VTT PUBLICATIONS 364

Method development in membrane inlet mass spectrometry. Air analysis and desorption techniques

Raimo Ketola

VTT Chemical Technology

Laboratory of Organic Chemistry, Department of Chemistry

Faculty of Science

University of Helsinki

ACADEMIC DISSERTATION

To be presented, with the permission of the Faculty of Science of the University of Helsinki, for public criticism in Auditorium of the Department of Chemistry, A.I. Virtasen aukio 1, Helsinki, on the 17th of October 1998.

TECHNICAL RESEARCH CENTRE OF FINLAND
ESPOO 1998

ISBN 951–38–5341–1 (soft back ed.)
ISSN 1235–0621 (soft back ed.)

JULKAISIJA – UTGIVARE – PUBLISHER

Valtion teknillinen tutkimuskeskus (VTT), Vuorimiehentie 5, PL 2000, 02044 VTT
puh. vaihde (09) 4561, faksi 456 4374

Statens tekniska forskningscentral (VTT), Bergsmansvägen 5, PB 2000, 02044 VTT
tel. växel (09) 4561, fax 456 4374

Technical Research Centre of Finland (VTT), Vuorimiehentie 5, P.O.Box 2000,
FIN–02044 VTT, Finland
phone internat. + 358 9 4561, fax + 358 9 456 4374

VTT Kemiantekniikka, Prosessitekniikka, Biologinkuja 7, PL 1401, 02044 VTT
puh. vaihde (09) 4561, faksi (09) 460 041

VTT Kemiteknik, Processteknik, Otsvängen 3 A, PB 1401, 02044 VTT
tel. växel (09) 4561, fax (09) 460 041

VTT Chemical Technology, Process Technology, Biologinkuja 7, P.O.Box 1401,
FIN–02044 VTT, Finland
phone internat. + 358 9 4561, fax + 358 9 460 041

Technical editing Leena Ukskoski

Oy Edita Ab, Espoo 1998

Ketola, Raimo. Method development in membrane inlet mass spectrometry. Air Analysis and desorption techniques. Espoo 1998, Technical Research Centre of Finland, VTT Publications 364. 90 p. + app. 46 p.

Keywords mass spectrometry, membrane inlet mass spectrometry, volatile organic compounds, semivolatile organic compounds, desorption, air, water

Abstract

Membrane inlet mass spectrometry (MIMS) is an established technique for the analysis of volatile organic compounds in aqueous solutions and in air. A thin membrane is the only interface between a liquid or gaseous sample at atmospheric pressure and the vacuum of a mass spectrometer. Since its introduction about 35 years ago MIMS has been applied mainly in biochemistry and environmental analysis.

In this work the applicability of MIMS in the analysis of volatile organic compounds (VOCs) in water samples was investigated, and improved MIMS methods for the analysis of VOCs in air samples were constructed. The possibilities of MIMS for the analysis of polar and/or semivolatile compounds in aqueous samples were enhanced with novel techniques.

It was demonstrated that the MIMS method is comparable with static headspace gas chromatography and purge&trap gas chromatography-mass spectrometry methods in the analysis of VOCs in water samples. The MIMS method was also shown to be very suitable for on-site measurement of water samples in a mobile laboratory.

A membrane inlet mass spectrometric method was developed for the analysis of volatile organic compounds, especially volatile sulfur compounds, in air samples. The method is very sensitive, i.e. detection limits are at sub or low $\mu g/m^3$ levels, and also very rapid: it is possible to analyze even 50 to 100 samples in one hour with a thin polydimethylsiloxane membrane because response times are only a few seconds. When MIMS is combined with a temperature-programmed desorption (TPD) technique it is possible to achieve

separation of compounds prior to mass spectrometric detection, still conserving a rapid analysis time per sample, 6–10 minutes, and low detection limits.

For the analysis of semivolatile and/or polar compounds in aqueous samples two trap&release (T&R) techniques were developed. In these techniques semivolatile compounds accumulated into a membrane are desorbed by heat from a filament and then analyzed by a mass spectrometer. In the traditional T&R-method a silicone membrane is used together with electron ionization, whereas in the desorption chemical ionization (DCI) method a hydrophilic membrane is used to allow a solvent chemical ionization with water as a reagent gas. With these techniques it is possible to measure e.g. caffeine and dicarboxylic acids directly from water samples.

Foreword

This work was carried out at VTT Chemical Technology, Espoo, Finland during the years 1994–1998 and at the Department of Biochemistry, Odense University, Odense, Denmark during the years 1997–1998.

I am most grateful to my supervisors Professor Risto Kostiainen, Docent Tapio Kotiaho and Professor Frants Lauritsen for their invaluable guidance and support throughout this work. I also thank my colleagues in this research project, Marja Ojala, Vesa Virkki, Harri Sorsa, Jouni Juujärvi, Taina Pyökäri, Tapio Honkanen, Timo Mansikka, Ismo Mattila, Thomas Graf and Søren Knudsen for their co-operation. I also thank the staff at VTT Chemical Technology and Odense University for their helpful assistance.

I wish to thank Professor Hans Degn and Professor Pirjo Vainiotalo for agreeing to review this manuscript and for their very valuable contributions. I am also very grateful to Research Directors Markku Auer, Veikko Komppa and Arto Kiviranta for providing me the opportunity to work in this research project. Specific thanks go to Mr. Michael Bailey for revising the language. I also thank Professor Tapio Hase, University of Helsinki, for the opportunity to do this work for the Department of Organic Chemistry.

I warmly thank my family for their support.

Financial support of Maj and Tor Nessling Foundation, IVO Foundation, Danish Research Academy, VTT Chemical Technology, Technology Development Center (TEKES) and Environics Oy is gratefully acknowledged.

Helsinki, September 1998

Raimo Ketola

List of publications

This thesis is based on the following publications, which are referred to by their Roman numerals:

I Ketola, R.A., Virkki, V.T., Ojala, M., Komppa, V., Kotiaho, T. Comparison of different methods for the determination of volatile organic compounds in water samples. *Talanta* **44** (1997), pp. 373–382.

II Virkki, V.T., Ketola, R.A., Ojala, M., Kotiaho, T., Komppa, V., Grove, A., Facchetti, S. On-site environmental analysis by membrane inlet mass spectrometry. *Anal. Chem.* **67** (1995), pp. 1421–1425.

III Ketola, R.A., Ojala, M., Sorsa, H., Kotiaho, T., Kostiainen, R.K. Development of membrane inlet mass spectrometric method for analysis of air samples. *Anal. Chim. Acta* **349** (1997), pp. 359–365.

IV Ketola, R.A., Mansikka, T., Ojala, M., Kotiaho, T., Kostiainen, R. Analysis of volatile organic sulfur compounds in air by membrane inlet mass spectrometry. *Anal. Chem*. **69** (1997), pp. 4536–4539.

V Ketola, R.A., Grøn, C., Lauritsen, F.R. Temperature-programmed desorption for membrane inlet mass spectrometry. *Rapid Commun. Mass Spectrom.* **12** (1998), pp. 773–778.

VI Lauritsen, F.R., Ketola, R.A. Quantitative determination of semivolatile organic compounds in solution using trap-and-release membrane inlet mass spectrometry. *Anal. Chem*. **69** (1997), pp. 4917–4922.

VII Ketola, R.A., Lauritsen, F.R. Detection of dicarboxylic acids in aqueous samples using membrane inlet mass spectrometry with desorption chemical ionization. *Anal. Chem.*, submitted.

Abbreviations

API-MS/MS	atmospheric pressure ionization tandem mass spectrometry
ASGDI	atmospheric-sampling glow-discharge ionization
CI	chemical ionization
CT-MIMS	cryotrap membrane inlet mass spectrometry
DCI-MIMS	desorption or direct chemical ionization/membrane inlet mass spectrometry
DDT	1,1,1-trichloro-2,2-bis(*p*-chlorophenyl)ethane
DL	detection limit
DMDS	dimethyl disulfide
DMS	dimethyl sulfide
DMSO	dimethylsulfoxide
EI	electron ionization
EMS	ethylmethyl sulfide
FIA	flow injection analysis
FTIR	Fourier transform infrared
GC-MS	gas chromatography/mass spectrometry
HPLC	high performance liquid chromatography
HPLC-MS	high performance liquid chromatography/mass spectrometry
HSGC	static headspace gas chromatography
i.d.	inner diameter

LDR	linear dynamic range
LLE	liquid-liquid extraction
MIMS	membrane inlet or introduction mass spectrometry
MS/MS	mass spectrometry/mass spectrometry or tandem mass spectrometry
N	number of theoretical plates
Nd	not detectable
o.d.	outer diameter
P&T-GC/MS	purge-and-trap gas chromatography/mass spectrometry
PAM-ECD	purge-and-membrane electron capture detection or detector
PAM-MS	purge-and-membrane mass spectrometry
PAN	polyacrylonitrile
PID	photoionization detector
ppb	parts-per-billion
ppm	parts-per-million
ppq	parts-per-quadrillion
ppt	parts-per-trillion
PVC	polyvinylchloride
PWHH	peak width at half height
R_S	resolution
RSD	relative standard deviation
SIM	selected ion monitoring

SPE	solid-phase extraction
SPME	solid-phase microextraction
SWIFT	stored wave form inverse Fourier transform
T&R-MIMS	trap-and-release/membrane inlet mass spectrometry
TMDA	thermal membrane desorption application
TPD-MIMS	temperature-programmed desorption/membrane inlet mass spectrometry
t_R	retention time
VOC	volatile organic compound
VOSC	volatile organic sulfur compound
w_i	bandwidth of a compound i

CONTENTS

APPENDICES of this publication are not included in the PDF version. Please order the printed version to get the complete publication (http://www.inf.vtt.fi/pdf/publications/1998)

1. Introduction

1.1 Theory of membrane inlet mass spectrometry

Membrane inlet mass spectrometry (MIMS) is an analytical technique in which a membrane is the only interface between a liquid or gaseous sample at atmospheric pressure and the vacuum of a mass spectrometer. The membrane, either non-porous or porous, creates a tight seal between the two sides. Through the membrane it is possible to achieve a rapid and selective transport of sample molecules into the vacuum. Typical membranes used are polyethylene and Teflon for gas analysis and polydimethylsiloxane for analysis of volatile organic compounds (VOCs). These membranes are hydrophobic and therefore strongly favor the transport of hydrophobic organic compounds as compared to water [1]. In the following studies polydimethylsiloxane membrane was mostly used and therefore only the theory of non-porous membranes is shortly presented here.

The transport of compounds through a non-porous membrane is a process, called pervaporation, involving three steps: sorption of sample molecules into the membrane, diffusion through the membrane, and desorption and evaporation of molecules into the vacuum. This process can be described by Fick's two equations for diffusion. The molecular flux, J(x,t) (mol/s cm^2), at depth x (cm) inside the membrane and time t (s) can be calculated from

$$J(x,t) = -D(c)\frac{\partial C(x,t)}{\partial x} \tag{1}$$

and the concentration, C(x,t) at depth x and time t from

$$\frac{\partial C(x,t)}{\partial t} + \frac{\partial}{\partial x}\left(-D(c)\frac{\partial C(x,t)}{\partial x}\right) = 0 \tag{2}$$

where D(c) is the concentration dependent diffusion coefficient (cm^2/s) and c is the concentration (mol/L). The operating parameters have been reported to have a significant effect on the responses and response times in the analysis of water samples. According to Eq.1 the dimensions of membranes (thickness and area) have a significant effect on the responses and the response times. This comes from the fact that Fick's diffusion equations (assuming that constants for solvation and diffusion are independent of partial pressure) can be expressed as

$$I_{SS} = ADS\left(\frac{P_s}{l}\right) \tag{3}$$

where I_{ss} is the steady-state flow through the membrane (mol/s), A is the membrane surface area (cm^2), D is diffusion coefficient (cm^2/s), S is the solubility constant (mol/torr cm^3), P_s is the vapor pressure of the analyte on the sample side of the membrane (torr) and l is the membrane thickness (cm). The diffusion coefficient D can be approximated from the response time $t_{(50)}$, which is the time required to achieve 50% steady-state permeation [1]:

$$D = 0.14 \text{ x } (l^2/t_{(50)}) \tag{4}$$

or from the response time $t_{(10\text{-}90)}$ which is the time required to achieve the signal from 10% to 90% of the steady state permeation [2]:

$$t_{(10\text{-}90)} = 0.237 \text{ x } l^2/D \tag{5}$$

From these equations it can be seen that the steady-state flux through the membrane is directly proportional to the product of the solubility constant and the diffusion constant, whereas the response time is inversely proportional to the diffusion constant but independent of the solubility constant. For most non-polar compounds the solubility increases and the diffusivity decreases in a homologous series of samples as the carbon chain length increases [3]. This

means that a high molecular weight compound such as *n*-octanol has a low limit of detection but a long response time, whereas a low molecular weight compound such as ethanol has a high limit of detection but a short response time [4]. From Eqs. (3) and (5) it can be seen that thickness of the membrane has a great effect on the response and the response time of a compound, because doubling the membrane thickness results in a fourfold increase in response time and a twofold reduction in sensitivity. However, the diffusion constant is not necessarily always constant, but it can depend on the concentration of the analyte or the composition of the matrix. In such cases the signal of one compound might influence that of another. This can occur when highly hydrophobic compounds are present in the sample at concentrations greater than 10 parts-per-million (ppm), or when hydrophilic compounds are present in the percentage range.

1.2 Applications and modifications of the MIMS method

1.2.1 Applications of the MIMS method

The first application of MIMS was presented in 1963 by Hoch and Kok [5], who used it for the measurement of O_2 and CO_2 during photosynthesis. Since its first introduction, MIMS has gained more interest every year among scientists, since the method is simple, fast, solvent-free, sensitive and well suited for on-line and real-time analysis. The main areas in which membrane inlet mass spectrometry has been exploited are biochemistry, especially fermentation monitoring, and environmental analysis.

Monitoring of reacting systems in aqueous solutions was introduced by Westover and Tou [6], who studied volatile organic compounds such as chloroform, hexane and methanol. Fermentation monitoring experiments were first reported by Reuss *et al.* [7] in 1975 and on-line feedback control of fermentation processes by Jørgensen and Degn [8] in 1987. The first applications of MIMS to the kinetics of biochemical reactions were reported by Calvo *et al.* [9, 10] and Degn and Kristensen [11]. In recent years many other biochemical applications of MIMS, especially monitoring of fermentation processes and reaction kinetics, have been reported [12–18] and the biochemical applications have also been reviewed [19–22]. Direct determination of

environmentally significant organic compounds from aqueous solutions by MIMS has become very popular in recent years [1, 23–37]. The applications of environmental analysis have also been thoroughly reviewed [2, 38–44].

1.2.2 Modifications of the MIMS method

MIMS was coupled with gas chromatography-mass spectrometry (GC-MS) as early as in the 1960s by Llewellyn and Littlejohn [45, 46]. Since then there have been different types of applications of MIMS together with GC-MS or gas chromatography (GC) [47]. Black *et al.* [48] constructed a membrane molecular separator for GC-MS interface. Melcher and Morabito [49] introduced an automated extraction analysis system, which combines membrane cell technology with a pneumatically operated pressurized rotary gas chromatographic injection valve. They used the system for the analysis of chlorinated aromatic compounds and pesticides in aqueous samples. Mitra *et al.* [50, 51] combined GC with an on-line membrane extraction system for continuous sampling of organic pollutants. They also added a microtrap for this system to concentrate the analytes permeated from the membrane extractor in order to achieve lower detection limits (low µg/L). Matz *et al.* [52, 53] developed a mobile mass spectrometer system for the on-line measurement of organic compounds in water. This system is based on a membrane separator which is combined with a GC or a GC-MS. MIMS has also been connected to high performance liquid chromatography/mass spectrometry (HPLC-MS), first by Jones and Yang [54] in 1975. Melcher *et al.* [55] developed a membrane interface for selective extraction and concentration of trace phenols in aqueous streams. The pH of the sample stream was adjusted to 2 to ensure that acidic phenols were absorbed into the membrane. The pH of the extractant stream on the other side of the membrane was adjusted to alkaline to selectively extract the phenols from the membrane. The extractant flow was then analyzed by HPLC. The membrane separator has also been connected on-line to HPLC [56, 57].

During the past few years there has been a great expansion of modifications of the standard MIMS method. Chemical ionization (CI) with tandem mass spectrometry was introduced to MIMS by Brodbelt and Cooks [58] in 1985. This combination is a very powerful tool for analyzing mixtures because with CI the spectrum of a mixture is simplified compared to an electron ionization (EI) spectrum but identification is enchanced by using mass spectrometry/mass

spectrometry (MS/MS). At early stages of MIMS the mass spectrometer was usually a simple quadrupole instrument but the membrane inlet can also be connected to an ion trap instrument [24]. With an ion trap MS/MS experiments are very easy to perform, and CI is also possible when using a silicone membrane because there is no need for high pressure in the ion trap for CI. Slivon *et al.* [26] introduced the idea of pneumatically assisted transport of the membrane permeate, the helium-purged MIMS. In this system the permeates are transported to the mass spectrometer with the help of a carrier gas, usually helium in the GC line. The direction of flow of the carrier gas is normally opposite to that of the sample stream. The carrier gas can be enriched further by a jet separator which removes most of the carrier gas and much of the water before the permeates reach the mass spectrometer [59]. In this way it is possible to obtain a two-stage enrichment for the analytes.

Degn and Kristensen [11] introduced a stopped flow MIMS in 1986 for direct measurements of CO_2 transients in the spontaneous and enzyme catalyzed hydrations of carbon dioxide. Using stopped flow MIMS they were able to determine rate constants and activation energies of these reactions at different temperatures. A very efficient method to reduce the amount of sample used is the flow injection analysis (FIA) technique [26, 60–62]. In the FIA-MIMS system the membrane is exposed to the sample only for a short period (10–20 s) and therefore the flow of the sample is interrupted by pure water before the steady-state permeation is reached. The height of the FIA peak can be used for quantitation because the total flux through the membrane at any time is linearly dependent on the sample concentration in the feed liquid. Due to short sampling times the FIA-MIMS technique is a very rapid method for analyzing organic compounds in aqueous solutions, and it can also be automated very easily.

The great majority of the MIMS studies are concerned with the measurement of environmentally or biologically important compounds from aqueous solutions. However, MIMS can also be used for measurement of dissolved compounds in organic solvents. This method is called reversed phase MIMS [54, 63–65]. It can be used e.g. for measurement of water activity in organic solution using a hydrophilic polyethylene terephthalate membrane which has high permeability for water but very low permeability for nonpolar organic compounds [64]. With this membrane a detection limit of 0.3 mg/L for water in octane was achieved. In addition to water, organic compounds as impurities in organic solvents can

also be measured by reversed phase MIMS [65]. When porous polypropylene membrane is used the flux of solvent is high enough for the vaporized solvent to be used as ionization gas in CI. The response times are short, about 10 seconds, but limits of detection (low or sub ppm) are higher than with non-porous silicone membrane because the enrichment process is missing. For example, limits of detection of 0.2 ppm and 0.15 ppm, respectively, were measured for methanol and acetone in hexane [65].

MIMS is a very sensitive technique, i.e. detection limits at ppq (parts-per-quadrillion) levels have been achieved by stored wave form inverse Fourier transform (SWIFT) and ion trap MS [66]. In this technique broad-band wave forms are created which are notched at the resonance frequencies of analyte ions of interest. A series of such pulses is applied during ionization to eject unwanted ions and store only analyte ions. This technique is used over a long ionization period to obtain very low detection limits at ppq levels. Another way to reach lower detection limits is to trap analytes to a cryotrap, followed by rapid heating of the trapped compounds, thus releasing them into the ion source of a quadrupole mass spectrometer [67]. An improvement factor of 100 is achieved by this cryotrap-MIMS (CT-MIMS) method compared to a standard MIMS method, and typical limits of detection for VOCs are 10–20 parts-per-trillion (ppt). A traditional purge-and-trap preconcentration technique has also been coupled with membrane inlet and GC-MS [68]. Analytes from a membrane inlet are swept into a solid adsorbent (Tenax), from which they are desorbed into a capillary column GC-MS system. Polar volatile organic compounds such as 2-propanol, 2-methyl-1-propanol and 1,4-dioxane are detected with this method at concentrations below 100 µg/L. The purging technique has also been applied in purge-and-membrane (PAM) methods, where the detector is either an electron capture detector (PAM-ECD) [69] or a mass spectrometer (PAM-MS) [70]. In these methods the sample (water or soil) is purged with an inert gas and the purged compounds are collected from the gas phase through a silicone membrane inlet to the analytical system. This method expands the applications of MIMS to soil samples which have not been measured before by MIMS. Detection limits are sub µg/L for water samples and at the low µg/kg level for soil samples. In sorption MIMS the analytes from the membrane inlet are adsorbed on a trap prior to the mass spectrometric detection [68, 71]. In all cases a relatively long trapping period combined with a rapid release of the analytes results in enrichment of the samples. A drawback of the sorption MIMS

techniques as compared to most other MIMS methods is the fact that real-time monitoring can no longer be conducted. Rivlin [71] introduced a system in which the trap was mounted in the vacuum between the membrane inlet and the ion source. A heating wire mounted inside the trapping material allowed the trapped sample to be thermally released into the mass spectrometer. Major drawbacks of this system were degradation of the sorbent material (Tenax) during the thermal desorption step and a considerable pressure increase inside the mass spectrometer due to the large amount of water trapped by the sorbent. The membrane itself can also be modified to trap organic molecules. In this method, called affinity MIMS, a chemically modified membrane is used to selectively adsorb analytes bearing a particular functional group and to concentrate them from solution [72]. Alkylamine-modified cellulose membrane was used to trap aldehydes at high pH as a result of imine formation. Release of the bound aldehyde was achieved by acid hydrolysis of the surface-bound imine at low pH. The results showed that e.g. benzaldehyde can be measured with excellent specificity at a concentration of 10 ppb.

The most common membrane material used in MIMS is polydimethylsiloxane. Other polymers have also been tested with MIMS to investigate whether they could be used instead of silicone. These materials include e.g. Teflon, PVC (polyvinylchloride), polyurethane, polyethylene and polyimide [73]. However, silicone is found to have the best overall performance in most situations. Some of the materials are more specific than silicone, e.g. polyurethane can be used for specific applications of fermentation monitoring. Similarly, liquid membranes are evaluated for use with MIMS [74, 75]. These liquids, such as Krytox (perfluorinated ether), polyphenyl ether and silicone oil, have low vapor pressure and can be used in the vacuum of the mass spectrometer. Usually their performance is not as good as that of silicone membrane but one of the advantages of liquid membranes is that they can formed to any desired thickness or shape. Another advantage is that they can easily be modified with different substrates to allow selective detection of compounds.

The first direct measurement of ions already present in a solution using membrane inlet mass spectrometry was published by Yakovlev *et al.* [76]. They used a strong electrical field to draw ions from a solution through a porous membrane and to stimulate their transport into the vacuum. They used polyethylene terephthalate membrane with small bores at high density. The

small bores prevented the liquid from flowing into the vacuum. In this manner they were able to detect nicotinic acid and acetylsalicylic acid in a glycerol/water matrix. Cisper and Hemberger [77] were able to detect metal-containing compounds using a membrane inlet with ion trap mass spectrometry. Ferrocene and molybdenum hexacarbonyl vapors were detected in air using a composite membrane (polydimethylsiloxane-polypropylene).

A normal drawback of MIMS methods is that all compounds are detected approximately at the same time. This means that the analysis of complex mixtures is very difficult if all the compounds in the sample permeate easily through the membrane. Several methods are proposed to achieve some sort of separation of compounds when samples are analyzed using MIMS. In order to get a maximum amount of data out of the measured multicomponent mass spectrum, different calculation methods have been used to identify and quantitate a range of compounds from one mass spectrum. Kotiaho *et al*. [78, 79] used a non-linear deconvolution algorithm for resolving a multicomponent mass spectrum. Ohorodnik *et al*. [80] coupled MIMS with multivariate calibration for the analysis of VOC mixtures. Overney and Enke [81, 82] used a modulated sample stream for mixture analysis in which the differences of diffusivities of different compounds can be used to resolve mixtures. Lauritsen *et al*. [83] also used differences in response times to obtain separation between different compounds. Cook *et al.* [84] exploited the time dependence of permeation of organic compounds through zeolite membranes to enhance mixture resolution.

1.3 Analysis of volatile and semivolatile organic compounds in air and/or aqueous samples

1.3.1 Volatile organic compounds in air

The quality of air in homes, urban areas, work places, etc. has become an important issue during recent years due to increased emissions and use of chemicals. The regulations for emissions have been made more stringent, since many pollutants have been found to be highly toxic. Reliable, sensitive, rapid and solvent-free analytical methods are needed for the control of air emissions. Volatile organic compounds (VOCs) can exist at very high concentrations in

indoor and outdoor air and therefore they are nowadays a very important environmental issue. A special class of VOCs is volatile organic sulfur compounds (VOSCs), which are released into the environment from both industrial and natural sources. The most important industrial sources are the pulp and paper industry, fossil fuels, solvent releases and waste dumping sites [85]. Of the natural sources the most important are oceans, soil, vegetation and volcanoes [86]. The most interesting volatile sulfur compound is dimethyl sulfide (DMS), produced by some marine algal species [87], since it is believed to be the principal sulfur carrier in the global sulfur cycle [88]. In terrestrial areas, DMS and methanethiol are mainly produced during the degradation of sulfur compounds of biological origin as, for example, the amino acids methionine and cysteine as well as their derivatives S-methylmethionine and S-methylcysteine [89]. DMS and many other organic sulfur compounds, such as thiols, sulfides and disulfides, are toxic and may cause health problems [90]. Furthermore, they can cause significant malodor problems even at low concentrations, since their odor threshold limit values are very low: 1.9–18 $\mu g/m^3$ for methanethiol, 1.7–100 $\mu g/m^3$ for DMS, 0.4–15 $\mu g/m^3$ for dimethyl disulfide (DMDS) and 33 $\mu g/m^3$ for carbon disulfide (CS_2) [89, 91].

1.3.2 Determination of volatile organic compounds in air by chromatographic methods

In order to achieve the required sensitivity level, preconcentration of air samples is often necessary before analysis of VOCs by conventional gas chromatographic (GC) and gas chromatography-mass spectrometric (GC-MS) methods. In the most frequently used methods the air samples are collected with adsorption tubes or passive samplers prior to chromatographic analysis. Denuder tubes, evacuated canisters or bubbling through a solvent have also been used in sampling. VOCs are thermally desorbed or extracted by solvents from adsorbents and are analyzed in a laboratory by gas chromatography (GC) or gas chromatography/mass spectrometry (GC-MS). Solid adsorbents are often used for preconcentration in connection with GC and GC-MS methods [51, 92–98]. In these systems the adsorbent trap is heated for several minutes at 200–300°C and the analytes are desorbed into a cryogenic trap prior to the analysis by GC or GC-MS [99]. Because of the preconcentration step in sampling, the method has low detection limits (at ppt (parts-per-trillion) levels) [96]. Various solid adsorbent materials have been used for trapping volatile organic compounds, and their properties have been thoroughly evaluated [99–103]. Stability and breakthrough volumes

of various adsorbents have also been measured on-line by MIMS [104]. Other preconcentration techniques used include chemisorption onto gold foil [105] and cryogenic trapping [106–110]. In some studies, volatile organic sulfur compounds (VOSCs) have also been analyzed after collecting air samples into gas canisters or gas bags [111]. In these studies detection limits for dimethyl sulfide at low ng/m^3 levels have been obtained [109, 110, 112–115]. These and many other methods of air analysis were reviewed by Fox [116] and Clement *et al.* [117].

However, both the trapping and the gas bag methods entail certain problems. The co-trapped water can often cause problems in the trapping methods, unless it is removed e.g. using a calcium chloride tube before the actual trapping step [118]. Degradation of solid adsorbents, such as Tenax TA, can interfere with the analysis of VOCs. [119] Substantial losses of highly volatile organic compounds may occur when using solid adsorbents [95]. A disadvantage of the conventional GC and GC-MS techniques is also the fact that they are in most cases fixed laboratory methods and the samples must be sent to a laboratory for the analysis. This can cause a decrease in the reliability of quantitative analysis due to loss of highly volatile analytes during sampling, transport and sample storage and due to variations in handling of samples in the laboratory [120]. Furthermore, many of the methods are time-consuming and do not offer sufficiently rapid analytical methods for reliable control of emissions. For these reasons on-line and real-time analytical methods are needed.

1.3.3 Analysis of air samples by MIMS

Although a large number of scientific papers concerning MIMS have been published, only a few of them have dealt with air analysis. The first applications of MIMS in air analysis were published already in the beginning of the 1970s [6, 121], but little attention was paid to air analysis until the 1990s. Recently LaPack *et al.* [1, 32] used a capillary MIMS in the analysis of VOCs in air. Hemberger *et al*. [122] described a two-stage membrane tube/jet separator combination to detect VOCs in air at ppt levels with an ion trap spectrometer. Gordon *et al.* [123] compared MS/MS measurements by MIMS to another direct air sampling method, atmospheric-sampling glow-discharge ionization (ASGDI). They achieved detection limits of low parts-per-billion in volume (ppbv) levels. Hemberger *et al.* [124, 125] reported sub ppbv detection limits in air using charge exchange ionization in conjunction with MIMS. An ion trap detector was

also used for real-time monitoring of volatile organic compounds in atmospheric samples in the low ppm or ppb ranges [126]. Lloyd *et al.* [127] constructed a membrane-covered gas miniprobe inlet for the direct simultaneous measurement of gas species. White *et al.* [128] developed a portable time-of-flight membrane inlet mass spectrometer for the analysis of environmental air samples. This instrument can be used either in one- or two-stage membrane inlet systems, and typical estimated detection limits are 2–3 ppbv.

1.3.4 Analysis of volatile organic compounds in aqueous samples by chromatographic methods

Static or dynamic headspace methods are the primary methods currently used to extract volatile organic compounds from water samples. These headspace methods rely on the establishment of equilibrium partitioning of an analyte between liquid and gas phases. In the static headspace method (HSGC), a water sample is placed in a headspace vial and an aliquot of the closed airspace above the water phase is sampled directly to a gas chromatographic column with split injection. Due to the high detection limits of the static headspace method, sample pre-treatment is often used, e.g. salting-out with sodium sulfate or chloride and adjustment of pH. In dynamic headspace methods, of which the purge-and-trap method (P&T-GC/MS) is the most common, the analytes are removed from the water phase by bubbling them with an inert gas such as helium or nitrogen, collecting them into an adsorbent trap, such as Tenax or activated charcoal, and desorbing them from the trap into a gas chromatographic column via a cold trap. The theory and construction of headspace methods have been reviewed by Koester *et al.* [129], Crompton [130], Soniassy *et al.* [131] and Poole *et al.* [132]. These techniques provide a clean sample, free from its matrix, and are best suited for the analysis of low molecular weight, slightly water-soluble volatile organic compounds. Theoretical considerations of the P&T-GC/MS method have been studied in detail by Pankow *et al.* [133–135] Both headspace methods can be automated by commercially available headspace autosamplers. Automation of the P&T-GC/MS method has been studied for rapid analysis of volatile compounds [136], and optimization of the parameters of the P&T-GC/MS [137] and HSGC methods [138] has also been studied.

Harland and Nicholson [30] compared the MIMS method with two purge-and-trap methods (GC-FID and GC-MS) in one comparison study in which six volatile halogenated hydrocarbons were analyzed from five environmental samples. The MIMS method was used in the selected ion monitoring mode. The concentration levels of the hydrocarbons were from less than 0.1 μg/L to 90 μg/L in the samples, and the analytical results were in good agreement. However, the comparison included only this intercomparison exercise and not a profound study of the characteristics of the methods.

1.3.5 Analysis of semivolatile and polar organic compounds in aqueous samples by chromatographic methods

Many different methods have been developed for the analysis of medium or less volatile organic compounds in water samples. A short overview of the most popular methods for chromatography and chromatography/mass spectrometry is presented in the following.

The most common technique for pretreatment of liquid samples for the analysis of semivolatile compounds by chromatography is liquid-liquid extraction (LLE) [139]. LLE is an equilibrium technique based on the distribution of a solute between two essentially immiscible solvents, usually an aqueous and an organic solvent. The benefits of the LLE technique are simplicity, low cost and the fact that the method is well known and understood. Considerable analyte enrichment can be achieved, e.g. concentration factors greater than 10 000 have been achieved using a micro-LLE procedure [140]. Furthermore, LLE is a very useful technique for eliminating interfering inorganic compounds present in aqueous samples. However, LLE is a typically non-selective pretreatment procedure. Although LLE is a versatile technique and requires only simple equipment, it is time-consuming, labor-intensive and not easily automated. For GC analysis, the analytes can be derivatized to increase their volatility but derivatization is also time-consuming and some of the analytes may be lost during the procedure.

If more selectivity and/or a higher sample enrichment is desired, solid-phase extraction (SPE) can be a good alternative to LLE. In SPE, the sample is loaded onto a porous packed bed. The trace-level analytes of interest, but also some less desirable sample constituents, accumulate on the sorbent while water is flushed to waste. After washing with water the analytes are desorbed by an organic

solvent, and the extract can be analyzed by GC, GC-MS or liquid chromatography (LC). Very large enrichment factors (typically 100–1 000) can be achieved by SPE. SPE can be performed off-line or on-line; off-line is simple and highly flexible, whereas on-line SPE provides the possibility of automation and a high sample throughput [141], but the greatest disadvantage is that the SPE technique is time-consuming.

Although solid-phase microextraction (SPME) has primarily been used for the determination of VOCs, it can also be applied for the determination of medium and less volatile compounds [142]. In SPME a fused silica fiber coated with a GC stationary phase such as polydimethylsiloxane or polyacrylate for polar compounds is inserted into a sample vial (either into the liquid, solid or headspace above the sample). The fiber coating is exposed to the sample for a predetermined time to extract analytes from the matrix. Once the sampling is completed, the fiber is directly transferred into a GC injector. Analytes are thermally desorbed from the coating and quantitatively analyzed by GC. SPME can be performed manually or by means of an autosampler. Because SPME is an equilibrium sampling technique, the limits of detection are higher than those of LLE and SPE, for example. The time to reach equilibrium depends on the nature of the analytes and ranges from 2 to 60 min. In the case of semivolatile compounds, the fiber coating must be inserted in the sample (not in the headspace above the sample), and general experience shows that, if reliable quantification is desired, the technique is only applicable to relatively pure samples [139].

High performance liquid chromatography (HPLC) is not only a powerful analytical method as such, but also allows effective sample preparation for GC. Semivolatile compounds can be directly analyzed by HPLC without any sample preparation and the analysis time can be very short, only a few minutes. The disadvantages are higher limits of detection than with extraction techniques (LLE, SPE) and difficulty of qualitative measurement of unknown samples unless the technique is used with mass spectrometry. HPLC can be combined with GC on-line for determination of some target compounds in highly complex matrices. This LC-GC method is highly specific but requires a complex instrument [143].

1.3.6 Analysis of semivolatile and polar organic compounds in aqueous samples by MIMS

Whereas the analysis of volatile organic compounds in aqueous samples has become routine for the MIMS system, the analysis of semivolatiles (boiling point above 250°C) has not. This is because the membrane inlets cannot be operated at temperatures much higher than 70°C before bubble formation in front of the membrane causes highly instable signals. At temperatures above 100°C the signal falls almost to baseline level because of the large volumetric expansion as water starts to boil [144]. The low inlet temperature limits the vaporization of the semivolatiles from the membrane surface and results in long membrane response times (> 5 minutes) for such compounds. Until recently, compounds with a boiling point between 200 and 300°C were best detected by the so-called direct insertion membrane probes (DIMP), in which the membrane is mounted inside [145] or in the immediate vicinity of the ionizing region [4, 144]. Using these inlets, problems with chromatographic effects on vacuum surfaces from the "cold" membrane surface to the ionizing region are almost eliminated. The capability of MIMS methods to measure polar compounds is also limited. The main reason for this is that the widely used polydimethylsiloxane membrane is hydrophobic and polar compounds do not easily diffuse through it at room temperature.

Recently, a completely new way of conducting the MIMS experiment, the so-called trap-and-release/MIMS (T&R-MIMS) [146] was introduced. In this method semivolatile organic compounds are preconcentrated inside the membrane before they are thermally released into the ion source by heat radiation from the filament. The system uses a standard membrane inlet with a silicone tube passing directly through the ion source. A long slit in the ion source parallel to both the tubular membrane and the filament allows heat radiation from the filament continuously to bombard the membrane surface. During a sampling period the membrane is kept cold by the sample liquid flowing through the inside of the silicone tube. However, during a short interruption of the liquid flow, the membrane is rapidly heated to more than 300°C and organic compounds dissolved in the membrane are released into the ion source. In this way a desorption peak is obtained. A similar system, a thermal membrane desorption application (TMDA), has been presented by the group of Matz [147, 148] for the on-line analysis of organics in water or

fermentation suspension by GC-MS. In the TMDA method a membrane separator is used to extract volatile organic compounds from the sample to direct analysis by GC-MS. Compounds which do not diffuse through the membrane during sampling but which are accumulated into the membrane, are then thermally desorbed from the membrane and transported to a GC-MS for the analysis. In this way it is possible to analyze both volatile and semivolatile compounds in one sampling. The major difference between TMDA and T&R-MIMS is that in T&R-MIMS the inlet is an integral part of the ion source, whereas TMDA forms a separate unit mounted at a short distance from the ion source. The physical principles of operation of the two systems are the same.

1.4 Aims of this work

The aims of this work were the following:

- to evaluate a standard MIMS method for the determination of VOCs in aqueous samples against the generally accepted methods for that purpose, static headspace gas chromatography (HSGC) and purge&trap gas chromatography-mass spectrometry (P&T-GC/MS)

- to investigate on-site capabilities of the MIMS method for the analysis of VOCs in water samples

- to develop simple and rapid analytical methods for the determination of VOCs, especially volatile organic sulfur compounds, in air

- to develop and evaluate new techniques for the determination of semivolatile and/or polar organic compounds in aqueous samples.

2. Materials and methods

2.1 Instrumentation

2.1.1 Standard MIMS

The mass spectrometer used [I–IV] was a Balzers QMG 421C quadrupole mass spectrometer with a mass range of 1 to 500 amu and equipped with an open cross-beam electron impact (70 eV) ion source. The mass spectrometer was equipped with a sheet membrane inlet, which was constructed on the basis of the design of Lauritsen [4]. The material of the sheet membrane was polydimethylsiloxane with dimensions: thickness 25 or 100 µm and contact area 28 mm^2. During operation of the system a stream of pure synthetic air (20% O_2 and 80% N_2; purity 99.999%) is continuously sucked to the membrane inlet via a gear pump, typically at a flow rate of 400 mL/min. Detection limits (signal to noise ratio 3:1) and linear dynamic ranges of the test compounds were measured by selected ion monitoring (SIM).

2.1.2 Temperature-programmed desorption MIMS

The mass spectrometer was a Balzers QMG 420 single quadrupole mass spectrometer with a mass range of 1–200 amu [V]. The ion source was a closed electron impact ion source with an ionization energy of 70 eV. The membrane inlet was a flow cell with a 25 µm thick polydimethylsiloxane membrane mounted in the vicinity of the ion source. The exposed area of the membrane was approximately 7 mm^2. The inlet was electrically heated and thermostatted by a heating controller to the desired temperature (110°C).

A schematic diagram of the temperature-programmed desorption unit is shown in Figure 2.1. It consists of an aluminum block, an adsorbent tube, a heating resistor and a thermocouple, all of which are mounted inside a protective stainless steel box. The aluminum block (52 x 27 x 16 mm) has an upper and a lower part, between which are the heating resistor and the adsorbent tube positioned in two cylindrical slots (diameters of 6 and 6.5 mm). Encapsulated in the aluminum block is also a thermocouple, and the thermocouple and the resistor are both connected to a heating. With this system the aluminum block and the adsorbent tube could be heated in a controlled manner from 30 to 300°C

at a temperature rate of 10–100°C/min. To cool down the aluminum block from high temperature to room temperature the protective stainless steel box was cooled by an ice bath and the aluminum block by a cool air blower. In this way it took less than 5 minutes to cool the system down from 250 to 30°C. The adsorbent tube was connected via Swagelok fittings and stainless steel tubings at one end to either a gas tight syringe for sampling or to a helium (99.996%) bottle. The other end of the adsorbent tube was connected directly to the MIMS system. The flow rate of the helium purge gas was measured at the outlet of the membrane inlet by a bubble meter.

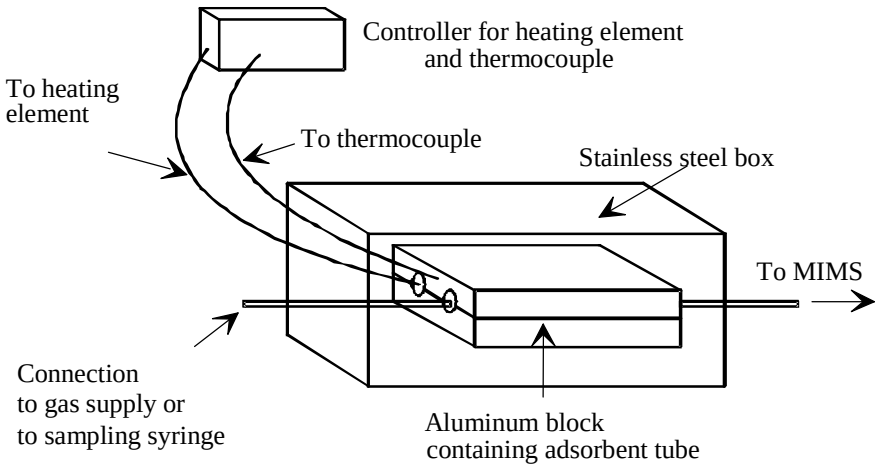

Figure 2.1. Schematic diagram of the TPD-MIMS system.

2.1.3 Trap-and-release MIMS

The mass spectrometer was a Balzers QMG 420 single quadrupole mass with a mass range of 1 to 500 amu [VI]. The ion source was a cross beam electron impact ion source, and ionization was performed using 50 eV electrons. The experiments were performed with an electron emission current of 1.0 mA. The membrane was a polydimethylsiloxane membrane with a wall thickness of 216 μm (i.d. 0.020 inc. and o.d. 0.037 inc.). It was soaked (expanded) in heptane and

then fitted to the steel tubes. Following evaporation of the heptane, a tight seal between the membrane and the steel tubes was obtained.

2.1.4 Desorption chemical ionization MIMS

The mass spectrometer used for the modification [VII] was a Balzers QMG 420 single quadrupole mass spectrometer with a mass range of 1–500 and supplied with two pumping systems: a turbomolecular pump TPU 062 with a rotary pump DUO 1,5 A and a turbomolecular pump TPU 240 with a rotary pump Edwards E2M2. The experiments were performed with an electron emission current of 0.7 mA and a rhenium filament. The membrane used was a polyacrylonitrile with dimensions: 1 mm i.d., 1.6 mm o.d., cut-off 30 kDa and length 15 mm.

2.1.5 On-site measurements

A gas chromatograph-quadrupole mass spectrometer, Fisons MD-800 GC-MS, was equipped with a helium purge type of membrane inlet [26] and operated under 70 eV electron ionization [II]. The inlet was constructed from two modified Swagelok reducing unions (1/4"–1/16", Swagelok SS-400-6-1ZV) and a 10 cm piece of 1/4" glass tube (2 mm i.d). A 4 cm Dow Corning silastic hollow fiber membrane (o.d. 0.635 mm and i.d. 0.305 mm) was mounted inside the glass tube. The membrane was soaked in *n*-hexane prior to mounting it over the two silica capillaries. A continuous flow of helium, about 1 mL/min, was supplied to the membrane inlet with a fused silica capillary restrictor (i.d. 0.22 mm). The sample/helium flow from the membrane inlet was directed to the ion source of the mass spectrometer using a deactivated fused silica capillary (i.d. 0.22 mm, length 40 cm). The normal GC-interface was used for introduction of a silica capillary of the membrane inlet to the oven of the GC.

2.1.6 High performance liquid chromatography (HPLC)

The high performance liquid chromatography (HPLC) method [VI]: the caffeine analysis (see Section 4.2) was carried out using a Model 510 liquid chromatograph equipped with a 50 μL sample loop and a Model 441 absorbance detector at a wavelength of 245 nm. The column used was a Hypersil 5 C_{18} column (4.0 x 300 mm), and the mobile phase was acetonitrile:water (8:92, v:v) at a flow rate of 2.0 mL/min.

2.1.7 Static headspace gas chromatography (HSGC)

Samples were analyzed using a Hewlett Packard 5890 Series II gas equipped with a Hewlett Packard 7694 head space sampler, two flame ionization detectors (FID) and two capillary columns [I]. The carrier gas was hydrogen. The temperatures of the sampler oven, the sample loop and the transfer line of the headspace sampler were 80°C, 120°C and 120°C, respectively. Analyses were carried out using the temperature program: 45°C (5 min), 10°C/min to 210°C (2 min). The temperature of the injector was 220°C and that of the detectors 250°C. Sample volumes were 10 mL in 20 mL headspace bottles.

2.1.8 Purge-and-trap gas chromatography/mass spectrometry (P&T-GC/MS)

Samples were analyzed using a system consisting of a Tekmar LSC 2000 purge-and-trap sampler, a Hewlett Packard 5890 Series II gas chromatograph equipped with a DB-1 capillary column, and a Jeol JMS-AX505WA mass spectrometer with electron impact ionization at 70 eV [II]. The GC temperature program was: 30°C (5 min), 20°C/min to 110°C (0 min), 10°C/min to 300°C (5 min). The carrier gas and purging gas was helium and the sample volume was 5 mL. Identification of compounds in the samples was accomplished by analyzing mass spectra obtained over the mass range from 29 to 400 amu.

2.1.9 Gas calibrator

A gas calibrator was developed for the production of a gas standard of volatile organic compounds [IV]. With the gas calibrator it was possible to produce accurate gas standards at a concentration level of 1 to 5 000 $\mu g/m^3$. It was not possible to test the accuracy of the gas calibrator below 1 $\mu g/m^3$ due to the detection limits of the MIMS method.

The gas calibrator also provides a convenient way to study the effect of humidity of the sample air on the response of the MIMS method. The response of trichloroethene (500 $\mu g/m^3$) clearly decreases when the relative humidity of air increases from 0 to 50%. At humidity levels higher than 50% the response did not decrease significantly. The observed decrease is probably due to the competition of “active” sites on the membrane surface between water and trichloroethene

molecules. At high humidity levels saturation of the membrane surface with water molecules hinders transportation of other molecules across the membrane. The results presented are in good agreement with those recorded earlier with dichloromethane at a concentration of 1 mg/m^3 prepared in ambient air supplemented with water at a concentration level of 0–25 g/m^3 [31]. The results also show that the calibration for quantitative analysis must be made at the same humidity level as the measurements of unknown samples, especially if very good accuracy is required.

2.2 Programs

2.2.1 Solver

The calculation program (Solver) [III, IV] for resolving a multicomponent mass spectrum was developed at VTT Chemical Technology. This program uses a modified algorithm of the general deconvolution method, which assumes that the intensity of any mass-to-charge ratio (m/z) is a linear function of the concentration of the chemical compounds which contribute to that particular m/z [78].

2.2.2 SIMION 3D 6.0

The program used for ion optics simulation was SIMION 3D 6.0 (Princeton Electronic Systems, Inc., Princeton, NJ, USA) [VII].

2.3 Chemicals, materials and samples

Reagents: the commercial reagents used were: trichloroethene, benzene, tetrachloroethene, carbon tetrachloride, xylenes (mixture of isomers), toluene, chloroform, 1,1,2,2-tetrachloroethane, 1,1,1-trichloroethane, 1,2,4-trichlorobenzene, 1,2-dichloroethene (mixture of isomers), 1,1-dichloroethane, 1,3-dichlorobenzene, chlorobenzene, benzaldehyde, 2-butanone, acetone, methyl-*tert*-butylether, carbon disulfide, ethanethiol, dimethyl sulfide (DMS), ethylmethyl sulfide, thiophene, dimethyl disulfide (DMDS), dimethylsulfoxide

(DMSO), methanol, *n*-hexane, caffeine, 1-naphthalenemethanol, acetonitrile, naphthalene, malonic acid and succinic acid.

Aqueous standards: aqueous standard solutions were prepared by volumetrically diluting stock standard solutions (typically 10 g/L in methanol) of commercial reagents using deionized water. If the compound was miscible with water, the stock solution was prepared in water. The final concentrations of the standard solutions were in the range of 0.1 to 5 000 µg/L.

Gas standards: gas standards were prepared either in 15 L gas bags prepared at VTT Chemical Technology from Tedlar®, or in 13 L gas bags from SKC Inc. The gas bags were flushed and filled with pure nitrogen (purity 99.998%) or with pure synthetic air (20% oxygen and 80% nitrogen, purity 99.999%). The stock solutions of test compounds were made by weighing one gram of the compound and dissolving it in 100 mL of methanol. Further dilutions of the stock solutions were made with methanol. The gas standards were made by injecting appropriate methanol dilutions (5–10 µL) of test compounds into the gas bag and allowing the bag to equilibrate for at least half an hour before use. All standards were made at room temperature. For gas standards made by a gas calibrator, see Section 2.1.9.

Samples: spiked water samples were prepared by diluting the stock standard solutions (10 g/L) with methanol to a concentration of 100 mg/L and diluting these solutions to a final specified concentration with deionized water. The content of methanol in the spiked samples was from 0.1 to 1%. All unknown and spiked samples were stored in 100 mL headspace vials and similar headspace vials were also used as sample vials in the MIMS method. The environmental water samples analyzed were obtained from various customers of VTT Chemical Technology. Appropriate dilutions of the samples were made with deionized water when needed for both the MIMS method and the P&T-GC/MS method. The groundwater samples for on-site measurements were obtained from an illegal chemical waste dump site in northern Italy. Spiked air samples were made in a similar way to the gas standards in gas bags. Five air samples were obtained from a paint shop.

Quantitative analysis of caffeine. 2.00 g of roasted coffee or a tea bag (weight approximately 2.0 g) was added to a 250 mL Erlenmeyer flask and 100 mL of

boiling water was added to the flask. After ten minutes the solution was filtered through a cheese cloth and 1.0 mL of an internal standard, 1-naphthalenemethanol (400 mg/L), was added to the solution in order to correct for instrumental drift. The sample solution was cooled to 0°C in an ice-water bath before analysis. Caffeine was analyzed using three different standard concentrations (100, 300 and 500 mg/L) and an internal standard method. Quantitation was achieved by comparing the signals of caffeine and 1-naphthalenemethanol in the samples with the signals obtained from the standard solutions prepared in a similar way.

Adsorbent materials and tubes for TPD-MIMS: 22–120 mg of solid adsorbent (Tenax TA 60/80 mesh, Tenax GR 60/80 mesh, HayeSepD 80/100 mesh, Chromosorb 105 60/80 mesh or Silica gel 60 70/230 mesh) was packed into a glass or stainless steel tube (length 85 mm, o.d. 6.1 mm, id. 4.5 mm) and trapped with a small plug of glass wool at each end. The length of the adsorbent plug inside the tube was usually 20 mm. Before preparing the adsorbent tubes, the tubes were sonicated with ethanol for 10 minutes and then heated at 150°C overnight. Each adsorbent tube was conditioned at 210–280°C for a few hours under a helium flow (99.996% purity) of 40 mL/min.

3. Evaluation of the standard MIMS method in water analysis against other analytical techniques

In order to demonstrate the capabilities of membrane inlet mass spectrometry in environmental analysis the MIMS method was compared with the routine water analysis methods, namely purge-and-trap gas chromatography-mass spectrometry (P&T-GC/MS) and static headspace gas chromatography (HSGC), in the analysis of environmentally significant compounds in water samples [I]. Here the standard MIMS method refers to a system in which the membrane inlet is a simple flow over or helium purge inlet.

The analytical characteristics of the MIMS, the P&T-GC/MS and the HSGC methods were studied with nine different volatile test compounds presented in Table 3.1, including six halogenated organic compounds and three aromatic compounds. Note that the P&T-GC/MS and the HSGC methods are the analytical methods used in routine water analysis at VTT Chemical Technology. All methods were optimized for routine analysis and for this reason the operation conditions used were a compromise between several different factors (e.g. detection limits required by government regulations, speed of analysis and capability to identify unknowns) and therefore the best performance characteristics of these techniques were not necessarily obtained. For example, lower detection limits can be attained with all three methods. Typical detection limits using the P&T-GC/MS method range from 0.01–0.1 μg/L [149], but detection limits below 1 ng/L have been reported [150]. With the MIMS method the typical detection limit range is 0.1–10 μg/L [2]. However, it has already been demonstrated that under optimum conditions detection limits at parts-per-quadrillion levels can be achieved, for example a detection limit of 500 ppq was measured for toluene in water [66]. With the HSGC method detection limits 0.1–1 μg/L are often obtained [151, 152]. Organic chlorocompounds, e.g. trichloroethene and chloroform, can be measured by the HSGC method even at lower levels under optimal conditions, i.e. at the level of 0.05–0.2 μg/L using an electron capture detector (ECD) [153]. The detection limits (signal-to-noise

ratio 3:1) for most of the compounds are comparable for the MIMS method and for the P&T-GC/MS method, being in the range of 0.1 to 1.0 µg/L. In the case of the HSGC method the detection limits are much higher, about 10 to 100 times higher than those for the other two methods. The linear dynamic ranges of test compounds measured by the MIMS method are three to four orders of magnitude, which is more than sufficient for the analysis of unknown samples with varying concentrations. For the P&T-GC/MS method the linear dynamic ranges are much narrower, about two orders of magnitude, due to the limited capacity of the adsorbent trap and the cryofocusing trap. These narrow linear dynamic ranges can cause problems in the analysis of samples containing analytes in a wide concentration range. However, with a different P&T-GC/MS configuration a linear dynamic range of up to 5 000 µg/L has been achieved [154]. The best performance in this respect was obtained with the HSGC method, for which linear dynamic ranges up to six orders of magnitude were measured, due to the very wide dynamic range of the flame ionization detector. The linear dynamic ranges of some compounds might be even wider, but for practical reasons the upper limit in the measurements was limited to 100 mg/L except in the case of toluene. For the HSGC method our results are in good agreement with those reported in the literature, e.g. using the HSGC method with photoionization detector (PID) followed by FID the useful working range has been reported to be 1 to 15 000 µg/L [151].

Table 3.1. Detection limits (DL) and linear dynamic ranges (LDR) of selected compounds (in µg/L) measured by MIMS, P&T-GC/MS and HSGC. The upper limit of LDR by the HSGC method is partly determined by the solubilities of the tested compounds in water.

Compound	MIMS		P&T-GC/MS		HSGC	
	DL	LDR	DL	LDR	DL	LDR
Toluene	0.1	0.3–1 000	0.2	0.2–15	3	3–380 000
Benzene	0.1	0.1–1 000	0.2	0.2–20	4	4–100 000
Xylenes	0.1	0.1–5 000	0.2	0.2–15	4	4–100 000
1,2-Dichloroethane	0.4	0.4–4 000	0.2	0.2–15	12	12–100 000
1,1,1-Trichloroethane	0.6	0.6–5 000	0.2	0.2–15	30	30–100 000
Trichloroethene	0.1	0.1–1 000	0.2	0.2–20	8	8–100 000
Tetrachloroethene	0.1	0.3–1 000	0.2	0.2–20	10	10–100 000
Chloroform	0.3	0.5–5 000	0.2	0.2–30	30	30–100 000
Carbon tetrachloride	0.5	0.5–5 000	0.2	0.2–20	40	40–100 000

The identification and quantitation capabilities of the three methods were compared by analyzing spiked samples. A good example of these results is presented in Table 3.2. The concentrations of the analytes were calculated for each method using external standards. Dimethyl- and trimethylbenzenes can be quantitated individually using the HSGC and P&T-GC/MS methods due to the chromatographic separation. The sum of these compounds can also be calculated with the MIMS method, but the identification of individual compounds is difficult.

Table 3.2. Analytical results of a spiked sample measured by the three methods and relative standard deviation (RSD) between calculated and measured concentrations. The notations C_2-benzenes and C_3-benzenes indicate the sum of benzene derivatives substituted by two (dimethylbenzenes and ethylbenzene) or three carbons (trimethylbenzenes, ethylmethylbenzenes, propylbenzene and isopropylbenzene), respectively.

Compound	Concentration, µg/L				RSD		
	Spiked	MIMS	HSGC	P&T-GC/MS	MIMS	HSGC	P&T-GC/MS
Toluene	50	51	41	44	1	13	8
Tetrachloroethene	8	12	8	12	35	0	35
1,2-Dichloroethene	98	120	110	90	16	9	6
C_2-benzenes	201	240	210	190	14	3	4
Benzene	20	26	19	12	21	4	28
C_3-benzenes	52	48	43	40	5	12	16
1,1,1-Trichloroethane	432	370	410	350	10	4	13
1,1-Dichloroethane	49	59	44	*	14	7	**
Dichloromethane	47	59	50	49	18	5	3
1,2-Dichloroethane	108	79	110	200	19	1	60
Trichloroethene	800	760	720	820	4	7	2
Sum[a]	1865	1820	1760	1800	2	4	2
Mean[b]					13	6	16

[a] Sum is the total amount of analytes.
[b] Mean is measured as an average of the RSDs between observed and spiked concentrations.
* Not found.
** Not measured.

The results presented in Table 3.2 show that compounds with low concentrations can be quantified reliably by all three methods, even if the concentration difference between some compounds is as much as two orders of magnitude. The small variations between observed and spiked concentrations are believed to be due to evaporation of the compounds during preparation of

the spiked sample and standards. The loss of volatile compounds in sample handling has been experienced before, e.g. by Wise *et al*. [155]. The smallest RSD between observed and spiked concentrations was obtained with the HSGC method (6 %) and the largest with the P&T-GC/MS method (16 %). The largest differences between observed and spiked concentrations with the MIMS and the P&T-GC/MS methods were observed in the case of tetrachloroethene, most probably due to its low concentration. The second error was due to the leakage of benzene from the Tenax adsorbent trap, which was observed in the analysis of blank samples. It can also be seen that the MIMS method gives the most accurate result for the total amount of volatile organic compounds in the sample.

One important parameter of an analytical method is the analysis time, i.e. the shorter the analysis time, the faster the results are obtained. The analysis time is shortest in the MIMS method, the cycle time from sampling of one sample to sampling of the next being 5–10 minutes. In the HSGC and P&T-GC/MS methods the analysis time depends on the GC run time and the head space parameters. In our experiments the analysis time was 43 minutes for the HSGC method and 40 minutes for the P&T-GC/MS method. In both cases the analysis time can be shortened by a few minutes, at the expense of accuracy and reproducibility. This comparison clearly shows that a much larger sample throughput can be obtained with the MIMS method than with the other two methods.

The repeatability of the analysis method was measured from three successive injections of the same sample and calculating the relative standard deviation (RSD) of the repeated injections from the measured concentrations of compounds in the sample. The RSDs obtained from ten measurements with each method ranged between 1 and 11 % (mean 8%) for the MIMS method, 1 and 8 % (mean 6 %) for the HSGC method and 2 and 13 % (mean 8 %) for the P&T-GC/MS method. The results obtained are very close to those reported earlier by Ho [149] (1–10 % with the P&T-GC/MS method) and Roe *et al.* [152] (2–8% with the HSGC method). As can be seen from these results, the repeatabilities of all three methods were good, demonstrating that minor changes in measurement conditions do not affect the analytical results.

The analytical characteristics of all three methods are summarized in Table 3.3. As can be seen, membrane inlet mass spectrometry in the analysis of volatile

organic compounds is a very comparable analysis method with the conventional methods, purge-and-trap gas chromatography-mass spectrometry and static head space gas chromatography. The main advantages of the MIMS method are low detection limits and short analysis time. The MIMS method is also the only method of these three which can be used for continuous on-line monitoring [27, 32, 156–159]. The major difficulty with the MIMS method is the lack of chromatographic separation of components, especially with heavily contaminated samples, but the recently developed deconvolution program for multicomponent mass spectra resolves this problem in many cases. The major advantages of the P&T-GC/MS method are low detection limits and the capability of analysis of very complex mixtures due to the gas chromatographic separation. In addition, identification of unknowns is relatively easy since commercial reference libraries of electron impact mass spectra can be used to assist the identification. The best qualities of the HSGC method are wide dynamic range, separation of compounds by GC and simpler instrumentation than for the other two methods. The major disadvantages of the HSGC method are poor detection limits compared to the other two methods and poor identification capability when a flame ionization detector is used. The measured results also showed that the reproducibilities of the methods are of the same order of magnitude and that agreement between the analytical results obtained by the three different methods is very good.

Table 3.3. Characteristics of the three analytical methods.

Quality	MIMS	P&T-GC/MS	HSGC
Detection limit	< 1 µg/L	< 1 µg/L	1–10 µg/L
Linear dynamic range	10^4	10^2	10^6
Repeatability	1–11 %	2–13 %	1–8 %
Analysis time	5–10 min	35–35 min	35–45 min
On-line monitoring capability	++++	+	+
Identification capability	++	+++	+[a]
Simplicity of instrumentation	++	+	+++

[a] Flame ionization detector (FID) used as a detector

+++ very good, ++ good, + fair

4. On-site capabilities of membrane inlet mass spectrometry

On-site chemical analysis is becoming more and more important due to growing knowledge of the toxicity of various chemicals and due to continuous tightening of the regulations of environmental legislation driven by increasing public awareness of environmental problems. In addition, the complexity of the environmental samples requires the development of new sophisticated analytical techniques and procedures for on-site environmental analysis.

The membrane inlet mass spectrometric method developed for on-site analysis of environmentally significant compounds from water samples [II] was constructed on the basis of a helium purge type of membrane inlet [26]. This type of membrane inlet was selected for the basis of development work mainly because it can be used together with commercial GC-MS instruments without any modifications to the instruments and because it can be installed very rapidly into the gas chromatograph oven of a GC-MS instrument. The detection limits of the used system were typically at sub µg/L levels and response times were between 1.5 and 2.0 minutes, which allows rapid identification of pollutants and screening of large numbers of environmental samples in a short period of time. The linear dynamic ranges with the GC-MS instrument started from the detection limit and extended to about 4 orders of magnitude higher concentrations. The good linearity and the freedom of matrix effects in a very wide concentration range clearly demonstrate that MIMS is an excellent analytical method for rapid on-site analysis of environmentally significant compounds from water.

A typical mass spectrum measured for one of the contaminated groundwater samples is presented in Figure 4.1. As can be seen from this figure toluene (ions m/z 91 and 92) was identified as the major volatile pollutant of the contaminated area studied in this work. Other pollutants which can easily be identified on the basis of their mass spectrum are benzene (ion m/z 78) and xylenes (ion m/z 106). On the basis of the relatively high intensity of the ion m/z 121 it was expected that the sample contained an aromatic nitrogen compound. This was latter confirmed by GC-MS measurements which showed that the samples contained N,N-dimethylbenzenamine or an isomer. The fact that the intensity ratio of ions m/z 120 and 121 in the mass spectra measured by MIMS is very similar to the intensity

ratio seen in the standard EI mass spectra of N,N-dimethylbenzenamine and its isomers further confirms that some of the samples contained this type of compound. Minor contaminants which could be identified in some of the samples were trichloroethene, dichloroethene and chlorobenzene. Characteristic ions of the latter two compounds can also be seen in Figure 4.1, i.e. ions m/z 96, 98 and 100 for dichloroethene and ions m/z 112 and 114 for chlorobenzene. The excellent signal to noise ratio of the measured mass spectrum and the very well reproduced chlorine isotope peak ratios seen in the mass spectrum should be especially noted, since they allow reliable identification of even some of the minor contaminants.

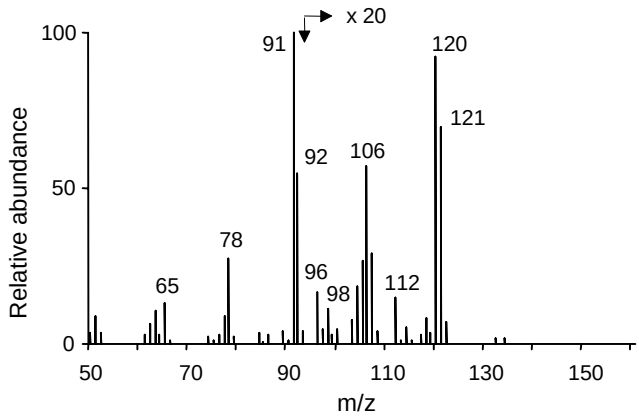

Figure 4.1. Background subtracted mass spectrum of a contaminated groundwater sample, W04, measured on-site with MIMS. Note that the ion intensities are multiplied by a factor of 20 after the ion m/z 93.

The quantitation results obtained during the on-site measurement campaign are shown in Table 4.1. The major compounds, toluene, benzene and xylenes, and one of the minor compounds, namely trichloroethene, were quantitated. The response of the standard solution was measured directly before or directly after analysis of the sample solution in order to minimize the effects of possible variations in instrumental conditions. As can be seen from Table 4.1 there are large variations in the concentrations of the pollutants. The highest concentrations of the pollutants were found near the suspected waste release site, and the further the sampling

point was from the suspected release site the lower were the concentrations of the pollutants. The results presented also provide a good indication that MIMS provides as good results in a mobile laboratory as in a fixed laboratory, since the results obtained for the sample W06 in a fixed laboratory were almost the same as those measured for the samples W03 and W04, which were taken from the nearest sampling points to the W06 sample.

Table 4.1. On-site quantitation results of the groundwater samples collected from 6 different sampling points. The quantitation limit of the compounds was 1 µg/L. All values are in µg/L.

Compound	W01	W02	W03	W04	W05	W06[a]
Toluene	370	11	58 000	16 000	2 900	27 000
Benzene	30	<1	7 400	5 300	170	4 100
Xylenes	28	1	1 400	620	190	850
Trichloroethene	1	<1	30	40	4	50

[a] Analyzed in a fixed laboratory.

5. Analysis of volatile organic compounds in air samples by MIMS

5.1 Determination of volatile organic compounds in air by standard MIMS

A method for the analysis of volatile organic compounds in air samples was developed using a sheet membrane inlet [III]. The performance of the sheet membrane inlet was characterized in detail because the earlier air analyses by MIMS were done with capillary membrane inlets [1, 32, 122–126]

The effects of membrane thickness on the responses and the response times of toluene and trichloroethene at a concentration level of 3.3 mg/m^3 were studied using the sheet polydimethylsiloxane membrane inlet. The measurements were made using membranes with thicknesses of 25 and 100 μm. The responses of the test compounds were almost inversely dependent on the thickness of the membrane, as predicted according to Eq. 3 (p. 14). This was demonstrated by the detection limits which were obtained using the two different membrane thicknesses. The detection limits with the thinner membrane were from three to six times better than those with the thicker membrane.

The theoretical response time-thickness relationship can be expressed as follows [32]:

$$\frac{t_{(50)2}}{t_{(50)1}} = \left(\frac{l_2}{l_1}\right)^2 \qquad (6)$$

where l_2 and l_1 are the thicknesses of membranes 2 and 1, and $t_{(50)2}$ and $t_{(50)1}$ are the response times required to achieve 50% steady-state permeation with membranes 2 and 1. Eq. 6 is correct if diffusivity is a constant for a given substance in a given polymer. In theory this response time ratio for membranes with thicknesses of 100 and 25 μm should then be 16. The measured ratios varied from 8.8 to 24.5 depending on the compounds (Table 5.1), but the average value of the response time ratios, 15.6, was very close to the theoretical value. Hayward *et al.* [73, 160] investigated the suitability of different sheet

membrane materials for MIMS analysis and the dependence of responses on the thickness of membranes in water analysis, and they obtained similar results to ours. Cooks *et al.* [161] characterized the performance of thin (10–50 μm) hydrophobic membranes for the on-line analysis of volatile compounds in solution by MIMS and flow injection analysis (FIA). Their results were different from our results as well as from those of Hayward, probably due to the use of membranes from different manufacturers, with variations in the composition of different membrane materials.

Table 5.1. Response times of some selected test compounds measured with the sheet membrane inlet. The thickness of the sheet membrane was either 25 or 100 μm. The ratio is the response time ratio of thick and thin membranes, and the average is the average response time ratio of all measured values.

Compound	Response time, s		Ratio
	25 μm, ±1	100 μm, ±5	
Carbon tetrachloride	1.1	20	18.2
Tetrachloroethene	1.4	21	15.0
Benzene	1.1	17	15.5
Chloroform	1.2	27	22.5
1,1,2,2-Tetrachloroethane	2.5	25	10.0
1,2-Dichloroethene	1.1	27	24.5
Xylenes	1.7	15	8.8
Chlorobenzene	1.9	19	10.0
Toluene	1.3	20	15.4
Trichloroethene	0.9	12	13.3
Average			15.6

The response of toluene with the sheet membrane inlet as a function of the sample gas flow rate is presented in Figure 5.1 [III]. The response of toluene increased about 35% when the flow rate was increased from 150 to 1 500 mL/min. It is believed that the increased response is due to better mixing at the sample/membrane interface at higher flow rates. The response times were slightly decreased (20–30%) when the sample flow rate was increased from 150 to 1 500 mL/min. This could be due to smaller boundary layer effects. The

optimum sample flow rate would be the highest value which could be obtained with the pump used (1 500 mL/min) but for practical reasons further experiments were performed with a sample flow rate of 400–500 mL/min, since at this flow rate the response was only 30% lower than its maximum and the response times were not significantly different from the their minimum values (measured at a flow rate of 1 500 mL/min).

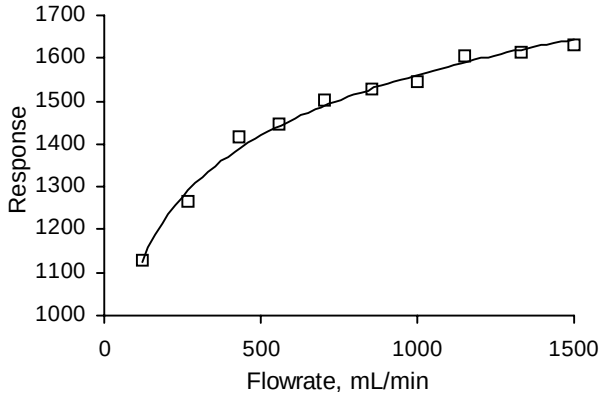

Figure 5.1. The response of toluene (m/z 91) as a function of the flow rate of the sample gas. The concentration of toluene was about 3.3 mg/m^3, and the thickness of the sheet membrane was 100 μm.

The responses of toluene (m/z 91) and trichloroethene (m/z 130) as a function of temperature of the membrane inlet are shown in Figure 5.2, which also shows the response of background (m/z 200). The ion m/z 200 is only an example of the background because in the scan mode the whole scanned mass range (m/z 50 to 200) behaved similarly at higher temperatures. In this study the sample flow rate was 500 mL/min. The responses slightly decreased until the temperature reached 140–160°C. This result is similar to that obtained by LaPack *et al.* [1] with a hollow fiber membrane, because the permeabilities of organic compounds in air decrease with increasing temperature. From 160°C to 200°C the responses appeared to increase considerably, by about one and a half orders of magnitude, but the background (the signal at m/z 200) was also greatly enchanced at high temperatures and there was actually a decrease in the signal to noise ratio with increase in temperature. This decrease was most probably due to physical changes in the membrane, since some changes were visually recognized after the experiment. Furthermore, the pressure in the vacuum

chamber increased from 1.0 x 10^{-6} mbar to 2.0 x 10^{-5} mbar with the increase in temperature of the membrane inlet from 40°C to 200°C. The response times decreased by 50% with temperature increase from 50°C to 80°C, but from 80°C to 180°C they remained approximately the same because there is a physical boundary limit to how fast molecules can diffuse through the membrane. Our results are in very good agreement with the results reported by LaPack *et al*. [1]. They observed a 46% decrease in response times in water analysis with a hollow fiber membrane in the temperature range 26–85°C. At a temperature of 200°C the signals were so high, and still increasing, that accurate determination of the response times was no longer possible. The optimum temperature of the membrane is about 80°C with respect to both responses and response times.

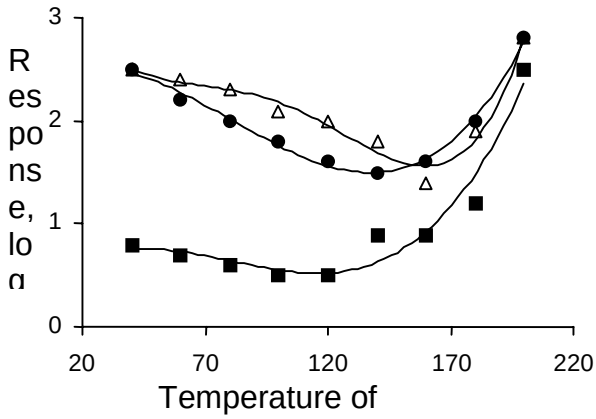

Figure 5.2. The responses of toluene (Δ, m/z 91), trichloroethene (●, m/z 130) and background (■, m/z 200) as a function of the temperature of the membrane inlet. The concentrations of the compounds were about 3.3 mg/m^3 and the thickness of the membrane was 100 μm.

Detection limits, linear dynamic ranges and response times for 18 volatile organic compounds (Table 5.2) were measured using the following parameters: the temperature and thickness of the membrane were 80°C and 25 μm, respectively, and the sample flow rate was 450 mL/min. The detection limits for most compounds are low, especially for the low molecular weight and more volatile compounds, being at the low or sub μg/m^3 level. The more polar or higher molecular weight the compound is, the higher is its detection limit. The

linear dynamic ranges were over four orders of magnitude, being even wider than the linear dynamic ranges measured from the water phase [II].

Table 5.2. Response times, detection limits and linear dynamic ranges of 18 selected volatile organic compounds in air measured by MIMS. The thickness and area of the sheet membrane were 25 µm and 28 mm^2, respectively. The sample flow rate was 450 mL/min and the temperature of the membrane inlet was 80°C.

Compound	Response time, s ± 1 s	Detection limit, µg/m³	Linear dynamic range, µg/m³
Toluene	1.3	0.5	1–10 000
Trichloroethene	0.9	0.5	1–10 000
Benzene	1.1	1	1–10 000
Chloroform	1.2	5	5–30 000
Tetrachloroethene	1.4	0.5	1–10 000
1,1,1-Trichloroethane	1.7	4	10–30 000
1,1,2,2-Tetrachloroethane	2.5	1	1–5 000
Xylenes	1.7	2	2–10 000
Carbon tetrachloride	1.1	3	10–20 000
1,1-Dichloroethane	1.0	1	1–5 000
1,2-Dichloroethene	1.1	1	5–10 000
Chlorobenzene	1.9	0.5	2–10 000
1,3-Dichlorobenzene	2.2	1	1–5 000
1,2,4-Trichlorobenzene	3.0	2	3–5 000
Ethanethiol	1.5	5	5–50 000
Dimethyl sulfide	1.3	1	1–40 000
Dimethyl disulfide	1.7	2	2–40 000
Ethylmethyl sulfide	2.0	3	3–60 000

The response times quoted here are the rise times, but in most cases the fall times were equal to or slightly shorter than the rise times. This result is opposite

to results obtained with water, when the fall times are usually longer than the rise times. It appears that the concentrations of compounds on the sample side of the membrane are decreased more slowly in water than in air analysis, resulting in longer fall times. The reason for this might be that sample water and pure water are mixed more slowly with each other than sample air and pure air with each other, so the sample water (analyte molecules) remains longer in the vicinity of the membrane. Another possible reason can be adsorption/desorption of compounds in the transfer line, thus resulting in longer fall than rise times. Wilson and Ottley [31] obtained similar response times (from 1.6 to 6.2 s) when analyzing industrial solvents in breath by a transportable mass spectrometer with a MIMS inlet. The very fast response times are demonstrated in Figure 5.3, in which three successive samplings of toluene (m/z 91) and trichloroethene (m/z 130) at a concentration level of 700 μg/m^3 were made with three different sampling times, namely five, ten and fifteen seconds. The repeatability of these samplings was below 2%, measured as an RSD of the responses of the compounds. With very short sampling times (five seconds) it is possible to analyze even four samples in one minute in the SIM mode. The rapid response

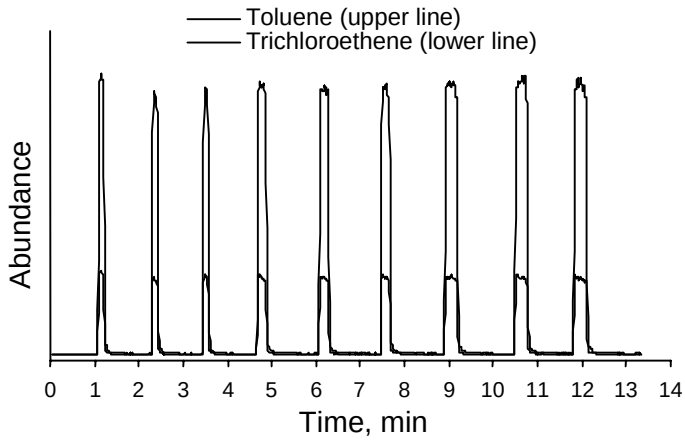

Figure 5.3. Three successive samplings of toluene (m/z 91) and trichloroethene (m/z 130) were made with three different sampling times of 5, 10 and 15 seconds. The thickness of the membrane was 25 μm and the concentrations of toluene and trichloroethene were 700 μg/m^3.

times obtained indicate that rapid changes in the concentrations can easily be monitored by the MIMS method. This is a very important property for example in the monitoring of chemical or biological reactions and processes.

Table 5.3. The analytical results of five air samples from a factory exhaust measured by MIMS and on-line FID analysis. The notations C_2-benzenes, C_3-benzenes and C_4-benzenes indicate the sum of benzene derivatives substituted by two, three and four carbons, respectively.

Compound	Sample, concentration mg/m^3				
	1	2	3	4	5
Benzene	3.4	2.4	3.0	4.4	1.4
C_2-benzenes	101	69	95	31	6.2
C_3-benzenes	25	18	22	11	2.5
C_4-benzenes	6.6	5.0	5.8	4.2	1.3
Naphthalene	0.16	0.09	0.13	1.7	0.88
1,1-Dichloroethane	10	7.1	9.1	11	4.6
Carbon tetrachloride	9.0	6.2	7.8	3.3	1.4
Other compounds [a]	18	13	17	28	12
Sum [b]	173	121	160	95	30
C_xH_y (FID) [c]	199	133	167	104	28

[a] The concentration of other compounds was estimated from the residual spectrum using toluene as a calibration standard.

[b] Sum of all compounds measured by MIMS.

[c] The result of on-line FID analysis.

Five air samples from a factory exhaust were analyzed by the MIMS method and by an on-line FID analyzer, and the analysis results are presented in Table 5.3. The on-line FID analyzer gave only the total amount of volatile organic compounds as C_xH_y using toluene as a calibration standard. The MIMS results were calculated from the multicomponent mass spectrum using the Solver program. The residual spectrum which contained the ions of unknown compounds contributed from 10 to 30% of the original spectrum measured from the m/z peak heights. The total amounts of unknown compounds were

calculated from the residual spectrum using toluene as a calibrant, which gave only an approximate estimation of the true amount due to possibly different responses of unknown compounds from that of toluene. However, the results of the on-line FID analyzer compared very well with the results of MIMS. The difference in the total amount of VOCs measured by the two methods varied from -6.7 to 15.0% (mean ±9.1%).

The capabilities of the developed MIMS method in the analysis of mixture samples were also studied by analyzing two spiked air samples in gas bags (Table 5.4) [IV]. The quantitative results were obtained from the multicomponent mass spectra of the samples using the Solver calculation program [78]. A spectrum of one spiked air sample, which contained 374 µg/m^3 of dimethyl sulfide, 371 µg/m^3 of dimethyl disulfide and 178 µg/m^3 of ethylmethyl sulfide, is presented in Figure 5.4. The quantitation of these compounds, when they all are present in a sample, is difficult in the selected ion monitoring mode, because the most intensive peaks of the spectrum of dimethyl sulfide, m/z 47 (relative abundance 100), m/z 62 (83) and m/z 61 (33), are also intensive peaks of the spectra of dimethyl disulfide (m/z 47 (26), m/z 61 (12) and m/z 62 (6)) and ethylmethyl sulfide (m/z 47 (39), m/z 61 (100) and m/z 62 (3)). The peaks m/z 79 and 94 belong to dimethyl disulfide and the peak m/z 76 belongs to ethylmethyl sulfide.

Table 5.4. The analytical results of two spiked samples measured by MIMS. Concentrations are in µg/m^3.

Compound	Sample 1			Sample 2		
	Spiked	Measured	Diff. (%)	Spiked	Measured	Diff. (%)
Dimethyl sulfide	1 333	1 343	0.8	667	653	- 2.1
Dimethyl disulfide	662	637	- 3.8	331	377	13.9
Ethylmethyl sulfide	634	710	- 12.0	529	507	- 4.2

The average of the absolute differences was 6.1 %.

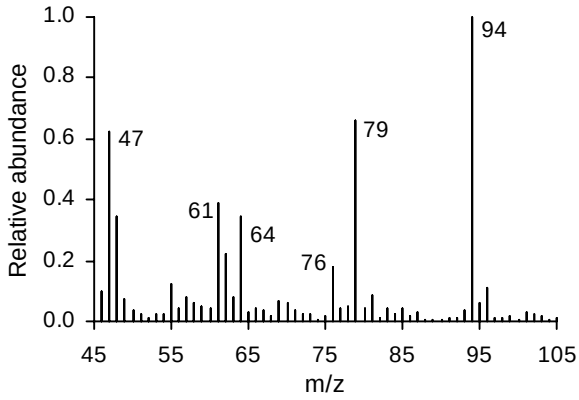

Figure 5.4. Mass spectrum of an air sample in a gas bag spiked with 374 µg/m³ of dimethyl sulfide, 371 µg/m³ of dimethyl disulfide and 178 µg/m³ of ethylmethyl sulfide.

However, in the scan mode the calculation program identifies compounds from the multicomponent mass spectrum by comparing the measured mass spectrum with the mass spectra of individual compounds in a reference library. At the same time as the program identifies compounds, it also searches for the minimum difference between the measured spectrum and the sum of the spectra of identified compounds [79]. From this iteration it calculates a response factor for each compound, and multiplying the response factor by a calculated concentration/response ratio from calibration runs, the concentration of each compound can be calculated. The differences between the calculated and measured concentrations varied from 0.8 to 14%. The results indicate that simple mixtures of VOSCs can be analyzed by MIMS in an accurate and reliable way.

For the sulfur compounds, diffusion coefficients were determined using a 100 µm thick polydimethylsiloxane membrane because there are no diffusion coefficients in the literature for these compounds with this membrane. The diffusion coefficients were calculated in two different ways: first, using the response times according to Eq. 5 and second, according to the method by Pasternak *et al.* [162] which uses the linear part of a permeation curve obtained by MIMS to determine the diffusion coefficient. The results (Table 5.5) were in rather good agreement, even though the response times were so short that measurement uncertainty was necessarily rather large (about 10–20%). With sulfides the diffusion coefficient diminishes as the size of the molecule

increases. With a thin membrane (25 µm) it was impossible to determine diffusion coefficients because response times were only a few seconds and the linear part of the permeation curve was so short that reliable calculation was not possible. Diffusion coefficients for non-polar VOCs with a silicone membrane have been reported to be in the range of 13 to 91×10^{-7} cm^2/s [3], i.e. 2 to 20 times higher than those obtained for VOSCs. The main reason for the difference was probably the difference in the membrane material (e.g. a degree of polymerization) since we found that response times with a silicone membrane of one manufacturer were clearly longer than those obtained with a similar membrane of another manufacturer, and diffusion coefficients of non-polar VOCs such as toluene in our measurements were similar to those of VOSCs.

Table 5.5. Diffusion coefficients of some VOSCs measured by MIMS. A: calculated from the permeation curve [162] ; B: calculated from response times according to Eq. 5.

Compound	Diffusion coefficient, 10^{-7} cm^2/s	
	A	B
DMS	5.3	6.7
DMDS	4.7	5.1
Ethylmethyl sulfide	4.6	4.4
Ethanethiol	5.1	5.8

5.2 Analysis of volatile organic compounds with temperature-programmed desorption MIMS

We developed a new MIMS method, temperature-programmed desorption/MIMS (TPD-MIMS), in order to achieve a rapid separation of volatile organic compounds in air samples prior to detection by MIMS [V]. In TPD a sample is collected into a solid adsorbent and the trapped analytes are desorbed from the adsorbent according to a temperature program. The desorption time, i.e. the time when an analyte desorbs from the adsorbent, is defined by the interaction between the analyte and the adsorbent. Temperature-programmed desorption of VOCs from a solid adsorbent has previously been investigated with other types of detectors. For example, Salvador and Merchán [163] investigated the thermal desorption of phenolic compounds from water

adsorbed onto activated charcoal and calculated the activation energies of desorption of those compounds. They used water as a carrier liquid at high pressure (0–300 atm) and a UV-visible spectrophotometer as a detector. Kovaleva *et al.* [164] used a short analytical column filled with a carbonaceous adsorbent to preconcentrate *p*-xylene from air, and then used this column for temperature-programmed desorption of adsorbed compounds. They used a flame ionization detector for the detection of desorbed compounds and obtained a detection limit of 10 ng for *p*-xylene in air. Peters and Bakkeren [104] measured breakthrough volumes of various adsorbents using MIMS, and for example with Tenax GR (250 mg) the breakthrough volumes were over 10 L.

Figure 5.5 shows the desorption profiles of *trans*-1,2-dichloroethene, chloroform, carbon tetrachloride, trichloroethene, toluene, tetrachloroethene, xylenes, styrene and 1,2-dibromo-1,2-dichloroethene, obtained from a mixture containing a few micrograms of each compound. In this experiment 25 mL of the gaseous mixture from the gas bag was injected into the adsorbent tube (HayeSepD adsorbent), which was then heated at a rate of 90°C/min. Trichloroethene was monitored in all three runs as an internal reference to check the stability of the system, and its desorption time was constant within 1% from run to run. With the HayeSepD adsorbent the first compound (*trans*-1,2-dichloroethene) started to desorb after approximately 60 seconds (120°C) and the last compound, 1,2-dibromo-1,2-dichloroethene, ended its desorption after approximately 160 seconds (270°C). The whole desorption process took place within 100 seconds (150°C) and with a typical width of the desorption profiles of 10 seconds (15°C) at half height, providing very good resolution. This is easily seen by comparing the desorption profiles from the closely related compounds *trans*-1,2-dichloroethene, trichloroethene, tetrachloroethene and 1,2-dibromo-1,2-dichloroethene (profiles 1, 4, 6 and 9 respectively), which are almost fully separated.

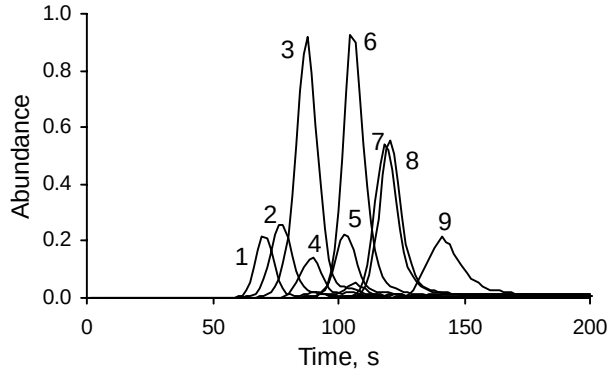

*Figure 5.5. TPD-MIMS desorption profiles of a gas sample (25 mL) containing a mixture of the following compounds: **1** trans-1,2-dichloroethene, **2** chloroform, **3** carbon tetrachloride, **4** trichloroethene, **5** toluene, **6** tetrachloroethene, **7** xylenes, **8** styrene and **9** 1,2-dibromo-1,2-dichloroethene.*

The separation properties of the temperature-programmed desorption system were tested using Tenax TA as a test adsorbent, because Tenax TA is the most widely used adsorbent. The parameters studied were the heating rate of the adsorbent tube and the purge gas flow rate through the tube. Figure 5.6 presents desorption profiles of toluene (11 µg) obtained with different heating rates and with a purge gas flow rate of 50 mL/min. Toluene desorbs very early from the adsorbent at rapid heating rates, as expected, and the peak is much narrower compared with the peak obtained at low heating rates. The area of each desorption profile was estimated from the height and the peak width at half height (PWHH), and it was found out that the area was the same (12.8 ± 1.6 s, RSD 13%) at all heating rates, i.e. the area of the desorption profile is independent of the heating rate. This means that compounds can be desorbed quantitatively from the adsorbent.

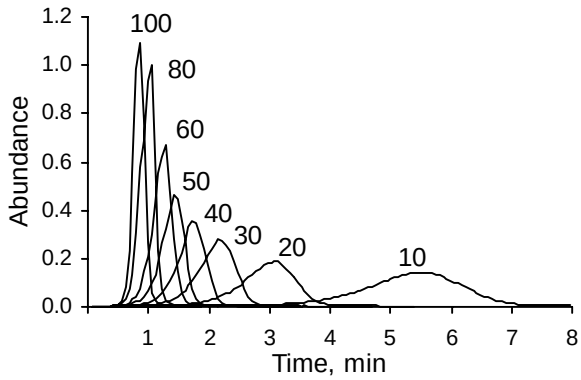

Figure 5.6. Desorption profile of toluene from Tenax TA at different heating rates (°C/min).

We found that the retention time (t_R) was proportional to the reciprocal of the heating rate V:

$$t_R = a + \frac{b}{V} \tag{7}$$

where a and b are constants. The peak width measured at half height (PWHH) also decreased with increase in the heating rate in a fashion similar to the retention time, but to a lower extent.

Consequently, the resolution R_S (Eq. 8) [165]

$$R_s = \frac{t_{R2} - t_{R1}}{\frac{1}{2}(w_1 + w_2)} \tag{8}$$

where t_{R1} and t_{R2} are retention times of two compounds and w_1 and w_2 are the bandwidths of the same peaks, slightly decreased with increasing heating rate (Figure 5.7).

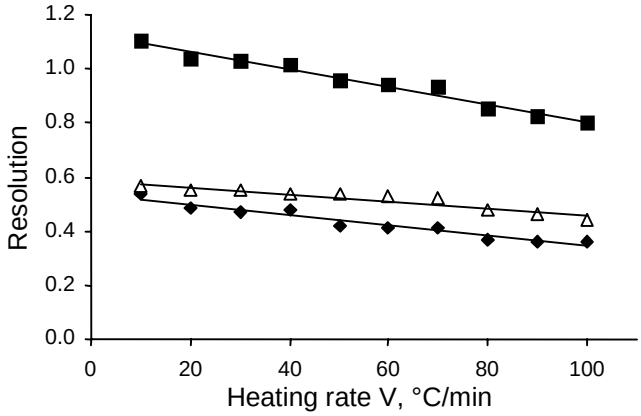

Figure 5.7. Resolution of chloroform, trichloroethene and toluene on Tenax TA as a function of heating rate (°C/min). ■ *Resolution of chloroform and toluene,* Δ *resolution of trichloroethene and toluene and* ◆ *resolution of chloroform and toluene. The helium flow rate for purging was 50 mL/min.*

The effect of the purge gas flow rate through the adsorbent tube on the separation properties was not as great as the effect of the heating rate. At flow rates below 40 mL/min the resolution improves with increasing flow rate, but at higher flow rates it stabilizes and becomes independent of the flow rate. A probable explanation for this behavior is that, at low flow rates (up to 40 mL/min), the resolution is a result of a combination of the temperature-programmed desorption and of the transport of desorbed compounds through the sorbent material. At high flow rates (> 40 mL/min) the desorbed compounds are rapidly purged through the sorbent material and the resolution reflects primarily the temperature-programmed desorption.

The most important parameter for study of the temperature-programmed desorption system is its ability to separate as many compounds as possible from a complex sample. This can be expressed by a parameter analogous to the number of theoretical plates, N, for chromatography [100]:

$$N = 5.545 \times \left(\frac{t_R}{PWHH} \right)^2 \qquad (9)$$

We tested various adsorbent materials (see Section 2.3) and found out that HayeSepD (polydivinylbenzene) adsorbent had the narrowest peaks, the highest number of theoretical plates and the best resolution in most cases. Overall, we found that the performance of the adsorbents depended both on the experimental conditions and on the compounds to be analyzed. The best choice of adsorbent material must therefore depend on the application. One advantage of the HayeSepD adsorbent was also that all the measured compounds behaved in a similar way whereas as with other adsorbents some compounds did not retain as well in the adsorbent and the desorption profiles were very broad.

The detection limits of the TPD-MIMS are rather low. For example, Figure 5.8 shows TPD-MIMS desorption profiles from a gas sample (25 mL) containing four compounds (dichloromethane, *trans*-1,2-dichloroethene, chloroform and tetrachloroethene) at low amounts using HayeSepD adsorbent. Table 5.6 shows the detection limits of a series of VOCs obtained in the selected ion monitoring (SIM) mode. Overall, the detection limits were at low or sub nanogram levels, depending on the permeability of the individual compound through the membrane and the background level at the particular ion detected. The detection limits are comparable to or slightly better than previously published data obtained by standard MIMS [III] and by temperature-programmed desorption with flame ionization detection (TPD-FID) [164]. However, when compared with the standard MIMS technique the TPD-MIMS system offers a fast and relatively efficient separation of compounds prior to MIMS detection and, when compared with TPD-FID, it offers a more selective detector (the mass spectrometer).

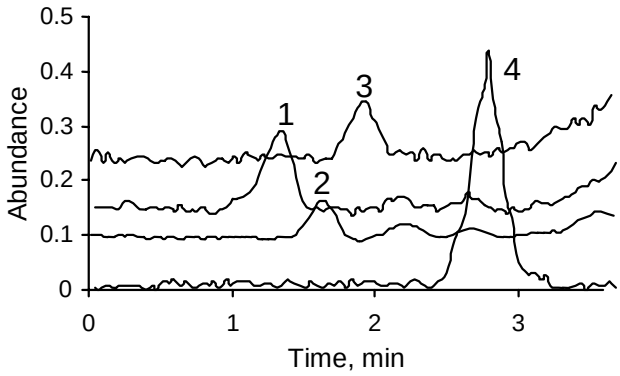

Figure 5.8. TPD-MIMS data of a gas sample (25 mL) containing the following compounds in low concentrations: ***1*** *dichloromethane 6.0 ng (m/z 84),* ***2*** trans-*1,2-dichloroethene 7.8 ng (m/z 61),* ***3*** *chloroform 5.5 ng (m/z 83) and* ***4*** *tetrachloroethene 3.6 ng (m/z 166) using HayeSepD adsorbent.*

Table 5.6. Detection limits of some selected VOCs by TPD-MIMS using HayeSepD adsorbent. The values were obtained from at least triplicate measurements in SIM mode in the range 1.2–18 ng. The detection limit was defined in terms of a signal-to-noise ratio of 3.

Compound	Detection limit, ng	Ion measured, m/z
Dichloromethane	1.1	84
trans-1,2-Dichloroethene	1.0	61
Chloroform	1.1	83
Benzene	0.4	78
Carbon tetrachloride	7.8	117
Trichloroethene	0.2	132
Toluene	0.2	92
Tetrachloroethene	0.3	166
Xylenes	1.0	106
Styrene	0.8	104

The linear dynamic range of the TPD-MIMS system was tested with toluene as an example. For samples containing total amounts of toluene between 0.5 ng and 50 ng, we obtained a linear relationship (linear regression coefficient 0.999) between the amount and the peak height of the desorption profiles. At sample amounts higher than 100 ng, we observed deviations from linearity. The linear dynamic range of the TPD-MIMS system was thus estimated to be 3 orders of magnitude. The memory effect/carry-over from one sample to the next is minimal, probably because of the continuous purging of pure helium through the adsorbent.

6. Analysis of semivolatiles from aqueous samples by MIMS

6.1 Trap-and-release membrane inlet mass spectrometry

6.1.1 Performance characteristics of trap-and-release MIMS

The new trap-and-release MIMS system [166] deviates from the original method [146] in the way it induces the rapid heating of the membrane. Instead of simply interrupting the liquid flow in the membrane inlet, an air plug is passed through it. In this way energy is not used for heating of the sample liquid inside the membrane, but only for heating of the membrane material itself. The result is a narrower desorption peak. As a practical demonstration of the technique the quantitative determination of caffeine in coffee and tea is presented [VI]. Because caffeine has low vapor pressure and high water solubility, the determination of caffeine represents the limits of the technique.

We found that the maximal desorption signal from caffeine was obtained after 20 minutes of sampling. This corresponds to the time required to reach a steady state flow [146], at which the number of caffeine molecules entering the membrane equals the number of molecules leaving it again either to the sample solution or into the vacuum. The sample flow rate was 1.0 mL/min. An increase in sample flow rate did not result in higher desorption intensities, and a reduction in sample flow rate resulted in insufficient cooling of the membrane.

Figure 6.1 shows the desorption profiles of three compounds with widely different melting and boiling points. In this experiment a standard solution containing 170 µg/L toluene (melting point (mp) -95°C, boiling point (bp) 110°C), 3.6 mg/L 1-naphthalene methanol (mp 64°C, bp 301 °C) and 270 mg/L caffeine (mp 238°C, bp not reported) was passed through the inlet for 20 minutes before the trapped molecules were released during the passage of a 50-second airplug. As expected, the low molecular weight and volatile compound toluene reached a steady state flow through the membrane before the airplug had passed through the system, as evidenced by its elevated level prior to 15 seconds in the Figure 6.1. 1-Naphthalene methanol did not diffuse through the

cold membrane (at room temperature) as fast as toluene and it did not reach a steady state flow within the 20 minutes before the passage of the airplug. Caffeine is expected to have a response time comparable to that of 1-naphthalene methanol, but because of its extremely low vapor pressure it was not observed at all during the 20-minute sampling period (standard MIMS mode). At the time when the airplug reaches the membrane (at time of 15 s in Figure 6.1) the signals from both toluene and 1-naphthalene methanol increase immediately because of the elevated temperature of the membrane, whereas the signal from caffeine has a delay of about 6 seconds before it starts to rise. The delay in the caffeine signal is probably the result of an interruption in the membrane heating at 100°C until residual liquid inside the membrane is vaporized [166]. Caffeine, with its extremely low vapor pressure, probably does not evaporate from the polydimethylsiloxane membrane at temperatures below 100°C. The measured width of the desorption profiles at half height was 8.5, 12.7 and 12.3 seconds for toluene, 1-naphthalene methanol and caffeine, respectively. The difference is a result of a slower diffusion of 1-naphthalene methanol and caffeine than of toluene in the silicone membrane.

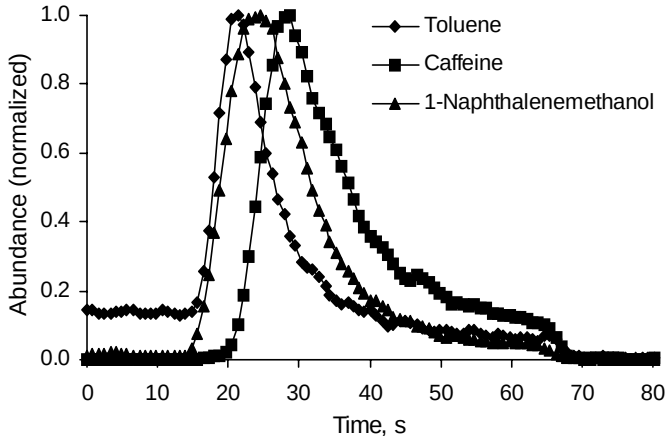

Figure 6.1. T&R-MIMS desorption profiles of toluene (m/z 91 monitored), 1-naphthalene methanol (m/z 158 monitored) and caffeine (m/z 194 monitored) obtained during the passage of a 50-second airplug.

Table 6.1 shows a comparison of measured detection limits for a variety of semivolatile compounds obtained with T&R-MIMS and standard MIMS. The improvement factors (detection limits with standard MIMS compared to those

with T&R-MIMS) are from 5 (fluoranthene) to >> 100 (caffeine). It is interesting that the improvement factors are largest (50 or higher) for the compounds which are most difficult to measure with standard MIMS. These are typically relatively polar compounds, which do not dissolve very well in the membrane. With the T&R-MIMS system, detection limits for relatively polar semivolatile compounds are lowered typically from mg/L to µg/L levels. On the other hand, VOCs such as toluene and trichloroethene are best measured with a standard MIMS system because with the VOCs the increase in the background is greater than the increase in the responses in the T&R-MIMS method.

Table 6.1. Comparison of detection limits with standard MIMS and T&R-MIMS.

Compound	Bp (°C)	Water solubility[b]	Ion monitored (m/z)	Detection limit[c] (µg/L)		Improvement factor
				Standard	T&R	
DDT	260	1	235	1 000	25	40
Phenoxyacetic acid[a]	285	3	107	10 000	100	100
4-Phenylphenol	305	2	170	100	2	50
Phenanthrene	340	1	178	4	0.5	8
Fluoranthene	385	1	202	25	5	5
Acetylsalicylic acid[a]	135[d]	3	120	20 000	250	80
Caffeine	238[d]	2	194	Nd	600	>>100

[a] pH adjusted to 2;

[b] 1=insoluble, 2=slightly soluble, 3=soluble, 4=very soluble;

[c] Standard MIMS: S/N=3; T&R-MIMS: the concentration that causes a 50% increase in signal as compared to a blank;

[d] Melting point;

Nd = not detectable.

In the experimental setup described here the membrane is continuously bombarded with light and electrons from the filament, also during the sampling period. This means that there must be a temperature gradient across the membrane, with the vacuum side warmer than the sample side. An obvious possibility to improve the detection limits would therefore be to turn off the filament during the sampling period or at least to deflect the electrons away from the ion source. In this way the membrane temperature at the vacuum side would be lowered and a greater sorption of sample molecules should result. The system was tested with an electron trap (active during the sampling period) mounted near the filament. However, this gave no difference in the performance of the system and it was concluded that most of the radiation energy hitting the membrane comes from the light and not from the electron bombardment. Only a few attempts were made to test the effect of turning the filament off during the sampling period, mainly because it is our experience with the Balzers quadrupole systems that the lifetime of the filament is drastically reduced when the filament is frequently turned on and off.

6.1.2 Quantitative analysis of semivolatiles by T&R-MIMS

One requirement for quantitative determination is a system with a low memory effect. The T&R-MIMS system is particularly sensitive to memory effects, since the part of the membrane which binds to the steel capillaries is not sufficiently heated during the desorption step. Sample molecules dissolved in this “cold” part of the membrane will therefore diffuse back to the center of the membrane and be released in the next desorption. The result is a carry-over from one sample to the next and cleaning between samples becomes necessary. In the T&R-MIMS system two parameters can be used in a cleaning process: (a) the duration of a flushing period with pure water and (b) the use of a cleaning airplug.

The duration of the cleaning airplug turned out to be much more important than that of the flushing period. Without the cleaning airplug the memory effect was almost 30% but it dropped to about 2% with a 50-second airplug. It is interesting to note that the duration of the airplug has only little effect up to 20 seconds, after which a fast drop is observed. This behavior probably reflects the temperature profile of the membrane during the passage of the airplug. At first the temperature rises rapidly to 100°C, where it stays for a short time until all

residual liquid inside the membrane has evaporated [166]. In this period, which takes about 10 seconds, the temperature is too low to vaporize the residual caffeine. With other compounds of higher volatility than caffeine, the cleaning step was even more effective, e.g. the residual amount of 1-naphthalene methanol in the second cleaning step was only 1.6% after sampling of 4 mg/L of 1-naphthalene methanol for 20 min (the signal height was equal to that of 300 mg/L caffeine).

Repeatability and stability are very important parameters of analytical methods. To test the T&R-MIMS system we analyzed a standard solution of 100 mg/L caffeine ten times according to a procedure in which the standard solution was passed through the inlet system for 20 minutes before an airplug of 50 seconds was used to release the caffeine. Between each analysis the system was cleaned two times with 130 seconds of pure water followed by an airplug of 50 seconds. The relative standard deviation was calculated as 4% and 3% using peak heights and areas, respectively. Linearity is another very important factor of an analytical system. In order to test this for the trap-and-release system we measured the intensities of the desorption profiles over a broad range of concentrations. We found that the T&R-MIMS system was linear over 3 orders of magnitude.

To test the capabilities of the T&R-MIMS technique with unknown samples, we analyzed the caffeine content in a typical cup of tea or coffee. Figure 6.2a shows the mass spectrum of a cup of tea obtained during desorption of the sample. The spectrum represents the average of six scans of the whole spectrum each recorded with a scan rate of 50 ms/amu. The molecular ion of caffeine (m/z 194) and the most abundant fragment ion (m/z 109) are clear, as are the peaks of m/z 147 (fragment from the membrane) and m/z 149 (phthalate plastizicers). The particular tea used (Earl Gray, Twinings of London) contains Bergamot flavor, a common tea additive, and the main constituents of that substance (linalool, linalyl acetate and limonene) produce the ions at m/z 105, 107, 119, 121 and 136. The results are consistent with those obtained by other methods, e.g. by SPME-GC/MS [167]. Overall, the spectrum is quite simple and the caffeine content can be selectively determined through single ion monitoring of the molecular ion. Figure 6.2b shows the mass spectrum of a typical cup of coffee recorded in a similar fashion to the tea spectrum in Figure 6.2a. The spectrum is very complex as compared with the tea spectrum. Oils present in coffee give a

huge background of ion clusters separated by fourteen mass units. However, the dominant ions (m/z 194 and 109) from caffeine are clear and can form the basis for a quantitation, although in the case of coffee the oil background prevents the detection of concentrations below 10 mg/L. A cup of decaffeinated coffee would typically contain residual caffeine at concentrations up to 10 mg/L. The selective determination of caffeine in such samples with the T&R-MIMS technique will require the use of tandem mass spectrometry. In general, the T&R-MIMS technique would probably gain considerably with respect to both selectivity and sensitivity if it were combined with tandem mass spectrometry.

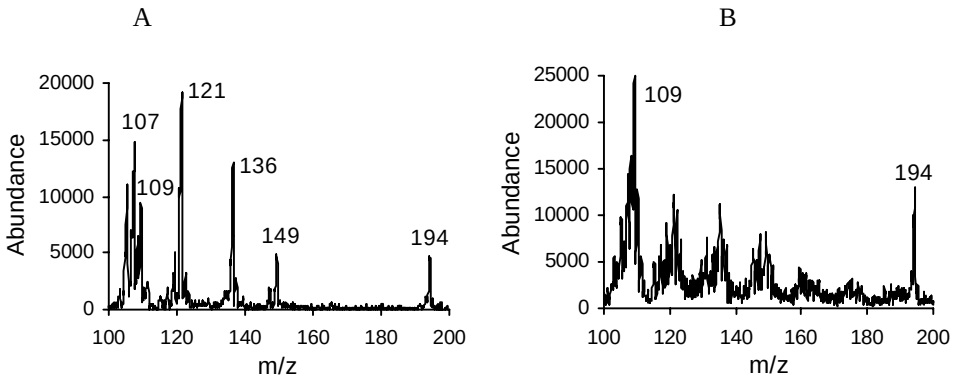

Figure 6.2. T&R-MIMS mass spectra of a typical cup of (a) tea and (b) coffee. The spectra represent the average of 6 successive scans obtained during the release period.

Table 6.2 shows the results of quantitative determinations of the caffeine content in various roasted coffee (all ecological) and tea brands obtained from a local supermarket. The concentrations are given both as the measured concentrations in the liquid (mg/L) and as the calculated amount of extractable caffeine in the pure coffee or tea (mg/g). All values represent the average of two duplicate measurements of the same sample and the relative standard deviations were below 10%. The results agree very well with reported values for the caffeine content in ground coffee and in tea bags [168, 169]. Good agreement between T&R-MIMS and HPLC determinations was found. Generally, the deviation between the T&R-MIMS and the HPLC values was slightly higher for the tea samples than for the coffee samples. This reflects problems with precipitations in some of the tea samples when they were cooled prior to the T&R-MIMS analysis.

Table 6.2. Quantitative analytical results of some coffee and tea brands.

Sample	Concentration of caffeine in extract (mg/L)	Caffeine content of the original brand (mg/g)	HPLC result (mg/L)	Difference[a] %
Coffee brand				
A	277	13.9	286	3.1
B	276	13.8	285	3.2
C	278	13.9	290	4.1
D	288	14.4	282	-2.1
E	277	13.9	287	3.5
Tea brand				
F	584	28.8	620	5.8
G	558	27.6	598	6.7
H	691	33.4	702	1.6
I	603	29.9	630	4.3
J	520	26.0	503	-3.4
K	606	30.1	577	-5.0
L	592	28.9	565	-4.8

[a] difference is (HPLC result - T&R-MIMS result)/HPLC result x 100 (as a %).

Overall, the performance characteristics of the two methods (T&R-MIMS and HPLC) were very similar. The detection limit for caffeine with the HPLC method was estimated to be somewhat lower than that of T&R-MIMS (0.1 mg/L as compared to 0.6 mg/L), but the T&R-MIMS method is still under development and it is expected that the detection limits will be improved by at least an order of magnitude. With both methods the analysis time was about 20 minutes with a sample throughput of approximately three per hour. The only real drawback of the T&R-MIMS method is the need for larger amounts of sample, 10–20 mL, as compared with HPLC, where only 0.5 mL or less is required to flush the sample loop.

6.2 Desorption chemical ionization membrane inlet mass spectrometry

6.2.1 Desorption chemical ionization and chemical ionization in MIMS

Direct or desorption chemical ionization (DCI) of relatively involatile compounds, e.g. oligopeptides, was first presented in 1973 by Baldwin and McLafferty [170]. In their experiments the sample was deposited on the surface of an extended tip of a conventional sample probe as a drop of dilute solution which was allowed to evaporate. The tip was in the middle of the ion plasma of the CI source and so it was possible to measure CI mass spectra of many oligopeptides at a temperature of 150°C, although these compounds did not show any $[M+H]^+$ ion with a conventional CI source even at 340°C. Since then, DCI has been widely applied as a desorption technique for nonvolatile, labile and polar compounds [171].

In membrane inlet mass spectrometry chemical ionization (CI) [172] has mainly been used to simplify multicomponent mass spectra, and the analyzed compounds have all been volatile compounds [28, 58, 145, 161, 173]. The first application of solvent chemical ionization in MIMS with water as a CI reagent gas was presented by Lister *et al.* [24], who used a silicone membrane and a quadrupole ion trap to analyze volatile organic compounds at ppb levels. Lauritsen *et al.* [174] used a microporous polypropylene membrane to reach CI conditions in a conventional CI ion source and glow discharge for ionization to analyze a range of organic compounds below 100 ppb levels.

We designed a new DCI source for MIMS in order to analyze polar and semivolatile organic compounds in aqueous solutions [VII]. The design of the ion source was based on simulations with the SIMION 3D 6.0 ion optical program, which was used to evaluate the performance of ion sources with different dimensions and designs (cylindrical, cubic).

6.2.2 Simulation of ion optics

The effect of the shape of the ion volume on the ion optics and on the efficiency of extraction of ions into a mass analyzer was tested with an ion source which

was used in the trap&release membrane inlet mass spectrometric studies [VI]. The number of ions used for calculations varied between 450 and 3 100 with even distribution inside the ion chamber in all experiments unless otherwise stated. The original ion volume was in the shape of a box which was open from both ends, from which the hollow fiber membrane passed through the ion volume (Fig. 6.3a). As can be predicted from this shape, it was not very effective for focusing ions formed inside the ion volume to a mass analyzer, i.e. the efficiency was about 10%. The ion volume was altered to have more closed ends, so that the space through which the membrane and the steel tubings entered the ion vacuum was much smaller (Fig. 6.3b). The membrane was so long that there was no bare steel tubing inside the ion volume. The efficiency was improved to 32% when the aperture was kept as small as possible. If the membrane was too short i.e. part of the steel tubing was inside the ion volume, the efficiency was slightly decreased, down to 25%. Whether the end of the membrane was located at the edge of the ion volume or outside the ion volume did not have any marked effect on the efficiency because the steel tubing was in any case outside the ion volume. When the ion volume was changed to a cylinder with the same dimensions as the cubic volume (Fig. 6.3c), there was a great improvement in the efficiency, to 60–70%. One reason for this was that ions produced in the corners of the cubic form were mainly expelled from the ion volume through the orifices for the membrane, not to the mass analyzer, and in the cylindrical form these corners were eliminated. In practice, the dead corners should have less impact on the efficiency because most of the ions would be produced in the middle of the ion volume, where the electrons from the filament are entering the ion volume.

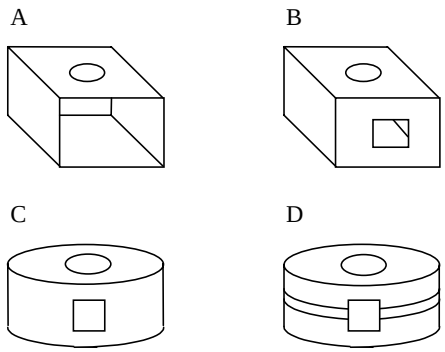

Figure 6.3. The configurations of the ion volume studied with the SIMION ion optical program.

The cylindrical ion volume was also studied in three different forms, i.e. the bottom plate of the cylinder was either open, a grid or closed. The grid and the closed ion volumes gave similar extraction efficiencies, about 67%. By contrast, as expected, the performance of the open volume was much poorer, i.e. it had an efficiency of only 19%. When the walls of the vacuum chamber were also presented in the simulations as grounded plates situated a short distance from the ion volume, the efficiency was still decreased to 10%. This shows that in simulations it is important to present the ion optics as a whole, not just the parts where voltages are applied because the grounded plates etc. might have a great impact on potentials in the system, especially if the system is very open. Of course, the open and grid forms of the cylindrical ion volumes cannot be used for chemical ionization because the pressure inside the ion volume would be too low for CI.

It was also investigated whether an ion volume made of two separate parts can be used more effectively for extracting ions to a mass analyzer. The cylindrical ion volume was split into two halves (Fig. 6.3d). In this way the potentials of the two parts can be adjusted separately, and in the simulations the lower part (opposite to the aperture) was fixed to the same or a higher potential than the upper part in order to deflect ions towards the aperture into the mass analyzer. When both parts had the same potential (+100 V) the extraction efficiency (57%) was decreased slightly from that of a closed ion volume because part of the ions escaped the ion volume from the narrow slit between the parts. The efficiency was improved to 61% when the potential of the lower part was fixed to +103 V so that the lower part acted as a deflector. A further increase in the potential of the lower part (+105 V) decreased the efficiency (55%). Although ions produced close to the lower part were more effectively deflected to the mass analyzer, ions produced close to the slit and the upper part escaped more easily through the slit. In practice this could be avoided by closing the slit with a ceramic insulator, and then the ion volume could also be used for CI, but this was never tested experimentally.

The basic idea for constructing a new inlet/ion source was to use the original parts of a single quadrupole mass spectrometer as much as possible. Because the analyzer needs a lower pressure than that normally found in a CI source, the vacuum chamber had to be split into two parts which could be pumped differentially. This was easy to achieve with a Balzers cross beam ion source

because there was a grounded plate between the extraction and focus lenses, and this plate could easily be extended to divide the vacuum chamber into two parts. The focus lens and the rest of the analyzer could then be the original ones but the parts of the ion source before the grounded plate had to be designed in a new way.

Figure 6.4 shows an example of the new design drawn in the SIMION program. This model was used in simulation experiments in which parameters such as the size of apertures and the potentials of different parts of the ion source were tested. The results of simulation concerning different potentials are presented in Table 6.3. The potentials of focus and field lenses did not have a great effect on the efficiency, as all the efficiencies were between 25 and 42%. The maximum efficiency was achieved at potential values of 105 V for the focus lens and 100 V for the field lens, and these values were very close to the optimum values for the original cross beam ion source. The efficiency increased with decreasing extraction voltage, i.e. the efficiency increased from 10% at -10 V to 50% at -200 V. This was as expected because more ions can be extracted through a small orifice if the power for extraction is greater. The size of the ion source apertures (an aperture for ionizing electrons from a filament and an orifice to the analyzer) had a predictable effect on the efficiency, i.e. the larger the orifice to the analyzer and the smaller the aperture for the electrons, the better was the efficiency because in that case the ions formed did not escape the ion source through the aperture for the electrons but were focused into the analyzer. The actual sizes of the apertures were determined, not by simulations, but by practice because the apertures should not be too large, otherwise the pressure inside the ion source would be too low for CI. However, these simulations gave only a prediction of how the ion source would work, not a real picture of the source, because the effects of the membrane and of the ion plasma could not be taken into account in these simulations. This means, of course, that the final design had to be made according to experiments with a real ion source, not with simulations.

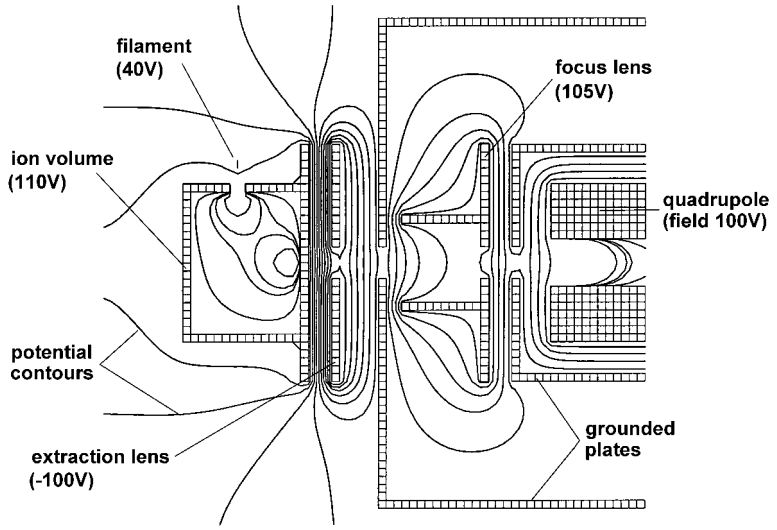

Figure 6.4. SIMION ion optics simulation of a new inlet/ CI source design.

Table 6.3. Effects of the potentials of focus, field and extraction lenses on the efficiency of extracting ions from the ion source to the analyzer. Values were obtained from SIMION simulations.

Focus		Field		Extraction	
Potential (V)	Eff. %	Potential (V)	Eff. %	Potential (V)	Eff. %
85	33	70	26	-10	11
95	28	80	25	-50	23
100	31	90	29	-100	34
105	42	100	42	-150	42
110	35	105	33	-200	50

6.2.3 Performance of the CI inlet/ion source

The final design of the CI inlet/ion source is presented in Figures 6.5 and 6.6. To simplify the illustration only half of the membrane is shown in the figure, and the filament is drawn on the same axis as the membrane although in reality it is located perpendicularly to the membrane. The ion volume was cylindrical, and the membrane was located as close to the exit orifice as possible in order to ensure effective transport of analyte ions to the analyzer. Pressure in the main

vacuum chamber (the left side in Figure 6.6) was between 5 x 10^{-4} and 5 x 10^{-3} mbar during operation, and so the estimated pressure inside the CI source was between 0.2 and 2.0 mbar as calculated from the conductances through the apertures. The length of the polyacrylonitrile (PAN) membrane inside the ion source was approximately 13 mm, and this length appeared to be sufficient for the flux of water through it to be high enough to achieve CI conditions in the ion source. As an example of this, Figure 6.7 shows a water CI plasma measured by DCI-MIMS. The major ions in the plasma are the water cluster ions $[H_3O]^+$ (m/z 19), $[(H_2O)_3H]^+$ (m/z 55), $[(H_2O)_2H]^+$ (m/z 37) and $[(H_2O)_4H]^+$ (m/z 73). The ion $[(H_2O)_5H]^+$ (m/z 91) was also present, but its abundance was rather low, below 1%. The structure of the plasma is rather similar to that obtained by Lauritsen *et al.* [174] using a microporous polypropylene membrane and glow discharge, except that the base peak was now m/z 19, not m/z 55. One reason for this difference might be collisions of the larger water clusters before reaching the analyzer, so that they were dissociated into smaller ions m/z 19, as evidenced by Lauritsen *et al.* [174]. Another reason could be that the pressure was lower in our experiments so that less water cluster ions were generated.

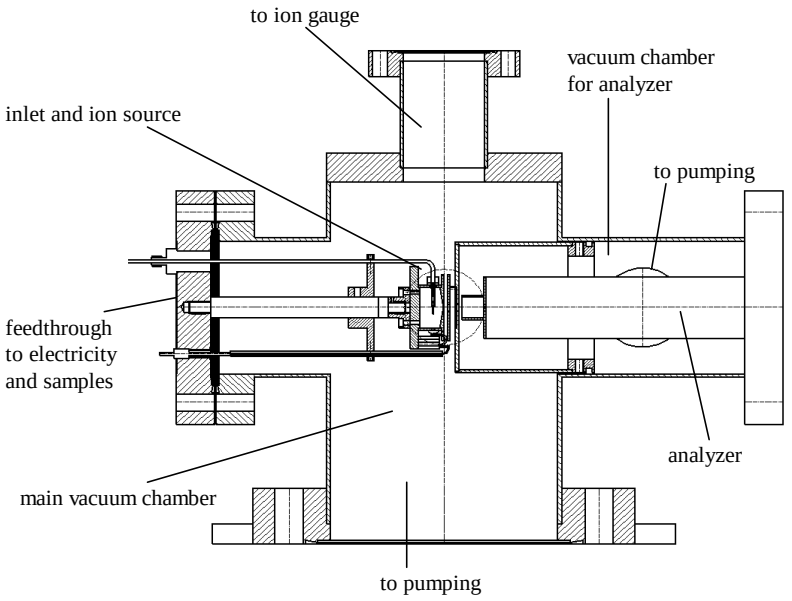

Figure 6.5. Vacuum chambers, membrane inlet, ion source and ion optics of the DCI-MIMS system.

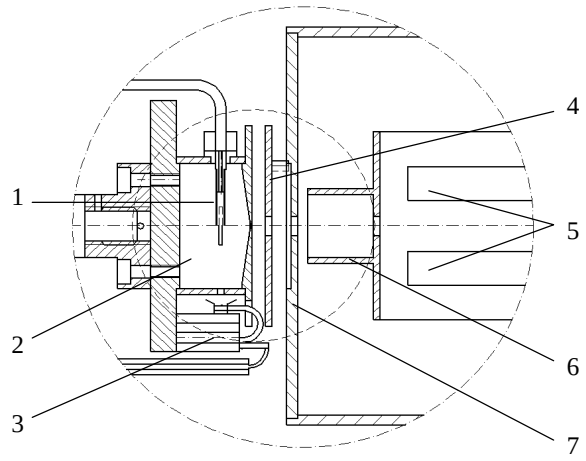

Figure 6.6. Enlarged figure of the inlet/ion source of the DCI-MIMS system. ***1*** *Membrane,* ***2*** *ion volume,* ***3*** *filament,* ***4*** *extractor lens,* ***5*** *field (quadrupole analyzer),* ***6*** *focus lens,* ***7*** *grounded plate (divider of the vacuum chamber).*

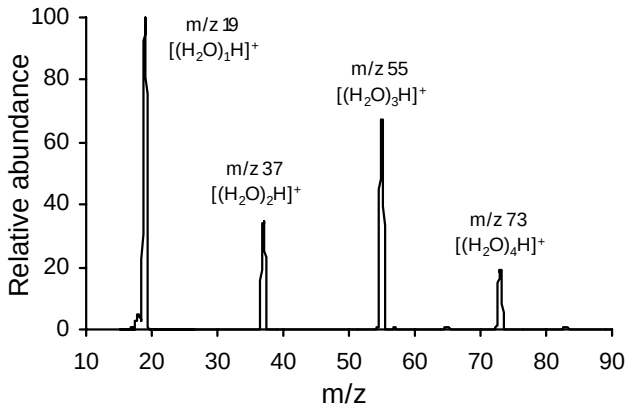

Figure 6.7. Water CI plasma measured by DCI-MIMS.

DCI-MIMS was used for analysis of polar and less volatile organic compounds in aqueous samples. It was possible to measure water CI mass spectra of dicarboxylic acids, malonic acid ($CO(OH)CH_2COOH$) and succinic acid ($CO(OH)CH_2CH_2COOH$), which have not been measured by MIMS before (Figure 6.8). Polar compounds such as monocarboxylic acids (e.g. 2-oxoglutaric acid [175]) have previously been measured by MIMS but usually the acids have first been esterified and then measured with a silicone membrane. However, low

molecular weight carboxylic acids such as acetic acid can be measured directly by MIMS [28]. With the DCI-MIMS system the dicarboxylic acids could be measured directly without any derivatization. In these experiments T&R-technique was applied to measure the mass spectra because in the T&R-mode the signal-to-noise ratio was better than in the standard MIMS mode. The pH of samples was adjusted to 1–2 to ensure that acids were not in an ionized form during the experiments. In the background subtracted CI mass spectrum of malonic acid (Fig. 6.8a), the base peak is the proton-bound water cluster of the molecule m/z 123 ($[M+H+H_2O]^+$) and another abundant peak is the protonated molecule m/z 105 ($[M+H]^+$). In contrast to the mass spectrum of malonic acid, the base peak in the mass spectrum of succinic acid (Fig. 6.8b) is the protonated molecule m/z 119 ($[M+H]^+$), and the second abundant peak is the ion m/z 101 ($[M+H–H_2O]^+$), which results from the protonated molecule loosing a water molecule. The monohydrated protonated molecule m/z 137 ($[M+H+H_2O]^+$) is also abundant (about 30% of the base peak). The reason for the difference between the mass spectra is the molecular size of the acids; the carbonyl groups in malonic acid are close to each other so that both groups can have a hydrogen bonding to the same water molecule, whereas the carbonyl groups in succinic acid are further away from each other and hydrogen bondings to the same water molecule are not so likely. On the other hand, protonated succinic acid looses water more easily than malonic acid because the lost of water molecule from succinic acid generates a stable anhydride.

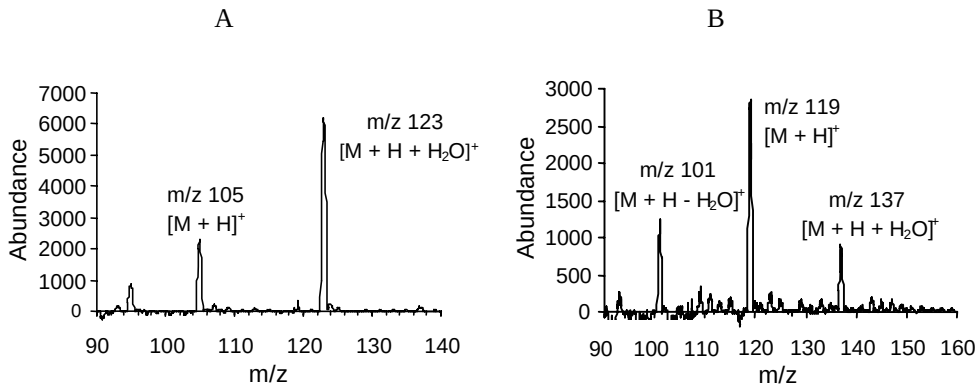

Figure 6.8. Water CI mass spectra of (A) malonic acid and (B) succinic acid. The mass spectra are background subtracted, and the concentrations of analytes are about 1 g/L.

Chemical ionization has normally been used in MIMS to analyze volatile organic compounds and the ionization process has occurred after the analytes have evaporated into the vacuum. In our system the membrane was in the middle of the water CI plasma, which is believed to assist the desorption of the analytes from the membrane in the standard MIMS mode and even more in the T&R-mode, in which the membrane is heated up by the filament during the passage of an airplug. The temperature of the ion source was between 100 and 140°C, depending on whether the ion source was heated by the filament or heated electrically. In some experiments the temperature was increased above 150°C to assist the desorption of molecules from the membrane but at these higher temperatures both the membrane and the glue by which the membrane was attached to the steel tubings were damaged and the pressure increased excessively.

DCI-MIMS was also applied to measure glucose, a nonvolatile monosaccharide, in aqueous solution because it has been shown that glucose can be measured by MIMS using a polyacrylonitrile membrane [39, 176] and it is of a great interest to measure glucose on-line in fermentation monitoring. However, it was not possible to measure glucose with DCI-MIMS. Firstly, the membrane in my experiments was much thicker (300 µm) than that in earlier experiments by Kotiaho [176] (45 µm). Secondly, during sampling of a glucose solution into the MIMS inlet, the pressure in the vacuum chamber continuously decreased while the background of the mass spectrometer continuously increased. This meant that glucose was trapped in the membrane, which prevented water from flowing through it. When less water flowed through the membrane, the pressure decreased and the temperature of the membrane increased, resulting in a higher background. When pure water was again flushed to the inlet, it dissolved the glucose residue from the membrane and both the pressure and the background returned to their original state. It appeared that the flux of glucose through the PAN membrane was diffusion dependent and that the temperature of the membrane and CI plasma was not sufficient to desorb glucose into the vacuum. A practical difficulty in the system was the filament (either rhenium or tungsten), which lasted only for a few hours before burning, due to reaction with water. Glow discharge ionization would be solution to this problem, but then

T&R technique could not be used efficiently. Other heating methods, such as laser desorption which has been used with MIMS in the analysis of polycyclic aromatic compounds (PAHs) [177], must be considered to enhance the desorption of semivolatile compounds from the membrane.

7. Conclusions and future perspectives

The standard MIMS method was compared to two other analytical methods, purge-and-trap gas chromatography-mass spectrometry (P&T-GC/MS) and static headspace gas chromatography (HSGC), for the analysis of volatile organic compounds in water samples as used in routine analysis. The performance characteristics of the MIMS method were rather similar to those of the other two methods, e.g. detection limits by MIMS are as low as those obtained by the P&T-GC/MS method and clearly lower than with the HSGC method, and the linear dynamic range is greater than with the P&T-GC/MS method. The main advantage of the MIMS method is the very short analysis time (only a few minutes), whereas the main disadvantage is the lack of separation of individual analytes. However, this problem can be solved by temperature-programmed desorption MIMS.

It was also demonstrated that membrane inlet mass spectrometry is an excellent analytical method for rapid on-site environmental analysis. The advantages of MIMS for this kind of application include sub or low μg/L detection limits directly from water samples without any preconcentration, short response times and simplicity of instrumentation. In addition, one important point to note is that even though standard MIMS does not provide any separation of the analytes in the usual chromatographic way, it allows rapid identification of the major pollutants of the contaminated samples, which is also a very important requirement during rapid on-site analysis. Due to the many demonstrated advantages of membrane inlet mass spectrometry it is foreseen that the application of MIMS in on-site environmental analysis or in on-site process control will become more popular.

Membrane inlet mass spectrometry is a very powerful tool for analyzing volatile organic compounds directly from air without any pre-treatment or preconcentration. VOCs can be detected in air at low or sub $\mu g/m^3$ levels, meaning that the method is suitable for the analysis of VOCs in indoor and outdoor air. Optimization of the membrane and sample flow parameters is essential as these parameters, e.g. temperature of the membrane/inlet, dramatically affect the behavior of the membrane and in this way the results of the analysis. Response times of only a few seconds were measured for the test compounds with the thin (25 μm) sheet membrane. With a short sampling time it is possible to analyze even 50–100 samples in one hour. Furthermore, the

MIMS method is believed to be suitable for on-line/on-site monitoring of VOC emissions of industry and transportation and for identification of malodor problems.

However, it can be difficult to analyze complex mixtures by standard MIMS because there is no separation of compounds prior to mass spectrometric detection. This problem was solved by combining MIMS with temperature-programmed desorption. A typical desorption time is 3 minutes with a heating rate of 50°C/min, and the whole cycle of analysis, i.e. sampling, temperature-programmed desorption and cooling of the adsorbent, can be completed in 6–10 minutes. The detection limits are comparable to those obtained with standard MIMS, i.e. at low or sub ng/L levels if one liter air samples are taken. The memory effects and risks of contamination are very low. In future the TPD-MIMS system may be improved with respect to resolution and detection limits through optimization of the dimensions of the TPD system and through closer study of the characteristics of various adsorbents. Furthermore, it will be investigated whether the whole TPD system can be simplified by mounting the adsorbent directly on top of the membrane and then temperature-programming both the adsorbent and the membrane inlet together. An expansion of the technique to include water samples will also be studied. The TPD system could also be connected to measurement devices other than MIMS, e.g. to a gas chromatographic detector or a Fourier transform infrared (FTIR) spectrophotometer, or even directly to a mass spectrometer when higher concentrations are to be measured.

Trap&release MIMS can be used for the quantitative determination of semivolatile organic compounds in solution. Detection limits for the semivolatiles are in the µg/L range and the linearity of the technique is 3 orders of magnitude. The reproducibility of the method is high. I expect that, with some optimization, the T&R-MIMS method could find applications such as the determination of pharmaceuticals in urine samples or pesticides in environmental samples. However, such applications will probably require the use of tandem mass spectrometry in order to improve selectivity and sensitivity. Another T&R technique for the analysis of semivolatile compounds was developed. In this technique, desorption chemical ionization MIMS, a hydrophilic polyacrylonitrile membrane is in a closed CI source and the water permeating through the membrane is used as a reagent gas. This system allows

the analysis of polar dicarboxylic acids from aqueous solution without any derivatization. The development of this system is still in its early stages, and the system could be improved e.g. by applying glow discharge or laser desorption instead of electron ionization and by making a more compact ion source in order to improve detection limits.

Overall, it was demonstrated that the standard MIMS method is very suitable for the analysis of volatile organic compounds both in air and in water, and that modifications of the standard MIMS method, i.e. T&R, TPD and DCI-MIMS methods, can extend the range of analyzable compounds to polar and semivolatile compounds. In future this trend will continue, i.e. the analysis of polar and semivolatile compounds by MIMS will become routine, and it will be possible to analyze even more complex mixtures.

References

1. LaPack, M.A., Tou, J.C., Enke, C.G. *Anal. Chem*. **62** (1990), p. 1265.
2. Kotiaho, T., Lauritsen, F.R., Choudhury, T.K., Cooks, R.G., Tsao, G.T. *Anal. Chem.* **63** (1991), p. 875A.
3. LaPack, M.A., Tou, J.C., McGuffin, V.L., Enke, C.G. *J. Membrane Sci.* **86** (1994), p. 263.
4. Lauritsen, F.R. *Int. J. Mass Spectrom. Ion Proc.* **95** (1990), p. 259.
5. Hoch, G., Kok, B. *Arch. Biochem. Biophys.* **101** (1963), p. 160.
6. Westover, L.B., Tou, J.C., Mark, J.H. *Anal. Chem.* **46** (1974), p. 568.
7. Piehl, H., Wagner, F., Reuss, M. *Eur. J. Appl. Microbiol.* **1** (1975), p. 323.
8. Jørgensen, L., Degn, H. *Biotechnol. Lett.* **9** (1987), p. 71.
9. Calvo, K.C., Weisenberger, C.R., Anderson, L.B., Klapper, M.H. *Anal. Chem.* **53** (1981), p. 981.
10. Calvo, K.C., Weisenberger, C.R., Anderson, L.B., Klapper, M.H. *J. Am. Chem. Soc.* **105** (1983), p. 6935.
11. Degn, H., Kristensen, B.J. *J. Biochem. Biophys. Meth.* **12** (1986), p. 305.
12. Kana, T.M., Darkangelo, C., Hunt, M.D., Oldham, J.B., Bennett, G.E., Cornwall, J.C. *Anal. Chem.* **66** (1994), p. 4166.
13. Lauritsen, F.R., Lloyd, D. In: *Mass Spectrometry for the Characterization of Microorganisms*, Fenselau, C. (ed.), *ACS Symposium Series*, **541**, American Chemical Society, Washington DC, 1994. Pp. 91–106.
14. Lauritsen, F.R., Gylling, S. *Anal. Chem.* **67** (1995), p. 1418.
15. Srinivasan, N., Kasthurikrishnan, N., Cooks, R.G., Krishnan, M.S., Tsao, G.T. *Anal. Chim. Acta* **316** (1995), p. 269.
16. Wong, P.S.H., Srinivasan, N., Kasthurikrishnan, N., Cooks, R.G., Pincock, J.A., Grossert, J.S. *J. Org. Chem.* **61** (1996), p. 6627.
17. Bohatka, S. *Rapid Commun. Mass Spectrom.* **11** (1997), p. 656.
18. Hansen, K.F., Degn, H. *Biotechnol. Techn.* **10** (1996), p. 485.
19. Heinzle, E., Reuss, M. In: *Mass spectrometry in biotechnological process analysis and control*, Plenum Press, New York, 1987. P. 241.

20. Willaert, R.G. In: *Membrane mass spectrometry, immobilised living cell systems*, R.G. Willaert, G.V. Baron, L. De Backer (eds.), John Wiley & Sons, Chichester, 1996. Pp. 177–193.

21. Degn, H. *J. Microbiol. Methods* **15** (1992), p. 185.

22. Lauritsen, F.R. In: *Proceedings of the COMETT II Course "Advanced Instrumentation, Data Interpretation, and Control of Biotechnological Processes"*, October 24–27, 1994, Gent, Belgium, Kluwer Academic Press 1998.

23. Harland, B.J., Nicholson, P.J.D., Gillings, E. *Water Res.* **21** (1987), p. 107.

24. Lister, A.K., Wood, K.L., Cooks, R.G., Noon, K.R. *Biomed Environ. Mass Spectrom.* **18** (1989), p. 1063.

25. Dheandhanoo, S, Dulak, J. *Rapid Commun. Mass Spectrom.* **3** (1989), p. 175.

26. Slivon, L.E., Bauer, M.R., Ho, J.S., Budde, W.L. *Anal. Chem.* **63** (1991), p. 1335.

27. Hansen, K.F., Lauritsen, F.R., Degn, H. *Biotechnol. Bioeng.* **44** (1994), p. 347.

28. Hayward, M.J., Kotiaho, T., Lister, A.K., Cooks, R.G., Austin, G.D., Narayan, R., Tsao, G.T. *Anal. Chem.* **62** (1990), p. 1798.

29. Gregori, A.M., Kephart, L.A. *Adv. Instrum. Control* **50** (1996), p. 1179.

30. Harland, B.J., Nicholson, P.J. *Sci. Total Environ.* **135** (1993), p. 37.

31. Wilson, H.K., Ottley, T.W. *Biomed. Mass Spectrom.* **8** (1981), p. 606.

32. LaPack, M.A., Tou, J.C., Enke, C.G. *Anal. Chem.* **63** (1991), p. 631.

33. Wong, P.S.H., Cooks, R.G. *Anal. Chim Acta* **310** (1995), p. 387.

34. Wong, P.S.H., Cooks, R.G., Cisper, M.E., Hemberger, P.H. *Environ. Sci. Technol.* **29** (1995), p. 215A.

35. Bauer, S.J., Cooks, R.G. *Talanta* **40** (1993), p. 1031.

36. Ojala, M., Ketola, R.A., Mansikka, T., Kotiaho, T., Kostiainen, R. *J. High Resolut. Chromatogr.* **20** (1997), p. 165.

37. Ojala, M., Ketola, R.A., Sorsa, H., Kotiaho, T. *Talanta* **44** (1997), p. 1253.

38. Cooks, R.G., Kotiaho, T. In: *Pollution Prevention in Industrial Processes: The Role of Process Analytical Chemistry*, Breen, J.J., Dellarco, M.J. (eds), *ACS Symposium Series*, **508**, American Chemical Society, Washington DC, 1992. Pp. 126–154.

39. Lauritsen, F.R., Kotiaho, T. *Rev. Anal. Chem.* **15** (1996), p. 237.

40. Kotiaho, T. *J. Mass Spectrom.* **31** (1996), p. 1.

41. Kotiaho, T., Ketola, R.A., Ojala, M., Mansikka, T., Kostiainen, R. *Am. Environ. Lab.* 3/97 (1997), p. 19.

42. Srinivasan, N., Johnson, R.C., Kasthurikrishan, N., Wong, P., Cooks, R.G. *Anal. Chim. Acta* **350** (1997), p. 257.

43. Bauer, S.J., Cooks, R.G. *Am. Lab.* **25** (1993), p. 36.

44. Bauer, S. *Trends Anal. Chem.* **14** (1995), p. 202.

45. Llewellyn, P.M., Littlejohn, D.P. In: *Pittsburgh Conference on Analytical Chemistry and Applied Spectroscopy*, Denver CO, USA, June 1966. Presentation.

46. Llewellyn, P.M., Littlejohn, D.P. U.S. Patent 3429105, 1969.

47. McFadden, W. In: *Techniques of Combined Gas Chromatography/Mass Spectrometry*, John Wiley and Sons, New York, 1973.

48. Black, D.R., Flath, R.A., Teranishi, R. *J. Chromatogr. Sci.* **7** (1969), p. 284.

49. Melcher, R.G., Morabito, P.L. *Anal. Chem.* **62** (1990), p. 2183.

50. Mitra, S., Zhu, N., Li, Z. In: *Proceedings in 18th Int. Symp. Capillary Chromatography*, Riva del Garda, Italy, 1996, Hüthig, Heidelberg. Pp. 154–162.

51. Xu, Y.H., Mitra, S. *J. Chromatogr. A* **688** (1994), p. 171.

52. Matz, G., Trinks, H. *Int. J. Mass Spectrom. Ion Phys.* **43** (1982), p. 79.

53. Matz, G., Kesners, P. *Analusis* **20** (1992), p. M53.

54. Jones, P.R., Yang, S.K. *Anal. Chem.* **47** (1975), p. 1000.

55. Melcher, R.G., Bakke, D.W., Hughes, G.H. *Anal. Chem.* **64** (1992), p. 2258.

56. Ouyang, S., Chen, Y.H., Xu, Y. *Anal. Chim. Acta* **337** (1997), p. 165.

57. Ouyang, S., Xu, Y., Chen, Y.H. *Anal. Chem.* **70** (1998), p. 931.

58. Brodbelt, J. S., Cooks, R.G. *Anal. Chem.* **57** (1985), p. 1153.

59. Dejarme, L.E., Bauer, S.J., Cooks, R.G., Lauritsen, F.R., Kotiaho, T., Graf, T. *Rapid Commun. Mass Spectrom.* **7** (1993), p. 935.

60. Canham, J.S., Pacey, G.E. *Anal. Chim. Acta* **214** (1988), p. 385.

61. Canham, J.S., Pacey, G.E. *Anal. Lett.* **21** (1988), p. 1619.

62. Tsai, G.-J., Austin, G.D., Sui, M.J., Tsao, G.T., Hayward, M.J., Kotiaho, T., Cooks, R.G. *Anal. Chem.* **63** (1991), p. 2460.

63. Bohatka, S., Degn, H. *Rapid Commun. Mass Spectrom.* **5** (1991), p. 433.

64. Degn, H., Bohatka, S., Lloyd, D. *Biotechnol. Techn.* **6** (1992), p. 161.

65. Lauritsen, F.R., Kotiaho, T., Choudhury, T.K., Cooks, R.G. *Anal. Chem.* **64** (1992), p. 1205.

66. Soni, M., Bauer, S., Amy, J.W., Wong, P., Cooks, R.G. *Anal. Chem.* **67** (1995), p. 1409.

67. Mendes, M.A., Pimpin, R.S., Kotiaho, T., Eberlin, M. *Anal. Chem.* **68** (1996), p. 3502.

68. Shoemaker, J.A., Bellar, T.A., Eichelberger, J.W., Budde, W.L. *J. Chromatogr. Sci.* **31** (1993), p. 279.

69. Kostiainen, R., Kotiaho, T., Ketola, R.A., Virkki, V. *Chromatographia* **41** (1995), p. 34.

70. Kostiainen, R., Kotiaho, T., Mattila, I., Mansikka, T., Ojala, M., Ketola, R.A. *Anal. Chem.* **70** (1998), p. 3028.

71. Rivlin, A.A. *Rapid Commun. Mass Spectrom.* **9** (1995), p. 397.

72. Xu, C., Patrick, J.S., Cooks, R.G. *Anal. Chem.* **67** (1995), p. 724.

73. Maden, A.J., Hayward, M.J. *Anal. Chem.* **68** (1996), p. 1805.

74. Baumgardner, J.E., Quinn, J.A., Neufeld, G.R. *J. Mass Spectrom.* **30** (1995), p. 563.

75. Johnson, R.C., Kock, K., Kasthurikrishnan, N., Plass, W., Patrick, J.S., Cooks, R.G. *J. Mass Spectrom.* **32** (1997), p. 1299.

76. Yakovlev, B.S., Talrose, V.L., Fenselau, C. *Anal. Chem.* **66** (1994), p. 1704.

77. Cisper, M.E., Hemberger, P.H. *Rapid Commun. Mass Spectrom.* **11** (1997), p. 1454.

78. Virkki, V., Ketola, R., Juujärvi, J., Oja, E., Sorsa, H., Ojala, M., Kotiaho, T. *Development of calculation program for the analysis of multicomponent spectra*, 29th Annual Conference of the Finnish Physical Society, Jyväskylä, Finland, March 1995. Presentation.

79. Ketola, R.A., Ojala, M., Komppa, V., Kotiaho, T. Juujärvi, J., Heikkonen, J. *Rabid Commun. Mass Spectrom.* 1998, submitted.

80. Ohorodnik, S., Shaffer, R.E., Callahan, J.H., Rose-Pehrsson, S.L. *Anal. Chem.* **69** (1997), p. 4721.

81. Overney, F.L., Enke, C.G. In: *Proceedings of the 45th ASMS Conference on Mass Spectrometry and Allied Topics*, Palm Springs, CA, USA, May 1997. P. 1227.

82. Overney, F.L., Enke, C.G. *J. Am. Soc. Mass Spectrom.* **7** (1996), p. 95.

83. Lauritsen, F.R., Nielsen, L.T., Degn, H., Lloyd, D., Bohatka, S. *Biol. Mass Spectrom.* **20** (1991), p. 253.

84. Cook, K., Falconer, J., Bennett, K. In: *Proceedings of the 45th ASMS Conference on Mass Spectrometry and Allied Topics*, Palm Springs, CA, USA, May 1997. P. 227.

85. Cullis, C.F., Hirschler, M.M. *Atmos. Environ.* **14** (1980), p. 1263.

86. Adams, D.F., Farwell, S.O., Robinson, E., Pack, M.R., Bamesberger, W.L. *Environ. Sci. Tech.* **15** (1981), p. 1493.

87. Challenger, F. *Adv. Enzymol. Relat. Areas Mol. Biol.* **12** (1951), p. 429.

88. Lovelock, J.E., Maggs, R.J., Rasmussen, R.A. *Nature* **237** (1972), p. 452.

89. Smet, E., Lens, P., Van Langenhove, H. *Crit. Rev. Environ. Sci. Technol.* **28** (1998), p. 89.

90. Marttila, O. *Outdoor and indoor malodorous sulfur compounds and their effects on health*, University of Helsinki, Finland 1995. PhD thesis.

91. Faith, W.L., Atkisson, A.A.Jr. (eds), *Air pollution*, 2nd edition, John Wiley and Sons, Inc., New York, 1972. Pp. 209–210.

92. Przyjazny, A. *J. Chromatogr.* **333** (1985), p. 327.

93. Steudler, P.A., Kijowski, W. *Anal. Chem.* **56** (1984), p. 1432.

94. Cropper, F.R., Kaminsky, S. *Anal. Chem.* **35** (1963), p. 735.

95. Rothweiler, H., Wäger, P.A., Schlatter, C. *Atmos. Environ.* **25B** (1991), p. 231.

96. Helmig, D., Greenberg, J.P. *J. Chromatogr. A* **677** (1994), p. 123.

97. Mitra, S., Yun, C. *J. Chromatogr.* **648** (1993), p. 415.

98. Mitra, S., Xu, Y.H., Chen, W., Lai, A. *J. Chromatogr. A* **727** (1996), p. 111.

99. MacLeod, G., Ames, J.M. *J. Chromatogr.* **355** (1986), p. 393.

100. Butler, L.D., M.F. Burke, *J. Chromatogr. Sci.* **14** (1976), p. 117.

101. Brown, R.H., Purnell, C.J. *J. Chromatogr.* **178** (1979), p. 79.

102. Helmig, D., Vierling, L. *Anal. Chem.* **67** (1995), p. 4380.

103. Pankow, J.F. *Anal. Chem.* **60** (1988), p. 950.

104. Peters, R.J.B., Bakkeren, H.A. *Analyst* **119** (1994), p. 71.

105. Braman, R.S., Ammons, J.M., Bricker, J.L. *Anal. Chem.* **50** (1978), p. 992.

106. Slater, P.G., Harling-Bowen, L. *Analyst* **111** (1986), p. 1059.

107. Farwell, S.O., Gluck, S.J., Bamesberger, W.L., Schutte, T.M., Adams, D.F. *Anal. Chem.* **51** (1979), p. 609.

108. Swan, H.B., Ivey, J.P. *J. High Resol. Chromatogr.* **17** (1994), p. 814.

109. Gaines, K.K., Chatham, W.H., Farwell, S.O. *J. High Resolut. Chromatogr.* **13** (1990), p. 489.

110. Ivey, J.P., Swan, H.B. *Anal. Chim. Acta* **306** (1995), p. 259.

111. Kangas, J. *J. Chromatogr.* **346** (1985), p. 405.

112. Rinken, M., Aydin, M., Sievers, S., König, W.A. *Fresenius Z. Anal. Chem.* **318** (1984), p. 27.

113. Bandy, A.R., Thornton, D.C., Driedger III, A.R. *J. Geophys. Res. [Atmos.]* **98** (1993), p. 23423.

114. Blomquist, B.W., Bandy, A.R., Thornton, D.C., Chen, S. *J. Atmos. Chem.* **16** (1993), p. 23.

115. Eisele, F.L., Berresheim, H. *Anal. Chem.* **64** (1992), p. 283.

116. Fox, D.L. *Anal. Chem.* **65** (1993), p. 156R.

117. Clement, R.E., Koester, C.J., Eiceman, G.A. *Anal. Chem.* **65** (1993), p. 85R.

118. Tangerman, A. *J. Chromatogr.* **366** (1986), p. 205.

119. Clausen, P.A., Wolkoff, P. *Atmos. Environ.* **31** (1997), p. 715.

120. Camel, V., Caude, M. *J. Chromatogr. A* **710** (1995), p. 3.

121. Collins, G.G., Utley, D. *Chem. Industry* **15** (1972), p. 84.

122. Cisper, M.E., Gill, C.G., Townsend, L.E., Hemberger, P.H. *Anal. Chem.* **67** (1995), p. 1413.

123. Gordon, S.M., Callahan, P.J., Kenny, D.V., Pleil, J.D. *Rapid Commun. Mass Spectrom.* **10** (1996), p. 1038.

124. Cisper, M.E., Garrett, A.W., Cameron, D., Hemberger, P.H. *Anal. Chem.* **68** (1996), p. 2097.

125. Cisper, M.E., Hemberger, P.H. *Rapid Commun. Mass Spectrom.* **11** (1997), p. 1449.

126. Cameron, D., Hemberger, P.H., Alarid, J.E., Leibman, C.P., Williams, J.D. *J. Am. Soc. Mass Spectrom.* **4** (1993), p. 774.

127. Lloyd, D., Thomas, K., Price, D., O'Neill, B., Oliver, K., Williams, T.N. *J Microbiol. Meth,* **25** (1996), p. 145.

128. White, A.J., Blamire, M.G., Corlett, C.A., Griffiths, B.W., Martin, D.M., Spencer, S.B., Mullock, S.J. *Rev. Sci. Instrum.* **69** (1998), p. 565.

129. Koester, C.J., Clement, R.E. *Crit. Rev. Anal. Chem.* **24** (1993), p. 263.

130. Crompton, T.R. In: *The Analysis of Natural Waters*, Vol. 2 Direct Preconcentration Techniques, Oxford, 1993.

131. Soniassy, R., Sandra, P., Schlett, C. In: HP Publication 5962-6216E: *Water Analysis, Organic Micropollutants*, 1994, Chapters 1 and 2. Pp. 11–72.

132. Poole, C.F., Schuette, S.A. *HRC&CC* **6** (1983), p. 526.

133. Pankow, J.F., Ligocki, M.P., Rosen, M.E., Isabelle, L.M., Hart, K.M. *Anal. Chem.* **60** (1988), p. 40.

134. Pankow, J.F., Rosen, M.E. *Environ. Sci. Technol.* **22** (1988), p. 398.

135. Pankow, J.F. *Environ. Sci. Technol.* **25** (1991), p. 123.

136. Badlings, H.T., de Jong, C., Dooper, R.P.M. *HRC&CC* **8** (1985), p. 755.

137. Kostiainen, R. *Chromatographia* **38** (1994), p. 709.

138. Penton, Z. *J High Resolut. Chromatogr.* **15** (1992), p. 834.

139. Goosens, E.C., de Jong, D., de Jong, G.J., Brinkman, U.A.Th. *Chromatographia* **47** (1998), p. 313.

140. Murray, D.A.J. *J. Chromatogr.* **177** (1979), p. 135.

141. Liska, I. *J. Chromatogr.* **655** (1993), p. 163.

142. Pawliszyn, J. *Trends Anal. Chem.* **14** (1995), p. 113.

143. Grob, K. *J. Chromatogr.* **626** (1992), p. 25.

144. Bier, M., Kotiaho, T., Cooks, R.G. *Anal. Chim. Acta* **231** (1990), p. 175.

145. Bier, M., Cooks, R.G. *Anal. Chem.* **59** (1987), p. 597.

146. Leth, M., Lauritsen, F.R. *Rapid Commun. Mass Spectrom.* **9** (1995), p. 591.

147. Matz, G., Kesners, P. *Anal. Mag.* **23** (1995), p. M12.

148. Matz, G., Lennemann, F. *J. Chromatogr. A* **750** (1996), p. 141.

149. Ho, J.S.-Y. *J. Chromatogr. Sci.* **27** (1989), p. 91.

150. Caron, F., Kramer, J.R. *Anal. Chem.* **61** (1989), p. 114.

151. Oliver, B.G. In: *Analysis of trace organics in the aquatic environment*, Afghan, B.K., Chau, A.S.Y. (eds), CRC Press, Boca Raton, FL, 1989. Pp. 2–29.

152. Roe, V.D., Lacy, M.J., Stuart, J.D., Robbins, G.A. *Anal. Chem.* **61** (1989), p. 2584.

153. Herzfeld, D., van der Gun, K.-D., Louw, R. *Chemosphere* **18** (1989), p. 1425.

154. Bianchi, A.P., Varney, M.S., Phillips, J. *J. Chromatogr.* **557** (1992), p. 429.

155. Wise, M.B., Thompson, C.V., Buchanan, M.V., Merriweather, R., Guerin, M.R. *Spectroscopy* **8** (1993), p. 14.

156. Hayward, M.J., Lister, A.K., Kotiaho, T., Cooks, R.G., Austin, G.D., Narayan, R., Tsao, G.T. *Biotechnol. Techniques* **3** (1989), p. 361.

157. Hayward, M.J., Riederer, D.E., Kotiaho, T., Cooks, R.G., Austin, G.D., Mei-Jywan, S., Tsao, G.T. *Process Control Qual.* **1** (1991), p. 105.

158. Ketola, R., Honkanen, T., Mansikka, T., Kostiainen, R., Kotiaho, T., Komppa, V., Wickström, K., Vahervuori, H. In: *Proceedings of the 44th ASMS Conference on Mass spectrometry and Allied Topics*, Portland OR, USA, May 1996.

159. Kotiaho, T., Kostiainen, R., Ketola, R.A., Mansikka, T., Mattila, I., Komppa, V., Honkanen, T., Wickström, K., Waldvogel, J., Pilviö, O. *Process Control Qual.* **11** (1998), p. 71.

160. Dongré, A.R., Hayward, M.J. *Anal. Chim. Acta* **327** (1996), p. 1.

161. Kasthurikrishnan, N., Cooks, R.G., Bauer, S. *Rapid Commun. Mass Spectrom.* **10** (1996), p. 751.

162. Pasternak, R.A., Schimscheimer, J.F., Heller, J. *J. Polymer Sci.* **8** (1970), p. 467.

163. Salvador, F., Merchán, M.D. *Carbon* **34** (1996), p. 1543.

164. Kovaleva, N.V., Nikitin, Y.S., Rudnitskaya, T.A. *J. Anal. Chem.* **52** (1997), p. 334.

165. Saunders, D.L. In: *Chromatography: a laboratory handbook of chromatographic and electrophoretic methods*, E. Heftmann (ed.), Van Nostrand Reinhold Company, New York NY, USA, 1975. Pp. 77–109.

166. Lauritsen, F.R., Leth, M. In: *Proceedings of the 44th ASMS Conference on Mass Spectrometry and Allied Topics*, Portland, OR, May 1996. Pp. 867–868.

167. Hawthorne, S.B., Miller, D.J., Pawliszyn, J., Arthur, C.L. *J. Chromatogr.* **603** (1992), p. 185.

168. Maier, H.G., Tillack, B., Höller, U. *Deutsche Lebensmittel Rundschau* **92** (1996), p. 283.

169. Blauch, J.L., Tarka Jr, S.M. *J. Food. Sci.* **48** (1983), p. 745.

170. Baldwin, M.A., McLafferty, F.C. *Org. Mass Spectrom.* **7** (1973), p. 1353.

171. Chapman, J.R. In: *Practical Organic Mas Spectrometry*, 2nd ed. John Wiley & Sons, Chichester, England, 1993. Chapter 5.

172. Harrison, A.G. In: *Chemical ionization mass spectrometry*, 2nd ed., CPC Press, Boca Raton, FL, USA, 1992.

173. Kasthurikrishnan, N., Cooks, R.G. *Talanta* **42** (1995), p. 1325.

174. Lauritsen, F.R., Choudhury, T.K., Dejarme, L.E., Cooks, R.G. *Anal. Chim. Acta* **266** (1992), p. 1.

175. Pungor, E. Jr., Pecs, M., Szigeti, L., Nyeste, L., Szilagyi, J. *Anal. Chim. Acta* **163** (1984), p. 185.

176. Kotiaho, T. *Methodology and applications of membrane introduction mass spectrometry*, Purdue University, USA, 1992. PhD thesis.

177. Soni, M.H., McElvany, S.W., Callahan, J.H. In: *Proceedings of the 46th ASMS Conference on Mass Spectrometry and Allied Topics*, Orlando, FL, USA, June 1998.

Appendices of this publication are not included in the PDF version. Please order the printed version to get the complete publication (http://www.inf.vtt.fi/pdf/publications/1998)