supported liquid membrane was then fixed between the polycarbonate blocks, forming the two channels (one serving as the donor and the other as the acceptor). Screws or clamps were used to make a leak-free seal in the case of reusable devices. Epoxy could also be used to bond the two wafers, but these devices could not be reused, as the membrane could not be replaced.

The sample containing the HAAs (donor), which was acidified by concentrated sulfuric acid to pH –0.3, flowed on one side of the membrane; while the acceptor, 0.05 M tris buffer (pH 10) was either held static or flowed counter-current on the other side. The donor and acceptor conditions have been optimized in our previous study [29]. The HAAs from the water sample were extracted into the membrane liquid held in the membrane pores, and then back-extracted into the acceptor in their ionic forms. The enriched acceptor was collected and injected into the HPLC for analysis.

The chromatographic separation was carried out by a Hewlett-Packard 1050 HPLC equipped with a Waters 486 Tunable Absorbance UV detector, which was set at 210 nm. The HPLC column used was a 150 mm × 4.6 mm I.D. YMC ODS-A C18 column with 3 μm packing. The HPLC mobile phase was 95:5 (v/v) 15 mM KH_2PO_4 buffer (pH 2.24) and acetonitrile with a flow rate of 1.0 mL/min. Complete separation of the nine HAAs was

achieved within 13 minutes. Minichrom V 1.62 software (VG Data System) was used for data acquisition and analysis.

4.3 Results and Discussion

The two important considerations were the enrichment factor (EF) and the extraction efficiency (EE). EF was determined as the ratio of the analyte concentration in the extract to that in the original donor:

$$EF = \frac{C_a}{C_d} \tag{4.1}$$

C_a was the analyte concentration in the acceptor; C_d was the analyte concentration in the donor. Extraction efficiency (EE) was determined as the fraction of analyte in the acceptor to that in the donor [14].

$$EE = \frac{n_a}{n_d} = \frac{C_a \times V_a}{C_d \times V_d} = EF \times \frac{V_a}{V_d} \tag{4.2}$$

n_a and n_d were the total mass of analyte in the acceptor and the donor, V_a and V_d were the volumes of the acceptor exiting the membrane and the donor entering, respectively.

In the continuous flowing system,

$$V_d = F_d \times t \tag{4.3}$$

Where, F_d was the flow rate of the water entering the microfluidic SLM module, and t was the run time. In SLM extraction, the acceptor was an aqueous phase and its loss was negligible, thus, the acceptor volume could be expressed as:

$$V_a = F_a \times t \tag{4.4}$$

where, F_a was the flow rate of the acceptor. Consequently equation (4.2) reduced to:

$$EE = EF \times \frac{F_a}{F_d} \tag{4.5}$$

If the acceptor was held stagnant, EE was expressed as:

$$EE = EF \times \frac{V_a}{F_d \times t} \tag{4.6}$$

where, V_a was the acceptor channel volume.

4.3.1 The Effect of Acceptor to Donor Ratio

The objective was to maximize EF, which was determined by the concentration gradient across the membrane and the volume ratio of the donor to the acceptor. A larger ratio provided a higher concentration factor. EFs from modules A and B were compared; the latter had a higher donor to acceptor ratio. The aqueous sample contained 1 μg/mL of the nine HAAs. The donor flow rate was 0.126 mL/min, while the acceptor (0.05 M tris buffer pH 10) was held static for 30 min before being collected for analysis. The results are shown in Figure 4.4 The EFs of the HAAs on Module A were from 3 to 5, while those on Module B were from 9 to 16. In Module B, the volumetric ratio of donor and acceptor was increased by having a shallower acceptor channel, as the width of the channels had to match for complete cross-channel permeation. This resulted in a more concentrated extract when the same amount of the aqueous sample was introduced into the two modules. Based on this observation, Module A was not used in subsequent experiments.

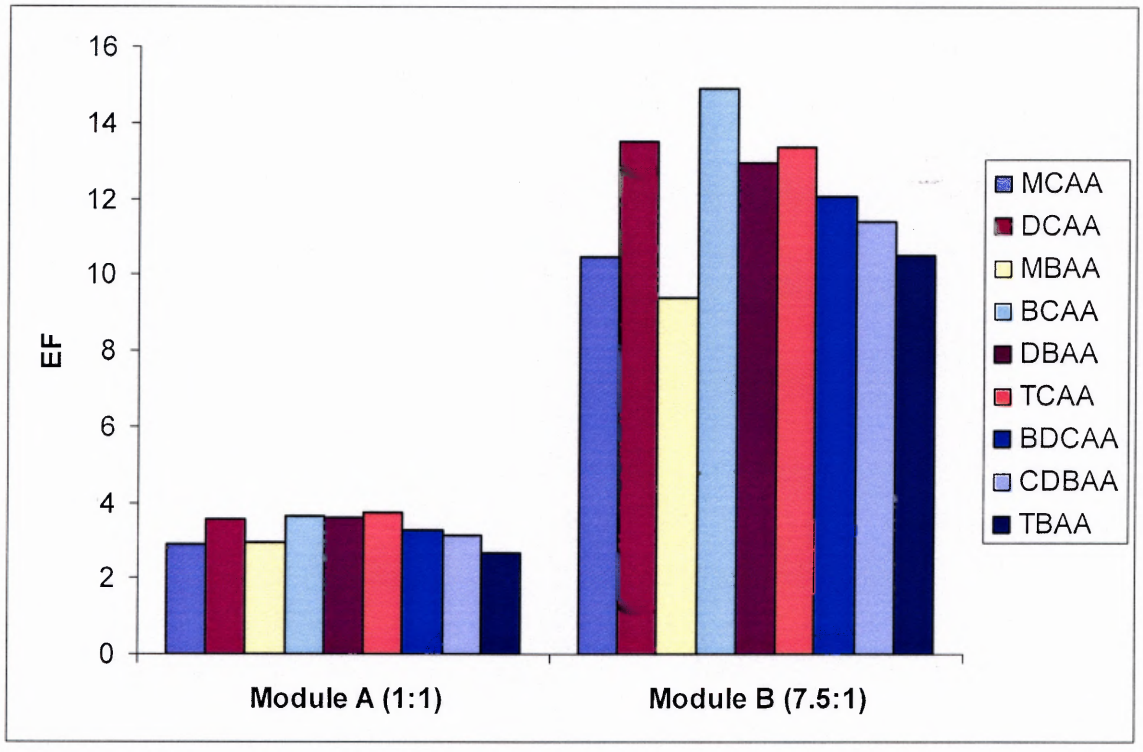


Figure 4.4 The effect of acceptor-to-donor ratio on the enrichment factor, the donor flow rate was 0.126 ml/min and the acceptor was stagnant for 30 min.

4.3.2 Static Versus Dynamic Acceptor

The extraction could be carried out in static, or in dynamic mode. In the former, the acceptor was not replenished as it continued to accumulate the analytes, and in the latter it flowed continuously. The effect of the acceptor status on EF and EE% was studied with Module B. The sample flow rate was 126 μL/min in both modes. In the static mode, the water sample flowed for 30 min before the extract was collected. In the dynamic mode, the acceptor flowed at 4.4 μL/min. The results are presented in Table 4.2. The EF varied from 9 to 13 in the static mode, and from 7 to 10 in the dynamic mode. The higher EF in the former was due to the smaller acceptor volume used. However, there was a big difference in EE by these two methods. The EE in the static mode varied from 7 to 11%, while in the

dynamic mode it ranged from 26 to 34%. The higher EE in the dynamic mode partially compensated for dilution to a large acceptor volume. In general, it was observed that the extraction could be carried out by either approach.

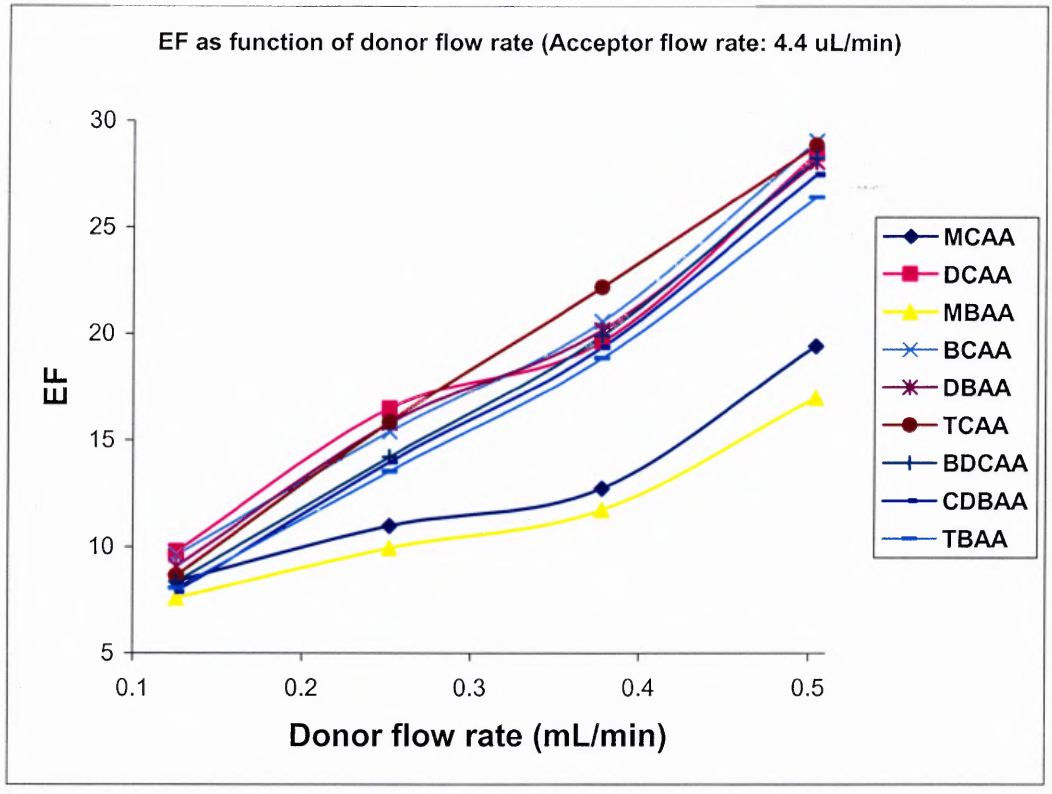
Table 4.2 Static versus Dynamic Extraction using Module B

Names	Abbreviations	Static* Mode		Dynamic** Mode	
		EF	EE%	EF	EE%
Monochloroacetic acid	MCAA	10.46	8.58	8.36	29.20
Dichloroacetic acid	DCAA	13.52	11.08	9.78	34.08
Monobromoacetic acid	MBAA	9.42	7.72	7.56	26.40
Bromochloroacetic acid	BCAA	14.92	12.24	9.59	33.50
Dibromoacetic acid	DBAA	12.96	10.63	9.05	31.61
Trichloroacetic acid	TCAA	13.39	10.98	8.63	30.12
Bromodichloroacetic acid	BDCAA	12.07	9.90	8.34	29.13
Chlorodibromoacetic acid	CDBAA	11.42	9.37	7.87	27.47
Tribromoacetic acid	TBAA	10.54	8.64	8.02	28.01

*The acceptor was held static for 30 min before being collected for analysis.
**The acceptor flow rate was 4.4 μL/min. The donor flow rate was 0.126 mL/min for both extraction modes.

4.3.3 Effect of the Flow Rates

The effect of the donor flow rate on EF and EE was studied with Modules B and C. The flow rate of the acceptor was kept constant at 4.4 μL/min with Module B and 2.64 μL/min with Module C. EF and EE (%), as a function of the donor flow rate is shown for the nine HAAs in Figure 4.5 and 4.6, respectively. EF increased with the donor flow rate. As more analytes contacted the membrane, more were extracted, leading to a higher EF. However, a larger fraction went unextracted, consequently EE decreased. These changes were consistent with Equations (4.7) and (4.8).



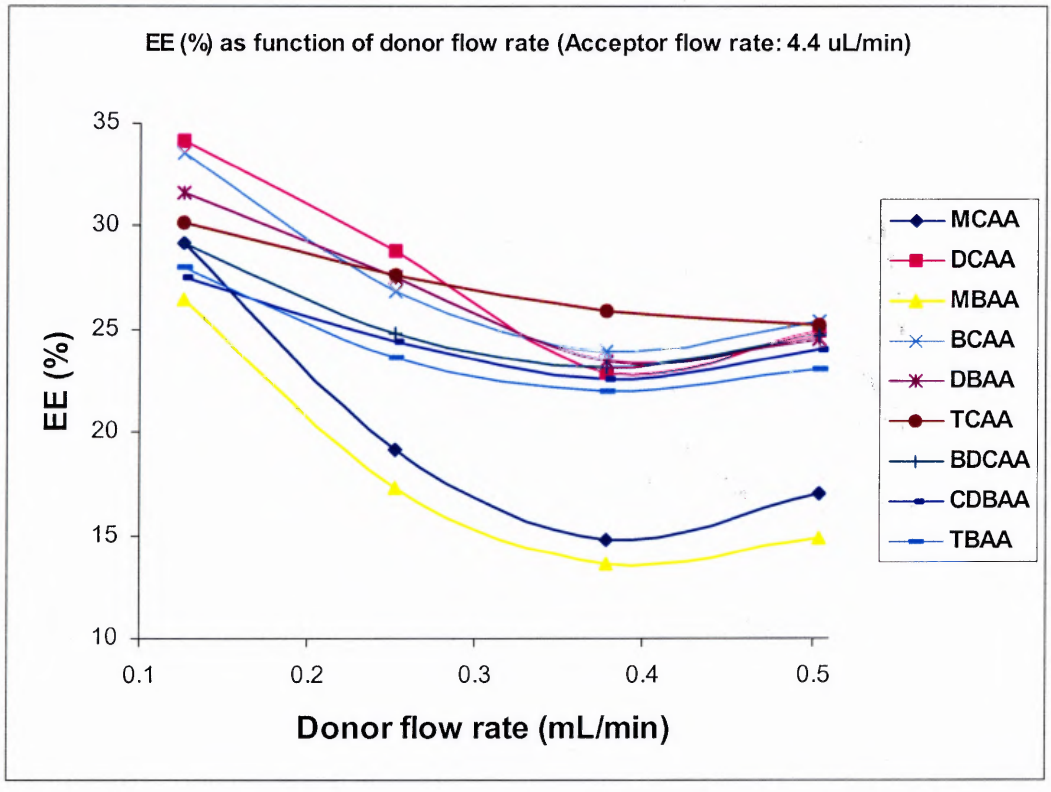
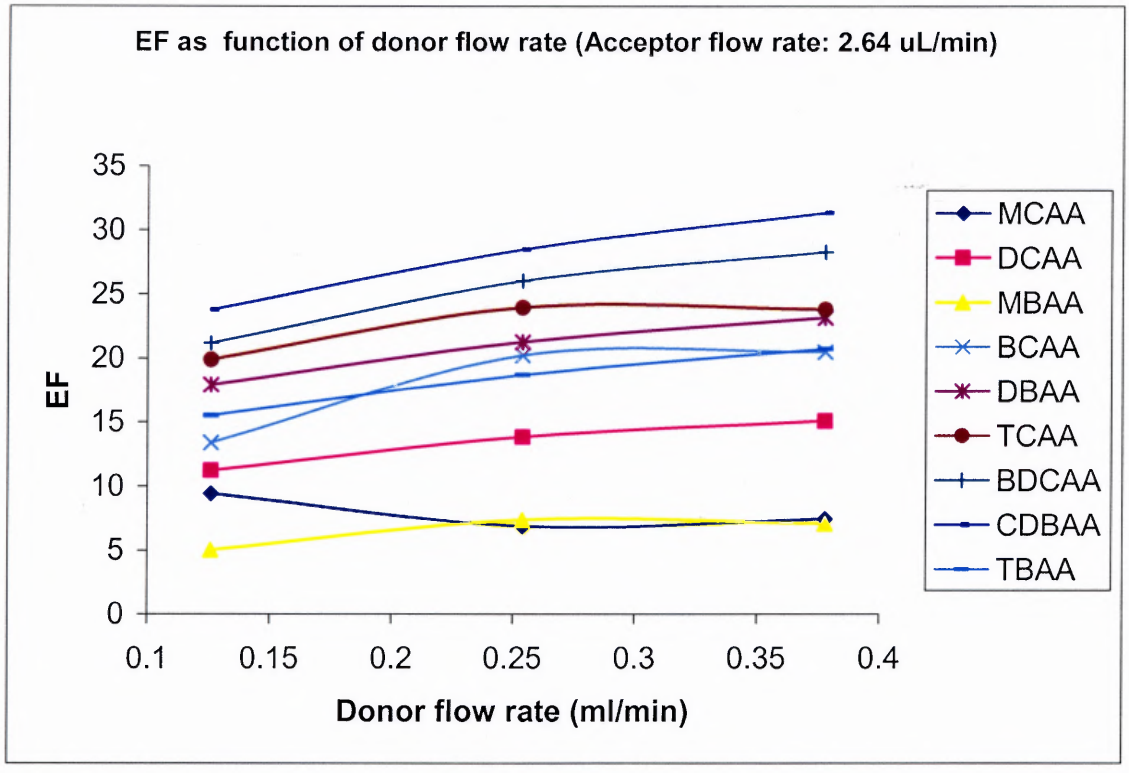


Figure 4.5 EF and EE% as a function of donor flow rate with Module B, the acceptor flow rate was kept constant at 4.4 μL/min.



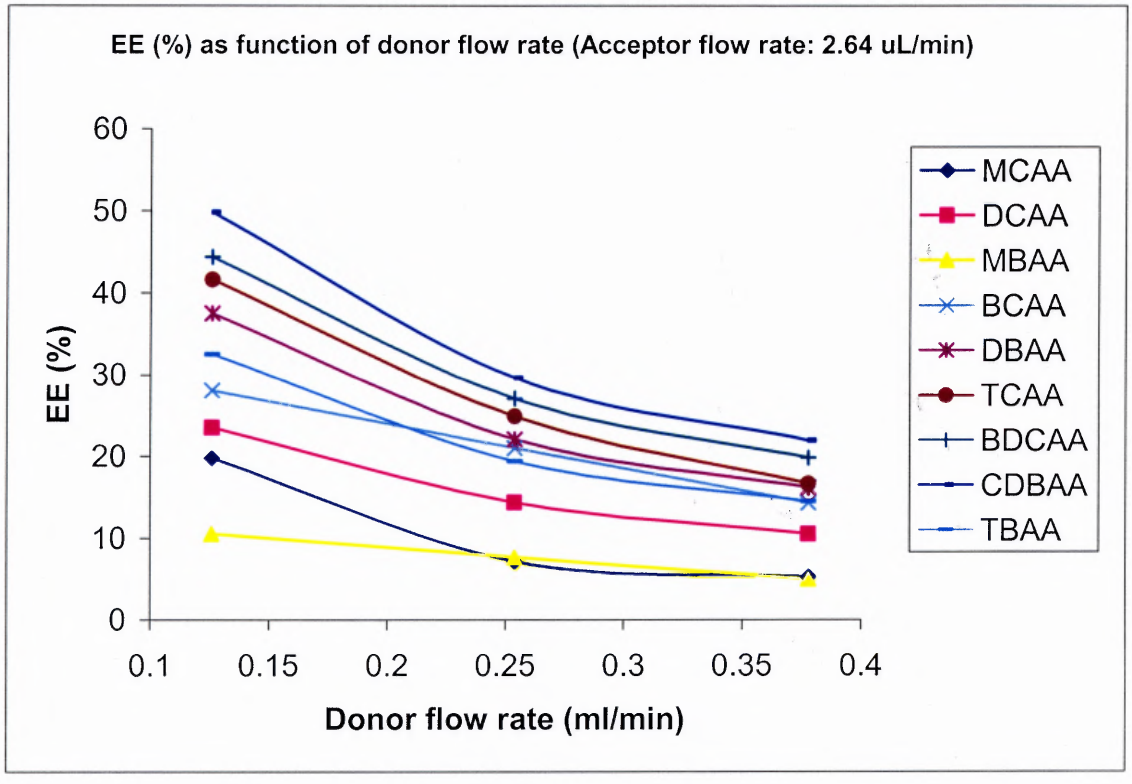


Figure 4.6 EF and EE (%) as a function of donor flow rate with Module C, the acceptor flow rate was kept constant at 2.64 μL/min.

The effect of the acceptor flow rate on EF and EE was also studied with modules B and C. With Module B the water sample flow rate was constant at 504 μL/min, while the acceptor flow rate was varied from 2.64 to 8.8 μL/min. With Module C the donor flow rate was kept constant at 126 μL/min while the acceptor flow rate varied from 0.88 to 2.64 μL/min. EF and EE (%) as functions of the acceptor flow rate with Module B and C are shown in Figure 4.7 and 4.8, respectively. EF decreased significantly with the increase of the acceptor flow rate, which also could be predicted by equation (4.8). At a lower flow rate, the acceptor volume was smaller, thus the analytes were more concentrated, leading to a higher EF. The effect of acceptor flow on EE was not significant. It appeared to increase marginally in the case of Module C. A lower acceptor flow rate increased the contact time, leading to a favorable EE.

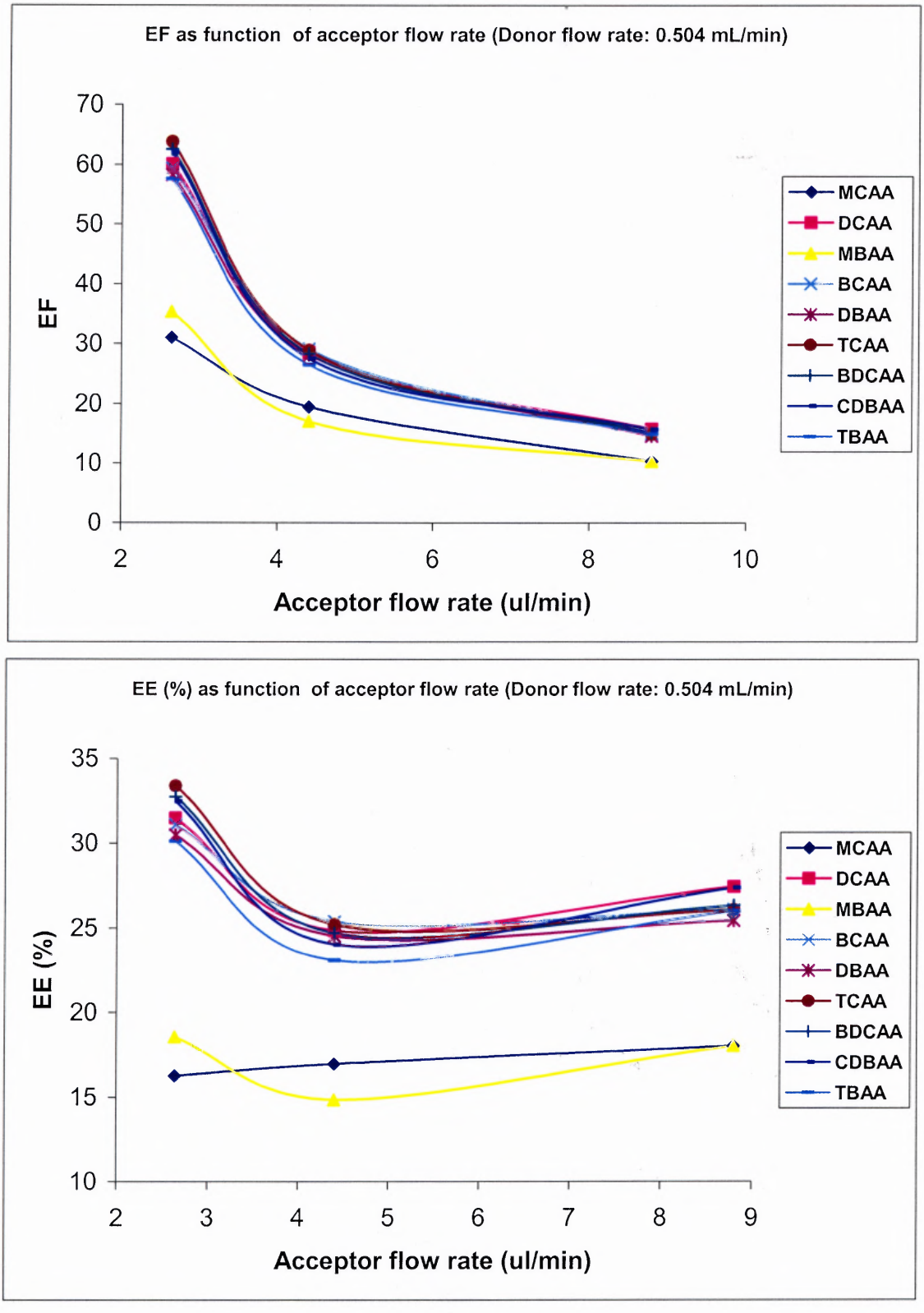


Figure 4.7 EF and EE% as a function of acceptor flow rate with Module B, the donor flow rate was kept constant at 0.504 ml/min.

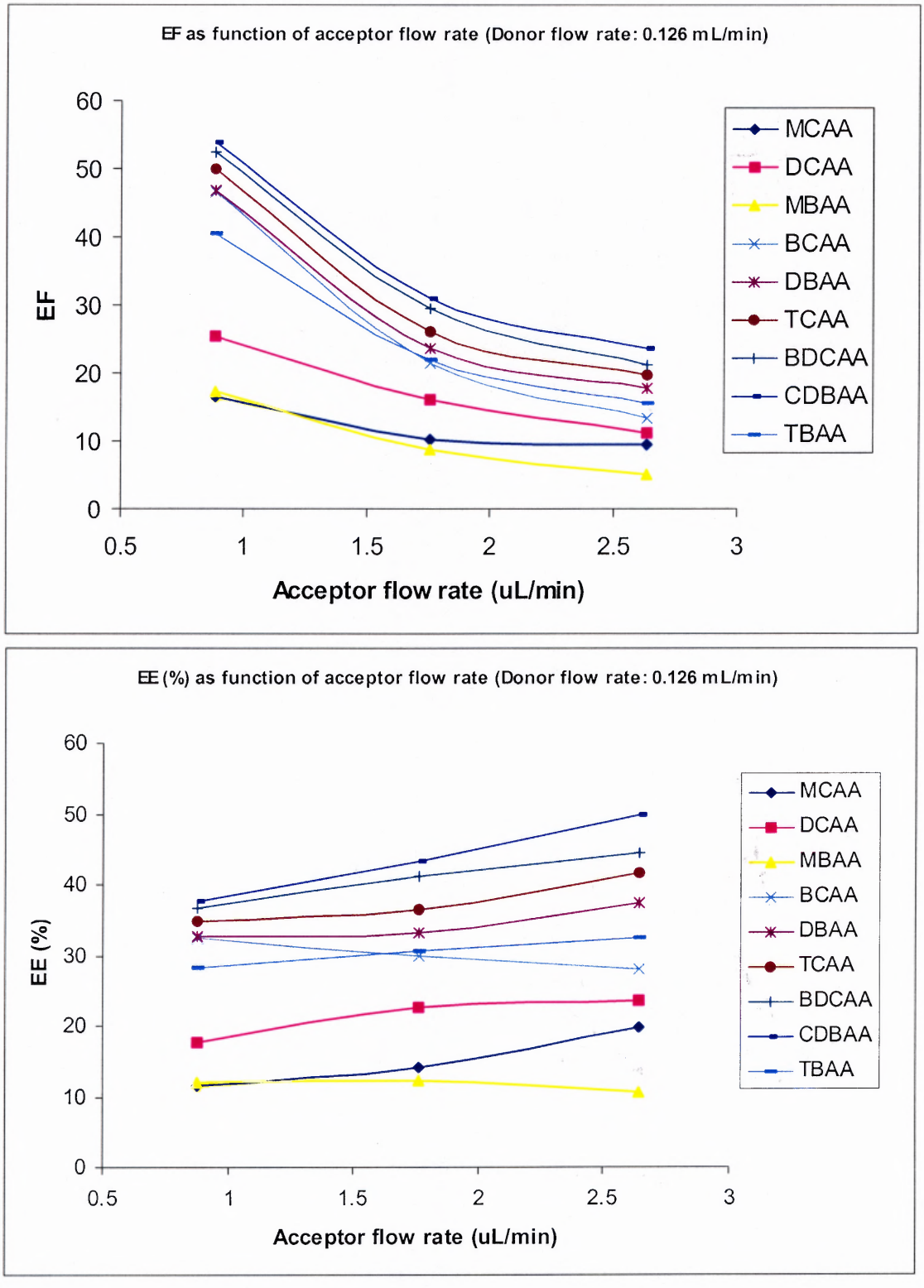


Figure 4.8 EF and EE (%) as a function of acceptor flow rate with Module C, the donor flow rate was kept constant at 0.126 mL/min.

Peak area as a function of experiment running time on Module B is show in Figure 4.9. The donor was an acidified water sample (5.6% H_2SO_4) containing 8 μg/ml MCAA and 1 μg/ml of the rest eight of HAAs. The donor flow rate was 0.504 ml/min. The acceptor was 0.05 M tris buffer (pH10) at a flow rate of 2.64 μl/min. As one can see, the peak area stopped increasing 45 min after the experiment started, in other words it needs 45 min to reach the extraction equilibrium.

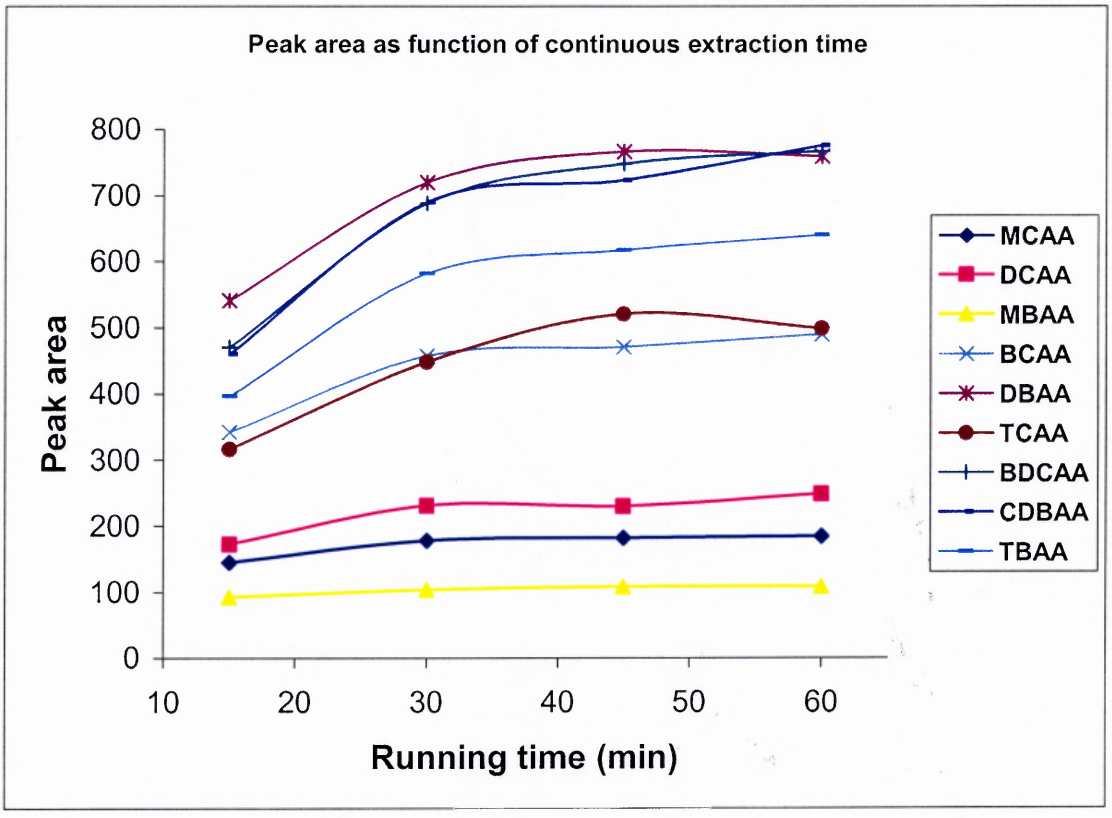


Figure 4.9 Peak area as a function of experiment running time, the donor flow rate was 0.504 ml/min and the acceptor flow rate was 2.64 μl/min.

4.3.4 Analytical Performance

The supported liquid membranes were stable for three weeks, after which they needed regeneration with the membrane liquid. Consequently the lifetime of the devices was long enough to be used either as a disposable or a re-usable device. Washing the membrane with the acceptor solution for two or three minutes between each run could eliminate the memory effect of the SLM.

The performance of Module B and C were comparable. As presented in Table 4.1, the maximum EF on Module C was 54 as compared to 65 on Module B. Analytical performance was tested with Module B. Chromatograms of a standard containing 10 μg/mL nine HAAs, and another after microfluidic SLM extraction of a water sample of 1 μg/ml of 9 HAAs are shown in Figure 4.10. The maximum EF and EE achieved with the three micro-fluidic SLM extraction modules are listed in Table 4.1. The data presented here were obtained in the dynamic mode. Based on the previous discussion, the EF (subsequently the detection limits) would have been higher in the static mode.

The linear dynamic ranges, regression coefficients (R^2), relative standard deviations (RSDs) and method detection limits (MDLs) are listed in Table 4.3. Being very polar, MCAA (log P of 0.22) showed performance that was not at par with the rest of the HAAs. This was consistent with the previous observation [29]. The detection limits determined by the standard EPA procedure [113] were at the ng/mL level, with six of them between 2.23 to 4.24 ng/mL. MDLs of MCAA, DCAA and MBAA were higher due to their high polarity (log P being less than 1). The response was linear with a fairly large linear dynamic range, the regression coefficient (R^2) were between 0.9895 and 0.9996. RSDs based on seven replicates were between 3.56 and 8.54 %.

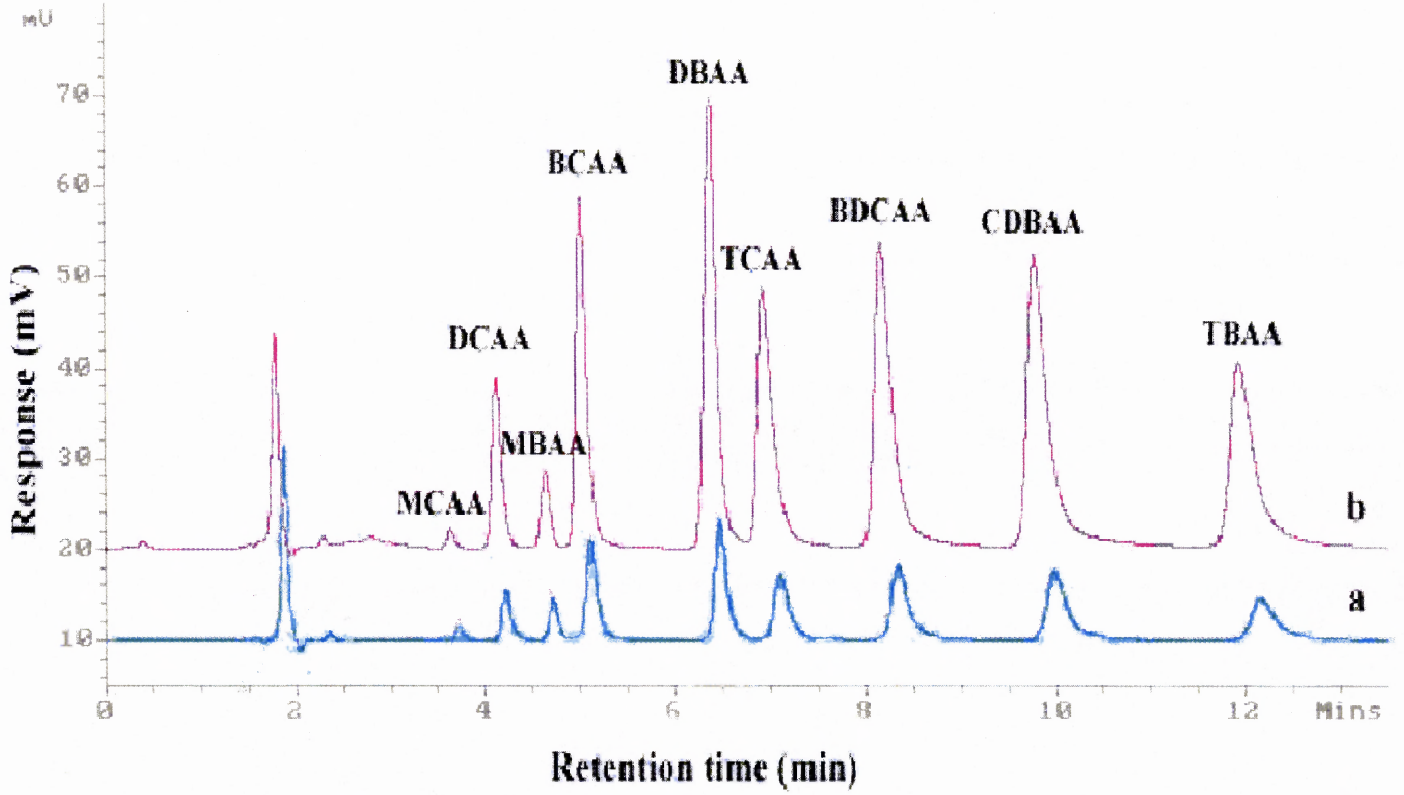


Figure 4.10 HPLC analysis of HAAs: a) Chromatogram of a standard containing 10 µg/mL of the nine HAAs, b) Chromatogram after microfluidic-SLME of a water sample containing 1 µg/mL of nine HAAs.

Table 4.3 Analytical Performance of Module B

HAAs	Log P*	Linear Dynamic Range (ng/mL)	Regression Coefficient (R^2)	RSD (%)	MDL** (ng/mL)
MCAA	0.22	400-20000	0.9987	8.54	106.87
DCAA	0.92	50-10000	0.9996	6.72	10.52
MBAA	0.41	100-5000	0.9995	7.95	24.87
BCAA	1.14	20-2000	0.9979	6.31	3.95
DBAA	1.69	20-2000	0.9980	6.77	4.24
TCAA	1.33	20-2000	0.9936	6.50	4.07
BDCAA	2.31	20-2000	0.9941	4.14	2.59
CDBAA	2.91	20-2000	0.9930	4.59	2.88
TBAA	3.46	20-2000	0.9895	3.56	2.23

* Log P: Octanol-water partition coefficient [29].
** MDLs were obtained with a water sample containing 400 ng/mL MCAA, 50 ng/mL DCAA, 100 ng/mL MBAA, and 20 ng/mL rest 6 HAAs.

4.4 Conclusion

Miniaturized supported liquid membrane extraction followed by HPLC-UV detection was designed and fabricated for sample enrichment. Microfluidic SLM modules with acceptor and donor channels of different depths were machined and the effects on the extraction performance were compared. Modules with shallower acceptor channels provided higher enrichment. With Module B enrichment factors as high as 65 were obtained along with relatively large linearity range and high precision. This micro-fluidic device can be integrated with other lab-on-a-chip components to develop a total analytical system (TAS).

SECTION B:

AUTOMATED, ON-LINE MEMBRANE EXTRACTION

CHAPTER 5

CONTINUOUS, ON-LINE MONITORING OF HALOACETIC ACIDS VIA MEMBRANE EXTRACTION

5.1 Introduction

The importance of the detection of haloacetic acids (HAAs) in water has been described in Chapter 3. We have reported an off-line method, supported liquid membrane micro-extraction, for the extraction and enrichment of nine HAAs in water [29]. However, currently there is no method for continuous, on-line monitoring of all of the nine HAAs. Automated online measurements are less expensive, provide real-time information and have better accuracy and precision [114]. Since there is less manual sample handling, these techniques tend to be less prone to contamination. Therefore the development of a continuous on-line method is needed. The goal of this study is to develop a novel automated method for such purposes.

Membrane extraction has recently emerged as a promising technique for sample enrichment. It has several advantages, such as simple instrumentation, requiring small solvent volumes and offering high enrichment factors. It allows continuous online extraction in a flow system, and can be coupled to a GC [15-17], HPLC [18-20, 24-26], mass spectrometry (MS) [21] and CE [23] for continuous on-line monitoring. There are two major approaches for membrane extraction, supported liquid membrane extraction (SLME) and liquid-liquid membrane extraction (LLME) [14]. SLME is a three-phase extraction system, where the analytes are extracted from an aqueous sample into an acceptor via an organic extractant held in the pores of the membrane. It works well for the extraction of highly polar and ionizable compounds [37]. Recently, we have reported

supported liquid membrane micro-extraction (SLMME) for the extraction of HAAs from water [29]. This technique provides high enrichment and relatively short analysis time. LLME is a two-phase system, where the analytes are extracted from an aqueous sample into an organic acceptor. Here, the organic solvent contacts the water sample across the membrane without direct mixing. Another advantage of the membrane interface is that there is no emulsion formation, which is a common occurrence in conventional liquid-liquid extraction.

The objective of this study is to develop a membrane extraction technique for continuous on-line monitoring of HAAs. Both continuous LLME and SLME followed by HPLC-UV detection were investigated in this research.

5.2 Experimental Section

The instrumentation used for SLME and LLME were quite similar and is shown in Figure 5.1. It includes a hollow fiber membrane module, two pumps and a HPLC system. The first pump (a Hewlett-Packard 1050 HPLC pump) was used for the delivery of the acceptor, and the other (a Beckman 110B pump) for the donor. An automated six-port HPLC injection valve (Valco Instruments Co. Inc., Houston TX, USA) was used to make repeat injections into the HPLC (Hewlett-Packard 1050) with a tunable absorbance UV detector (Waters 486). The wavelength was set at 210 nm. Donor flowed on the shell side of the membrane module, while the acceptor flowed inside the hollow fiber lumens. This approach has been described before [18-21]. The HAAs in the donor were extracted and enriched into the acceptor. The extract was injected (20 μL injection volume) into the HPLC for analysis. Minichrom V 1.62 software (VG Data System) was used for data acquisition.

Nine individual HAAs were purchased from Supelco (Supelco Park, PA, USA). All other chemicals used in this study were ACS reagent grade (Sigma Chemical Co., St. Louis, MO, USA). Deionized water was obtained from a Milli-Q® water purification system (Millipore Co., Bedford, MA, USA).

5.2.1 Continuous Liquid-Liquid Membrane Extraction

The membrane module for LLME was made by packing six pieces of 100 cm long composite hollow fiber membranes into a PTFE tube. Each end of the tube was connected to a tee union (Supelco Inc., PA, USA). Epoxy (Resin Technology Group, LLC, S. Easton, MA, USA) was used to seal the space between the membranes and the tee, preventing the mixing of water and the acceptor. The membrane used was Celgard X10 (Hoechst Celanese, Charlotte, NC, USA) with an I.D. of 0.240 mm and an O.D. of 0.290 mm, which was made of polypropylene. The acceptor used in LLME was 99.8% methyl tert-butyl ether (MTBE) (Fluka, Milwaukee, WI, USA). The HPLC column was a 3.9 mm I.D., 150 mm long Waters Resolve C_{18} with 5 μm spherical packing (Waters, Milford, MA, USA). The HPLC mobile phase was a 0.4 M ammonium sulfate solution and the flow programming was as follows: flow rate was held constant at 0.5 mL/min during the first five minutes, and was then increased gradually to 2.0 mL/min in the next three minutes, the flow rate was kept constant at that level between 8 to 13 minute.

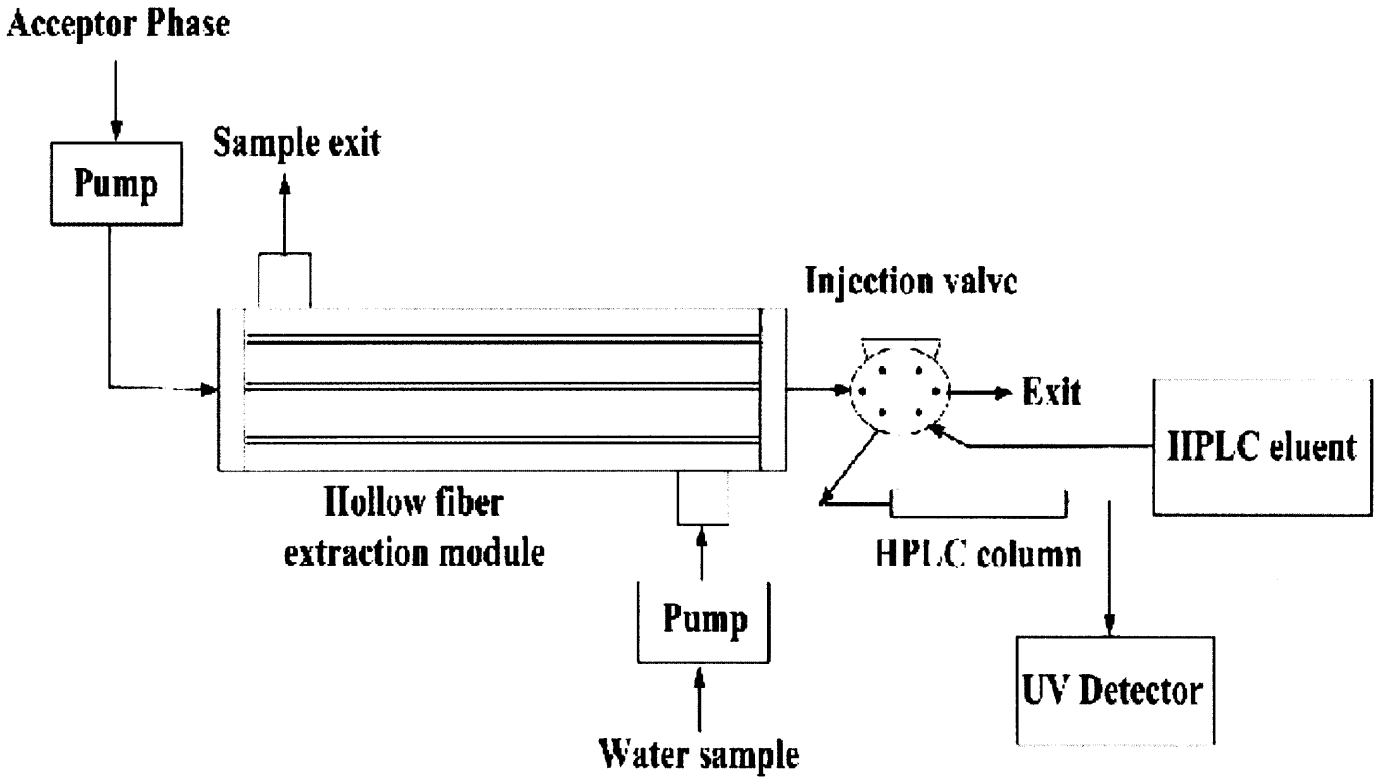


Figure 5.1 Schematic diagram of continuous membrane extraction followed by HPLC-UV detection.

5.2.2 Continuous Supported Liquid Membrane Extraction

The membrane module for SLME was made by packing three pieces of 130 cm long hollow fiber membranes into a PTFE tube. The membrane was Celgard X20 (Hoechst Celanese, Charlotte, NC, USA). It had an I.D. of 400 μm and an O.D. of 460 μm, with an average pore size of 0.03 μm and porosity of 40%. The membranes were soaked in the membrane liquid, 5% trioctylphosphine oxide in dihexyl ether, which was optimized in a previous study [30] before the analysis. The HPLC column used here was a 150 mm × 4.6 mm YMC ODS-A C_{18} column with 3 μm packing. The HPLC mobile phase was 95:5 (v/v) 15 mM KH_2PO_4 (pH 2.2): Acetonitrile at a flow rate of 1.0 mL/min.

The membrane module can be regenerated in this way: first, clean the membrane by flowing the acceptor, then dry it with nitrogen, and finally soak with the membrane liquid.

5.3 Results and Discussions

5.3.1 Continuous Liquid-Liquid Membrane Extraction

Continuous LLME was carried out across the hollow fiber. The donor consisted of 0.12 μg/mL (ppm) MCAA and DCAA, 0.08 ppm MBAA, BCAA and BDCAA, 0.04 ppm DBAA and TCAA, 0.2 ppm CDBAA and 0.4 ppm TBAA. The acceptor was 99.8% methyl tert-butyl ether (MTBE). During HPLC analysis MTBE co-eluted with CDBAA and TBAA. Consequently, only seven of the HAAs could be quantified. An enrichment factor of about 50 was achieved. However, the objective of this study was to develop a method for continuous on-line monitoring of all the nine HAAs at trace levels, thus continuous SLME was investigated in the following study.

5.3.2 Continuous Supported Liquid Membrane Extraction

SLME was carried out by two different modes: stop-and-flow SLME and continuous SLME. The former is a semi-static approach and is referred to as SF-SLME. In SF-SLME, the donor flows continuously while the acceptor is held stagnant, and then collected after a certain period. The donor contained 83.2 μg/L (ppb) of nine HAAs. Sulfuric acid (0.26%) was added to the donor to keep the acids in their uncharged molecular form. The donor was pumped at the rate of 1 mL/min for 60 minutes. The acceptor used here was 0.01 M sodium hydroxide (NaOH). The acceptor was stagnant in the membrane lumens. The membrane module was made by packing two pieces 128 cm Celgard X20 hollow fiber membranes into a PTFE tube. The internal volume of the membrane lumens was 0.322 mL. The membrane was soaked with 5 % trioctylphosphine oxide (TOPO) in dihexyl ether (DHE) before extraction. After extraction, the acceptor was pumped at 0.1 mL/min and collected into HPLC vial inserts every minute. Results are shown in Figure 5.2. The EF was found to decrease with time. The concentration gradient in the acceptor along the length of the hollow fiber was evident. The concentration was higher in the static acceptor segment near the entrance point of the donor, which corresponded to the exit point of the acceptor. During counter-current contact across the membrane, the entering donor had high concentration and it first contacted the extract near the acceptor outlet and the extraction was initiated. As it flowed further, and toward the acceptor inlet, its concentration decreased. Consequently, the extract near the donor inlet was more enriched than that near the outlet.

SF-SLME provided a higher enrichment factor but lower extraction efficiency as the acceptor solution was kept stagnant in the hollow fiber lumens. There existed a

concentration gradient in the acceptor for the SF-SLME. Shorter membrane fibers may decrease the concentration gradient across the hollow fiber.

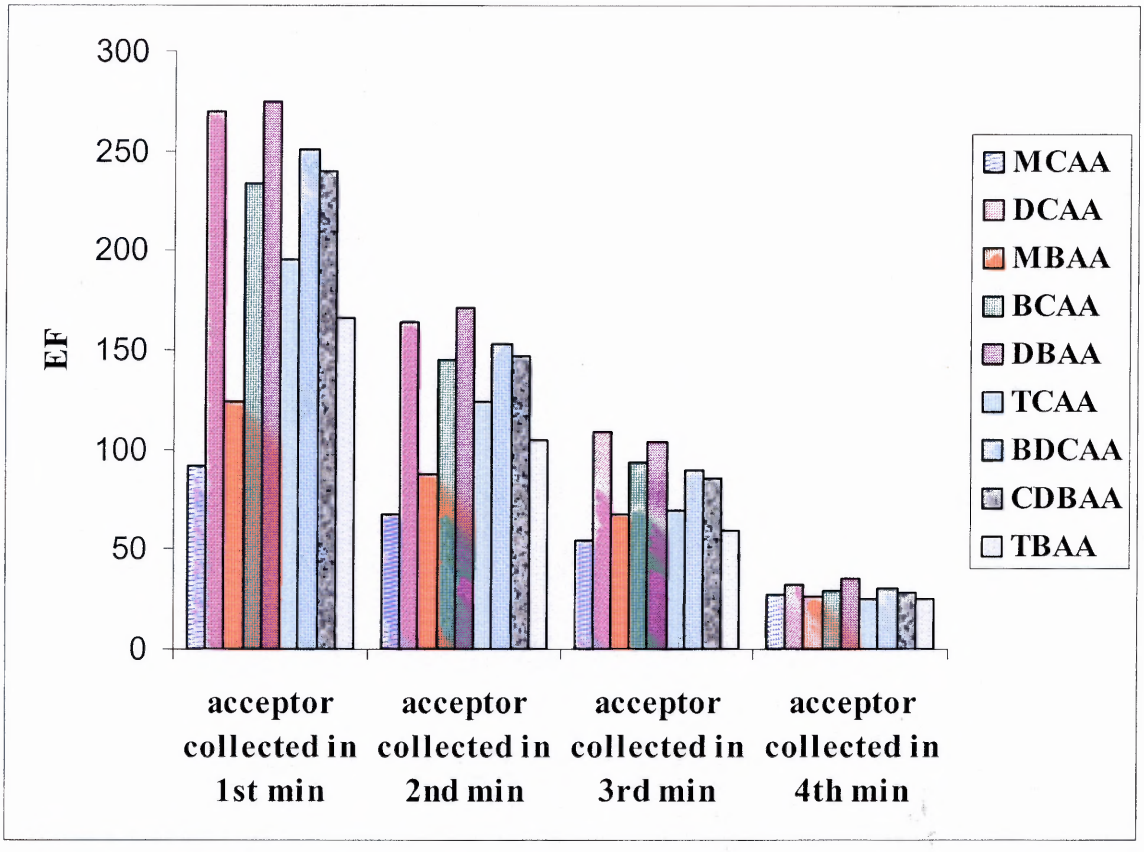


Figure 5.2 EF as a function of acceptor collecting time, the donor was flowed at 1 ml/min for 60 minutes, while the acceptor was kept stagnant in the membrane lumens. After the extraction the acceptor was pumped to flow at 0.1 ml/min. The acceptor exiting the membrane module was collected at 1^{st}, 2^{nd}, 3^{rd} and 4^{th} minute.

To obtain real-time information, both the donor and acceptor need to flow continuously, which is referred as continuous SLME. In continuous SLME, the donor contained 80 ng/mL (ppb) of the nine HAAs in DI water, which was acidified with sulfuric acid to pH 1.9. The donor pH is acceptable even though it is somewhat higher than the pKa values of the acids, as the protonation equilibrium is dynamic and quite fast. Lower pH

may result in higher extraction efficiency, however very acidic solution is more corrosive, which may destroy the pump. The acceptor was 0.05 M tris buffer adjusted to pH 8.7, which was more than 3.3 units higher than the pKa values that were required to prevent the extracted acids from re-entering the membrane. The membrane module was made of three pieces 130 cm Celgard X20 hollow fiber membranes. The membrane was soaked with 5 % TOPO in DHE prior to use.

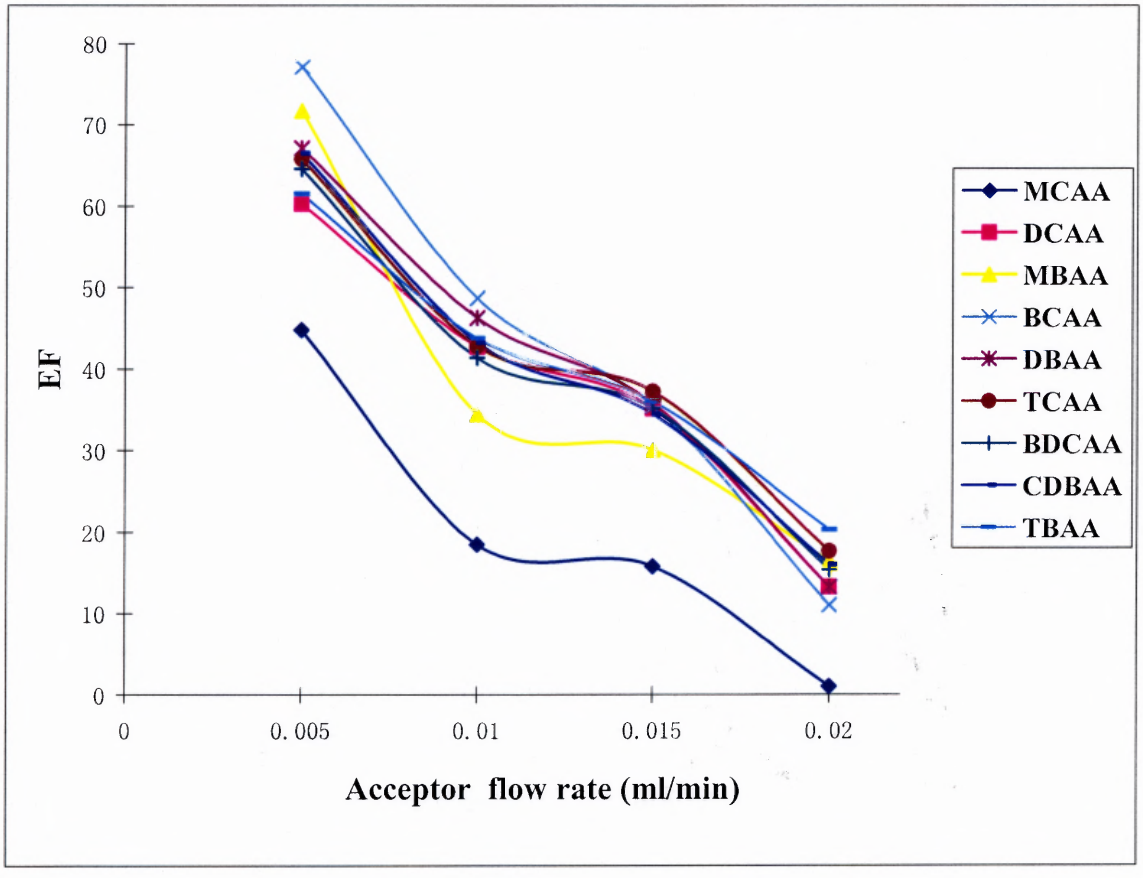


Figure 5.3 EF as a function of acceptor flow rate, the donor flow rate was kept constant at 1 ml/min.

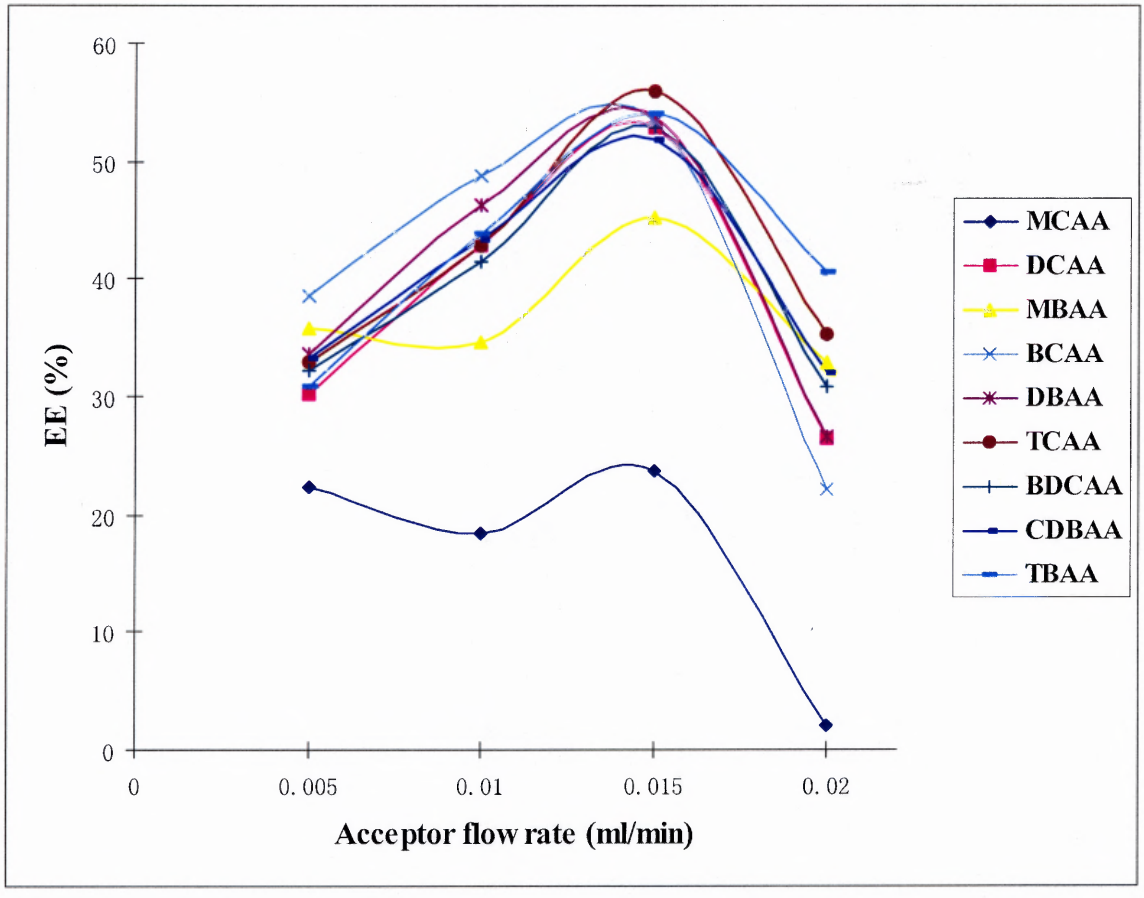


Figure 5.4 EE (%) as a function of acceptor flow rate, the donor flow rate was kept constant at 1 ml/min.

The effects of the acceptor flow rate on EF and EE in continuous SLME were studied. The flow rate of the water sample was kept constant at 1 ml/min, while the acceptor flow rate was varied from 0.005 to 0.02 mL/min. It was found that the loss of acceptor during extraction was negligible. Thus the experiment was carried out online with HPLC-UV detection. EF and EE (%) as a function of the acceptor flow rate are shown in Figure 5.3 and 5.4, respectively. EF decreased with the increase in the acceptor flow rate. At a lower flow rate, the contact time was longer and more analytes could be trapped into the acceptor. Moreover, higher acceptor flow rate diluted the extract, so the overall effect was the reduction in EF. According to Equation 4.5, EE increases with the increase of the

EF and the acceptor flow rate, and decreases with the donor flow rate. However, EF decreases with the increase of the acceptor flow rate. In Figure 5.4 the EE increased with acceptor flow rate increase from 0.005 to 0.015 mL/min, then decreased as the acceptor flow rated increased to 0.02 mL/min due to the dramatic decrease of the EF.

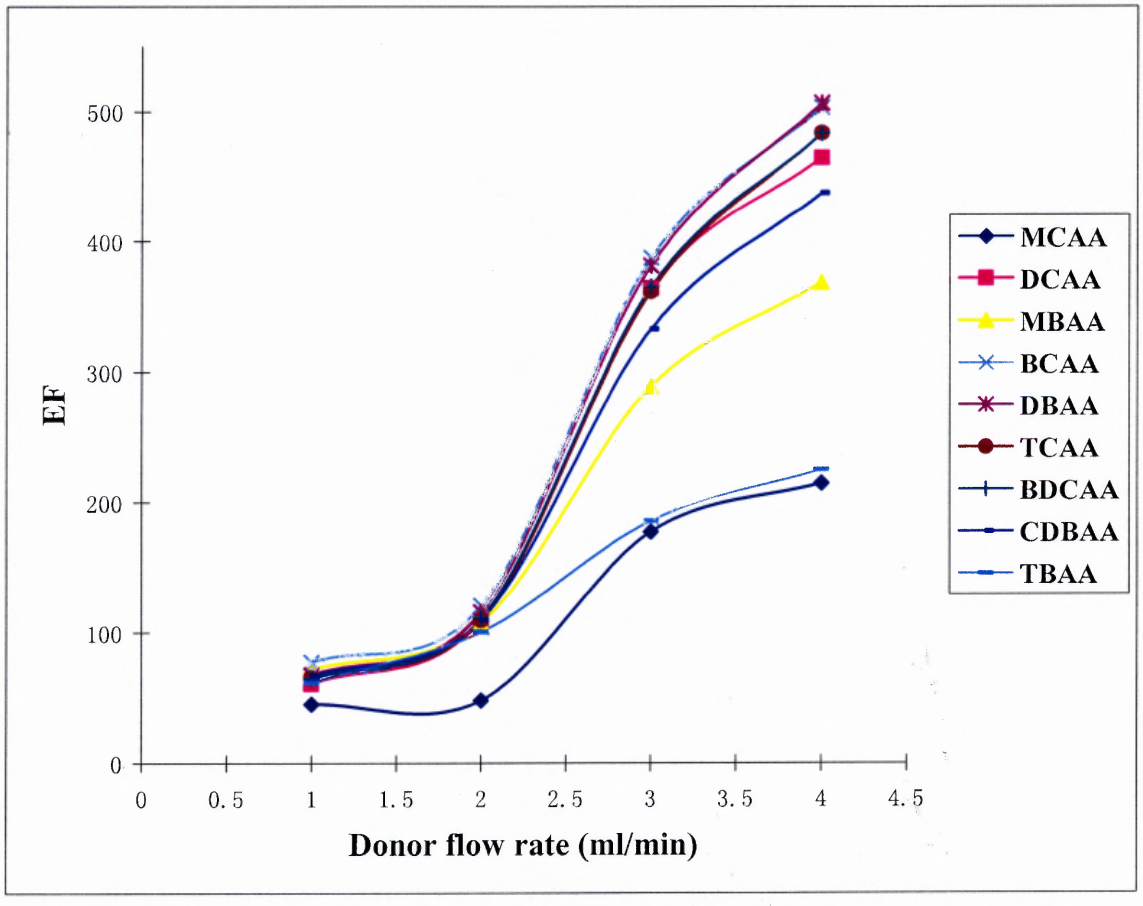


Figure 5.5 EF as a function of water sample (Donor) flow rate, the flow rate of acceptor was kept constant at 0.005 ml/min.

The effects of the donor flow rate on EF and EE was also studied. The acceptor flow rate was kept constant at 0.005 mL/min, while the donor flow rate increased from 1 to 4 mL/min. EF and EE as a function of the donor flow rate is shown in Figures 5.5 and 5.6. EF increased dramatically as the flow rate of water sample increased from 1 to 4 ml/min.

With a higher donor flow rate, more analytes contacted the membrane, thus resulting in more analytes trapped in the acceptor, which led to higher EF. EE increases with the increase of EF but decreases with the increase of the donor flow rate and is shown in Figure 5.6.

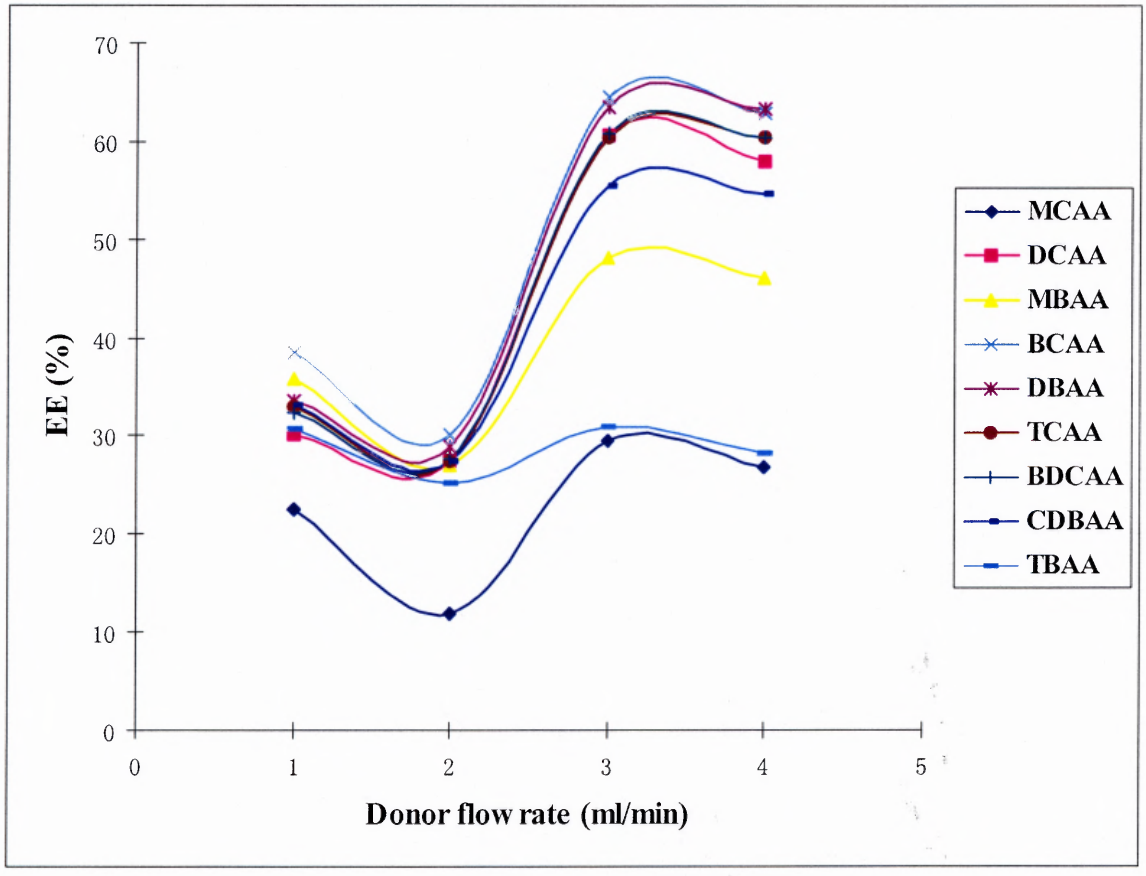


Figure 5.6 EE (%) as a function of water sample (Donor) flow rate, the flow rate of acceptor was kept constant at 0.005 ml/min.

5.3.3 Analytical Performance of Continuous SLME

SLME showed higher EF compared to LLME, and its analytical performance was evaluated. The HAAs in acidified donor were extracted and trapped into the alkaline

acceptor. The enriched acceptor was automatically injected into the HPLC-UV system every fifteen minutes by automated injection valve. Sequential chromatograms were obtained and shown in Figure 5.7. Good reproducibility in peak shape and retention time was observed.

Relative standard deviations, enrichment factors, extraction efficiencies and method detection limits (MDL) are listed in Table 5.1. The donor used was a water sample (pH 1.9) containing 21 ppb MCAA, 3 ppb MBAA and 1 ppb of each of the other seven HAAs. The donor flow rate was 4 ml/min. The acceptor was 0.05 M tris buffer (pH 8.7) at a flow rate of 0.005 ml/min. Figure 5.8 shows the chromatogram obtained under such low concentrations. The RSDs were between 3.3 and 10.3 % and the MDLs were at sub-ppb levels. The MDL of MCAA was higher than other HAAs. This is because MCAA is a very polar compound and its octanol-water partition coefficient (log P) is considerably lower than the other HAAs. This is consistent with previous observation [25-26].

The memory effect of the membrane was tested by flowing reagent water as the donor. The HAAs concentrations were about 2% of that obtained with a normal experiment. Thus the retention by the membrane itself was minor, and could be eliminated by washing the membranes with the acceptor for one minute. The supported liquid membrane module worked well for about 4 weeks, after which it needed regeneration.

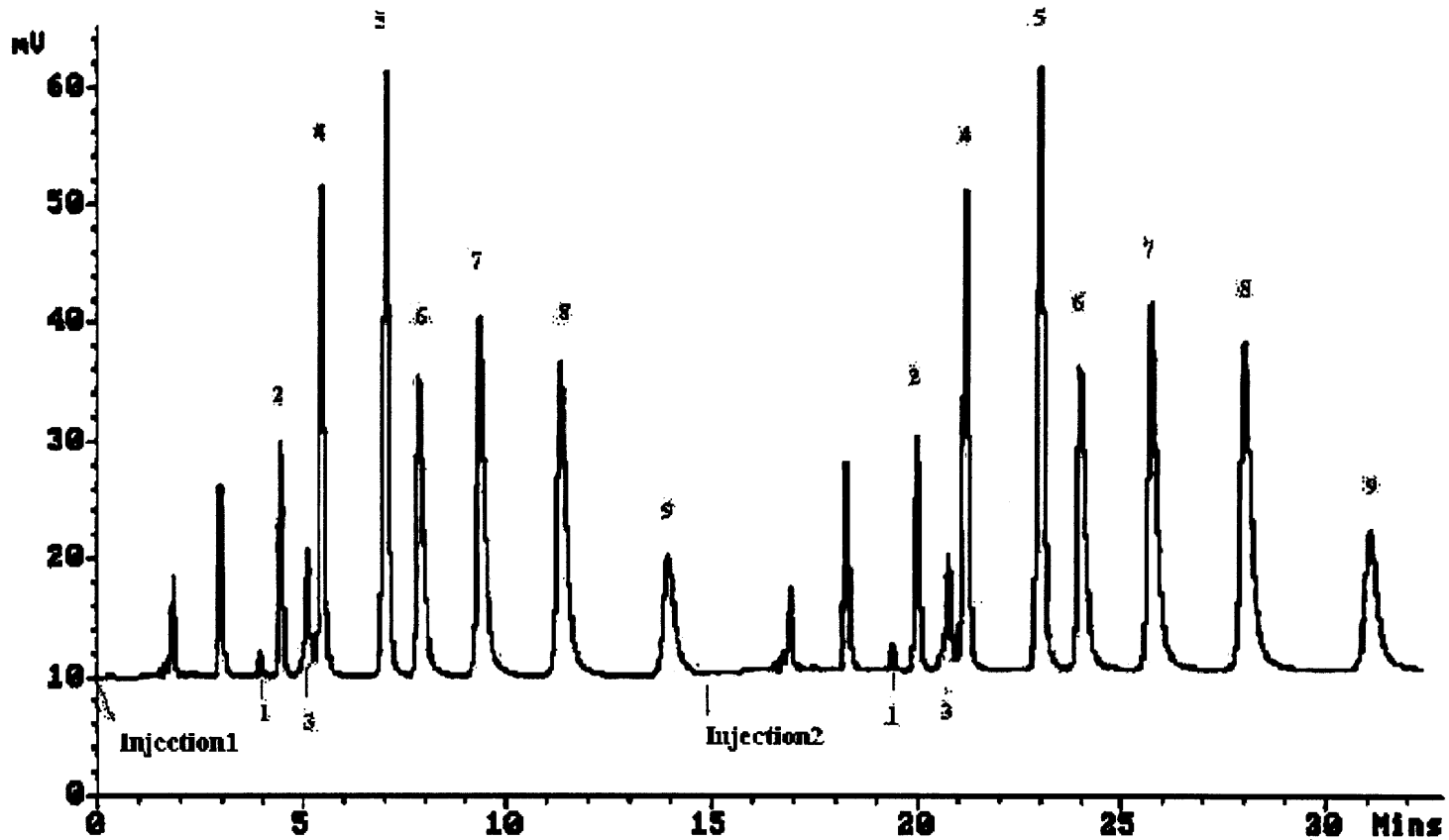


Figure 5.7 Chromatogram of continuous SLME of a water sample (pH 1.9) containing 80 ng/mL (ppb) nine HAAs: 1: MCAA, 2: DCAA, 3: MBAA, 4: BCAA, 5: DBAA, 6: TCAA, 7: BDCAA, 8: CDBAA, and 9: TBAA. The water sample flow rate was 4 mL/min. The acceptor was 0.05 M tris buffer (pH 8.7) at a flow rate of 0.005 mL/min. Injections were made every 15 min.

Table 5.1 Analytical Performance of Continuous SLME-HPLC

Names	Abbreviation	LogP*	pKa*	RSD** (%)	EF	EE (%)	MDL*** (ng/mL)
Monochloroacetic acid	MCAA	0.22	2.87	10.3	71.3	8.9	6.84
Dichloroacetic acid	DCAA	0.92	1.26	10.3	335.5	41.9	0.32
Monobromoacetic acid	MBAA	0.41	2.89	3.5	335.9	42.0	0.33
Bromochloroacetic acid	BCAA	1.14	1.39	4.2	273.6	34.2	0.13
Dibromoacetic acid	DBAA	1.69	1.47	4.8	412.1	51.5	0.15
Trichloroacetic acid	TCAA	1.33	0.51	5.7	383.4	48.0	0.18
Bromodichloroacetic acid	BDCAA	2.31	1.09	5.9	412.3	51.5	0.18
Chlorodibromoacetic acid	CDBAA	2.91	1.09	3.3	428.4	53.6	0.10
Tribromoacetic acid	TBAA	3.46	3.13	8.8	305.5	38.2	0.28

*logP values are from Ref. [29].
**Relative Standard Deviations (RSD) based on seven replications were obtained with continuous SLME, the water containing 21 ppb MCAA, 3 ppb MBAA, and 1 ppb rest 7 HAAs flowed at 4 mL/min and the acceptor at 0.005 mL/min.
***The Method Detection Limits (MDLs) were obtained following a standard EPA procedure [98].

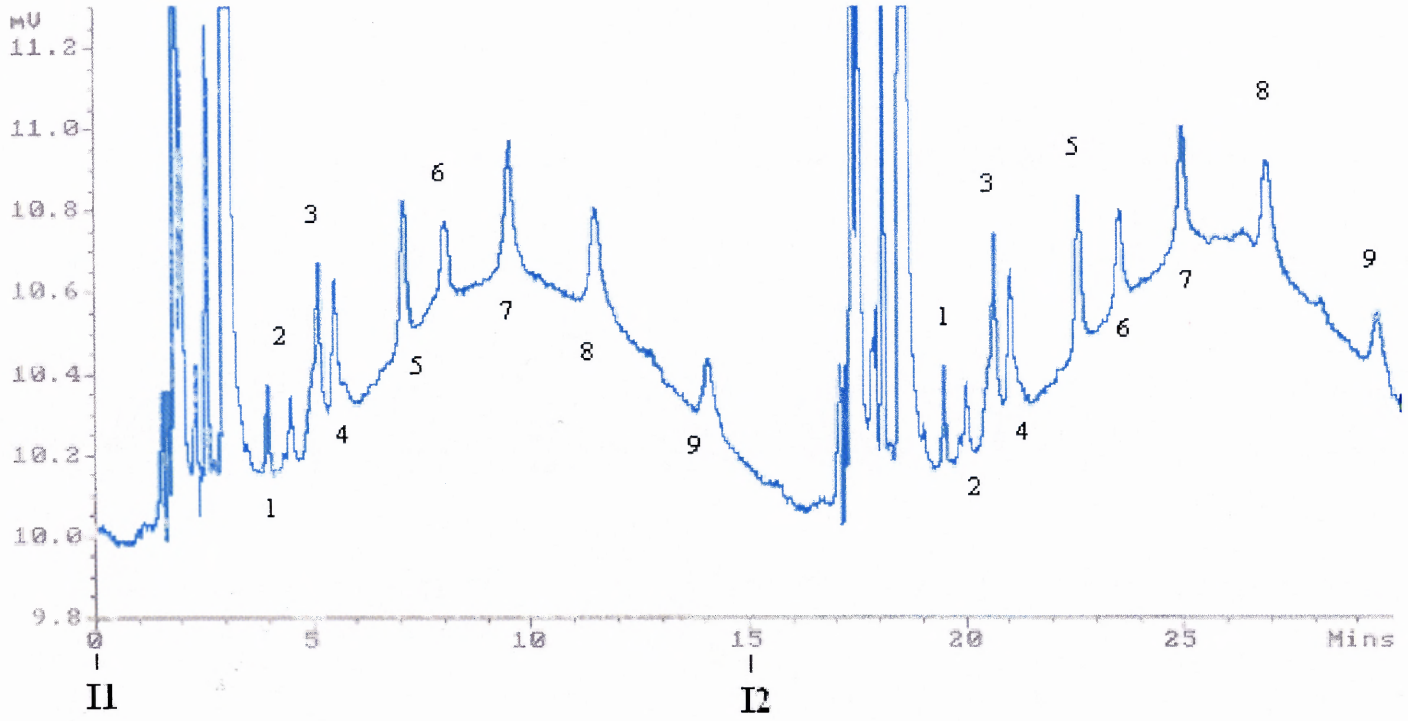


Figure 5.8 Chromatogram of continuous SLME of a water sample (PH 1.9) containing: (1) 21 ppb MCAA, (2) 1 ppb DCAA, (3) 3 ppb MBAA, (4) 1 ppb BCAA, (5) 1ppb DBAA, (6) 1 ppb TCAA, (7) 1 ppb BDCAA, (8) 1 ppb CDBAA, and (9) 1 ppb TBAA. The donor flow rate was 4 ml/min. The acceptor was 0.05 M tris buffer (PH 8.7) with a flow rate of 0.005 ml/min. I1: Injection 1, I2: Injection 2.

5.4 Conclusion

Both LLME and SLME were developed for the continuous analysis of the HAAs. Seven HAAs could be analyzed by LLME and the EF was about 50. Continuous SLME could be used to monitor all of the nine HAAs and enrichment factors were as high as 500 with MDLs at sub-ppb levels. The HPLC method used in continuous SLME is free from the interference of humic acid presented in real-world water. On the whole, continuous SLME was found to be more effective and is recommended for the applications of real-time monitoring of HAAs in water treatment plants.

CHAPTER 6

INTERFACING MEMBRANE EXTRACTION, PERVAPORATION AND HPLC DETECTION INTO AN AUTOMATED TOTAL ANALYTICAL SYSTEM

6.1 Introduction

There has been much interest in integrating different analytical functions onto a single platform. The efforts have been mainly confined to the bio-analytical arena, where procedures, such as, cell lysis, extraction, PCR and electrophoresis have been integrated in a micro total analytical system (μ-TAS) [115-116]. These ideas are equally valid for conventional laboratory techniques. Typical approaches for inorganic, organic, metals and biological analysis involve extraction and concentration followed by analytical detection. The development of a total analytical system (TAS) requires the hyphenation of these steps so that continuous, on-line analysis can be carried out without manual intervention.

Let us take the example of the analysis of semi-volatile organics in water. Liquid-liquid extraction (LLE), solid phase extraction (SPE) and solid phase micro-extraction (SPME) [117-121] are the conventional extraction procedures. Although they have some excellent merits, there exist limitations when it comes to direct interfacing with instruments. Classical LLE is labor intensive, uses large amounts of solvents and is difficult to couple directly to an analytical instrument. LLE coupled with flow injection (FI) analysis was reported for the first time in the late 1970s [122] and since then it has developed quickly [123-125]. It minimizes reagent consumption, and can be carried out continuously followed by on-line or off-line detection. In SPE the analytes are extracted from an aqueous sample onto a solid sorbent, and then eluted with a suitable solvent. It has been automated on-line involving multiple batch processes such as conditioning, washing,

and elution [126]. SPME, where the analytes are adsorbed onto a fused-silica coated fiber and then desorbed at a high temperature prior to analysis, is simpler, but only suitable for high concentration analysis due to its low sensitivity. The extracts, especially those from SPE and LLE may need further concentration. Conventional methods for this include nitrogen blowing, rotary evaporator, or Kuderna-Danish concentrators. It is evident that both the extraction and concentration procedures involve several discreet batch operations, and are either time-consuming or labor intensive. Thus automated continuous sampling systems are needed.

In the realm of continuous, on-line extraction procedures, membrane extraction is one of the most promising techniques. It is simple, inexpensive, requires small solvent volumes and offers high enrichment. It allows on-line extraction, and has been coupled to gas chromatography, high performance liquid chromatography, mass spectrometry, GC-MS and other analytical instruments [15-26]. Liquid phase membrane extraction can be classified into supported liquid membrane extraction (SLME) and liquid-liquid membrane extraction (LLME) [14]. SLME is a three-phase system in which the analytes are extracted from an aqueous sample into an acceptor phase via an organic extractant held in the pores of the membrane by capillary force. It is suitable for analyzing highly polar and ionizable compounds. LLME is a two-phase system where the analytes are extracted from an aqueous sample to an organic acceptor. The extraction occurs across a membrane, so that the two phases contact through the membrane pores without direct mixing. LLME can be used in any application as long as the compounds can be extracted into an organic solvent [24, 61, 63]. The driving force in LLME is the partition of analytes between the aqueous phase and the organic acceptor. The presence of the membrane in LLME prevents

emulsion formation, and other complex phenomena due to the physicochemical instability of the organic-aqueous interface, which occurs when the two phases are directly contacted, such as in LLE.

In membrane pervaporation, a liquid mixture contacts a membrane, the volatile species selectively permeate through, and are removed by a vacuum or an inert stripping gas. It has been used in the analysis of volatile organics by selectively stripping them from an aqueous medium [93], and for solvent removal in various industrial applications. In this paper analytical-scale membrane pervaporation was carried out continuously for the removal of solvent from the membrane extraction step. The extract is passed through the lumens of hollow fiber membranes while a counter-current inert gas selectively removes the solvent, resulting in the enrichment of the analytes in the lumens. Temperature is one of the important variables in membrane pervaporation and the temperature effect has been studied in previous research [94], thus it was not investigated in this study.

The objective of this study is to develop an automated and simple TAS by interfacing LLME, membrane pervaporation and HPLC-UV detection. These steps will perform extraction, concentration and detection respectively. The automated TAS will have the capability of continuous on-line monitoring of trace analytes in water.

6.2 Experimental Section

6.2.1 Experimental System

The experimental system is shown in Figure 6.1. It included two hollow fiber membrane modules, two pumps (Hewlett-Packard 1050 HPLC pump) and a HPLC system (Hewlett-Packard 1050). The first pump was used for the delivery of the organic extractant,

and the other for the water sample. An automatic six-port injection valve (Valco Instruments Co. Inc., Houston TX, USA) was used to make repeat injections into the HPLC. The two membrane modules were structurally similar. Hollow fiber membranes were selected because they provided higher surface area per unit volume. The modules were made in the shell and tube format. The first one served as the extraction module, and the latter as the pervaporation module. Water sample flowed through the shell side of the extraction module while the organic extractant flowed inside the hollow fiber lumens. The target analytes from the aqueous sample were extracted into the organic solvent in the membrane pores and then into the acceptor phase in the lumens. The extract continued to flow through the membrane lumen of the pervaporation module, where the nitrogen stripping gas flowed counter-current on the shell side. The evaporation of the solvent into the nitrogen flow concentrated the extract. The enriched extract was injected directly into the HPLC for analysis. The injection volume was 20 μL, and the injections were made automatically by a timer controlled six-port injection valve every five minutes.

6.2.2 Membrane Module Construction

The hollow fiber membrane modules were made with six pieces of composite membrane fibers packed in a Teflon tube. The length of the membrane used in the extraction and pervaporation modules were 128 and 144 cm respectively. These could be rolled into 3-4" diameter spools. Each end of the Teflon tubing was connected to a tee union (Supelco Inc., PA, USA). Epoxy (Resin Technology Group, LLC, S. Easton, MA, USA) was used to seal the space between the membranes and the tee, thus preventing the mixing of the water sample and the organic extractant. The hollow fiber membrane used in this study was a composite membrane with 0.260 mm O.D. and 0.206 mm I.D. (Applied Membrane

Technology, Minnetonka, MN, USA). It consisted of a 1 micron thick homogeneous siloxane as the active layer deposited on micro-porous polypropylene tubing as the support.

6.2.3 Reagents

The model analytes used in this study were naphthalene and biphenyl, which were purchased from Aldrich Chemicals (Milwaukee, WI, USA). HPLC grade n-hexane (Fisher Scientific, NJ, USA) was selected as the organic extractant. Prior experiments in the laboratory have shown good pervaporation of n-hexane through non-polar membranes [57]. All other chemicals in this study were ACS grade reagents. Deionized water was obtained from a Milli-Q® water purification system (Millipore Co., Bedford, MA, USA).

6.2.4 Chromatographic Separation

Chromatographic separation was carried out by a Hewlett-Packard 1050 HPLC with a Waters 486 Tunable Absorbance UV detector. The HPLC column was a 15 cm long Nova-Pack (Waters, Medford, MA, USA) C_{18} column with an I.D. of 3.9 mm. A mixture of 45:55 (v/v) Acetonitrile-10mM K_3PO_4 solutions was used as the mobile phase at a flow rate of 2 mL/min. The absorbance wavelength was set at 254 nm. Minichrom V 1.62 software (VG Data System) was used for data acquisition.

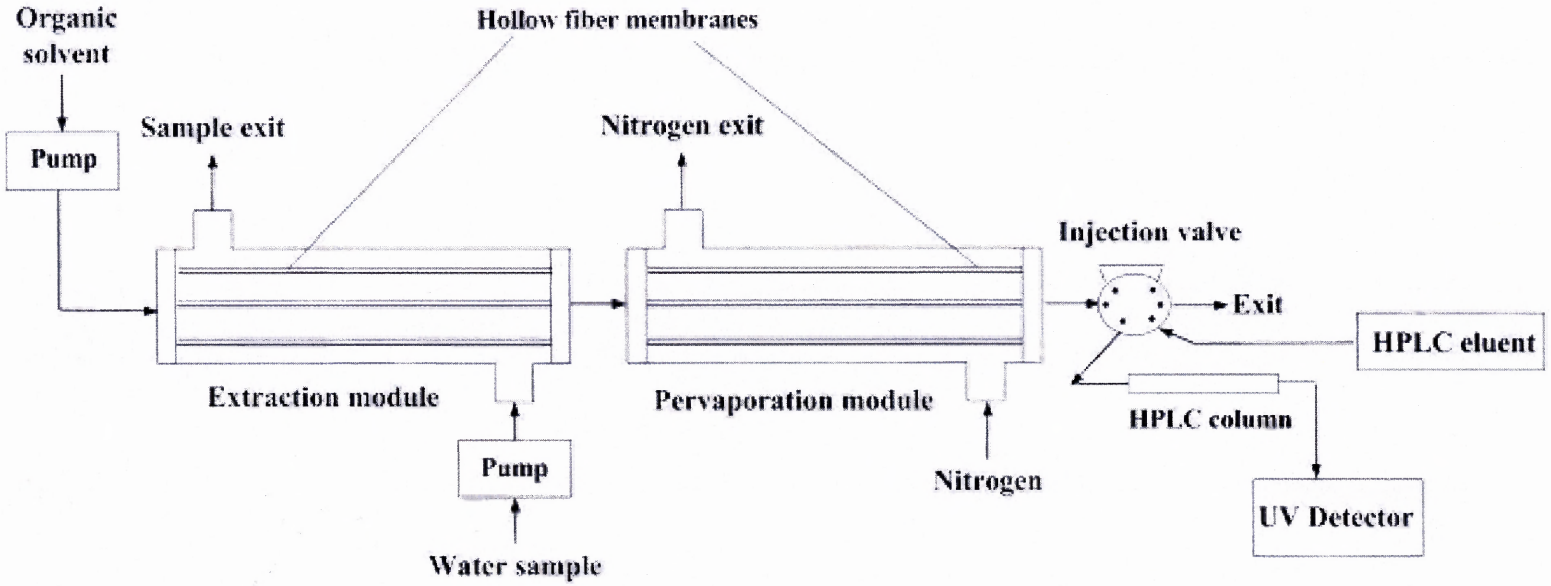


Figure 6.1 On-line interfacing membrane extraction, pervaporation and HPLC-UV detection into a TAS.

6.2.5 Three Experimental Modes

The experiments were carried out in three different experimental modes, namely, 1, 2 and 3. In Mode 1, only the membrane extraction module was used. In Mode 2, both extraction and pervaporation modules were used, but without the flow of stripping gas in the pervaporation step. This limited the solvent loss in the pervaporation step. In Mode 3, the membrane extraction was followed by membrane pervaporation with a flow of stripping gas, which resulted in a large solvent loss and led to a high degree of enrichment. Mode 2 and 3 are referred as the TAS modes. Any prior memory effect in the membranes was eliminated by flowing/washing the hollow fiber membranes with n-hexane for 2 minutes.

6.3 Results and Discussion

Figure 6.2 shows the concept of on-line interfacing of membrane extraction and pervaporation. The organic solvent, which is also referred to the acceptor, flowed inside of the lumens of the hollow fiber membrane extractor, while the water sample (donor) flowed counter-current on the shell side. Analytes were extracted into the organic solvent. Some of the solvent could be lost by dissolving in water, thus concentrating the extract. It has been reported that this preconcentration approach was possible with polar, water-soluble solvents such as butyl acetate and isopropyl acetate, but not with non-polar solvents such as hexane [61]. In this study, pervaporation was used for analyte preconcentration. This approach is more universal and should work with all solvents with reasonable volatility. As the extract flowed in, the stripping gas selectively removed some of the solvent, resulting in a more concentrated extract for HPLC analysis. In general, the combination of the two

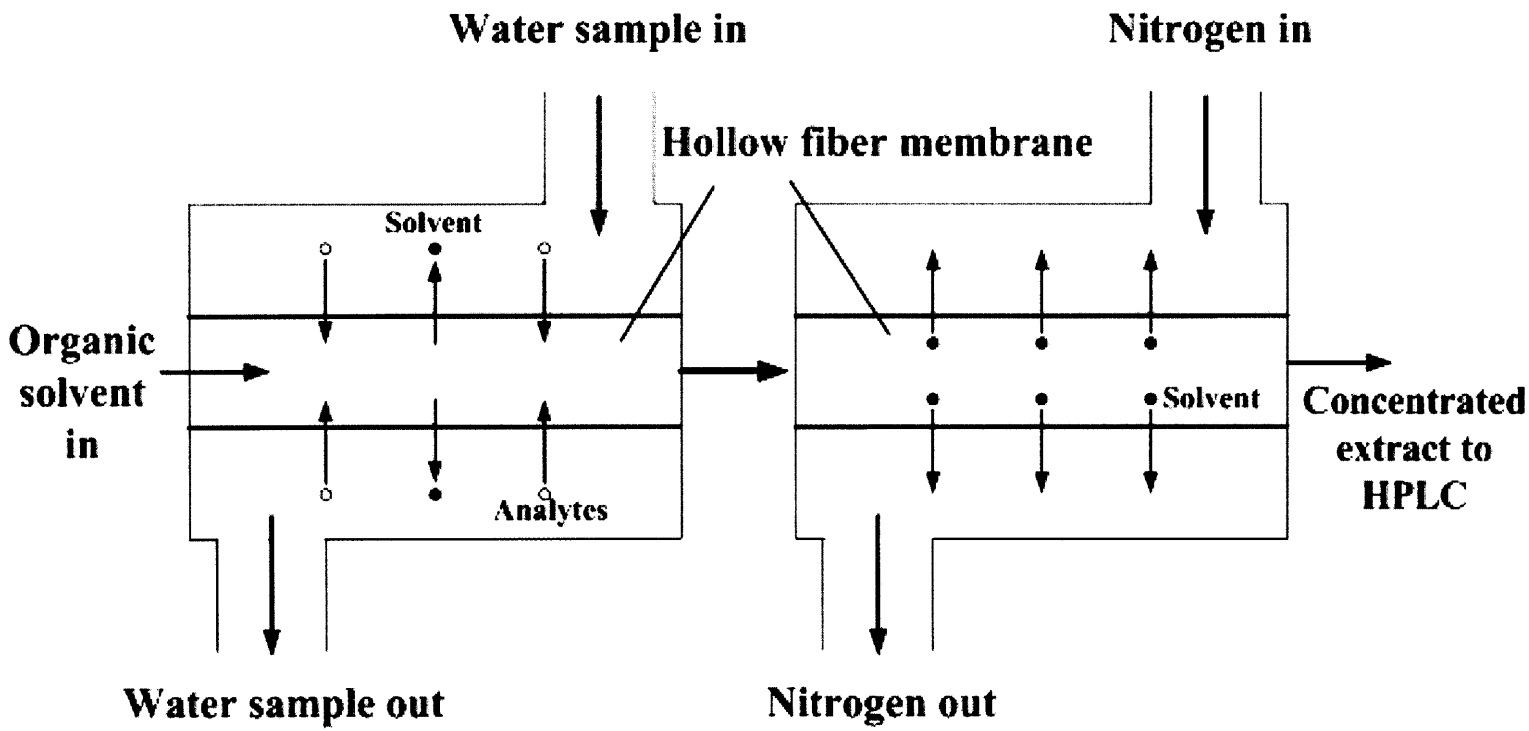


Figure 6.2 The concept of membrane extraction and pervaporation.

membrane modules allowed both extraction and concentration to be carried out on-line, and continuously.

6.3.1 Extraction Efficiency and Enrichment Factor

Enrichment factor (EF) is defined as the ratio of analyte concentration in the final extract to that in the original water sample:

$$EF = \frac{C_s}{C_w} \tag{6.1}$$

C_s is the analyte concentration in the final extract, and C_w is the analyte concentration in the original water sample. A higher EF leads to a higher sensitivity and a lower detection limit. Extraction is usually quantified as extraction efficiency (EE), which is the fraction of analytes removed by the acceptor from the original water sample [14]. EE is computed as:

$$EE = \frac{n_s}{n_w} = \frac{C_s \times V_s}{C_w \times V_w} = EF \times \frac{V_s}{V_w} \tag{6.2}$$

n_s and n_w are the analyte mass in the final extract and in the original water sample, V_s and V_w are the volume of the concentrated extract and the original water sample respectively.

In the continuous flowing system, the volume of the water entering in time t is expressed as:

$$V_w = F_w \times t \tag{6.3}$$

where, F_w is the water flow rate. The volume of organic solvent is different throughout the system, as a portion of the solvent is lost during the experiment. It is expressed as:

$$V_s = V_{si} - V_{ls} = V_{si} - V_{si} \times L_s = V_{si} \times (1 - L_s) \tag{6.4}$$

V_{si} is the initial volume of the organic solvent entering the extraction module while V_{ls} is the solvent lost during the extraction and pervaporation, and L_s is the fraction of the solvent lost. The volume of the organic solvent entering the extraction module over time t is given

as:

$$V_{si} = F_s \times t \tag{6.5}$$

where, F_s is the flow rate of the entering organic solvent. According to equation (6.4) and (6.5) the fraction of the solvent lost can be expressed as:

$$L_s = 1 - \frac{V_s}{F_s \times t} \tag{6.6}$$

Based on equations (6.2), (6.3), (6.4), and (6.5) EE is expressed as:

$$EE = EF \times (1 - L_s) \times \frac{F_s}{F_w} \tag{6.7}$$

Thus, EE can be computed from the enrichment factor, flow rates of solvent and water sample, and the fraction of the solvent lost.

6.3.2 Comparison of the EF in the Three Experimental Modes

Three different experimental modes (Mode 1, 2 and 3, which were described in Section 2.5) were compared. The donor sample used was 0.5 μg/mL naphthalene and 0.1 μg/mL biphenyl in water. The water sample flow rate was 5 mL/min. The organic solvent (n-hexane) flowed at 0.1 mL/min. The flow rate of the stripping nitrogen in the pervaporation module in Mode 3 was 45 mL/min. The results are shown in Figure 6.3. The EF somewhat increased (less than 2 times) when the pervaporation without nitrogen stripping was coupled to membrane extraction module. The enrichment factor increased significantly (4 to 9 times) in the presence of nitrogen stripping in the pervaporation module. Enrichment factors for naphthalene and biphenyl in Mode 3 were 93 and 188 respectively. The advantage of the TAS is clearly evident.

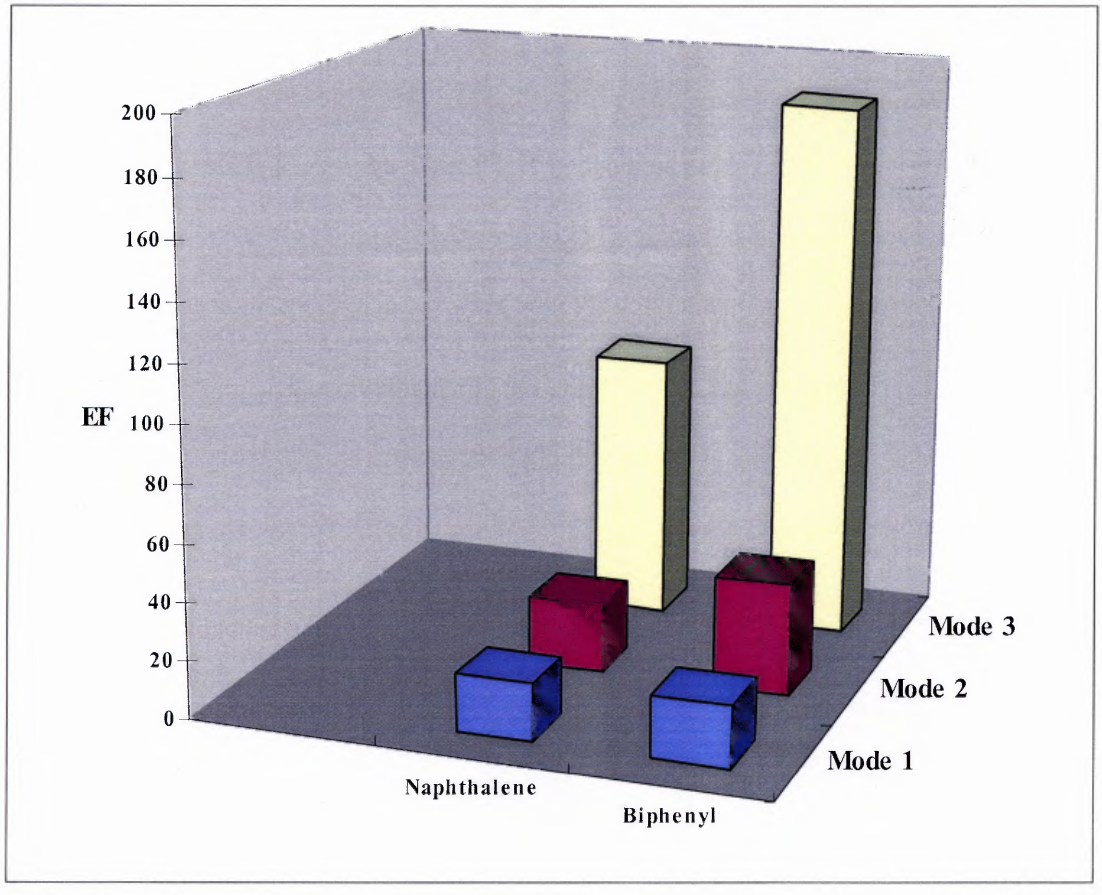


Figure 6.3 Comparison of EF in the three experimental modes. Mode 1:membrane extraction only; Mode 2: membrane extraction and pervaporation without N_2 stripping; Mode 3: and membrane extraction and pervaporation with a N_2 flow rate of 45 mL/min. In each case the sample contained 0.5 μg/mL naphthalene (Nap) and 0.1 μg/mL biphenyl (Bph), the water flow rate was 5 mL/min, and the extractant (n-hexane) flow rate was 0.1 mL/min.

6.3.3 Influence of the Water Sample Flow Rate

The water sample flow rate played an important role in the determination of enrichment factor, extraction efficiency and solvent loss. The experiment was performed in mode 2, the hexane flow rate was kept constant at 0.1 mL/min, while the water flow was changed from 1 to 5 mL/min. The concentration of naphthalene and biphenyl in the water sample was 0.1 μg/mL. The extract volume at the outlet of the pervaporation module was

measured for time t, and the solvent loss was calculated using equation 6.6. The results are shown in Figure 6.4.

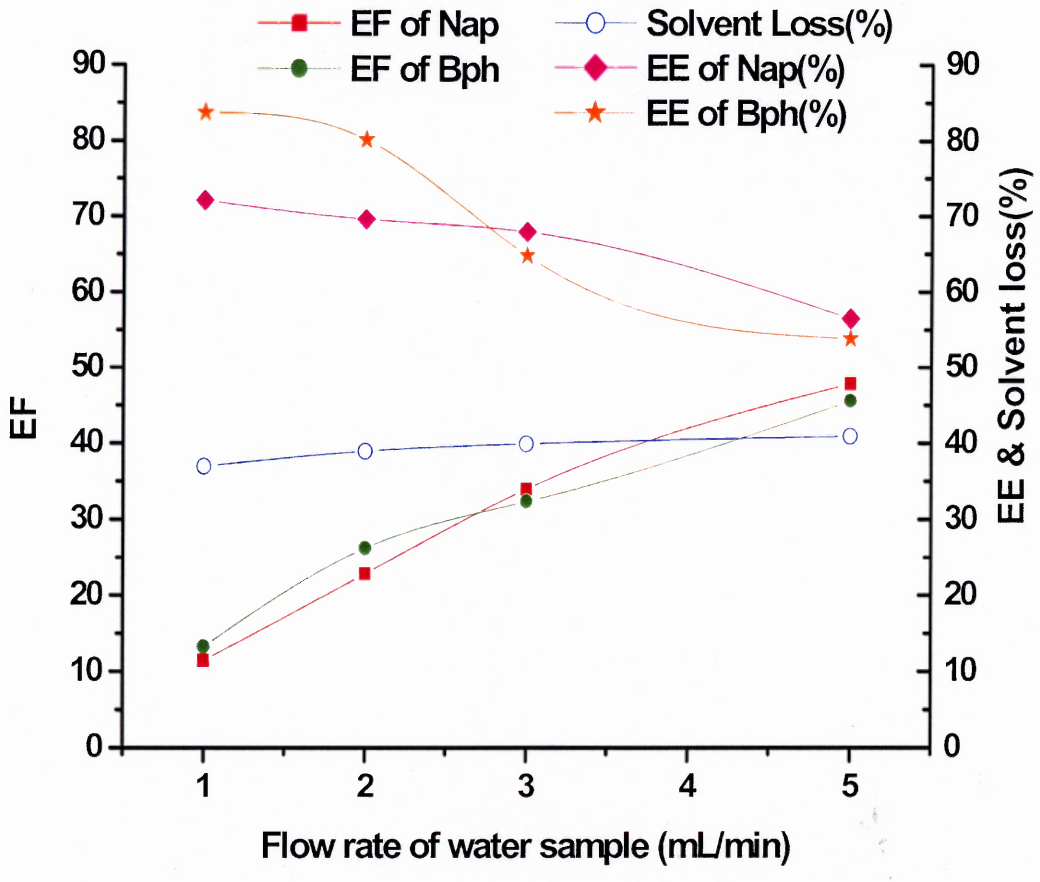


Figure 6.4 EF, EE (%) & Solvent loss (%) as a function of flow rate of the water sample (0.1 µg/mL Nap and Bph). The extractant flowed at 0.1 mL/min. This was operated in Mode 2.

The EF of naphthalene (Nap) and biphenyl (Bph) increased four times as the water flow increased from 1 to 5 mL/min. With an increased water flow rate, more analytes contacted the membrane, which led to a higher EF. The solvent loss increased slightly with the increase in water flow rate. The extraction efficiency decreased with the increase of

water flow rate. Although more analytes were brought in, a larger fraction went unextracted. Higher flow rates were not employed as the high pressure across the membrane could decrease the lifetime of the extraction module. A donor flow rate of 4 mL/min was selected as a compromise between high enrichment and long lifetime.

The organic solvent in the extract could be lost either by dissolution into the aqueous phase or evaporation in the pervaporation module. The poor water solubility of n-hexane and no stripping gas in the pervaporation module resulted in relatively low solvent loss in Mode 2.

6.3.4 Influence of Nitrogen Flow Rate in the Pervaporation Module

The effect of the nitrogen flow in the pervaporation module was tested in mode 3, by varying the nitrogen flow rate from 10 to 60 mL/min. The inlet concentration of naphthalene and biphenyl was 0.1 μg/mL. The water and the extraction solvent flow rates were 4 and 0.1 mL/min respectively. EF, EE and solvent loss as a function of N_2 flow rate are shown in Figure 6.5. The EF increased with the N_2 flow rate, which was attributed to higher solvent removal. The EE decreased with the increase in N_2 flow rate as some analyte molecules were lost with the solvent in the pervaporation step. The goal of this study was to achieve higher enrichment, thus nitrogen flow rate of 60 mL/min was selected in the following study.

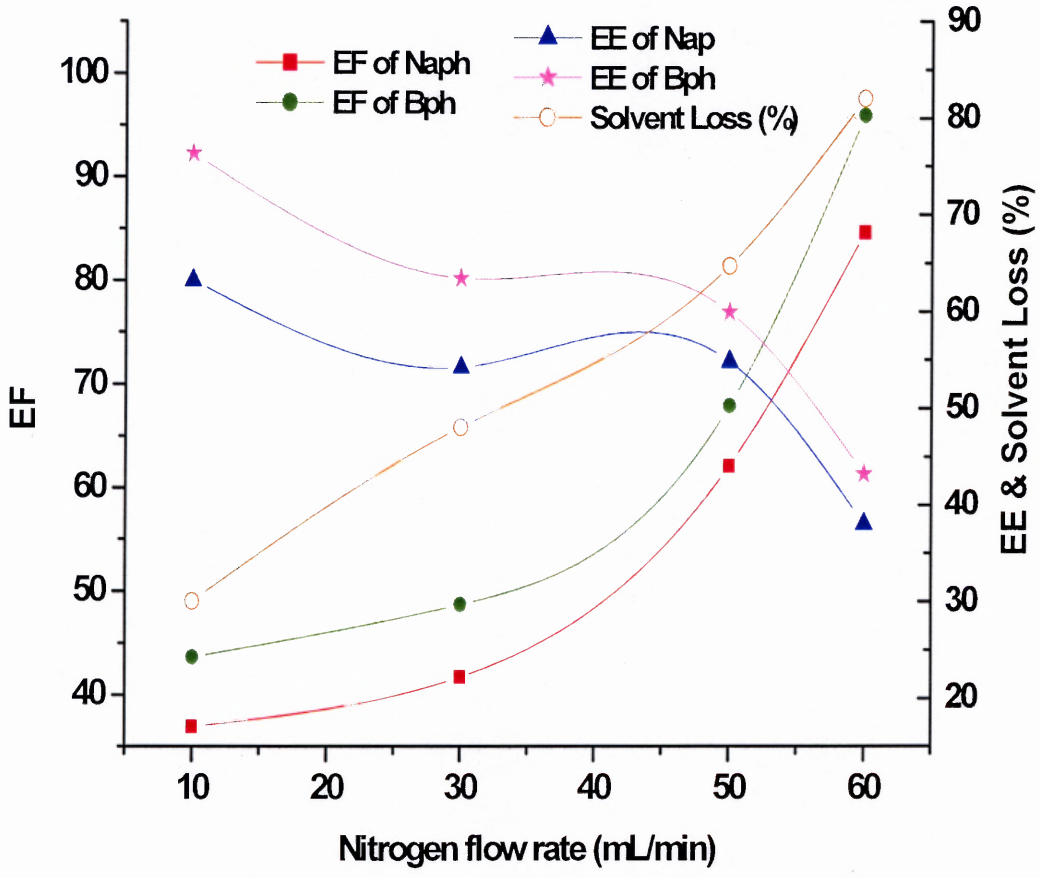


Figure 6.5 EF, EE (%) and solvent loss (%) as a function of N_2 flow rate. Water sample containing 0.1 μg/mL Nap and Bph flowed at 4 mL/min. The extractant flow rate was 0.1 mL/min. This was operated in Mode 3.

6.3.5 Influence of the Acceptor Flow Rate

The effect of the organic solvent flow rate on EF was tested by operating Mode 3. The nitrogen flow rate was 60 mL/min. The sample contained 0.1 μg/mL each of naphthalene and biphenyl in water. The water flow rate was constant at 4 ml/min, while the extractant (n-hexane) flow rate was varied from 0.075 to 0.25 mL/min. The whole process was carried out on-line and the extract was injected into the HPLC every five minutes. The results are shown in Figure 6.6.

EF decreased with the increase of the extractant flow rate. At lower flow rate, the contact time of the analytes with the solvent increased, thus more analytes could be extracted. In the pervaporation module also, the lower flow rate led to higher solvent removal, leading to higher EF. At flow rates lower than 0.075 mL/min, all the solvent was lost in the pervaporation module and no extract could be obtained for analysis. Therefore 0.075 mL/min was chosen as the acceptor flow rate. Enrichment factors as high as 192 were obtained under these conditions.

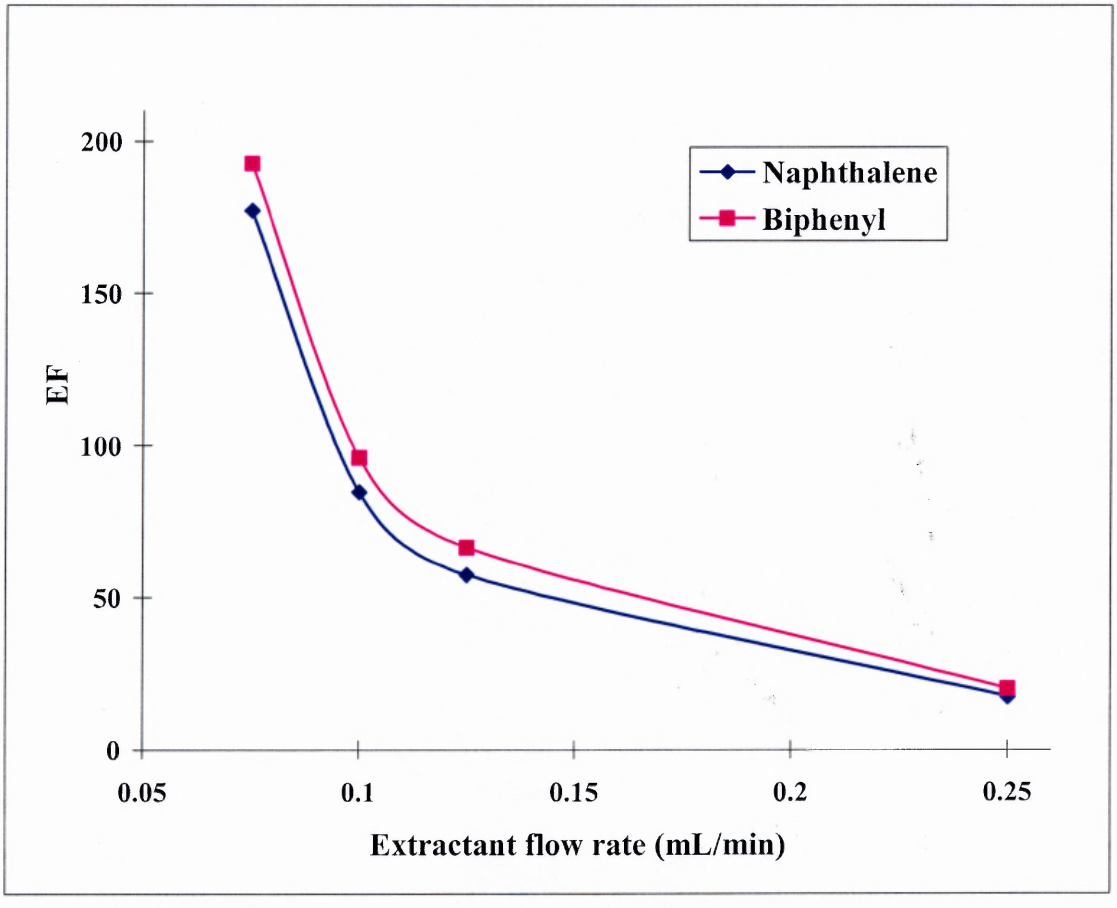


Figure 6.6 EF as a function of the extractant flow rate. The sample containing 0.1 μg/mL Nap and Bph flowed at 4ml/min. The N_2 flow rate was 60 mL/min. This was operated in Mode 3.

6.3.6 Analytical Performance

Different analyte concentrations were assayed in Mode 3. The flow rates of the water sample, the organic solvent and the stripping nitrogen were 4 ml/min, 0.075 mL/min and 60 mL/min respectively. The linear dynamic ranges were found to be 25-100 and 5-100 ng/mL for naphthalene and biphenyl respectively, with correlation coefficient (R^2) above 0.998.

Continuous automated monitoring was carried out at a relatively high frequency. The analytes were extracted into the organic solvent (n-hexane) and the solvent was continuously removed from the extract by pervaporation. The enriched extract was injected into the HPLC every five minutes by the automatic six-port injection valve. The sequence of chromatograms obtained during continuous on-line operation is shown in Figure 6.7. The concentration of naphthalene and biphenyl in the water sample was 50 ng/mL. Good reproducibility in peak shape and retention time was observed. The relative standard deviations (RSD) of naphthalene and biphenyl were 3.7% and 5.0% based on seven replicates, measured over three different days. The method detection limits (MDLs), which were obtained following a standard EPA procedure [113], were 5.8 and 7.9 ng/mL for naphthalene and biphenyl respectively.

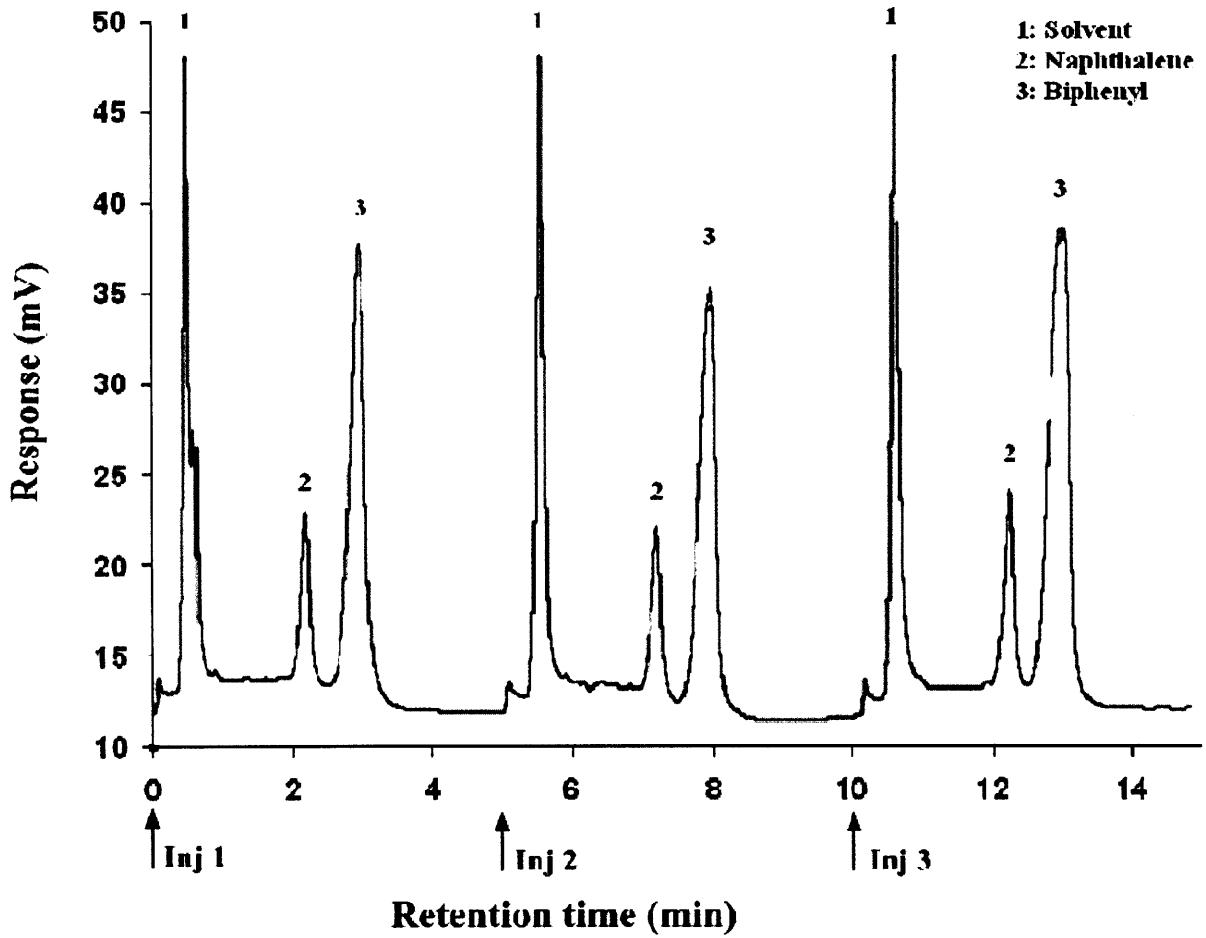


Figure 6.7 Continuous monitoring of a water sample containing 50 ng/mL Nap and Bph. The flow rates of the water sample, the extractant and the N_2 were 4, 0.075 and 60 mL/min respectively. Automatic injections were made every 5 min at Inj 1, Inj 2 and Inj 3.

6.4 Conclusion

Interfacing on-line membrane extraction, pervaporation and HPLC-UV detection led to the development of a relatively simple and an effective TAS for the on-line monitoring of trace semi-volatile/non-volatile organic compounds. Membrane extraction served as the separation and the initial enrichment step, while pervaporation was used as the final enrichment step. This system demonstrated an enrichment factor as high as 192 and good analytical performance during the continuous monitoring. The important operational variables were the different flow rates, namely, those of the water sample, the solvent and the stripping gas.

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