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Green Analytical Methods For The Determination Of Perfluorocarboxylic Acids (pfcas) And Fluorotelomer Alcohols (ftohs) In Water

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IN WATER

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Abstract

Per- and polyfluoroalkyl substances (PFAS) are a large group of synthetic organic compounds manufactured for their heat, water, and stain-resistant properties. PFAS can be found ubiquitously in the environment because they are widely used in everyday consumer products such as fast-food wrappers, non-stick cookware, stain-resistant products, cosmetics, aqueous film-forming foams, etc. As a result, PFAS are commonly detected in surface water, wastewater, and biosolids from wastewater treatment plants (WWTPs). These are the direct sources of PFAS contamination in drinking water supplies, which are substantial sources of human exposure. Among these PFAS chemicals, two major groups are perfluoroalkyl carboxylic acids (PFCAs) and their precursors, fluorotelomer alcohols (FTOHs). Even though studies have been conducted nationwide to evaluate the degree of these PFAS in the environment, research is lacking in our region. To fill the knowledge gap, we aimed to investigate the occurrence and transport of PFCAs and FTOHs in wastewater and biosolids. Furthermore, it is crucial to have a simple, fast, green, and reliable detection technique that can monitor the trace amount of PFCAs and FTOHs in water and biosolid matrices. In this study, we developed and validated a simple, low-cost, no clean-up, and sensitive method for the determination of PFCAs and FTOHs in water by applying 'green chemistry' based extraction named stir bar sorptive extraction (SBSE) coupled with thermal desorption-gas chromatography-mass spectrometry (TD-GC-MS).

Three commonly detected FTOHs (6:2 FTOH, 8:2 FTOH, and 10:2 FTOH) were selected as the model compounds to develop an enhanced SBSE-TD-GC-MS for the analysis of FTOHs in water. Factors such as extraction time, stirring speed, solvent composition, salt addition, and pH were investigated to achieve optimal extraction efficiency. This "green chemistry" based extraction provided good sensitivity and precision with low method limits of detection ranging

from 2.16 ng/L to 16.7 ng/L and with an extraction recovery ranging 55% to 111%. The developed methods were tested on tap water, brackish water, and wastewater influent and effluent. In two wastewater samples, 6:2 FTOH and 8:2 FTOH were detected at 78.0 and 34.8 ng/L, respectively. This optimized SBSE-TD-GC-MS method stands as a valuable alternative to investigate FTOHs in water matrices.

In addition, we developed an enhanced SBSE-TD-GC-MS for the analysis of PFCAs in water. Our study provides a comprehensive evaluation of the method's linearity, recovery, sensitivity, repeatability, and spiked recovery across diverse water matrices. The method demonstrates linearity with coefficients of determination (R^2) spanning from 0.9892 to 0.9988. Sensitivity metrics showed low limits of detection (LOD) in the low ng/L (ppt) range for all analytes, achieving LODs between 21.2 ng/L to 74.0 ng/L. The recoveries for the method varied from 47-97%, suggesting an efficient extraction process. Additionally, the method’s robustness across various water matrices (tap water, wastewater influent, and effluent) reflected by the spiked recovery experiment underscored the method’s efficiency in real-world applications. In comparison with traditional PFCAs analysis methods, our optimized SBSE technique requires only a minimum sample volume of 1 mL and minimal solvent usage, enhancing eco-friendliness and reducing potential contamination and handling errors. Repeatability assessments at two concentration levels produced %RSD (Relative Standard Deviation) values at 14% or less for any target PFCA compounds, indicating good precision. These attributes showcase the developed method's capability to serve as a precise, eco-friendly, and reliable tool for the analysis of PFCAs across diverse water matrices.

This study also presents a comprehensive exploration into the presence and transport behavior of FTOHs and PFCAs in biosolid samples collected from wastewater treatment plants

(WWTPs) in El Paso, Texas. We optimized an ultrasonic extraction method for efficient recovery of FTOHs and PFCAs compounds from biosolids followed by SBSE-TD-GC-MS analysis. The results showed specific concentrations of FTOHs compounds in biosolid samples from the different WWTPs. 6:2 FTOH was not detected in any of the samples, while 8:2 FTOH was found in three WWTPs at varying concentrations: 100.30 ng/g in WWTP-1, 62.87 ng/g in WWTP-2, and 56.41 ng/g in WWTP-4. Additionally, 10:2 FTOH was detected in WWTP-1 at a concentration of 68.07 ng/g. Interestingly, despite the sensitive analytical approach employed, none of the targeted PFCAs were detected in any of the biosolid samples. These findings provide important insights into the distribution and prevalence of specific FTOHs in biosolids from WWTPs, that highlight the inherent variability in their occurrence.

Through the development and validation of a cost-effective, environmentally friendly, and sensitive analytical method, this dissertation represents a reliable alternative analytical technique for monitoring PFCAs and FTOHs in aquatic matrices and contributes valuable data to the ongoing efforts to monitor and manage emerging contaminants in wastewater treatment systems.

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Chapter 1: Introduction and Literature Review

1.1 PER- AND POLYFLUOROALKYL SUBSTANCES (PFAS)

Per- and Polyfluoroalkyl substances (PFAS) are a family of more than 4700 highly fluorinated compounds manufactured for diverse applications [1]. They are well-known for their water and oil-repellant properties, thermal stability, environmental mobility, resistance to biochemical degradation, bioaccumulative effects, and toxicity. PFAS can be found in common consumer products like non-stick cookware, clothing, leather, upholstery, carpets, etc. They can also be used in fire-fighting foams and in industrial applications such as wetting agents, additives, coatings, emulsifiers, paints, waxes, and polishes [2–5]. Their useful properties are due to their structure, which includes a totally fluorinated carbon chain that is both hydrophobic and oleophobic and hydrophilic charged functional groups (for example, carboxylate or sulfonate groups) attached to the structure [4,6,7]. In general, PFAS contains at least one fluorine atom in replacement of hydrogen atoms [4]. A per-fluoroalkyl compound is expressed as $C_nF_{(2n+1)}$-R, where $C_nF_{(2n+1)}$ represents the per-fluoroalkyl portion of the molecular structure, while poly-fluoroalkyl compound contains at least one carbon atom that is non-fluorinated [4,8]. A poly-fluoroalkyl compound can be biotically or abiotically transformed into a per-fluorinated compound by removing its non-fluorinated component in its structure. Under other classifications, PFAS can also be categorized based on chain length into long- and short-chain compounds and also classified into numerous groups and sub-groups depending on their terminal functional groups [4].

As aforementioned, PFAS are widely produced and used. They have high resistance to heat and chemical reactions [4] and therefore, often referred to as "forever chemicals." Their persistence in the environment has resulted in pro-long human and wildlife exposure to PFAS [9,10]. Recognizing these dangers, the U.S. Environmental Protection Agency (EPA) has taken steps to regulate and

monitor PFAS levels, particularly in drinking water. Thus, in 2016, US EPA established a health advisory level of 70 ng/L (ppt) for two of the most conventional types of PFAS, i.e., perfluorooctanoic acid (PFOA) and perfluorooctanesulfonic acid (PFOS) in drinking water [11]. Most recently, in June 2022, The Environmental Protection Agency (EPA) recently revised its drinking water health advisories for the four predominant PFAS found in drinking water. In its Interim health advisories, EPA set the levels for PFOA and PFOS at 0.004 ppt and 0.02 ppt, respectively. Final health advisories set the levels of GenX chemicals (PFOA replacement) and PFBS (PFOS replacement) at 10 ppt and 2000 ppt, respectively [12]. Hence, EPA has finalized the UCMR 5 (Fifth Unregulated Contaminant Monitoring Rule) to initiate nationwide monitoring for 29 PFAS in drinking water. This measure aims to address the public health and environmental impacts of PFAS and mitigate their risks.

Perfluoroalkyl acids (PFAA) are a significant subset of the extensive family of PFAS. PFAA encompasses two main types: perfluoroalkyl carboxylic acids (PFCAs, with the chemical formula $C_nF_{2n+1}COOH$) and perfluoroalkyl sulfonic acids (PFSA, $C_nF_{2n+1}SO_3H$) [4,13]. These compounds have garnered considerable attention from both regulatory bodies and the scientific community due to their unique and concerning characteristics. A critical aspect of PFAAs is their extraordinary persistence in the environment, coupled with their potential for bioaccumulation and the toxicity they have demonstrated in laboratory tests on animals [14,15]. Two of the most notable compounds within this group are eight-carbon chain perfluorooctanoic acid (PFOA) and perfluorooctane sulfonic acid (PFOS), which are the most commonly used, detected, and studied PFAAs [16–18]. PFAAs enter the environment through both direct and indirect sources. Direct sources involve the immediate discharge of PFAAs into the environment, whether intentional or accidental. Indirect sources, on the other hand, pertain to the formation of PFAAs through the

biotic or abiotic degradation of other PFAS compounds, often termed as precursors to PFAA [16,18,19].

Precursors to PFAS typically refer to compounds that can transform into PFAAs through oxidation reactions [2,4]. A major group of these precursors includes fluorotelomer-based compounds, such as Fluorotelomer alcohols (FTOHs). These compounds have structures that facilitate easy oxidation under the presence of strong oxidants [16,20,21]. The current scientific literature features an extensive array of research focused on both the qualitative and quantitative analysis of PFAS precursors. These studies extensively explore the mechanisms through which these precursors undergo transformation into PFAAs within environmental contexts [14,22–24]. Thus, this study focuses on PFCAs and their major precursor compounds, i.e., FTOHs.

1.2 PFAS Toxicity and Human Exposure

PFAS, known for its structural thermal and chemical stability, is persistent and has the potential for bioaccumulation in the environment. This leads to widespread detection in wildlife and humans [25–28]. Human exposure to PFAS mainly occurs through ingestion pathways such as diet and drinking water, especially in areas proximal to extremely contaminated sites. Reports have shown that human exposure to high levels of certain PFAS has been linked to various health conditions such as reproductive disorders, developmental effects in children, liver and kidney disease, immunotoxicity, hepatotoxicity, and thyroid hormone disruption immune system depression [29–35]. Initially, the understanding of PFAS toxicity was largely based on animal studies. However, growing concerns over their adverse health effects have spurred extensive human-focused research. The impact of PFAS on human health varies based on several factors, including the duration and concentration of exposure, the exposure method, and individual variables like age, sex, ethnicity, and existing health conditions [26,28,36]. High exposure levels to PFAS have been

linked to increased blood lipid levels, with studies indicating a correlation between higher blood concentrations of PFOA and PFOS and elevated low-density lipoprotein levels (high cholesterol) [27,28,36].

According to the ASTDR (Agency for Toxic Substance and Disease Registry), PFAS may lead to potential health impacts such as liver damage, high cholesterol, thyroid disease, reduced vaccine antibody responses, asthma, reduced fertility, and lower birth weights [37]. PFOA, in particular, is associated with developmental, reproductive, hepatic, and immunological issues, both acutely and over the long term. High concentrations of PFOA in lab mice have shown neurodevelopmental and skeletal effects [25]. Communities near Teflon manufacturing plants at Parkersburg, West Virginia (WV), have shown higher serum levels of PFOA, which are associated with increased rates of various cancers, including testicular, kidney, prostate, and ovarian [38]. The liver, being instrumental in processing PFAS for excretion, is particularly susceptible to bioaccumulation [39].

Other PFAAs like Perfluorononanoic acid (PFNA) have been linked to reduced body weight and developmental delays in mice. The Center for Disease Control has highlighted PFAS as a significant public health challenge [37]. **Figure 1.1** represents that by August 2023, PFAS contamination had impacted 3,186 locations in the U.S. alone [40]. Community studies have shed light on the broader public health implications of PFAS. For instance, in the Cape Fear Region of North Carolina, USA, long-term residents exposed to water contaminated by a fluorochemical plant had detectable levels of PFAS, including fluorotelomers, in their serum [41]. Additionally, a notable correlation has been found between PFAS levels in drinking water and proximity to industrial and military sites [42].

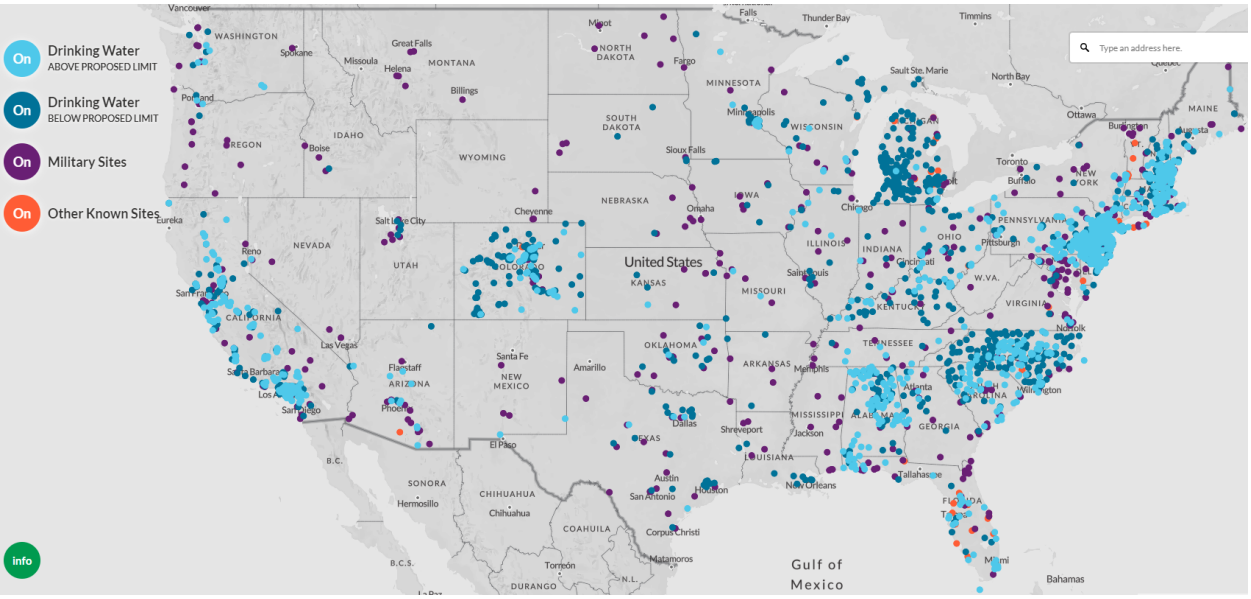

Figure 1.1: PFAS Contamination in the U.S. (August 2023)
Source: https://www.ewg.org/interactive-maps/pfas_contamination/map/

1.3 Occurrence, Fate, and Transport of PFAS in Water

Over the concerns of persistence and health impacts, many uses of some PFAS were phased out by U.S. manufacturers. After the phaseout of the two most concerned PFAS (i.e., PFOA and PFOS), shorter carbon-chain ionic and neutral PFAS are now being gradually used as replacements [43]. Neutral/semi-volatile PFAS are largely considered to be precursors of the ionic PFAS that include fluorotelomer alcohols (FTOHs), perfluorooctane sulfonamides (FOSAs), perfluorooctane sulfonamidoethanols (FOSEs), fluorotelomer acrylates (FTACs), and fluorotelomer methacrylates (FTMACs) [4,16,17,44]. However, the high volatility of fluorotelomer-based compounds can undergo long-range environmental transport. This can contribute to PFAS contamination in remote regions such as the Arctic, where they are degraded and contribute to potential perfluoroalkyl carboxylate contamination [17,45]. The study identified PFOA as the predominant PFCAs commonly found in the Arctic region which is the degradation product of FTOHs. The short-chain PFAS are also highly

mobile and can be bioaccumulated in the environment, while long-chain PFAS are easily accumulated in humans, animals, soils, and sediments [46].

Over the past few decades, PFAS have been ubiquitously present in the soil, water, air, food, and biological matrices [13,47]. Particularly, PFAS and their precursors have been found in all types of waters throughout the world, including surface, ground, tap, bottled, wastewater influents and effluents, industrial waste influents and effluents, rivers, lakes, and seas [48–52]. In a study by Pan and colleagues expanded the scope of investigation by evaluating PFCAs and PFSA in 160 surface water samples collected from various countries, including China, the United States, United Kingdom, Sweden, Germany, Netherlands, and South Korea [53]. Their findings revealed that several PFAS compounds were consistently present across all these nations, which points out the ubiquitous presence of PFAS in surface waters worldwide. In another study conducted by Kaboré and team, a comprehensive analysis of 133 PFASs from different PFAS groups were carried out on drinking water samples [54]. The study concluded that the levels of PFOS and PFOA in the collected drinking water samples did not exceed 70 parts per trillion (ppt), the health advisory values previously set by the U.S. Environmental Protection Agency (EPA). Yamashita et al., conducted an extensive investigation into the presence and distribution of perfluorooctanoic acid (PFOA) and perfluorooctane sulfonate (PFOS) in 71 water samples collected from different oceans. The concentrations of these chemicals ranged from less than 5 ppt to as high as 439 ppt [55]. Their study shed light on the widespread occurrence of these substances in oceanic environments. In a separate study by Yeung and his group, [56] they examined the circulation of PFAS in the Arctic Ocean. They routinely detected 13 different PFASs in this remote region in the range of 5−343 pg/L. This highlights the global reach of these compounds and their presence even in such pristine environments.

1.4 Role of Wastewater Treatment Plants in PFAS Transport

Wastewater treatment plants (WWTPs) play a crucial role in the transport of PFAS from human activities into the environment. These facilities receive wastewater streams from a wide range of sources, including residential, industrial, and commercial sources [57,58]. As a result, PFAS compounds are now detected in nearly every wastewater system worldwide [59,60]. Wastewater Effluent is one significant route for the release of PFAS in the environment. Although the effluent is regularly monitored and discharged into nearby water bodies, it often contains unregulated PFAS levels. As a result, wastewater effluent could be a significant contributor to PFAS contamination in aquatic environments. Gallen and co-wokers examined 14 WWTPs and found that only three of them managed to reduce the total PFAS concentration from influent to effluent [61]. Additionally, another research group led by Dauchy, investigated the mass flow of the most abundant PFAS in individual WWTPs [58] and found that PFCAs mass flow increased after oxidative conversion during secondary biological treatment processes, indicating that PFCAs precursors can transform during treatment [57,58,62,63]. According to the literature, the total concentration of PFAS in wastewater ranges from not detected level to 143 µg/L (ppb); in river water, PFAS concentration ranges from a few ng/L to 496 µg/L; in surface water levels were up to 84 µg/L and in drinking water ranges up to 8300 ng/L [64]. Relatively, there have been inadequate studies on the effects of wastewater effluent for FTOHs contamination on the environment. Some investigation on FTOHs in wastewater effluents of WWTPs found containing FTOHs ranging from <0.13−6.67 ng/L [65,66].

Another critical route for PFAS release from WWTPs is through sludges and biosolids. WWTP biosolids the solid waste generated primarily through biological treatment processes [57]. Notably, these sludges can contain high concentrations of long-chain and precursor PFAS [57,62]. The land application of municipal biosolids represents a significant pathway through which PFAS

in sewage return to the soil environment. Considering that biosolids are recycled through land applications and landfills, it is important to monitor the composition and concentration of PFAS in biosolids. Biosolids are often applied to land as fertilizer. There have been reports that PFAS, which are present in biosolids, can leach into the soil and accumulate in plants [58,63]. Moreover, certain PFAS compounds, particularly those with longer chain lengths, tend to adhere to biosolids due to their higher hydrophobicity. This introduces a concept of fractionation, altering the PFAS profile in both effluent and biosolids [61]. Due to the limited information available regarding PFAS levels in biosolids, there is a pressing need to determine the levels of both identified and unidentified PFAS species in these samples to better grasp the extent of the issue.

1.5 METHODOLOGIES FOR PFAS DETECTION

The extensive use of PFAS has led them as emerging contaminants in the environment. Thus, monitoring them is essential to help with environmental management and remediation [13]. Numerous instrumental separation methods for PFAS have been developed. A variety of chromatographic techniques such as Liquid chromatography (LC), Gas chromatography (GC), High-performance Liquid chromatography (HPLC), and Ultra-high performance liquid chromatography (UHPLC) are frequently used for the determination of PFAS in water matrices [13,14,67–70]. Less commonly used methods include nuclear magnetic resonance, Fourier transform infrared spectroscopy, and ion chromatography [69,71,72]. In the last decade, research has been conducted on developing new on-site detection techniques that are convenient and reliable. For example, there are several sensor-based methods such as electrochemical sensors, ion-selective electrodes (ISE), fluorescence sensors, and smartphone app-based monitoring systems have been developed [73,74]**.** However those techniques usually detect high concentrations of PFAS (> 10 ppb). Moreover, methods such as the total oxidizable precursor assay have determined that a significant

fraction of the total PFAS present in environmental samples consists of unidentified compounds [75,76].

Chromatographic techniques are essential tools in analytical laboratories due to their versatility and effectiveness. These methods find extensive application in separating and analyzing diverse compound mixtures. Chromatography allows for precise separation, analysis, and purification, often requiring only small sample volumes. These advantages make chromatography a better choice for PFAS determination in water over other analytical techniques.

1.5.1 Liquid chromatography-tandem mass spectrometry (LC-MS/MS)

Liquid chromatography-tandem mass spectrometry (LC-MS/MS) stands as a versatile and robust analytical approach, demonstrating remarkable precision and accuracy across a broad spectrum of sample types and analytes. Typically, when dealing with PFAS analytes, a crucial preliminary step involves their preconcentration using solid-phase extraction (SPE) prior to LC-MS/MS analysis [22,47,77]. As LC-MS/MS is a predominant technology[73], US EPA has developed and validated a series of methods (EPA Method 533, 537.1, 8327) for PFAS using LC-MS/MS with solid-phase extraction in sample preparation [76].

When chromatography is combined with mass spectrometry, its power is greatly enhanced. In this configuration, the LC system is often coupled with tandem mass spectrometers, particularly either triple quadrupole MS or linear ion trap MS, using an electrospray ionization interface [13,74,78,79]. This tandem mass spectrometry setup allows for multiple reaction monitoring (MRM), which enables the selective monitoring of specific analytes of interest [22]. Recent advancements in chromatographic analysis have made it possible for researchers to detect and quantify PFAS at extremely low concentration levels (pg/L) [80]. **Table 1.1** provides an overview of LC methods used for tracing PFAS in water samples. An impressive example of the power of LC-MS/MS in PFAS

analysis is demonstrated in the work of Gamo and colleagues, who determined the concentration of perfluoroalkyl acids (PFAAs) in oceanic water [81]. In this study, LC-MS/MS was employed to measure PFAA concentrations at sub-parts per trillion (sub-ppt) to low parts per quadrillion (low ppq) levels in samples collected at the ocean's surface [81].

But overall, LC techniques have some limitations. Yamashita and collegues, [80] reported background contamination issues in PFAS monitoring. Specific attention should be paid to auto-sampler vial caps made of Teflon or fluoropolymers, fluoropolymer tubing, solvent inlet filters and a variety of other laboratory products containing Teflon and perfluoroalkoxy compounds, as they are sources of PFAS in blanks. A well-known problem in electrospray ionization (ESI), as is commonly used in LC-MS, is ion suppression [82]. Moreover, Liquid chromatography techniques require highly sophisticated and expensive instruments, and their analysis is generally time-consuming [83].

However, it is also worth noting that to analyze PFASs at trace levels, large sample volumes are typically needed for the preconcentration of target analytes. Unfortunately, when using SPE, the analysis time increases linearly as the sample volume increases. This presents a challenge in terms of analysis efficiency. As a result, there is a pressing need to develop improved methods that can lower the limits of detection (LODs) while reducing analysis times, particularly when working with smaller sample volumes.

Table 1.1: Liquid Chromatographic Techniques for Determination of PFAS in Water

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
PFHxA, PFHpA, PFOA, PFDoA	Groundwater	Methyl iodide, Diazomethane	200	SPE	GC-ECNI-MS	SPB-1 SULFUR column (30m×0.32mm i.d., 4.0µm filmthickness), (Supelco, Bellefonte, USA)	35-90	18-36 µg/L	84
PFBA, PFPeA, PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Wastewater, Seawater	Tetrabutylammonium, Butanol, Thionyl chloride	5	IP-SPME	GC–NCI-MS	ZB-624 column (60m×0.25mm i.d., 1.4µm filmthickness), (Phenomenex, Torrence, CA, USA).	35-90	0.02-0.75 µg/L	85
PFPrA, PFBA, PFPeA, PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA, TFA	Surface Water, Lake water, Precipitaion	2,4- difluoroaniline, N,N-dicyclohexylcarbodiimide	300	SPE	GC-EI-MS	ZB-5 Zebron fused silica capillary column (30 m × 0.25 mm i.d.), (Phenomenex, Torrence, CA, USA)	25-137	0.01-0.5 ng/L	86
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	River Water	Isobutyl chloroformate (IBCF), pyridine, Isobutanol	500	LLE	GC-ECD	SPB-5 column (15m×0.25mm i.d., 0.25 µm filmthickness), (Supelco, Bellefonte, USA)	N.R.	0.1–-1.8 µg/ml	87

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
PFOA, FHUEA, FOUEA, FNUEA	River Water	Benzyl bromide	500	IP-LLE	GC-EI-MS	BPX35 column (30m×0.25mm i.d., 0.25µm film thickness)	89-101	0.2-1 µg/ml	[88]
PFOA, PFNA, PFDA, PFUnA, PFDoA	Surface water, Precipitation	2,4- difluoroaniline and DCC	500	IP-LLE	GC-NCI-MS	RTX-35 column (105 m × 0.25 mm i.d., 0.50 µm film thickness), (Restek Corporation, Brockville, ON).	91-98	0.3-5.9 ng/L	[89]
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Treated Water	2,4- difluoroaniline and DCC	10	LLE	GC-FID	RTX 5 column (30m×0.25mm i.d., 0.1µm film thickness), (Restek, Bellefonte, PA, USA)	82-110	0.127 µg/ml	[90]
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	River Water	Propyl chloroformate, Propanol	10	HS-SPME	GC–QqQ–MS/MS	TR-5MS column (30m×0.25mm i.d., 0.25µm film thickness)	85-117	0.08-6.6 ng/L	[91]
PFPeA, PFHxA, PFHpA, PFOA, PFDA, PFNA, , PFUnA, PFDoA	River Water	Isobutyl chloroformate (IBCF), Pyridine, Isobutanol, pH 2.5	250	SPE	GC-NCI-MS	Rtx-200MS column (30m×0.25mm i.d., 0.25µm film thickness), (Restek, USA)	53-111	0.1-24 pg/ml	[92]

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
PFHpA, PFOA, PFNA, PFDA	Surface Water	Tetrabutylammonium hydrogen sulfate (TBAHS)	10	IP-DLLME	GC-NCI-MS/MS	DB-624 column (30 m × 0.25 mm, 1.4µm film thickness), (J&W, Folsom, CA, USA)	95-98	37-51 ng/L	93
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, & PFOS	Drinking Water/ Wastewater	IBCF, DCC in Pyridine, Isobutanol, pH 1.0	250	SPE	GC-DSQ II-MS	DB-5MS column (30 m × 0.25 mm, 1.4µm film thickness), (J&W, Folsom, CA, USA)	94-98	0.1-0.5 ng/L	94
FTOH (4:2, 6:2, 8:2, and 10:2), N-EtFOSE, N-MeFOSE, N-MeFOSA, N-EtFOSA	Wastewater, River water	N/A	1000	SPE	GC-APCI-MS/MS	TG-WaxMS column (30m×0.25mm i.d., 0.25µm film thickness), (Thermo Scientific, USA)	80-97	1-5 pg/L	95
4:2 FTI, 6:2 FTI, 8:2 FTI, 6:2 FTOH, 8:2 FTOH, 10:2 FTOH, , 6:2 FTAC, 8:2 FTAC, 6:2 FTMAC, 8:2 FTMAC, MeFOSA, EtFOSA	Tap Water, Surface water	N/A	10	HS-SPME	GC-EI-MS	Rxi-624SilMS column (30 m × 0.25 mm i.d.; 1.4µm film thickness) (Restek, Bellefonte, PA, USA).	76-126	20-100 ng/L	96

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
PFPrA, PFBA, PFPeA, PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Wastewater	Triethylsilanol (TES), H_2SO_4	250	SPE	GC-EI-MS	DB-5MS column (30m×0.25mm i.d., 0.25µm film thickness), (Agilent J&W Scientific)	93-108	4-48 ng/L	97
PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA, PFTeA	River Water, Lake Water	Isobutyl chloroformate (IBCF), pyridine, Isobutanol	1	DLLME	GC-EI-MS	TR-5MS column (30m×0.25mm i.d., 0.25µm film thickness), (Thermo Fisher, Shanghai, China)	83.7–117	0.9-3 ng/mL	98
PFBA, PFPeA, PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Surface water	2,4-difluoroaniline (DFA) and DCC	500	SPE	GC-µECD	HP-5 column (30m×0.32mm i.d., 0.25µm film thickness)	62-118	1.14–6.32 µg/L	99
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Tap Water	2,3,4,5,6-pentafluorobenzyl bromide (PFBBr)	500	SPE	GC-EI-MS	DB-5MS column (30m×0.25mm i.d., 0.25µm film thickness), (Agilent J&W Scientific)	40.1-101.8	0.1-.28 ng/L	100

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
4:2 FTO, 6:2 FTO, 8:2 FTO, 4:2 FTOH, FTOH (6:2, 8:2, and 10:2), N-MeFOSE, N-EtFOSE, N-MeFOSA, N-EtFOSA, 7-Me-6:2 FTOH	River Water	N/A	500	SPE	GC-EI-MS, GC-CI-MS, GC-NCI-MS	DB-624 column (60 m × 0.25 mm, 1.4µm film thickness), (Agilent Technologies, CA, USA)	(90-100)	0.06-6 µg/L	[101]
TFA, PFPrA, PFBA, PFPeA, PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA, PFTrDA, PFTeDA	Wastewater, Tap water	Isobutyl chloroformate (IBCF), Isobutanol, Pyridine,	250	SPE	GC-ECNI-MS	HP-5MS column (20m×0.18mm i.d., 0.18µm film thickness), (Agilent, CA, USA)	(83-130)	0.06-14.6 ng/L	[102]

1.5.2 Gas chromatography-mass spectrometry (GC-MS)

As an alternative, Gas chromatography (GC) is a convenient instrument for volatile and semi-volatile PFAS analysis (i.e., fluorotelomer alcohols, FTOHs; perfluorinated sulfonamido ethanols, FASE [70,82]. However, for a sample to be suitable for GC-MS analysis, it must be both volatile and thermally stable. As a result, many PFAS compounds, which are generally non-volatile, require a chemical derivatization process to convert them into volatile derivatives for analysis [70,82]. There are several studies have been done on PFCAs derivatization prior GC-MS analysis [70,82,103–108]**.** Gołebiowski et al., applied 2,4 difluoroaniline for derivatization of PFCAs (PFHxA, PFHpA, PFOA, PFNA, and PFDA) in the presence of N, N′-dicyclohexylcarbodiimide (DCC) [109]. But this method consists of several sample preparation steps: pH adjustment, phase separation and washing the organic phase with HCl, $NaHCO_3$, and NaCl solution. In another study, Dufkova et al. developed a fast derivatization procedure for PFCAs by using isobutyl chloroformate to quantify various chain lengths of PFCAs in water [110]. This technique allowed them to test river water samples for PFCAs. Similarly, Jurado-Sánchez and colleagues investigated perfluoroalkyl acids in water using GC-MS. They first preconcentrated samples using solid-phase extraction and then derivatized them with isobutyl chloroformate in the presence of pyridine and isobutanol. Through optimization of derivatizing agent amounts, low limits of detection (LOD) were achieved ranging from 0.1 to 0.5 parts per trillion (ppt) [104]. Another research group led by Strozynska developed two derivatization processes using triethylsilanol and N,N-Dimethylformamide dimethylacetal for the separation of PFCAs through GC-MS [108]. Overall, isobutyl chloroformate-based derivatization provides rapid and simple procedure and is commonly used for the determination of PFAS in water by GC-MS. **Table 1.2** provides an overview of

derivatization techniques and gas chromatographic methods used for tracing PFAS in water samples.

Table 1.2: Gas Chromatographic Techniques for Tracing PFAS in Water

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
PFHxA, PFHpA, PFOA, PFDoA	Groundwater	Methyl iodide, Diazomethane	200	SPE	GC-ECNI-MS	SPB-1 SULFUR column (30m×0.32mm i.d., 4.0µm filmthickness), (Supelco, Bellefonte, USA)	35-90	18-36 µg/L	84
PFBA, PFPeA, , PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Wastewater, Seawater	Tetrabutylammonium, Butanol, Thionyl chloride	5	IP-SPME	GC–NCI-MS	ZB-624 column (60m×0.25mm i.d., 1.4µm filmthickness), (Phenomenex, Torrence, CA, USA).	35-90	0.02-0.75 µg/L	85
PFPrA, PFBA, PFPeA, PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA, TFA	Surface Water, Lake water, Precipitaion	2,4- difluoroaniline, N,N-dicyclohexylcarbodiimide	300	SPE	GC-EI-MS	ZB-5 Zebron fused silica capillary column (30 m × 0.25 mm i.d.), (Phenomenex, Torrence, CA, USA)	25-137	0.01-0.5 ng/L	86
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	River Water	Isobutyl chloroformate (IBCF), pyridine, Isobutanol	500	LLE	GC-ECD	SPB-5 column (15m×0.25mm i.d., 0.25 µm filmthickness), (Supelco, Bellefonte, USA)	N.R.	0.1-1.8 µg/ml	87

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
PFOA, FHUEA, FOUEA, FNUEA	River Water	Benzyl bromide	500	IP-LLE	GC-EI-MS	BPX35 column (30m×0.25mm i.d., 0.25µm film thickness)	89-101	0.2-1 µg/ml	[88]
PFOA, PFNA, PFDA, PFUnA, PFDoA	Surface water, Precipitation	2,4- difluoroaniline and DCC	500	IP-LLE	GC-NCI-MS	RTX-35 column (105 m × 0.25 mm i.d., 0.50 µm film thickness), (Restek Corporation, Brockville, ON).	91-98	0.3-5.9 ng/L	[89]
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Treated Water	2,4- difluoroaniline and DCC	10	LLE	GC-FID	RTX 5 column (30m×0.25mm i.d., 0.1µm film thickness), (Restek, Bellefonte, PA, USA)	82-110	0.127 µg/ml	[90]
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	River Water	Propyl chloroformate, Propanol	10	HS-SPME	GC–QqQ–MS/MS	TR-5MS column (30m×0.25mm i.d., 0.25µm film thickness)	85-117	0.08-6.6 ng/L	[91]
PFPeA, PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA	River Water	Isobutyl chloroformate (IBCF), Pyridine, Isobutanol, pH 2.5	250	SPE	GC-NCI-MS	Rtx-200MS column (30m×0.25mm i.d., 0.25µm film thickness), (Restek, USA)	53-111	0.1-24 pg/ml	[92]

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
PFHpA, PFOA, PFNA, PFDA	Surface Water	Tetrabutylammonium hydrogen sulfate (TBAHS)	10	IP-DLLME	GC-NCI-MS/MS	DB-624 column (30 m × 0.25 mm, 1.4µm film thickness), (J&W, Folsom, CA, USA)	95-98	37-51 ng/L	93
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, & PFOS	Drinking Water/ Wastewater	IBCF, DCC in Pyridine, Isobutanol, pH 1.0	250	SPE	GC-DSQ II-MS	DB-5MS column (30 m × 0.25 mm, 1.4µm film thickness), (J&W, Folsom, CA, USA)	94-98	0.1-0.5 ng/L	94
FTOH (4:2, 6:2, 8:2, and 10:2), , N-MeFOSE, N-EtFOSE, N-MeFOSA, N-EtFOSA	Wastewater, River water	N/A	1000	SPE	GC-APCI-MS/MS	TG-WaxMS column (30m×0.25mm i.d., 0.25µm film thickness), (Thermo Scientific, USA)	80-97	1-5 pg/L	95
FTOH (6:2, 8:2, and 10:2), 4:2 FTI, 6:2 FTI, 8:2 FTI, 6:2 FTAC, 8:2 FTAC, 6:2 FTMAC, 8:2 FTMAC, MeFOSA, EtFOSA	Tap Water, Surface water	N/A	10	HS-SPME	GC-EI-MS	Rxi-624SilMS column (30 m × 0.25 mm i.d.; 1.4µm film thickness) (Restek, Bellefonte, PA, USA).	76-126	20-100 ng/L	96

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
PFPrA, PFBA, PFPeA, PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Wastewater	Triethylsilanol (TES), H_2SO_4	250	SPE	GC-EI-MS	DB-5MS column (30m×0.25mm i.d., 0.25µm film thickness), (Agilent J&W Scientific)	93-108	4-48 ng/L	[97]
PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA, PFTeA	River Water, Lake Water	Isobutyl chloroformate (IBCF), pyridine, Isobutanol	1	DLLME	GC-EI-MS	TR-5MS column (30m×0.25mm i.d., 0.25µm film thickness), (Thermo Fisher, Shanghai, China)	83.7–117	0.9-3 ng/mL	[98]
PFBA, PFPeA, PFHpA, PFHxA, , PFOA, PFNA, PFDA, PFUnA, PFDoA	Surface water	2,4-difluoroaniline (DFA) and DCC	500	SPE	GC-µECD	HP-5 column (30m×0.32mm i.d., 0.25µm film thickness)	62-118	1.14–6.32 µg/L	[99]
PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Tap Water	2,3,4,5,6-pentafluorobenzyl bromide (PFBBr)	500	SPE	GC-EI-MS	DB-5MS column (30m×0.25mm i.d., 0.25µm film thickness), (Agilent J&W Scientific)	40.1-101.8	0.1-.28 ng/L	[100]

Target PFAS	Matrix/ Sources	Derivatizing agent	Sample Volume (mL)	Extraction method	Analytical method	GC Column	% Recovery	LOD	Reference
4:2 FTO, 6:2 FTO, 8:2 FTO, FTOH (4:2, 6:2, 8:2, and 10:2), N-MeFOSE, N-EtFOSE, N-MeFOSA, N-EtFOSA, 7-Me-6:2 FTOH	River Water	N/A	500	SPE	GC-EI-MS, GC-CI-MS, GC-NCI-MS	DB-624 column (60 m × 0.25 mm, 1.4µm film thickness), (Agilent Technologies, CA, USA)	(90-100)	0.06-6 µg/L	[101]
TFA, PFPrA, PFBA, PFPeA, , PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA, PFTrDA, PFTeDA	Wastewater, Tap water	Isobutyl chloroformate (IBCF), Isobutanol, Pyridine,	250	SPE	GC-ECNI-MS	HP-5MS column (20m×0.18mm i.d., 0.18µm film thickness), (Agilent, CA, USA)	(83-130)	0.06-14.6 ng/L	[102]

1.5.3 Untargeted analysis and other methods for PFAS

Untargeted analysis is a powerful approach that plays an important role in quantifying the total concentration PFAS. The main difficulty lies in the fact that, for all PFAS compounds, standardized reference materials or analytical standards are not available [111]. Therefore, untargeted analysis methods focus on quantifying PFAS as a compound class, allowing for a more comprehensive assessment of their presence. In order to overcome the challenges posed by reference materials and the constraints of targeted analytical methods, high-resolution mass spectrometry (HRMS) has emerged as a valuable tool [112,113]. HRMS equipment, such as quadrupole time-of-flight (QToF) or orbitrap mass analyzers, offers significantly higher resolution compared to traditional quadrupole-based instruments [112]. One of the key advantages of HRMS in PFAS detection is its ability to detect and characterize unexpected or new-generation PFAS compounds with unknown fragmentation patterns [114,115]. By analyzing the measured elemental formulae of unknown ions, researchers can uncover novel PFAS variants [116]. Wang and colleagues conducted both targeted and non-targeted screening of PFAS in wastewater from a fluorochemical manufacturing park [117]. Through their investigation, they successfully identified 90 PFASs belonging to 15 different chemical classes using HRMS to identify previously unknown PFAS compounds. Furthermore, researchers like Ruan and his team [118], Nakayama et al., [119], and Liu et al., [111], all reviewed the present state of non-target PFAS detection methods. Liu research group highlighted that non-targeted HRMS-based approaches have led to the discovery of more than 750 PFAS compounds across 130 diverse chemical classes [111]. They summarized various strategies for non-targeted PFAS discovery, emphasizing the significant contribution of HRMS to this expanding field of research. Untargeted analysis by HRMS, has revolutionized the detection and identification of PFAS compounds.

Another method for untargeted PFAS analysis is total organic fluorine (TOF) analysis. TOF analysis is a method used to measure the overall amount of fluorinated organic compounds in a sample [112]. To perform TOF analysis, a dedicated combustion ion chromatography (CIC) system is employed [120]. In this process, PFAS compounds are first adsorbed onto materials like activated carbon or other sorbents. Then, combustion is used to release fluoride ions from the absorbed PFAS, and the concentration of fluoride ions is determined using CIC [120,121]. However, TOF analysis has certain limitations. One major drawback is that it lacks specificity for the chain length of PFAS compounds and doesn't target specific PFAS precursors. This means that TOF analysis cannot distinguish between different PFAS molecules based on their structure or size [120]. To identify specific PFAS compounds and their origins, additional PFAS-specific extraction methods are necessary. Another important limitation of TOF analysis is its non-selectivity to PFASs. This means that it measures the total fluorine content in a sample, which can include fluorine contributions from naturally occurring organofluorines that are not PFAS [112]. As a result, TOF analysis tends to overestimate the presence of PFASs in the sample matrix, as it cannot differentiate between PFAS and other sources of organic fluorine. Overall, TOF analysis provides a measure of the total organic fluorine content in a sample but lacks specificity in terms of PFAS identification, chain length differentiation, and source discrimination. These limitations should be taken into account when employing TOF analysis to assess the presence of PFAS compounds in environmental or analytical studies.

1.6 EXTRACTION, PRECONCENTRATION, AND CLEAN-UP

Regardless of the method used to detect PFAS, it is always necessary to employ efficient extraction, purification, and concentration techniques to accurately identify PFAS in water, especially when they are present in very trace amounts. Typically, PFAS are extracted from

different water samples through a method known as solid-phase extraction (SPE) [119,122,123]. This method is widely favored for its efficiency. However, there are alternative methods that have been employed in numerous research studies such as liquid-liquid extraction (LLE), ion-pair extraction (IPE), solid-phase microextraction (SPME), and dispersive liquid-liquid microextraction (DLLME). LLE involves the separation of compounds based on their solubilities in two different immiscible liquids [124]. IPE is a technique that pairs ions to make them more extractable in organic solvents [125]. SPME method is a solvent-free technique that uses a coated fiber to absorb and concentrate analytes [126]. DLLME is a rapid and solvent-minimizing method where a disperser solvent helps create a fine emulsion for extracting the analytes [127]. Each of these methods has unique advantages and has been applied based on the particular needs of the study. **Table 1.1** and **Table 1.2** provide examples of the extraction techniques that are frequently used to extract PFAS from water samples.

Solid-phase extraction (SPE) is predominant method for preconcentration or clean-up prior to LC-MS/MS analysis for PFAS determination in aquous samples [47,82]. An alternative to SPE is liquid-liquid extraction (LLE), but its automatization is quite limited. In the present era of "green chemistry," the sample preparation methods that produce large amounts of toxic solvents, i.e., liquid-liquid extraction (LLE) and solid phase extraction (SPE), are difficult to justify for multi-residue determinations of PFAS in water samples [128]. Moreover, the widely used solid-phase extraction (SPE) is in some cases tedious, time- consuming and could present some disadvantages, i.e., sample extracts being insufficiently clean, poor recovery, the breakthrough of large sample volumes, essentially at the ultra-trace level [82].

Modern approaches to sample preparation are more devoted to solventless extraction methods, where miniaturization has become an essential role in analytical chemistry.

Microextraction techniques were introduced as a modern and more efficient alternative to traditional extraction methods such as LLE or SPE. Arthur and Pawilszyn research group first proposed Solid-phase microextraction (SPME). SPME is growing in popularity due to its ease of use, high sensitivity, and reproducibility [65,129]. It requires neither solvents nor previous sample preparation, is simple to automate, and has been successfully applied for environmental analysis, in particular to water samples [130]. Similar to SPME, Stir bar sorptive extraction (SBSE) was also developed to screen priority organic micro-pollutants in water samples [131,132]. Because of the higher volume of absorbent material in SBSE, this new sampling method enables to increase in the sensitivity by a factor of 1000 as compared to SPME, decreasing the detection limits at the sub-ng/L level [132]. Up to date, only very limited number of studies have been reported that used the SBSE method for extracting perfluoroalkyl acids in water [133–135]. Villaverde-de-Sáa and co-workers first studied polydimethylsiloxane (PDMS) and polyethersulfone (PES) based materials as stir bar coating for extraction of perfluoroalkyl acids (PFAA) from water samples [135]. Later, Aparicio and Yao research groups studied commercial ethylene glycol-modified silicone (EG-silicone), and polydimethylsiloxane (PDMS) coated stir bars [133,134]. All three studies have used LC-MS/MS method for the determination of perfluoroalkyl acids. And even few examples of the application of SBSE with GC/MS were reported [136]. Moreover, none of those methods have been evaluated for the determination of a wider range of PFAS precursors, such as FTOHs.

1.7 CURRENT ANALYTICAL CHALLENGES AND BRIDGING THE RESEARCH NEED

The research in this study addresses a critical need for alternative and more accessible analytical methods to monitor FTOHs and PFCAs; two major groups of PFAS, those are mostly detected in the water matrices. Currently, the predominant technique for PFAS analysis involves

the use of LC-MS/MS. But the high cost associated with using LC-MS/MS as the primary method for PFAS analysis poses a significant barrier, limiting accessibility for many laboratories, especially those with limited resources. Moreover, the labor-intensive as well as time-consuming preconcentration and cleanup procedures required by existing extraction methods further complicate the analysis. Recognizing these challenges, there is a clear and pressing need for the development of alternative analytical methods that are not only simple and cost-effective but also sensitive enough to detect FTOHs and PFCAs at trace levels in water matrices. By addressing these research needs, this study aims to enhance the accessibility and efficiency of PFAS analysis, which will ultimately contribute to more comprehensive environmental monitoring and protection efforts.

1.8 RESEARCH GOAL, OBJECTIVES, AND HYPOTHESIS

The overarching goal of this research is to develop simple, sensitive, green, and cost-effective methods to study perfluorocarboxylic acids (PFCAs) and fluorotelomer alcohols (FTOHs). Two major groups of PFAS, perfluorocarboxylic acids (PFCAs) and fluorotelomer alcohol (FTOHs), were studied; and green analytical techniques were employed. To achieve this goal, we specify two aims.

Aim 1: Develop an improved method for FTOHs and PFCAs detection in water by using SBSE-TD-GC-MS.

a) Apply and test the use of Stir Bar Sorptive Extraction (SBSE) coupled with thermal desorption-gas chromatography-mass spectrometry (TD-GC-MS) to detect and quantify FTOHs and PFCAs in water.

b) Optimize the SBSE-TD-GC-MS method to achieve a detection limit within the parts per trillion (ppt) range.

c) Study the concentration of FTOHs and PFCAs in biosolids.

Aim 2: Investigate the occurrence and transport of FTOHs and PFCAs in wastewater treatment process.

a) Establish a baseline of FTOHs and PFCAs levels in wastewater in the El Paso region.

b) Study the level of FTOHs and PFCAs in biosolid to understand the fate of PFAS in wastewater treatment process

We hypothesize that Stir Bar Sorptive Extraction (SBSE) coupled with TD-GC-MS offers a sensitive and fast analytical alternative for PFAS determination at ppt level. We also hypothesize that PFAS will be detected in wastewater collected from all WWTPs, and the wastewater treatment process cannot remove PFAS completely. Thus, wastewater is a source of PFAS contamination in our drinking water supply.

1.9 FUTURE CHAPTERS AND APPROACHES OF EACH STUDY

This dissertation is structured into five chapters, with a focus on methodological development using three FTOHs and nine PFCAs as the primary model compounds. The dissertation commences with a comprehensive review of the existing literature in this Chapter 1, setting the stage for the subsequent research. The research findings of the dissertation are included in Chapters 2, 3, and 4, each dedicated to a specific study. These chapters are organized in manuscript format in preparation for manuscript submission. These three chapters contains sections of introduction, methodology, results and discussion, and references. Finally, Chapter 5 offers an overall conclusion, summarizing the findings from the three studies and directions for future research.

The three detailed studies in chapters 2-4 are summarized as follows.

Chapter 2: Rapid, Efficient, and Green Analytical Technique for Determination of Fluorotelomer Alcohol in Water by Stir Bar Sorptive Extraction

This chapter outlines the development and validation of a novel method for detecting Fluorotelomer Alcohols (FTOHs) in water. The technique utilizes Stir Bar Sorptive Extraction (SBSE) combined with Thermal Desorption-Gas Chromatography-Mass Spectrometry (TD-GC-MS). It emphasizes a straightforward, rapid, and eco-friendly approach with minimal solvent use and no clean-up requirement. The method's sensitivity is highlighted through the analysis of three prevalent FTOHs. Key factors such as extraction time, stirring speed, solvent composition, salt addition, and pH levels were optimized for maximum extraction efficiency.

Chapter 3: Green Analytical Method for Determination of Perfluorocarboxylic Acids (PFCAs) in Water by Stir Bar Sorptive Extraction coupled with GC-MS.

This chapter discusses the development of an improved SBSE method, combined with TD-GC-MS, for the analysis of Perfluorocarboxylic Acids (PFCAs) in water. The study thoroughly evaluates the method's performance in terms of linearity, recovery, sensitivity, repeatability, and spiked recovery across various water matrices. The findings demonstrate the method's precision, environmental friendliness, and reliability for PFCAs analysis in wastewater samples.

Chapter 4: Investigating FTOHs and PFCAs in Biosolids Applying Stir Bar Sorptive Extraction Followed by GC-MS Analysis

In this chapter, the focus shifts to the development of an efficient method for extracting PFCAs and FTOHs from biosolid samples, followed by analysis utilizing the previously established SBSE-TD-GC-MS techniques. The study presents the analysis results of biosolids samples from four different wastewater treatment facilities, demonstrating the effectiveness of the method in these specific environmental contexts.

Through each chapter, the dissertation presents innovative, sustainable, and effective methods for environmental analysis.

Chapter 2: Rapid, Efficient, and Green Analytical Technique for Determination of Fluorotelomer Alcohol in Water by Stir Bar Sorptive Extraction

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2.1 ABSTRACT

Fluorotelomer alcohols (FTOHs) are one of the major classes of per- and polyfluoroalkyl substances (PFAS). Due to their potential toxicity, persistence, and ubiquitous presence in the environment, some common PFAS are voluntarily phased out; while FTOHs are used as alternatives to conventional PFAS. FTOHs are precursors of perfluorocarboxylic acids (PFCAs) and therefore they are commonly detected in water matrices, which eventually indicate PFAS contamination in drinking water supplies and thus a potential source of human exposure. Even though studies have been conducted nationwide to evaluate the degree of FTOHs in the water environment, robust monitoring is lacking because of the unavailability of simple and sustainable analytical extraction and detection methods. To fill the gap, we developed and validated a simple, rapid, minimal solvent use, no clean-up, and sensitive method for the determination of FTOHs in water by stir bar sorptive extraction (SBSE) coupled with thermal desorption-gas chromatography-mass spectrometry (TD-GC-MS). Three commonly detected FTOHs (6:2 FTOH, 8:2 FTOH, and 10:2 FTOH) were selected as the model compounds. Factors such as extraction time, stirring speed, solvent composition, salt addition, and pH were investigated to achieve optimal extraction efficiency. This "green chemistry" based extraction provided good sensitivity and precision with low method limits of detection ranging from 2.16 ng/L to 16.7 ng/L and with an extraction recovery ranging 55% to 111%. The developed method were tested on tap water, brackish water, and wastewater influent and effluent. Two FTOHs (6:2 FTOH and 8:2 FTOH) were detected in two

wastewater samples at 78.0 and 34.8 ng/L, respectively. This optimized SBSE-TD-GC-MS method will be a valuable alternative to investigate FTOHs in water matrices.

Keywords: Per- and polyfluoroalkyl substances (PFAS), Fluorotelomer alcohol (FTOHs), Stir bar sorptive extraction (SBSE), Method Development, GC-MS, Wastewater.

2.2 INTRODUCTION

Per- and polyfluoroalkyl substances (PFAS) are a family of more than 4700 highly fluorinated compounds manufactured for diverse applications [1]. They are organic chemicals manufactured for their heat, water, and stain-resistant properties. PFAS can be found ubiquitously in the environment because they are widely used in everyday consumer products such as non-stick cookware, food wrapper, cosmetics, all stain-resistant products, aqueous fire-fighting foams, etc. [2–5]. Fluorotelomer alcohols (FTOHs) are one of the major classes of PFAS, with a fluorinated tail ranging from 4-14 carbons and an alcohol head group [4]. They are denoted by the nomenclature m:n FTOH, where 'm' is the length of the nonpolar tail and 'n' is the length of the non-fluorinated carbon linkage [137,138] (**Figure 2.1**). FTOHs find extensive applications in numerous consumer and industrial goods, serving as essential components in non-stick cookware [139], cleaning agents [17], food packaging [139], aqueous film-forming foam (AFFF) [16,140], as well as in the production of fluoropolymers where FTOHs act as surfactants, lubricants, and intermediate products in the manufacturing processes [141–144]. As a result, FTOHs have become widespread contaminants in the environment. Over the past few decades, FTOHs have been found ubiquitous in water [44,144–146], air [147–151], soil [44,138,152], food [153], and biological matrices [154,155]. Studies have also shown that FTOHs can break down into other persistent, bioaccumulative, and toxic perfluorinated compounds especially perfluorocarboxylic acids (PFCA) in water by various biotransformation mechanisms [18,143,152,156,157]. Therefore, FTOHs could be considered an indirect source of PFCAs in the

environment. Moreover, the high volatility of fluorotelomer-based compounds can undergo long-range environmental transport contributing to PFAS contamination in remote regions such as the Arctic, where they are degraded and contribute to potential perfluoroalkyl carboxylate contamination [17,45].

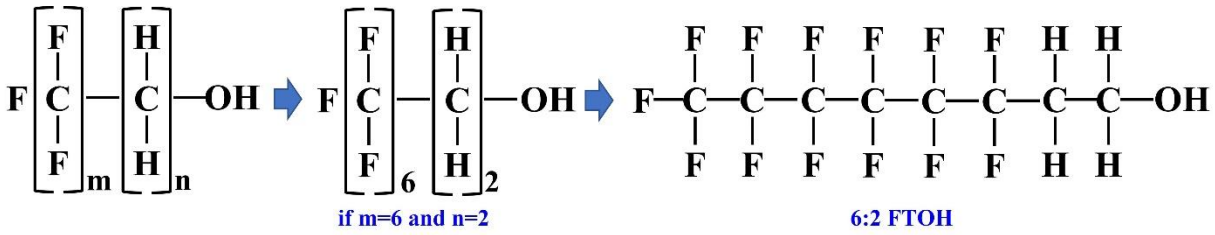

Figure 2.1: An illustration of FTOH molecular structure.

Reports have shown that human exposure to high levels of PFAS (*e.g.*, mg/L levels in human blood has been linked to various health conditions such as reproductive disorders, developmental effects in children, liver and kidney disease, immunotoxicity, hepatotoxicity, and thyroid hormone disruption immune system depression [76,140,158,159]. Being a major precursor of common PFCAs such as PFOA, FTOHs cause similar adverse health impacts on human health and the environment [160]. Human exposure to FTOHs mainly occurs through ingestion pathways such as diet and drinking water [44,145,153]. Because they are widely used, FTOHs have been found in various types of water sources throughout the world, including tap water [44,145], municipal wastewater influents and effluents [17,144,146,161], industrial wastewater influents and effluents [145,161,162], river water [44,163,164], and rainwater [163,165]. Therefore, the determination of FTOHs in all water matrices is important to monitor their presence at trace levels, and thus a simple, sensitive, and robust analytical method is necessary to identify and quantify FTOHs in water matrices.

Analytical methods for FTOHs in water are mainly gas chromatography-mass spectrometry (GC-MS) [164]. There are several GC-MS based well-established methods for the

analysis of FTOHs in water matrices [166,167]. As an alternative to GC-MS methods, the analysis of FTOHs by liquid chromatography-tandem mass spectrometry (LC-MS/MS) was reported [140,144,153,161]. LC techniques have some limitations, mostly regarding background contamination issues [80]. Specific attention needs to be paid to sample vial caps made of Teflon or fluoropolymers, fluoropolymer tubing, solvent inlet filters, and other laboratory products containing fluoropolymer compounds, as they are sources of FTOHs contamination in laboratories [80]. Moreover, LC-MS techniques require highly sophisticated and expensive instruments, and their analysis requires some sample pre-treatments, which include time-consuming extraction, clean-up, and pre-concentration steps. In aqueous matrices, solid-phase extraction (SPE) is the prevailing enrichment or clean-up method prior to LC-MS/MS analysis [144,161,168], and in some cases, it is tedious and time-consuming. SPE also encounters other disadvantages, such as sample extracts being insufficiently clean, having poor recovery and breakthrough problems which are critical at the ultra-trace level [169]. Another technique is liquid-liquid extraction (LLE), but its automatization is quite limited. In the present era of "green chemistry," this sample preparation method is difficult to justify its large solvent consumption for multi-residue monitoring of PFAS in water samples [128]. Modern approaches to sample preparation are more devoted to solventless extraction methods, where miniaturization has become a vital role in analytical chemistry. More recently, microextraction techniques were introduced as an alternative to classical extraction techniques, *i.e.*, LLE or SPE. Belardi and Pawilszyn research group [170,171] first proposed solid-phase microextraction (SPME). It requires neither solvents nor prior sample preparation, is simple to automate, and has been successfully applied for environmental analysis in particular to water samples [130]. Two separate research group by Bach [44] and Ayala-Cabrera, successfully applied SPME to determine different fluorotelomer compounds in water[145]. Similar to SPME, stir bar

sorptive extraction (SBSE) was also developed to screen priority organic micro-pollutants in water samples [131,132,172]. SBSE is an environmentally friendly and low-cost sample preparation technique that can easily handle large sample volumes with minimum labor. SBSE has been used in numerous fields, including environment and food analysis, forensic analysis, pharmaceuticals, and cancer research [173,174]. Because of the higher volume of absorbent material in SBSE, this sampling method enables to increase in the sensitivity by a factor of 100 as compared to SPME, decreasing the detection limits at the sub-ng/L level [132,172]. SBSE is growing in popularity due to its ease of use, high sensitivity, and reproducibility [172,173,175]. Moreover, SBSE matches the main principle of green chemistry. Limited research has utilized the SBSE method to extract perfluoroalkyl acids from environmental water samples [15,133–135]. These studies employed the LC-MS/MS method to analyze the extracted compounds. However, the specific application of the SBSE method for extracting FTOHs from water samples and analyzing them using GC-MS has not been explored.

The purpose of this research was take advantage of the green aspect that SBSE offers to develop and validate a rapid, simple, and improved method for the determination of FTOHs in water using SBSE coupled with thermal desorption-gas chromatography-mass spectrometry (TD-GC-MS). Three commonly detected FTOHs (6:2 FTOH, 8:2 FTOH, and 10:2 FTOH) in the environment were used as the model compounds to develop the method, which can be applied to other known/unknown FTOHs in the water. The study focused on evaluating the selectivity, sensitivity, precision, and accuracy of the method. The developed method was applied to the analysis of water samples collected from different sources, such as tap water, brackish water, and four municipal wastewater treatment plants. Additionally, the study compared the performance of the developed method with other commonly used methods for the detection of FTOHs in water.

Based on our literature review, this study is the first of its kind in applying the SBSE technique for FTOHs analysis in water.

2.3 EXPERIMENTAL

2.3.1 Standards and Reagents

The analytical standard of studied fluorotelomer alcohols, as listed in **Table 2.1**, were supplied by Fisher Scientific, USA. Sodium chloride (98%) was obtained from Fisher Scientific, USA. HPLC-grade acetonitrile and methanol were bought from J.T.Baker®, USA. Mirex from Fisher Scientific, USA, was used as the internal standard. Mirex stock solutions at 1000 µg/mL and 10 µg/mL were prepared in acetonitrile. Ultra-pure deionized (DI) water from the Milli-Q system (Millipore, Bedford, MA) was used in dilutions and sample preparations. The analytical standard substances were dissolved in HPLC-grade acetonitrile to prepare standard stock solutions of 1000 µg/mL FTOHs. These stock solutions were stored in amber glass vials at 4°C.

Table 2.1: List of the studied fluorotelomer alcohol.

Sl. No.	Compound Name	Acronym	Molecular Formula	Molecular Weight	CAS No.
1	1H,1H,2H,2H-Perfluorooctan-1-ol	6:2 FTOH	$C_8H_5F_{13}O$	364.10	647-42-7
2	1H,1H,2H,2H-Perfluorodecan-1-ol	8:2 FTOH	$C_{10}H_5F_{17}O$	464.12	678-39-7
3	1H,1H,2H,2H-Perfluorododecan-1-ol	10:2 FTOH	$C_{12}H_5F_{21}O$	564.13	865-86-1

2.3.2 Sample Collection

Wastewater samples were provided in September 2022 by El Paso Water Laboratories, El Paso, Texas, USA. Wastewater samples (influent and effluent) from four municipal wastewater treatment plants (labeled as WWTP-1, WWTP-2, WWTP-3, and WWTP-4) were collected in a 500 mL polypropylene bottle without leaving headspace before being stored in a refrigerator at 4

°C. To prepare for further analysis, each wastewater sample was centrifuged at 4000 revolutions per minute (rpm), and the supernatant was collected and stored at 4 °C. All the wastewater samples were analyzed for FTOHs within 14 days. We collected tap water directly from our laboratory's water supply. The brackish water samples were provided by the Brackish Groundwater National Desalination Research Facility located in Alamogordo, New Mexico.

2.3.3 Sample Preparation and Stir Bar Sorptive Extraction (SBSE)

For method development, 50 ng/L, 100 ng/L, and 500 ng/L FTOHs mixture solutions were used. Briefly, to prepare a 500 ng/L FTOHs mixture solution, 100 µL of 100 µg/L FTOHs mixture, 19.88 mL of D.I. water, and 20 µL of 10 µg/mL Mirex (as the internal standard) were added into a 20 mL amber vial. Then, a commercially available stir bar (Twister™, 10 mm × 1 mm, Gerstel, USA) coated with polydimethylsiloxane (PDMS), was placed into the vial, and the solution was stirred at 1000 rpm for a pre-determined period of time. The stir bar was removed after stirring. It was then rinsed with deionized water. Finally, it was dried using lint-free tissue paper and placed in a thermal desorption tube for TD-GC-MS analysis. **Figure 2.2** illustrates the overall SBSE-TD-GC-MS technique.

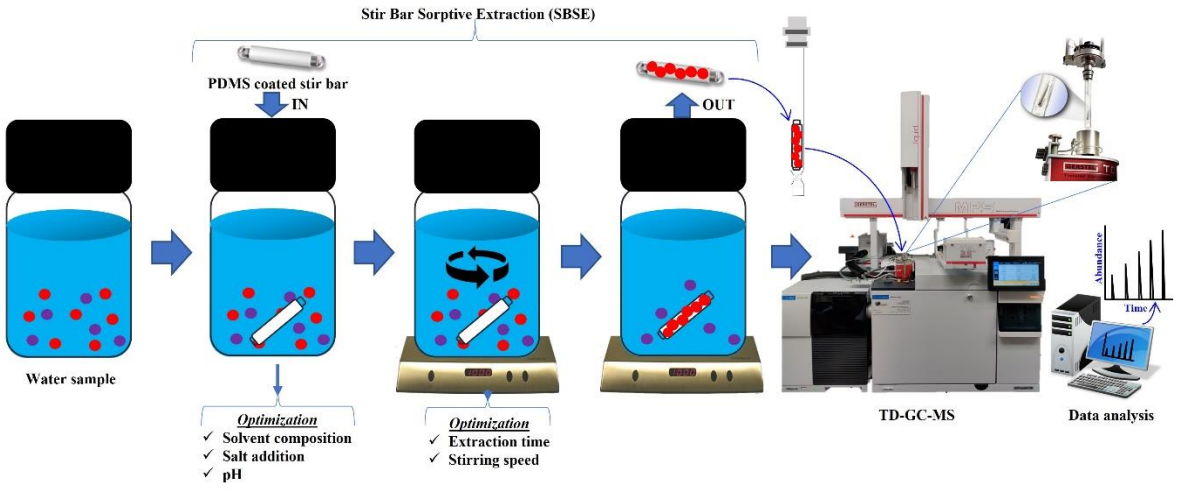

Figure 2.2: Schematic representation of experimental approach for SBSE-TD-GC-MS method.

2.3.4 Thermal Desorption-Gas Chromatography-Mass Spectrometry (TD-GC-MS) Analysis

All targeted FTOHs were analyzed in a thermal desorption unit (TDU, Gerstel, MD, USA), coupled with an 8890-Gas Chromatography (GC) and a 5977B Mass Selective (MS) Detector (Agilent Technologies, CA, USA). The thermal desorption process was programmed as follows. The initial temperature was set at 50°C holding for 0.5 min; the temperature was increased to 280°C at 60°C/min and held for 7.0 min. Ultra-high purity helium was used as the carrier gas with a constant flow of 1.2 mL/min. The transfer line temperature was maintained at 290°C. During desorption, all the desorbed compounds were concentrated in a cold injection system, CIS4/TDU baffled liner (Gerstel, USA), at -40°C prior to GC injection. Once the desorption process was completed, the CIS4 was heated to 300°C at 12°C/sec and held for 5 min in splitless mode. The FTOHs were separated and analyzed by GC-MS using solvent vent mode through an HP-5MS UI column (30 m × 0.25 mm i.d., 0.25 µm film thickness) (Agilent, CA, USA).

The GC oven temperature was programmed as follows: Initial temperature 40°C, held for 2 minutes; ramped at 10°C/min to 170°C and then held for 2 min; finally, ramped at 25°C/min to 300°C and held for 4.8 min (total run time of 27 min). The transfer line temperature was maintained at 280°C. The ionization energy of the electron ionization (EI) source was 70 eV, and the solvent delay was set at 3 min. For qualitative and quantitative analysis, the mass spectrometer was operated in the SIM (selected ion monitoring) mode at m/z values 69, 95, 131, 169, 181, and 272. The identification of compounds was conducted by ChemStation Mass Spectral Search Program, and the National Institute of Standards and Technology Library (NIST17) was used for the identification of FTOHs profiles. **Figure 2.3** shows the chromatogram of 10 ng of FTOHs

analytical standard generated using the optimized oven program in the selected ion monitoring (SIM) mode.

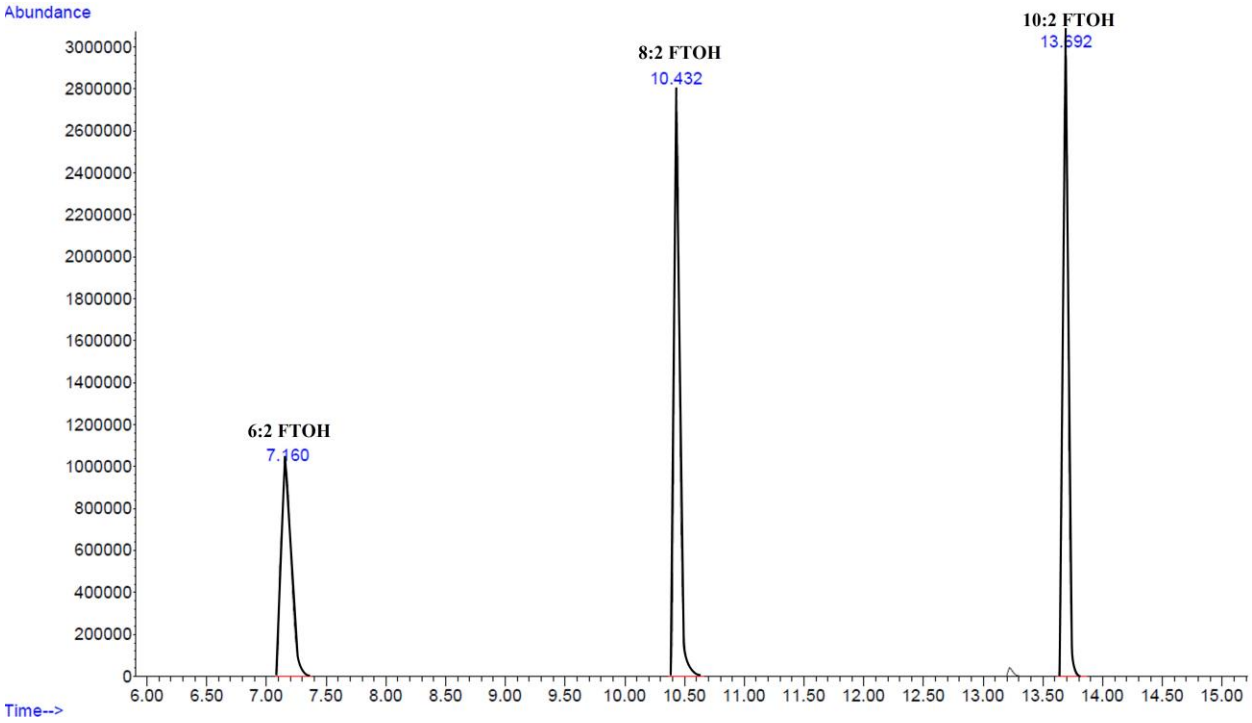

Figure 2.3: A chromatogram of 10 ng of three FTOHs analytical standards in SIM mode.

2.3.5 Method Validation and Quality Control

To minimize the contamination of samples, we avoided the use of products that may contain fluorotelomer-based substances during sampling, sample preparation, and instrumental analysis. One distilled water blank was incorporated into the analytical procedures for every batch of samples. Each sample was measured thrice. If the relative standard deviation (RSD) of the replicates is greater than 25%, a 4^{th} sample was analyzed. Limit of detection (LOD) was calculated based on the standard deviation of the minimum detectable response (Sy) and the slope of the calibration curve (S) according to the formula: LOD = (3Sy)/S, and limit of quantification (LOQ) was calculated according to the formula: LOQ = (10Sy)/S. The extraction efficiency of SBSE-TD-GC-MS was evaluated by spiking FTOHs standards into the water samples at two spiking levels:

100 ng/L and 500 ng/L. The spiked recovery experiments (n=3) were performed using the optimized SBSE method in four different water matrices; tap water, brackish water, wastewater influent, and wastewater effluent. The matrix effect was calculated using the signal intensity of FTOHs in the sample matrix versus the signal of the same concentration in DI water. Recoveries of FTOHs in wastewater samples were calculated to assess the accuracy of the method in the real water samples, and the relative standard deviation (RSD) was used to evaluate precision.

2.3.6 Statistical Analysis

During the optimization of SBSE parameters, the instrument responses of FTOHs (presented as abundance as shown in **Figure 2.4**) were recorded and presented as means of the replicate measurements in the bar plots. The Tukey Honest Significance Difference test (Tukey HSD) was used for multiple pair-wise-comparison between the means of FTOHs abundances under the conditions used for analysis. The effect of each tested parameter on each FTOHs recovery was considered significant based on a probability of $p<0.05$ except when otherwise stated. The statistical analysis was performed using RStudio (version 1.4.1564).

2.3.7 Analysis of FTOHs in Real Water Samples

In this study, we investigated real water samples by applying our developed and optimized SBSE-TD-GC-MS method for the determination of FTOHs. In brief, 20 mL of water sample, 1 mL of methanol and 400 mg of NaCl, 20 µL of 10 µg/mL Mirex, and one stir bar were added to a 20 mL amber vial. Then the solution was stirred at 1000 rpm for 90 minutes. The stir bar was removed after stirring. It was then rinsed with deionized water. Finally, it was dried using lint-free tissue paper and placed in a thermal desorption tube for TD-GC-MS analysis. A calibration curve (with concentrations ranging from 5 to 500 ng/L) was constructed for the quantification of targeted FTOHs.

2.4 RESULTS AND DISCUSSIONS

2.4.1 Optimization of Stir Bar Sorptive Extraction (SBSE) Parameters

The primary aim of this study was to develop, optimize, and validate a reliable extraction method for the determination of FTOHs in water samples. SBSE was the extraction technique selected in the present work. Several operational parameters in SBSE, such as extraction time, stirring speed, solvent composition, salt addition, and water pH value, were assessed and optimized prior to GC-MS analysis.

2.4.1.1 Optimization of Extraction Time

In the SBSE process, sufficient extraction time is needed to reach equilibrium and can result in a significant improvement in SBSE. The effect of extraction time for the extraction of FTOHs was investigated in the range of 30–240 min. As shown in **Figure 2.4 A**, the signal intensity of target FTOHs significantly increased with the increase of extraction time from 30 to 90 min. For 6:2 FTOH, 8:2 FTOH and 10:2 FTOH, extraction equilibrium was achieved in 90 min, and no significant increase in extraction recoveries was observed from 90 min to 240 min. 8:2 FTOH kept almost constant regardless of the increase of extraction time. In contrast, for 6:2 FTOH, after achieving equilibrium in 90 min, the response decreased with increasing extraction time to 240 min. Based on the results, it is concluded that 90 min was satisfactory for the three FTOHs compounds, hence was selected as the optimum extraction time and used for subsequent experiments.

2.4.1.2 Optimization of Stirring Speed

The stirring speed influences the SBSE efficiency since agitation controls the mass transfer of the analytes from the aqueous media toward the PDMS phase of the stir bar during the equilibrium process [128]. However, high stirring speed can affect the integrity of the stir bar coating

and promote unstable movements, which reduces extraction efficiency. To avoid excessive or too little agitation, the studied speeds were tested at 750, 1000, 1250, and 1500 rpm with 90 minutes of stirring time. As shown in **Figure 2.4 B**, all three FTOHs were best extracted at a stirring speed of 1000 rpm. Thus, 1000 rpm was chosen as the optimum stirring speed and used for consequent experiments.

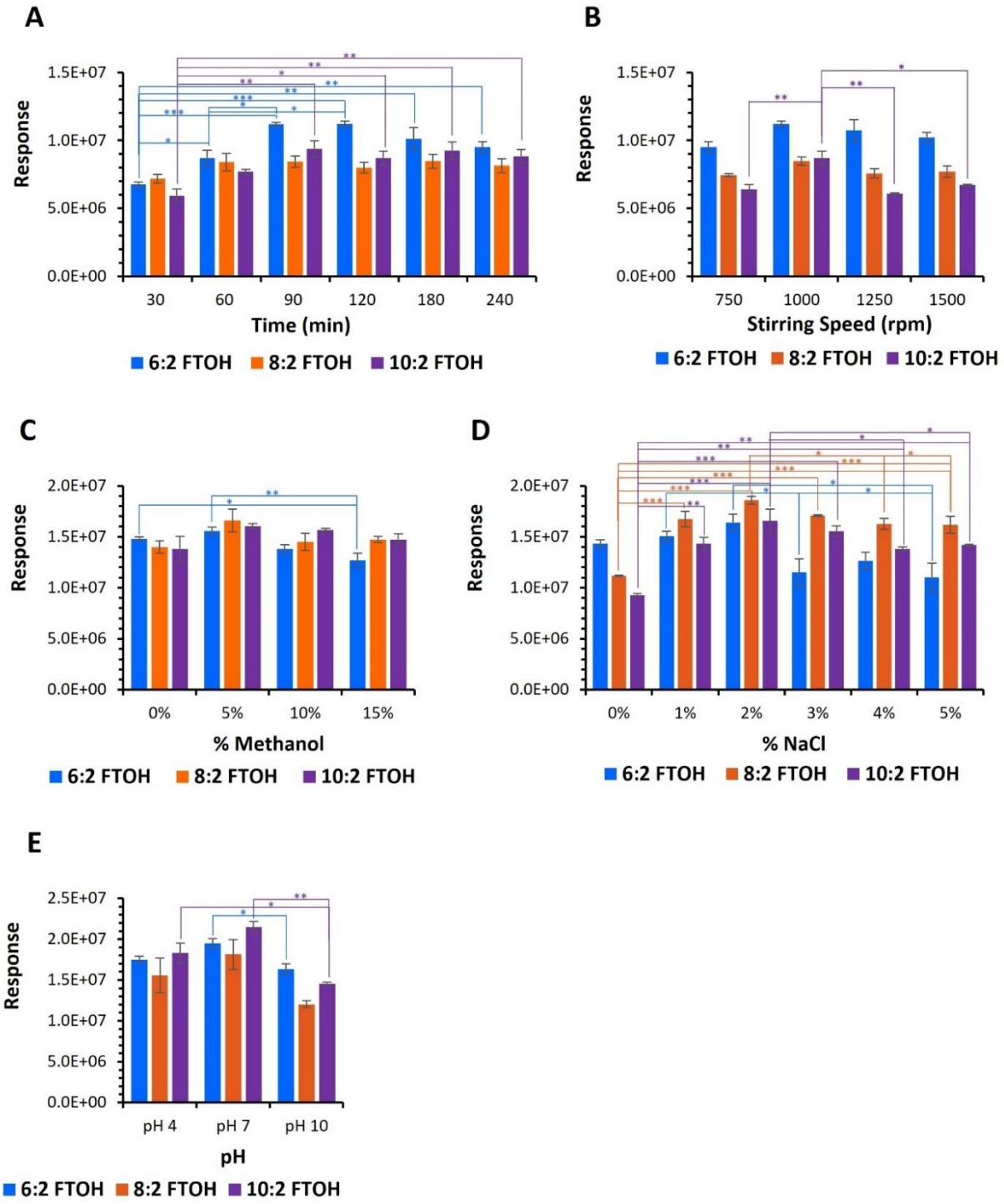

Figure 2.4: Optimization of SBSE parameters.

A: *instrument response (i.e., extraction recovery) of FTOHs with various extraction time.* ***B:*** *the effect of stirring speed on FTOHs extraction recovery.* ***C:*** *the response of FTOHs under various methanol compositions.* ***D:*** *the effect of salt addition on FTOHs extraction.* ***E:*** *the influence of pH on FTOHs extraction. Error bars represent standard deviations of duplicate measurements.*

*Asterisks indicate differences of statistical significance within the groups determined by the Tukey test: *P < 0.05, **P < 0.01, ***P < 0.001.*

2.4.1.3 Optimization of Solvent Composition

A considerable loss of efficiency can occur by the adsorption of the analytes onto the glass vial, known as the 'wall-effect'. This effect can play a negative role in leading to analyte loss and decreasing the recovery by SBSE [176]. By adding methanol to the solution, it can increase the solubility of non-polar compounds (log K_{ow}>3) in the solution and improve their extractability by SBSE [177]. To minimize the adsorption of these compounds onto the glass walls and increase the extraction efficiency, methanol was the solvent of choice for these experiments. The addition of methanol to the sample matrix was studied at 0%, 5%, 10%, and 15% of the total solvent composition. As shown in **Figure 2.4 C**, the instrumental response of each three analysts depicts that maximum recovery yields were obtained with 5% methanol which composition was therefore selected as the optimum solvent composition.

2.4.1.4 Optimization of Salt Addition

The characteristics of the aqueous matrix play a very important role in SBSE efficiency. Typically, ionic strength in the sample matrix was adjusted by the addition of different amounts of salt. When salt is added, there are two opposite effects, *i.e.* salting-out (favoring the extraction) and salting-in (resisting the extraction) effects [178]. Extraction efficiency for most of the compounds, especially those with log K_{ow} values lower than 3.5, increases with increasing ionic strength. To increase the ionic strength of the sample matrix, sodium chloride (NaCl) was used for the optimization of this parameter. The effect of the salt addition on the extraction efficiency was studied at six NaCl levels between 0% and 5% (w/v). As shown in **Figure 2.4 D**, significant impacts of NaCl contents on FTOHs extractions were observed. The extraction efficiency of all three FTOHs increased with increasing NaCl content from 0 to 2% (w/v) but started to decrease

when the salt fraction increased to 5% (w/v). Therefore, 2% NaCl was selected as an optimized parameter for the extraction of all three FTOHs.

2.4.1.5 Optimization of pH

In many pre-treatment procedures, pH was adjusted to improve the extraction efficiency. In this work, the influence of sample pH on the extraction performance was investigated at three pH conditions (pH 4, pH 7, and pH 10). As shown in **Figure 2.4 E**, the signal intensity of target FTOHs decreased in both the acidic and basic sample matrix. The best extraction recoveries were obtained at pH 7, with no pH adjustment required. Considering that the average pH values of real water samples range from 6.5 to 7.5, the result indicated that there was no need for adjusting the sample pH during SBSE. This is also considered a cost benefit when applying this method to water monitoring.

2.4.2 Method Repeatability and Accuracy

Method repeatability was evaluated at three spiking levels (50 ng/L, 100 ng/L, and 500 ng/L) with seven replicates at each level. Repeatability was calculated based on relative standard deviation (RSD). A lower RSD indicates a higher degree of repeatability and lower variability in the measurements or experimental results. **Table 2.2** shows the RSD was less than 10% for all compounds at each three-concentration level indicating good repeatability and reliability of the method for the determination of FTOHs in water.

Table 2.2: Method repeatability test in DI water and Tap water at three spiking levels 50 ng/L, 100 ng/L, and 500 ng/L; (n=7).

Matrix	Analyte	Repeatability (RSD; n = 7) Spiked concentration		
		50 ng/L	**100 ng/L**	**500 ng/L**
DI water	6:2 FTOH	5.9%	8.8%	5.2%
	8:2 FTOH	4.7%	8.1%	6.1%
	10:2 FTOH	6.4%	3.9%	6.8%
Tap water	6:2 FTOH	9.5%	7.9%	1.5%
	8:2 FTOH	6.8%	7.7%	5.9%
	10:2 FTOH	8.1%	6.6%	7.6%

2.4.3 Method Recovery Experiment

The recoveries of FTOHs by the optimized SBSE method were tested. For this purpose, 2 µL of 5 mg/L of a standard mixture containing 6:2 FTOH, 8:2 FTOH, and 10:2 FTOH (*i.e.*, 10 ng of each FTOH) were placed in a thermal desorption tube, and the FTOHs were analyzed by GC-MS. To determine the SBSE recoveries, the same amount of the standard mixture containing 6:2 FTOH, 8:2 FTOH, and 10:2 FTOH were spiked in 20 mL of DI water, and extraction was performed using the optimized SBSE method. All recovery experiments were performed in triplicate. The ranges of absolute recoveries of standards were as follows; 6:2 FTOH (91 – 111%), 8:2 FTOH (60 – 81%), and 10:2 FTOH (55 – 75%). The results indicate that SBSE could still be effective in extraction FTOHs from water within 90 min.

The second recovery experiment was conducted to evaluate the SBSE performance in extracting FTOHs in different water matrices as compared to DI water. The recovery experiments were performed at two spiking levels: 100 ng/L and 500 ng/L of FTOHs using the optimized SBSE method in four different water matrices; tap water, brackish water, wastewater influent, and wastewater effluent. As shown in **Table 2.3**, SBSE provided good recoveries of FTOHs in tap

water, brackish water, and wastewater effluent with 81%-115%, 96 – 122%, and 64 – 120%, respectively. Recoveries of FTOHs in wastewater influent (12 – 87%) were affected due to the matrix effect of wastewater influent. There were suspended solid materials observed in the influent samples that might have affected the extraction recovery by the SBSE method. Therefore a removal of the suspended solid in wastewater influent may need to be in place prior to the SBSE extraction of FTOHs.

Table 2.3: Results of spiked recoveries of target FTOHs in various types of water samples at two spiking levels (100ng/L and 500 ng/L) by SBSE method. Results shown as mean recovery (±Standard Deviation) in percent; (n=3).

	6:2 FTOH		**8:2 FTOH**		**10:2 FTOH**	
Water type	**100 ng/L**	**500 ng/L**	**100 ng/L**	**500 ng/L**	**100 ng/L**	**500 ng/L**
Tap Water	101 (±4)	81 (±3)	115 (±4)	109 (±4)	88 (±1)	93 (±6)
Brackish Water	104 (±5)	96 (±4)	122 (±3)	107 (±4)	113 (±3)	105 (±3)
Wastewater Influent	36 (±3)	50 (±5)	87 (±9)	56 (±3)	21 (±4)	12 (±1)
Wastewater Effluent	64 (±7)	85 (±3)	120 (±6)	94 (±4)	73 (±3)	80 (±6)

2.4.4 Analytical Performance of the Method

Table 2.4 represents the linearity, coefficient of determination, limit of detection (LOD), and limit of quantification (LOQ) for the studied FTOHs. Method linearity was studied by extracting the spiked FTOHs in the ranges of 25–500 ng/L. Satisfactory linearity of each target FTOHs in the water matrix was obtained with a coefficient of determination (R^2) greater than 0.996. The linear range experiments provide the necessary information to estimate the limit of detection (LOD) and limit of quantification (LOQ). LOD of 6:2 FTOH, 8:2 FTOH, and 10:2 FTOH were found to be 2.16 ng/L, 10.8 ng/L, and 16.7 ng/L respectively. The LOQ for 6:2 FTOH, 8:2 FTOH and 10:2 FTOH are 23.3 ng/L, 28.4 ng/L and 40.1 ng/L respectively.

Table 2.4: Analytical merits of proposed SBSE-TD-GC-MS method for FTOHs determination.

Analyte	Linear ranges (ng/L)	Coefficient of determination (R^2)	Recovery (%)	LOD (ng/L)	LOQ (ng/L)
6:2 FTOH	25 - 500	0.996	91 - 111	2.16	23.3
8:2 FTOH	50 - 500	0.994	60 - 81	10.8	28.4
10:2 FTOH	50 - 500	0.996	55 - 75	16.7	40.1

In comparison to other analytical techniques (**Table 2.5)**, the results showed that the developed method is feasible for determining trace FTOHs in water matrices. As shown in **Table 2.5**, our developed method requires only 20 mL of water sample which makes the method simple to operate. Moreover, our SBSE method requires less organic solvents (only 1 mL of methanol to enhance the extraction efficiency), which makes the method more eco-friendly and greener than most of the other methods listed in **Table 2.5**.

SBSE may encounter challenges in the presence of complex sample matrices, such as high levels of dissolved solids, suspended particulates, or high concentrations of organic matter. These matrix interferences can affect the extraction efficiency and result in inaccurate quantification of the target analytes. But a matrix-matched calibration approach can help compensate for the matrix effects present in water samples. This can lead to improved accuracy and recovery without altering the method itself. Overall, The study incorporates "green chemistry" principles by minimizing the use of organic solvents, reducing environmental impact, and emphasizing sustainability in the extraction process. Moreover, the specific application of the SBSE method for extracting FTOHs from water samples and analyzing them using GC-MS has not been explored.

Table 2.5: Comparison of analytical parameters of the proposed method with the reported method for the analysis of FTOHs in water.

Sample matrix	Extraction Method	Analytical Technique	Sample volume	Organic solvent usage	LOD (ng/L)	Recovery (%)	Reference
Rainwater, River water, Wastewater	LLE	GC-MS	500 mL	200 mL MTBE, 10 mL ethyl acetate	0.2~0.5	58~78	[163]
Rainwater	LLE	GC-MS	500 mL	75 mL MTBE	6.9~8.7	38~59	[165]
River water, Wastewater	SPE	GC-MS/MS	1000 mL	20 mL methanol, 4 mL ethyl acetate	1.0~2.0	80~97	[164]
Tap water, River water	SPME	GC-MS	10 mL	N/A	50.0~100.0	94~124	[44]
Wastewater	SPE	UPLC-MS/MS	250 mL	14 mL ACN	0.03~0.12	83~116	[144]
Wastewater	LLE	GC-MS	400 mL	400 mL ethyl acetate	0.59~0.85	82~87	[17]
Wastewater	SPE	UPLC-MS/MS	250 mL	10 mL ACN	0.05~0.12	84~112	[161]
Tap water, Brackish water, Wastewater	SBSE	GC-MS	20 mL	1 mL methanol	2.16~16.7	55~111	This work

2.4.5 Quantification of FTOHs in Water Samples

The concentrations of FTOHs in real wastewater samples were tested using optimized SBSE-TD-GC-MS, and the results are shown in **Table 2.6**. Out of the three FTOHs tested, only 6:2 and 8:2 FTOH were detected. In the influent of WWTP-1, 34.8 ng/L of 8:2 FTOH were detected; the influent of WWTP-4 contains 78.0 ng/L of 6:2 FTOH. These observations are consistent with a previous study by Ma et al. [161] that 6:2 FTOH and 8:2 FTOH are the most dominating FTOHs in water matrices, as both of them are the precursor for common PFCAs.

Table 2.6: Results of determination of target FTOHs in real water samples; (n=2).

Water samples	**Sampling point**	Detected (ng/L)/ Not detected (N.D.)		
		6:2 FTOH	**8:2 FTOH**	**10:2 FTOH**
Tap Water		N.D.	N.D.	N.D.
Brackish Water		N.D.	N.D.	N.D.
WWTP-1	Influent	N.D.	34.8	N.D.
	Effluent	N.D.	N.D.	N.D.
WWTP-2	Influent	N.D.	N.D.	N.D.
	Effluent	N.D.	N.D.	N.D.
WWTP-3	Influent	N.D.	N.D.	N.D.
	Effluent	N.D.	N.D.	N.D.
WWTP-4	Influent	78.0	N.D.	N.D.
	Effluent	N.D.	N.D.	N.D.

2.5 CONCLUSIONS

We developed and validated a simple, rapid, and robust analytical method (SBSE-TD-GC-MS) for the efficient extraction and determination of FTOHs in water matrices. Using three FTOHs (6:2, 8:2, and 10:2) as the model compounds, our SBSE-TD-GC-MS method has low method limits of detection ranging from 2.16 ng/L to 16.7 ng/L, and good linearity (25 to 500 ng/L), repeatability (%RSD below 10%) and recoveries (55% to 111%). These method characteristics indicate that the method is a reliable alternative for FTOHs monitoring in the aquatic matrices. Applying the

optimized method, two FTOHs, 6:2 FTOH and 8:2 FTOH, were detected in two wastewater treatment plants at the parts per trillion (ppt) level. Overall, the SBSE is a sensitive and green analytical technique that requires a low volume of sample (*i.e.*, 20 mL) and minimum amount of organic solvent (1 mL methanol). Moreover, it offers simplicity and ease of use. Its compatibility with a highly sensitive and specific analytical technique, GC-MS, allows the detection of FTOHs at very low concentrations in water samples. This will provide a more comprehensive understanding of the presence and distribution of FTOHs in water samples.

2.6 ACKNOWLEDGMENTS

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Chapter 3: Green Analytical Method for Determination of Perfluorocarboxylic Acids (PFCAs) in Water by Stir Bar Sorptive Extraction Coupled with GC-MS

3.1 ABSTRACT

Perfluoroalkyl carboxylic acids (PFCAs) represent a significant category within the broader group of Per- and polyfluoroalkyl substances (PFAS). They are man-made persistent organic chemicals manufactured for their resistance to heat, water, and stains. PFCAs can be found ubiquitously in the environment because they are widely used in everyday consumer products. As a result, PFCAs are commonly detected in surface water and wastewater. Therefore, drinking water supplies are contaminated directly from these sources which is a substantial source of human exposure. Thus, sensitive and effective analytical methods are needed for monitoring water quality. In this study, we developed an enhanced Stir Bar Sorptive Extraction (SBSE) method coupled with Thermal Desorption-Gas Chromatography-Mass Spectrometry (TD-GC-MS) for the analysis of PFCAs in water. This study summarizes a detailed evaluation of the method's linearity, recovery, sensitivity, repeatability, and spiked recovery across different water matrices. The method demonstrates linearity with coefficients of determination (R^2) spanning 0.9892 to 0.9988. Sensitivity metrics revealed low limits of detection (LOD) and quantification (LOQ) in the low ng/L (ppt) range for all analytes, achieving LODs between 21.2 ng/L to 74.0 ng/L. The recoveries for the method varied from 47-97%, suggesting an efficient extraction process. In comparison with traditional PFCAs analysis methods, the developed SBSE technique requires a notably lesser sample volume of 1 mL and minimal solvent usage, enhancing eco-friendliness and reducing potential contamination and handling errors. Repeatability assessments at two concentration levels produced %RSD values at 14% or less for any target PFCA compounds, indicating good precision. Additionally, the method's robustness across various water matrices reflected by the spiked

recovery experiment underscored the method's efficacy in real-world applications. These attributes showcase the developed method's capability to serve as a precise, eco-friendly, and reliable tool for the analysis of PFCAs across diverse water matrices.

Keywords: Perfluorocarboxylic acids (PFCAs), PFAS, Stir bar sorptive extraction (SBSE), GC-MS, Wastewater.

3.2 INTRODUCTION

PFAS stands for Per- and Polyfluoroalkyl substances, encompass a vast group of over 4,700 synthetic compounds characterized by their multiple fluorine atoms [1]. They are well-known for their water and oil-repellant properties, and thermal stability; while their environmental mobility, resistance to biochemical degradation, bioaccumulative effects, and toxicity are causing increasing concerns. PFAS can be found in common consumer products like non-stick cookware, clothing, leather, upholstery, and carpets etc. [4,179]. They can also be used in fire-fighting foams and in industrial applications such as wetting agents, additives, coatings, emulsifiers, paints, waxes, and polishes [2–5]. Their useful properties are due to their structure, which includes a fluorinated carbon chain that is both hydrophobic and oleophobic and hydrophilic charged functional groups (such as carboxylic or sulfonic acid) attached to the structure [4,6,7]. Among the PFAS compounds, perfluorocarboxylic acids (PFCAs) stand as a significant subgroup, which have gained attention due to their ubiquitous occurrence and persistence in the environment, especially in the water matrices [122]. A PFCA compound is expressed as $C_nF_{(2n+1)}$-COOH, where $C_nF_{(2n+1)}$ represents the per-fluoroalkyl portion of the molecular structure [4,8,180]. PFCAs predominantly exist in water environments owing to their low pKa values, which make them more soluble [181]. Human exposure to PFCAs primarily occurs through consumption, including food and water intake, particularly near heavily contaminated locations [10]. Recent studies suggest that exposure to elevated levels of

PFCAs is associated with a range of adverse health outcomes, including reproductive and developmental problems, liver and kidney damage, immunological effects, and disturbances in thyroid function and overall immune system health [29–35].

In the past few years, research has increasingly revealed that PFAS are found in a wide range of aquatic environments worldwide. Specifically, the presence of PFCAs has been confirmed in a variety of sources. These substances have been detected in surface waters [182,183], underground aquifers [184], oceans [185], and even in the water we consume from faucets [186] and in bottled form [187,188]. Furthermore, they have been identified in the water entering and exiting wastewater treatment plants, showcasing their widespread distribution in both natural and treated water systems. Wastewater treatment plants (WWTPs) are known to accept industrial, household, and commercial waste streams. Due to their uses in our daily lives, PFCAs are being found in most wastewater systems globally. The concentrations of PFCAs in wastewater have been reported ranging from not detected level to 143 µg/L (ppb) [189]. As wastewater treatment processes are not designed to remove PFCAs, these compounds were found in wastewater effluents ranging from <0.13−6.67 ng/L. The US EPA (United States Environmental Protection Agency) has recognized the severity of this issue and has established interim updated minimum reporting levels as low as 4 parts per trillion (ppt) for PFOA [190] indicating the critical need for vigilant monitoring of these substances in our water.

Numerous methods for analyzing PFCAs in aqueous environments have been established. A variety of chromatographic techniques such as Liquid chromatography (LC), Gas chromatography (GC), High-performance Liquid chromatography (HPLC), and Ultra-high performance liquid chromatography (UHPLC) are frequently used for the determination of PFCAs in water matrices [68–70,122,191,192]. Less commonly used methods include nuclear magnetic resonance,

Fourier transform infrared spectroscopy, and ion chromatography [69,71,72]. In the last decade, research has been conducted on developing new on-site detection techniques that are convenient and reliable. For example, several sensor-based methods such as electrochemical sensors, ion-selective electrodes (ISE), fluorescence sensors, and smartphone app-based monitoring systems have been developed [193,194]**.** However, those techniques usually detect high concentrations of PFCAs (> 10 ppb). Moreover, methods such as the total oxidizable precursor assay have determined that a significant fraction of the total PFCAs present in environmental samples consists of unidentified compounds [75,76]. The gold standard for PFCA analysis has been liquid chromatography coupled with tandem mass spectrometry (LC-MS/MS) [193]**.** US EPA has developed and validated a series of methods for PFAS using LC-MS/MS with sample preparation using solid-phase extraction [76]**.** Despite these advancements, the most common methods, centered around LC, are not without their challenges. Yamashita et al., [80] reported background contamination issues in PFAS monitoring. Specific attention should be paid to auto-sampler vial caps made of Teflon or fluoropolymers, fluoropolymer tubing, solvent inlet filters, and a variety of other laboratory products containing Teflon and perfluoroalkoxy compounds, as they are sources of PFAS in blanks. A well-known problem in electrospray ionization (ESI), as is commonly used in LC-MS, is ion suppression [82]. Moreover, while LC methods are invaluable for PFCA detection, they require sophisticated and costly instrumentation, and the analytical process can be time-intensive. This underscores the need for ongoing innovation and improvement in analytical techniques to enhance our ability to monitor PFCAs efficiently and effectively [83].

As an alternative, Gas chromatography (GC) is a convenient instrument for volatile and semi-volatile PFAS analysis (for example, fluorotelomer alcohols, FTOHs; perfluorinated sulfonamido ethanols, FASE [70,82,136]. In order to decrease the polarity and increase the volatility of

PFCAs, a derivatization process is often performed. There are several studies have been done on PFCAs derivatization prior GC-MS analysis [70,82,103–108]. Gołebiowski research group applied 2,4 difluoroaniline for derivatization in the presence of N, N′-dicyclohexylcarbodiimide (DCC) [109]. However, this method consists of several sample preparation steps: pH adjustment, phase separation and washing the organic phase with HCl, $NaHCO_3$, and NaCl solution. Dufkova et al., developed a fast derivatization procedure for PFCAs by using isobutyl chloroformate to quantify PFAS in water [110]. Strozynska and colleagues developed two derivatization processes using triethylsilanol and N,N-Dimethylformamide dimethylacetal for the separation of PFCAs through GC-MS [108]. Overall, due to rapid and simple derivatization steps, isobutyl chloroformate-based derivatization appeared to be commonly used for the determination of PFAS in water by GC-MS.

In the analysis PFCAs in water, both liquid chromatography (LC) and gas chromatography (GC) require robust pre-treatment processes to detect these compounds at ultra-trace levels. Solid-phase extraction (SPE) is the conventional choice for concentrating and purifying samples before LC-MS/MS analysis, owing to its widespread application [82,119]. However, liquid-liquid extraction (LLE), despite its potential, is less favored due to its limited potential for automation. As the scientific community shifts towards sustainable practices, traditional sample preparation techniques like SPE and LLE, which rely heavily on harmful solvents, are becoming less attractive, especially for analyzing multiple residues of PFCAs in aqueous samples [128]. The application of SPE in PFCA analysis, while established, can sometimes be labor-intensive and less effective, with potential pitfalls such as incomplete removal of matrix components, inconsistent recovery rates, and issues with processing large volumes of water samples when targeting ultra-trace level contaminants. This is particularly challenging given the stability and persistence of PFCAs, which

demand highly efficient extraction and clean-up methods to achieve the required sensitivity and specificity [82].

In recent years, microextraction techniques have emerged as a modern alternative to traditional extraction methods such as liquid-liquid extraction (LLE) and solid-phase extraction (SPE). Alzaga et. al. [85] and Monteleone at. el. [91] successfully applied solid-phase microextraction (SPME) to determine multiple PFCAs in water matrices. Typically, SPME require less solvent and are designed to be more sensitive, more selective, and are generally easier and safer to operate. Dispersive liquid-liquid microextraction (DLLME) has been employed by researchers such as Liu et al., [93] and Hu et al., [98] as an extraction technique specifically for extracting PFCAs from aqueous environments. Similar to SPME, Baltussen research group first introduced stir bar sorptive extraction (SBSE) as a potent technique for screening organic micro-pollutants in water samples [195]. SBSE is well known for its environmentally friendly approach and low operational costs, excels in handling large volumes of samples with minimal labor, and seamlessly fits into the standard of green chemistry [136]. It stands out in its higher absorbent volume compared to SPME, amplifying sensitivity by an order of magnitude and facilitating the detection of compounds at sub-ng/L concentrations [136,195,196]. SBSE's robust adsorption capacity and high extraction efficiency address the limited exploration of its application in extracting PFCAs for subsequent thermal desorption - gas chromatography-mass spectrometry (TD-GC-MS) analysis. Previous studies have indeed utilized SBSE for perfluoroalkyl acid extraction, coupled with liquid chromatography-tandem mass spectrometry (LC-MS/MS) for analysis [197–200]. However, our research fills a gap in the existing literature by employing this method with TD-GC-MS, a novelty in the field. Additionally, employing a thermal desorption unit coupled with GC-MS can eliminate the organic solvent usage for desorption in LC-MS/MS methods, as reported in the previous studies. By

optimizing the SBSE conditions, we can enhance the extraction effectiveness, decrease the time required for extraction, and address the issue of low recovery reported in earlier research.

The study aimed to develop and validate a method that is both straightforward and highly sensitive, involves handling smaller sample volumes, and requires minimal manual effort by utilizing green chemistry-based SBSE techniques to extract and quantify PFCAs in water samples through TD-GC-MS. The scope of the research included the development and validation of the SBSE-TD-GC-MS method for nine targeted PFCAs. We employed the developed method in investigating PFCAs in water samples sourced from various origins, including tap water; influents, and effluents from four local municipal wastewater treatment facilities. Furthermore, this research contrasted the efficacy of the newly developed method against other prevalent extraction techniques in conjunction with GC-MS for detecting PFCAs in aquatic environments. To our knowledge from the existing literature, this research pioneers the application of the SBSE technique for PFCAs extraction followed by solvent-free desorption using the thermal desorption unit prior to GC-MS analysis.

3.3 EXPERIMENTAL

3.3.1 Standards and reagents

The analytical standard of PFCAs, as listed in **Table 3.1**, was purchased by Fisher Scientific, USA; and standard stock solutions of 1000 µg/mL of PFCA were prepared in acetonitrile. Isobutyl chloroformate (98%), pyridine (99%), and isobutyl alcohol were supplied from Fisher Scientific, USA. HPLC-grade acetonitrile, hexane, and methanol were purchased from J.T.Baker®, USA. Mirex (Fisher Scientific, USA) was used as the internal standard and 1000 µg/mL and 10 µg/mL Mirex stock solutions were prepared in acetonitrile. Deionized (DI) water, purchased from J.T.Baker®, USA, was used in dilutions and sample preparations.

Table 3.1: List of the studied PFCAs.

	Compound Name	Acronym	Molecular Formula	Molecular Weight	CAS No.
1	Perfluoroheptanoic acid	**PFHpA**	$C_7HF_{13}O_2$	364.06	375-85-9
2	Perfluorooctanoic acid	**PFOA**	$C_8HF_{15}O_2$	414.07	335-67-1
3	Perfluorononanoic acid	**PFNA**	$C_9HF_{17}O_2$	464.08	375-95-1
4	Perfluorodecanoic acid	**PFDA**	$C_{10}HF_{19}O_2$	514.08	335-76-2
5	Perfluoroundecanoic acid	**PFUnA**	$C_{11}HF_{21}O_2$	564.09	2058-94-8
6	Perfluorododecanoic acid	**PFDoA**	$C_{12}HF_{23}O_2$	614.1	307-55-1
7	Perfluorotetradecanoic acid	**PFTeDA**	$C_{14}HF_{27}O_2$	714.11	376-06-7
8	Perfluorohexadecanoic acid	**PFHxDA**	$C_{16}HF_{31}O_2$	814.13	67905-19-5
9	Perfluorooctadecanoic acid	**PFODA**	$C_{18}HF_{35}O_2$	914.1	16517-11-6

3.3.2 Sample Collection

In this study, wastewater samples were obtained from El Paso Water in El Paso, Texas, USA in September 2023. Both wastewater influent and effluent samples were taken from four municipal wastewater treatment plants, denoted as WWTP-1, WWTP-2, WWTP-3, and WWTP-4. The samples were collected in 500 mL polypropylene bottles, ensuring no headspace remained, and were then promptly stored at 4 °C. Upon their arrival at the analytical laboratory, wastewater samples were centrifuged using the Sorvall Legend X1R (Thermo Scientific, USA) at 4000 rpm and 4 °C. The supernatant was separated and stored at the same temperature. All samples were analyzed for PFCAs within 7 days from their collection. Additionally, tap water samples were procured directly from our laboratory for comparative analysis.

3.3.3 Sample Preparation, Derivatization, and Stir Bar Sorptive Extraction (SBSE)

For the determination of PFCAs, a derivatization process was employed according to a methodology developed by Dufková *et al.* [110] with modification. To elaborate, a 2.0 mL polypropylene (PP) vial was used for the derivatization reaction. The vial was filled with a mixture

comprising 1000 µL of 1 µg/mL of nine targeted PFCAs, 50 µL of isobutyl alcohol, 20 µL of pyridine, and 50 µL of isobutyl chloroformate (IBCF). To ensure the uniform mixing and derivatization reaction of these components, the vial was placed in an ultrasonic bath and sonicated for 30 seconds. After sonication, the vial was set aside for 5 minutes to allow the formation of the isobutyl ester derivatives of each PFCAs.

The next step involved the extraction of these PFCAs derivatives from the water sample. For this purpose, the Stir Bar Sorptive Extraction (SBSE) technique was employed. Briefly, a 20 mL amber vial was prepared with a mixture of 20 µL of the derivatized PFCAs solution (with a concentration of 1 µg/mL), 19.96 mL of deionized (D.I.) water, and an addition of 20 µL of Mirex at a concentration of 10 µg/mL, which served as the internal standard. The ultimate concentration of the PFCAs in the vial was 1 ng/mL in a total volume of 20 mL. A commercially available stir bar coated with polydimethylsiloxane (Twister™, 10 mm × 1 mm, Gerstel, USA) was then introduced into the vial. The solution was subsequently stirred at a speed of 1000 rpm for 120 minutes. Post stirring, the stir bar was carefully retrieved, washed with D.I. water to remove any residual sample, and then dried using a lint-free tissue. The stir bar was then placed in a thermal desorption tube (TDT), and ready for the final TD-GC-MS analysis. **Figure 3.1** illustrates the entire SBSE-TD-GC-MS procedure.

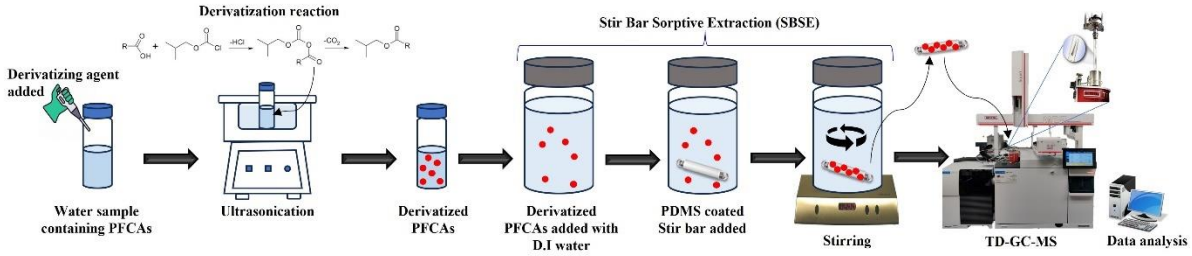

Figure 3.1: Schematic representation of experimental approach with SBSE coupled to TD-GC-MS.

3.3.4 Analysis Using Thermal Desorption-Gas Chromatography-Mass Spectrometry (TD-GC-MS)

PFCAs analysis was conducted using a thermal desorption unit (TDU, Gerstel, MD, USA), integrated with an 8890-Gas Chromatograph system and a 5977B Mass Selective Detector (Agilent Technologies, CA, USA). The thermal desorption program was set up as follows. The starting temperature was set at 40°C and maintained for 0.5 minutes; thereafter the temperature was ramped at 60°C/minute to 280°C and held for 5.0 minutes. Ultra-high pure helium served as the carrier gas, flowing steadily at 1.2 mL/minute. The temperature of the transfer line was kept steady at 290°C. As desorption occurred, the compounds were concentrated in a cold injection setup, CIS4/TDU, with a baffled liner (from Gerstel, USA), at -40°C before the GC step. After completing the desorption process, the CIS4 was heated at 12°C /s to 300°C and held for 5 minutes in splitless mode. PFCAs were then separated and assessed by GC-MS. They were analyzed using solvent vent mode. The analysis was conducted through an HP-5MS capillary column (30 m × 0.25 mm i.d., 0.25 um film thickness, Agilent, CA, USA).

The temperature for GC oven was configured as follows: Initial temperature was set to 40°C and held for 2 minutes. Then it ramped at 10°C/min to 200°C, where it was held for another 2 minutes. Lastly, it ramped to 300°C at 25°C/min. Hold time was set for 3 min (total run time, 27 min). The temperature for the transfer line was set at 280°C. The electron ionization (EI) source had ionization energy set at 70 eV. The solvent delay time was set to 1 min. The mass selective detector (MSD) was set at scan mode (40-980 m/z) for PFCAs identification. For qualitative and quantitative analysis, the mass spectrometer was configured in the selected ion monitoring (SIM) mode at m/z values 69, 131, 169, 181, and 272. Compounds were identified using ChemStation

Mass Spectral Search Program. The NIST17 (National Institute of Standards and Technology) library was used for the identification of PFCA profiles.

3.3.5 Optimization of Stir Bar Sorptive Extraction Parameters

In this study, our foremost objective was to establish and validate a reproducible method for extracting PFCAs from water samples. We opted for the Solid-Phase Stir Bar Sorptive Extraction (SBSE) technique as our method of choice for extraction. We evaluated various operational parameters that could influence the extraction process. Factors such as the duration of extraction, the speed of stirring, the composition of the solvent, the addition of salt, and the pH value of the water were meticulously assessed. These parameters were then optimized to ensure the most efficient extraction possible by using SBSE. Following this optimization, the samples underwent Gas Chromatography-Mass Spectrometry (GC-MS) analysis for further evaluation.

3.3.6 Quality Control and Quality Assurance

For quality assurance, a D.I. water blank was processed with each batch of samples to serve as a procedural blank. Each sample underwent duplicate measurements, and a third analysis was conducted if the relative standard deviation (RSD) exceeded 25%. A nine-point calibration curve was constructed using calibration standards (25 ng/L, 50 ng/L, 100 ng/L, 200 ng/L, 400 ng/L, 800 ng/L, 1000 ng/L, and 2000 ng/L) spanning from 0 to 2000 ng/L. The detection limit (LOD) was determined using the standard deviation (Sy) of the lowest measurable response and the calibration curve's slope (S), applying the equation LOD = (3Sy)/S. Similarly, the quantification limit (LOQ) was derived using LOQ = (10Sy)/S. We assessed the method's repeatability by analyzing seven replicate samples at concentrations of 100 ng/L and 1000 ng/L. The extraction efficiency of the SBSE-TD-GC-MS method was tested by adding known amounts (10 ng and 20 ng) of PFCAs standards to various water samples (tap water, wastewater influent, and wastewater effluent), and

these spiking experiments were conducted in triplicates. We also quantified the matrix effects by comparing the signal intensities in the sample matrix to those in deionized water. Finally, the accuracy of our method was verified by calculating the recoveries of PFCAs in wastewater, and precision was determined through the relative standard deviation of the recovery data.

3.3.7 Analysis of PFCAs in real water samples

In our research, we analyzed real-world water samples for PFCAs using a carefully developed and optimized SBSE-TD-GC-MS method. The process commenced with the derivatization of the water samples, following SBSE procedure. We utilized a 2.0 mL polypropylene vial containing 1 mL of the water sample for this derivatization reaction. The derivatization was facilitated by adding 50 µL of isobutyl alcohol, 20 µL of pyridine, and 50 µL of isobutyl chloroformate (IBCF). To achieve a consistent and thorough reaction, the vial underwent ultrasonic agitation for 30 seconds and was subsequently left to stand for 5 minutes. The SBSE phase followed in a 20 mL amber vial, where we mixed 17.98 mL of deionized water, 1 mL of methanol, and 1 mL of the derivatized water sample. We added 20 µL of a 10 µg/mL Mirex solution as an internal standard. A PDMS coated stir bar was placed into the vial. The sample was stirred for 120 minutes at 1250 rpm. After extraction, the stir bar was cleaned with D.I. water, dried with lint-free tissue, and placed in a thermal desorption tube, readying it for the final TD-GC-MS analysis.

3.4 RESULTS AND DISCUSSIONS

3.4.1 Optimization of Stir Bar Sorptive Extraction

To optimize the SBSE for extraction PFCA derivatives in water, various parameters were tested.

1) Five different extraction times (60 min, 90 min, 120 min, 180 min, and 240 min) were tested for the extraction time optimization;
2) Four stir bar stirring speeds were investigated (750 rpm, 1000 rpm, 1250 rpm, and 1500 rpm);
3) Five methanol concentrations (0%, 5%, 10%, 15%, and 20%) were tested for the solvent composition optimization during SBSE;
4) Five salt (NaCl) weight (w/v) percentages (0%, 1%, 2%, 3%, and 4%) were tested upon obtaining the optimized conditions in (1) to (3); and finally
5) Three different pH conditions (pH 4, no pH adjustment, and pH 10) were tested.

3.4.1.1 Optimization of Extraction Time

The efficiency of SBSE depends largely on the duration of extraction. To determine the ideal time frame for effectively extracting PFCAs, the process was evaluated over periods ranging from 60 to 240 minutes. Extractions were performed in duplicate, under a constant stirring speed of 1000 rpm at room temperature. As illustrated in **Figure 3.2**, there was significantly higher ($P < 0.05$) escalation in the extraction of all targeted PFCAs as the extraction time expanded from 60 to 120 minutes. Specifically, for PFHpA, extraction equilibrium was reached in 120 min and this equilibrium persisted up to 240 minutes without gains in recovery rates from 90 to 240 minutes. Interestingly, for the remaining PFCAs, their response began to decrease beyond 120 minutes as the duration of extraction increased. This decline post-equilibrium is likely a result of the analytes back into the sample matrix as stirring is prolonged beyond the equilibrium point [136]. Consequently, 120 minutes was determined to be the most effective extraction period and was therefore employed in all further experiments.

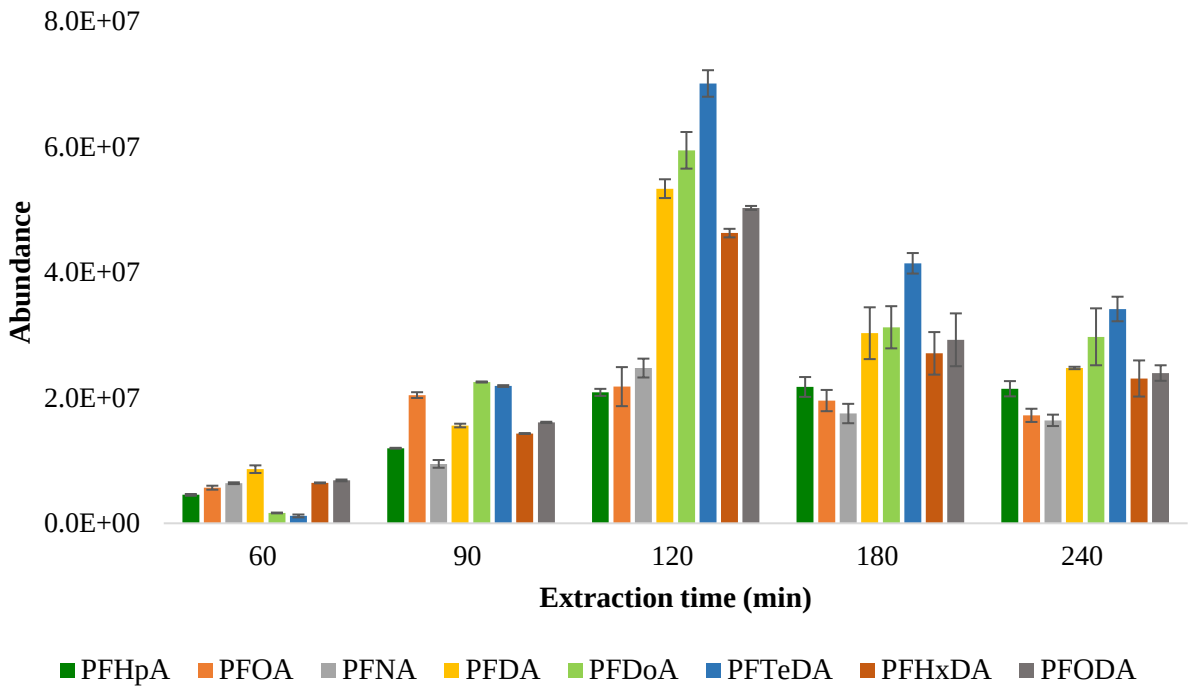

Figure 3.2: Optimization of extraction time.
Instrument response (i.e., extraction recovery) of PFCAs with various extraction time. Error bars in this bar chart represent the standard deviations of duplicate measurements.

3.4.1.2 Optimization of Stirring Speed

The efficacy of SBSE is closely linked to the stirring speed, which is a critical factor in managing the transfer of analytes from the water to the polymer coating on the stir bar [136]. This transfer is essentially a diffusion process, influenced by the agitation of the medium: too little agitation leads to slow mass transfer due to a limited diffusion gradient, while too much can create turbulence that disrupts the delicate equilibrium [64]. Higher stirring speed could damage the polymeric coating of the stir bar or create air bubbles, both of which can result in less efficient extraction. To find out the optimal balance, stirring speeds were methodically evaluated at 750, 1000, 1250, and 1500 revolutions per minute (rpm) over a 120-minute extraction period. As shown in **Figure 3.3**, the extraction of all studied PFCAs peaked at 1250 rpm. This specific speed presumably offers the best balance between ensuring rapid mass transfer and maintaining the

integrity of the stir bar's coating. Subsequently, the speed of 1250 rpm was selected as the most effective for the extraction process and was consistently applied in further experiments to ensure reproducibility and accuracy.

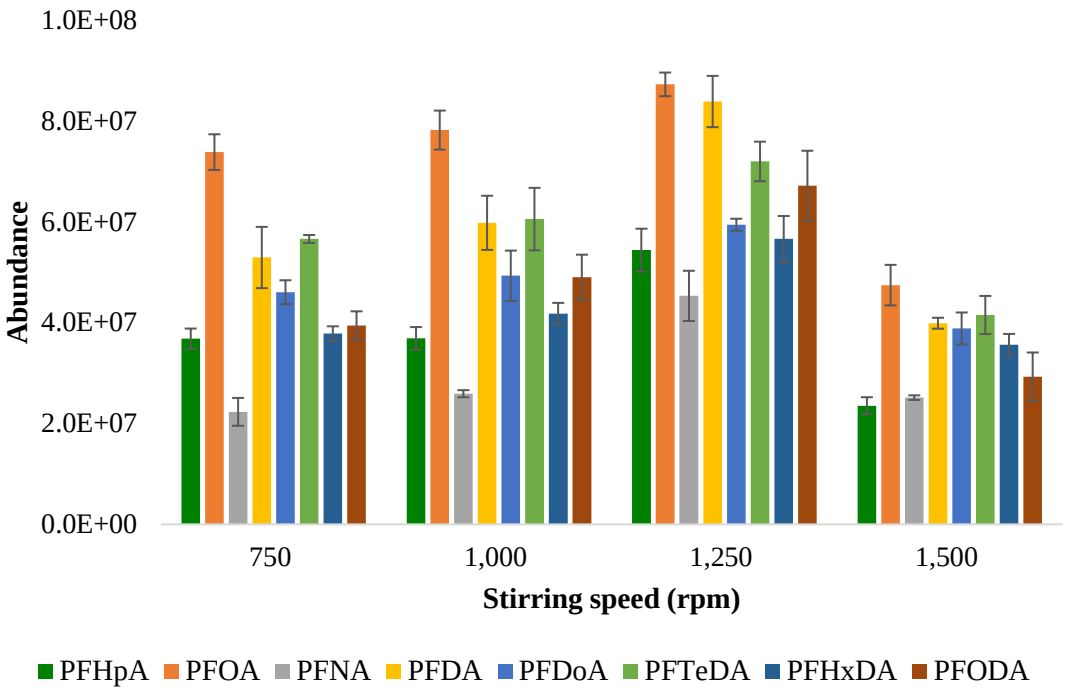

Figure 3.3: Optimization of stirring speed.
The effect of stirring speed on PFCAs extraction recovery. Error bars in this bar chart represent the standard deviations of duplicate measurements.

3.4.1.3 Optimization of Solvent Composition

The "wall effect" in the SBSE procedure refers to the tendency of analytes to adhere to the glass surfaces of the extraction vial, this may result in a significant decrease in their recovery [201]. This adhesion is particularly problematic for hydrophobic compounds, which have a strong affinity for surfaces like glass compared to the aqueous phase. To mitigate this issue, methanol can be added to the aqueous solution as a modifier; it increases the solubility of non-polar compounds, especially those with a log Kow (octanol-water partition coefficient) greater than 3, reducing their propensity to stick to the glass walls and enhancing their availability in the solution for extraction

[136]. The use of methanol offers a two-fold benefit: it acts as a solubilizing agent for the analytes and as a competing solvent against adsorption to the glass vial. By increasing the water solubility of the non-polar analytes, methanol decreases their interaction with the glass, allowing for a greater proportion of the analytes to remain in the liquid phase where they can be extracted by the polymeric phase on the stir bar. The optimization of methanol concentration is crucial, as too much methanol could lead to a decrease in extraction efficiency due to changes in the solvent properties of the matrix. The addition of methanol to the sample matrix was evaluated at various percentages of the total solvent composition (0%, 5%, 10%, 15%, and 20%). The experimental data, presented in **Figure 3.4**, indicates that a methanol composition of 5% yielded the significantly higher ($P < 0.05$) recovery rates for all the analytes. Thus, 5% was determined to be the optimal methanol concentration for enhancing the extraction efficiency in subsequent SBSE procedures.

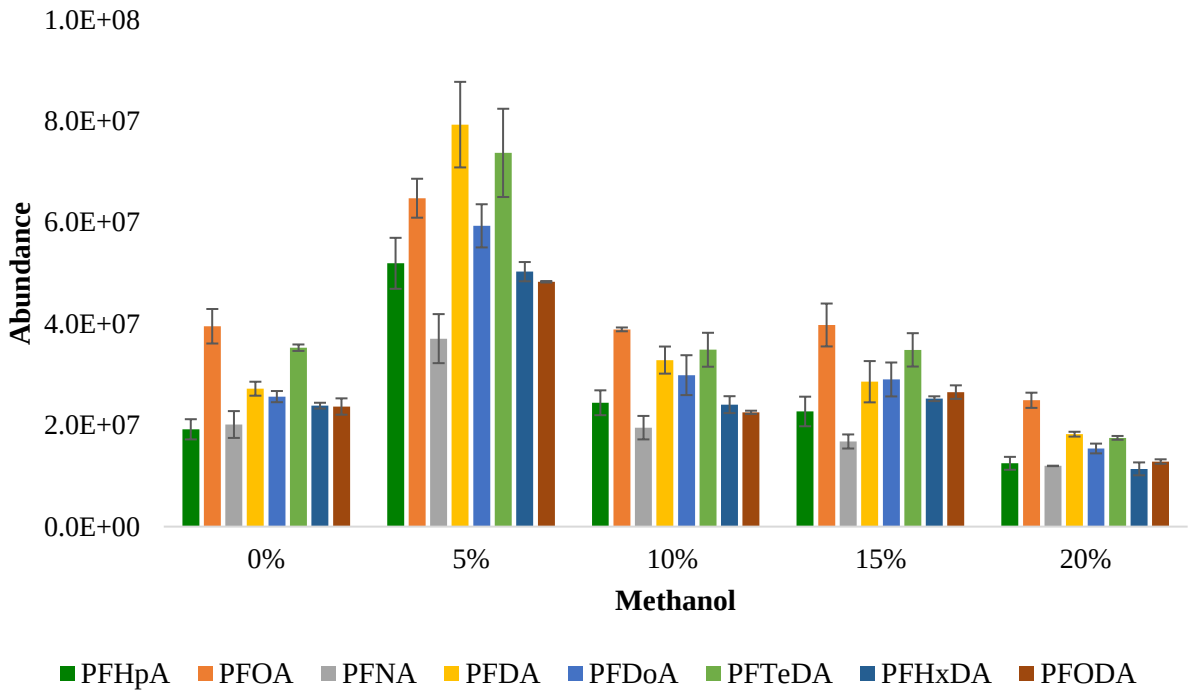

Figure 3.4: Optimization of solvent composition: the response of PFCAs under various methanol compositions.
Error bars in this bar chart represent the standard deviations of duplicate measurements.

3.4.1.4 Optimization of Ionic Strength

In aqueous solutions, the ionic strength, adjusted by adding varying amounts of salt, has a significant impact on the extraction efficiency. Incorporating salt, such as sodium chloride (NaCl), to the aqueous matrix is a critical step in enhancing the SBSE process, as it regulates the ionic strength of the sample matrix [198]. The presence of salt induces a 'salting-out' effect, which promotes the transfer of analytes from the aqueous phase to the sorbent by reducing the solubility of hydrophobic compounds in water. Conversely, too much salt can also lead to a 'salting-in' effect, where the increased ionic strength of the solution can hinder the extraction process, potentially by modifying the physical properties of the aqueous matrix and the sorbent [136]. In optimizing the SBSE for PFCAs, NaCl was added in incremental concentrations, ranging from 0% to 4% (w/v). It was observed that the extraction efficiency significantly improved ($P < 0.05$) as the NaCl concentration was raised to 1% (w/v), suggesting an optimal salting-out condition **(Figure 3.5)**. However, further increasing NaCl to 4% (w/v) led to a reduction in efficiency, likely due to an over-enhanced ionic strength which could negatively influence the extraction dynamics. Hence, after evaluating the effects at various levels, a 1% (w/v) NaCl addition was determined to provide the most favorable conditions for the SBSE of PFCAs. This optimized concentration was thus established as a standard parameter for subsequent extractions to ensure efficient and consistent recovery of the targeted analytes.

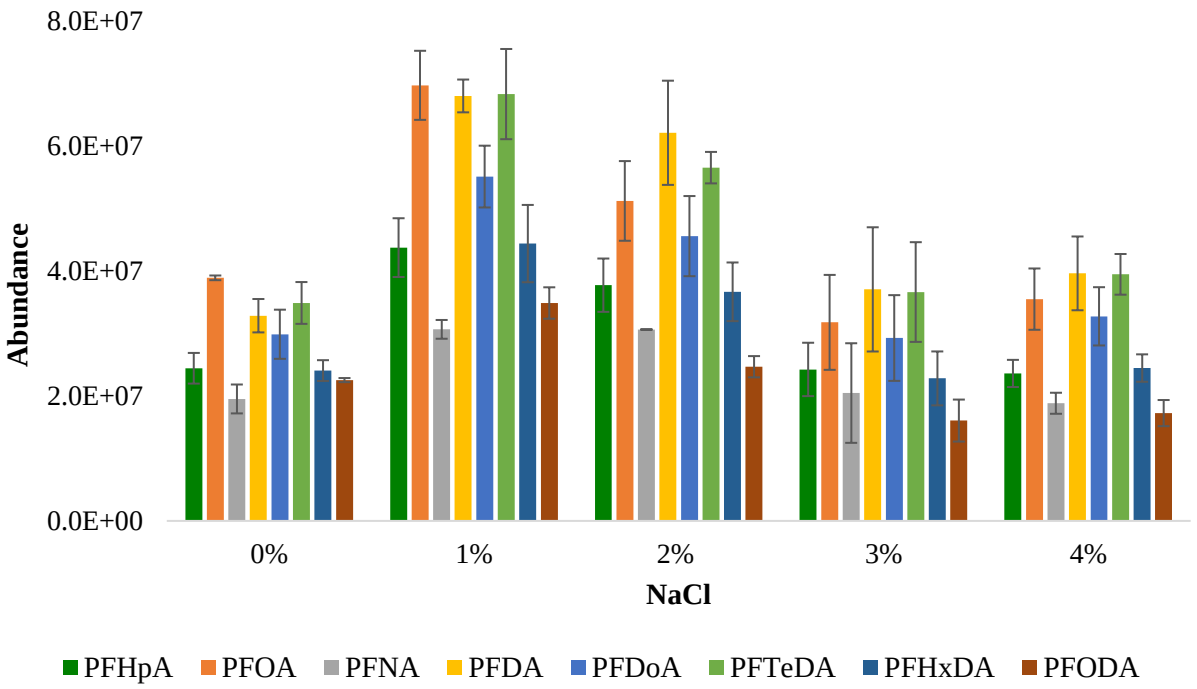

Figure 3.5: Optimization of ionic strength: the effect of salt (NaCl) addition on PFCAs extraction.
Error bars in this bar chart represent the standard deviations of duplicate measurements.

3.4.1.5 Optimization of pH

The pH of the sample is a critical factor in the SBSE method, as it affects the analytes existing forms and consequently their interaction with the sorbent. Considering that the typical pH range for actual water samples is between 6.5 and 7.5, in this study, the influence of sample pH on extraction efficiency was evaluated at three pH levels (pH 4, no pH adjustment and pH 10). **Figure 3.6** demonstrates that significantly higher ($P < 0.05$) recovery rates for extractions were achieved when the pH was not adjusted. At a neutral pH, typically around pH 7, PFCA-esters are more likely to be in their neutral, non-ionic form, which has a higher affinity for PDMS coating on the stir bar due to its hydrophobic nature. This neutral form is less soluble in water and more inclined to be sorbed onto the PDMS surface, resulting in enhanced extraction efficiencies. When the sample pH is either acidic (pH 4) or basic (pH 10), the PFCA esters can become ionized. Acidic

conditions can lead to additional protonation, while basic conditions can result in deprotonation, yielding charged forms of the PFCA-esters. These charged species are more soluble in water and less likely to adhere to the hydrophobic PDMS coating, which significantly decreases the efficiency of the SBSE process (**Figure 3.6**). Therefore, for subsequent extractions, no pH modification was carried out. This approach simplifies the sample preparation process and avoids potential complications associated with pH modification.

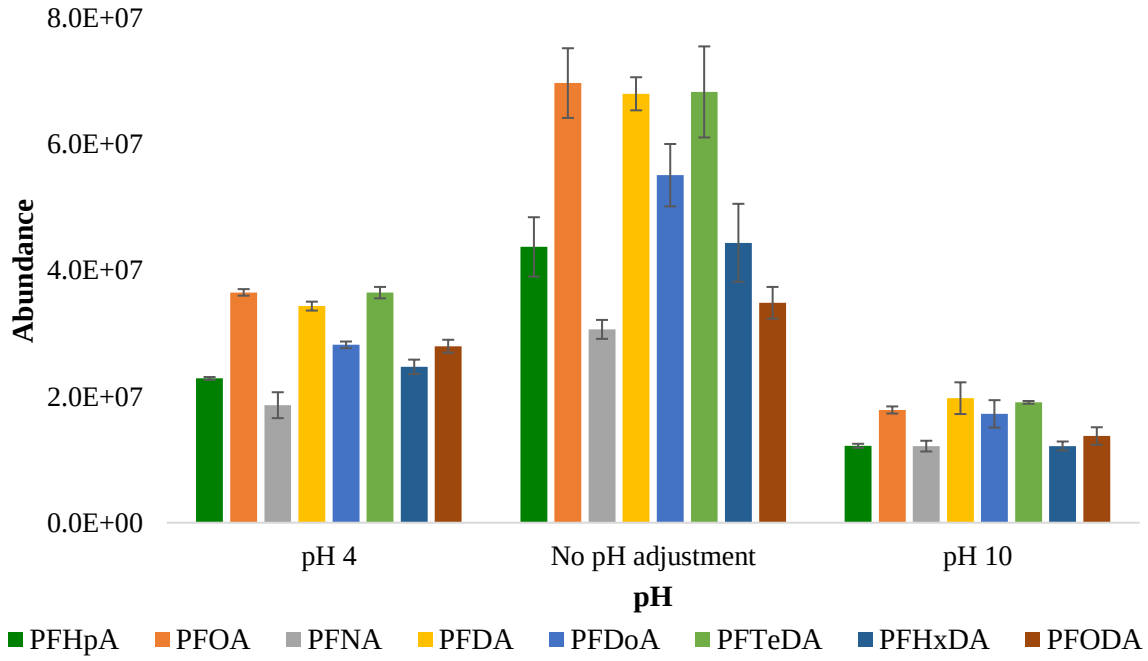

Figure 3.6: Optimization of pH: the influence of pH on PFCAs extraction.
Error bars in this bar chart represent the standard deviations of duplicate measurements.

3.4.2 Evaluating SBSE-TD-GC-MS Performance: Linearity, Recovery, and Sensitivity Metrics

In the development of a robust SBSE method followed by TD-GC-MS analysis for PFCAs, we have established a comprehensive set of analytical parameters that underscore the efficacy and

reliability of this approach. As shown in **Table 3.2**, the retention times for the PFCAs ranged from 5.884 minutes for PFHpA to 13.943 minutes for PFODA, showcasing the method's ability to efficient chromatographic separation and analyze these compounds in a timely manner. The linearity of the method was investigated through the extraction of PFCAs that were spiked into samples at concentrations varying from 75 to 2000 ng/L indicating good linearity range. The coefficients of determination (R^2) ranged from 0.9892 to 0.9988, representing a high level of precision in the quantitative analysis. The method's sensitivity, as demonstrated by the limits of detection (LOD) and limits of quantification (LOQ), were notably in lower ppt range across all analytes. LODs ranging from 21.2 ng/L to 74.0 ng/L. Furthermore, the LOQs remained well under 200 ng/L, with PFODA demonstrating the lowest LOQ of 87.74 ng/L. As documented in the study by Bansal and colleagues, [189], the concentrations of PFCAs in wastewater span a range from below detectable levels up to 143 µg/L (ppb). The LOQs established by our developed method have enabled dependable detection and precise quantification of PFCAs,, even at very low levels. Collectively, these analytical merits not only validate the effectiveness of the SBSE-TD-GC-MS method but also emphasize its potential as a reliable tool for monitoring PFCAs in water matrices.

Table 3.2: Performance Parameters of SBSE-TD-GC-MS for PFCAs Analysis - Linearity, Recovery, and Sensitivity Data

Analyte	Retention time (min)	Parent ion (m/z)	Coefficient of determination (R^2)	% Recovery (±SD)	LOD (ng/L)	LOQ (ng/L)
PFHpA	5.88	405	0.998	97 (±11)	21.2	138
PFOA	6.64	455	0.995	94 (±11)	21.2	103
PFNA	7.61	505	0.996	86 (±5)	30.1	90.8
PFDA	8.44	555	0.989	91 (±10)	54.3	170
PFUnA	9.41	605	0.989	88 (±10)	34.4	133
PFDoA	10.0	655	0.993	85 (±8)	74.0	192
PFTeDA	11.4	755	0.991	85 (±3)	35.0	107
PFHxDA	12.7	855	0.999	79 (±7)	51.8	128
PFODA	13.9	955	0.993	77 (±6)	42.1	87.7

In comparison with established analytical methods for PFCA analysis, as detailed in **Table 3.3**, the newly developed SBSE-TD-GC-MS technique showcases a significant advancement in the field. This method distinguishes itself by requiring only a 20 mL sample volume, which is markedly less than the 200-500 mL typically needed by other techniques. This reduced sample volume is not only a logistical advantage but also a methodological improvement, potentially leading to less sample handling efforts. Furthermore, the SBSE method uses just 1 mL of methanol to enhance the extraction process, thereby supporting the method's eco-friendliness. This reduced solvent use is aligned with the principles of green chemistry, and it makes our approach not only more sustainable but also less hazardous compared to traditional methods that utilize larger volumes of organic solvents. **Table 3.3** summarizes methods, such as SPE and LLE, with recovery rates from 25% to 137%, and limits of detection (LODs) ranging widely from ng/L to µg/L. Our SBSE method achieves a consistent recovery rate of 47-97% with LODs from 21.2 to 74.0 ng/L. This demonstrates a competitive sensitivity and efficiency in detecting trace levels of PFCAs.

Overall, our developed method maintains a balance between methodological simplicity and analytical performance, avoiding the need for more complex and costly MS/MS systems.

Table 3.3: Analytical performance of SBSE-TD-GC-MS for PFCAs detection compared to existing water analysis methods.

Sl No.	Target PFCAs	Matrix/ Sources	Derivatizing agent	Extraction Method	Sample Volume	Instrumentation	Recovery %	LOD (ng/L)	Reference
1	PFHxA, PFHpA, PFOA, PFDoA	Groundwater	Methyl iodide, Diazomethane	SPE	200 mL	GC-ECNI-MS	35-90	18-36 µg/L	[84]
2	PFBA, PFPeA, PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Wastewater, Seawater	Tetrabutylammonium, Butanol, Thionyl chloride	IP-SPME	5 mL	GC–NCI-MS	35-90	20-750	[85]
3	TFA, PFPrA, PFBA, PFPeA, PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Surface water, Lake water, Sewage WTP, Precipitation	2,4-difluoroaniline and N, N-dicyclohexylcarbodiimide	SPE	300 mL	GC-EI-MS	25-137	0.01-0.5	[86]
4	PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	River water	Isobutyl chloroformate, Pyridine, Isobutanol	LLE	500 mL	GC-EI-MS	N/A	200-2200 µg/L	[87]
5	PFOA, PFNA, PFDA, PFUnA, PFDoA	Surface water, Precipitation	2,4-difluoroaniline and DCC	IP-LLE	500 mL	GC-NCI-MS	91-98	0.3-5.9	[89]

Sl No.	Target PFCAs	Matrix/ Sources	Derivatizing agent	Extraction Method	Sample Volume	Instrumentation	Recovery %	LOD (ng/L)	Reference
6	PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	River water	Propyl chloroformate, Propanol	HS-SPME	10 mL	GC–QqQ–MS/MS	84.4-116.8	0.08-6.6	[91]
7	PFPeA, PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA	River Water	Isobutyl chloroformate, Pyridine, Isobutanol	SPE	250 mL	GC-NCI-MS	53-111	0.1-24	[92]
8	PFHpA, PFOA, PFNA, PFDA	Surface water	Tetrabutylammonium hydrogen sulfate	IP-DLLME	10 mL	GC-NCI-MS/MS	95-98	37-51	[93]
9	PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA	Drinking water/ Wastewater	Isobutyl chloroformate, DCC in Pyridine, Isobutanol	SPE	250 mL	GC-DSQ II-MS	94-98	0.1-0.5	[94]
10	PFPrA, PFBA, PFPeA, PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Wastewater	Triethylsilanol, H_2SO_4	SPE	250 mL	GC-EI-MS	93-108	4 - 48	[97]

Sl No.	Target PFCAs	Matrix/ Sources	Derivatizing agent	Extraction Method	Sample Volume	Instrumentation	Recovery %	LOD (ng/L)	Reference
11	PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA, PFTeA	River water, Lake water	Isobutyl chloroformate, Pyridine, Isobutanol	DLLME	1 mL	GC-EI-MS	83.7-117	0.9-3	98
12	PFBA, PFPeA, PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Surface water	2,4-difluoroaniline and DCC	SPE	500 mL	GC-μECD	62-118	1140–6320	99
13	PFHxA, PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA	Tap water	2,3,4,5,6-pentafluorobenzyl bromide	SPE	500 mL	GC-EI-MS	40.1-101.8	0.1-.28	100
14	TFA, PFPrA, PFBA, PFPeA, PFHpA, PFHxA, PFOA, PFNA, PFDA, PFUnA, PFDoA, PFTrDA, PFTeDA	Tap water, Precipitation, Ocean water	Diphenyl diazomethane	SPE	250 mL	GC-ECNI-MS	83-130	0.06-14.6	202

Sl No.	Target PFCAs	Matrix/ Sources	Derivatizing agent	Extraction Method	Sample Volume	Instrumentation	Recovery %	LOD (ng/L)	Reference
15	**PFHpA, PFOA, PFNA, PFDA, PFUnA, PFDoA, PFTeDA, PFHxDA, PFODA**	**Wastewater, Tap water**	**Isobutyl chloroformate, Isobutanol, Pyridine,**	**SBSE**	**20 mL**	**GC-EI-MS**	**47-97**	**21.17-73.96**	**This work**

3.4.4 Method Repeatability

The method repeatability has been rigorously evaluated through a series of tests at two spiked concentration levels, 100 ng/L and 1000 ng/L, with seven replicates for each level. The resultant data **(Table 3.4)** demonstrates the method's robust precision, with the percentage relative standard deviation (%RSD) below 14% for all target compounds, regardless of the concentration level. These results are consistently replicated in tap water analyses, with PFNA displaying a notably low %RSD of 7.2% at the 1000 ng/L concentration. This data emphasizes the method's reliable performance and its high repeatability which highlights its suitability for accurate and consistent monitoring of PFCAs.

Table 3.4: Method repeatability test in DI water and Tap water at two spiking levels, 100 ng/L and 1000 ng/L; (n=7).

Analyte	Repeatability (RSD %; n = 7)			
	DI Water		Tap Water	
	100 ng/L	1000 ng/L	100 ng/L	1000 ng/L
PFHpA	8.6%	6.5%	13.1%	9.7%
PFOA	13.3%	10.3%	9.4%	8.6%
PFNA	10.0%	9.5%	11.4%	7.2%
PFDA	6.2%	8.2%	12.3%	6.2%
PFUnA	12.5%	10.2%	9.6%	12.5%
PFDoA	13.4%	12.1%	7.8%	12.0%
PFTeDA	12.9%	8.3%	8.9%	10.2%
PFHxDA	11.6%	12.7%	12.0%	9.0%
PFODA	6.7%	9.6%	12.8%	9.4%

3.4.5 Spiked recovery experiment

In our study, we thoroughly assessed the recovery efficiency of our newly developed SBSE-TD-GC-MS method for PFCAs analysis across various water matrices. This was accomplished by spiking two levels (10 ng and 20 ng) in 20 mL of tap water, wastewater influent, and wastewater effluent samples. The recovery percentages, along with their respective standard deviations, were calculated based on duplicate measurements (n=2) for each condition. In tap water, recovery rates were fairly high, ranging from 68% (±8) to 90% (±10) at the 10 ng level, and improved at the 20 ng level with rates from 61% (±1) to 96% (±6) **(Table 3.5)**. This indicates a degree of matrix-related enhancement in recovery at the higher spiking level. On the other hand, the recovery rates in wastewater influent were significantly lower, with values from 29% (±12) to 44% (±11) at the 10 ng level, and only a slight increase at the 20 ng level, achieving between 21% (±7) and 31% (±10). This indicates the substantial matrix effect likely due to the presence of interfering substances. Wastewater effluent displayed intermediate recovery rates. The 10 ng spikes yielded 43% (±6) to 65% (±14) recoveries and the 20 ng spikes showing 44% (±11) to 74% (±8) recoveries. These findings further confirm the impact of matrix complexity on the extraction efficiency of PFCAs. The effluent samples showed a lesser degree of interference compared to influent samples, likely due to the partial removal of contaminants during the wastewater treatment process. To address the matrix effect encountered, particularly in the more complex wastewater samples, the standard addition method could be considered as a viable solution. By adding known quantities of PFCAs directly to the samples, this method compensates for the matrix-induced deviations. This allows for more accurate quantification of the analytes.

Table 3.5: Results of spiked recoveries of target PFCAs in tap water, wastewater influent, and wastewater effluent samples at two spiking levels (10 ng and 20 ng). Results shown in % recovery (±SD); (n=2).

	Tap water		Wastewater Influent		Wastewater Effluent	
Analyte	**10 ng**	**20 ng**	**10 ng**	**20 ng**	**10 ng**	**20 ng**
PFHpA	68 (±8)	81 (±7)	29 (±12)	27 (±9)	45 (±8)	64 (±11)
PFOA	74 (±5)	77 (±13)	31 (±7)	26 (±8)	46 (±7)	53 (±3)
PFNA	75 (±6)	73 (±10)	35 (±4)	21 (±7)	59 (±12)	44 (±11)
PFDA	90 (±10)	87 (±12)	31 (±9)	23 (±6)	56 (±6)	49 (±10)
PFUnA	69 (±3)	61 (±1)	29 (±9)	29 (±10)	43 (±6)	45 (±11)
PFDoA	82 (±13)	93 (±7)	32 (±6)	27 (±9)	56 (±11)	65 (±10)
PFTeDA	85 (±15)	96 (±6)	41 (±10)	30 (±6)	53 (±4)	74 (±8)
PFHxDA	84 (±8)	93 (±1)	44 (±11)	31 (±10)	57 (±11)	62 (±6)
PFODA	82 (±8)	92 (±6)	42(±2)	24 (±10)	65 (±14)	61 (±10)

3.4.6 Determination of PFCAs in real water samples

In our deployment of the novel SBSE-TD-GC-MS methodology, comprehensive testing was conducted on various water matrices, including tap water and both influent and effluent samples from four distinct wastewater treatment facilities. Despite the method's enhanced sensitivity and specificity for PFCAs detection, none of the analytes were identified above the method's quantification limit in the real water samples examined. This absence of detectable PFCAs may reflect the efficiency of the wastewater treatment processes, which seem to be effective in removing these compounds or reducing their concentrations to sub-threshold levels. It's also important to consider the potential influence of matrix effects, which could lead to signal suppression or enhancement, affecting the detection of PFCAs in complex matrices like wastewater. However, due to the rigorous method validation, including standard addition strategies to counter matrix effects, the absence of detectable PFCAs in the tested samples likely reflects

their true scarcity or absence in the sampled environments rather than an artifact of analytical interference.

3.4.7 Method Strengths and Limitations

The SBSE-TD-GC-MS method for PFCAs analysis presents a novel approach that integrates the principles of green chemistry and marks a significant advancement in environmental analytical methodologies. Its strength lies in minimizing environmental impact by drastically reducing the reliance on organic solvents traditionally required for extraction processes. This not only aligns with green chemistry goals but also decreases the potential for introducing solvent-related contamination. Another considerable strength of the method is its ability to effectively analyze PFCAs without the need for a clean-up and pre-concentration step. This method requires only 20 mL of water sample. This feature is particularly advantageous for field studies where sample volume is often limited or in situations where the transportation of large volumes of samples is impractical. This aspect of the method greatly enhances its application in remote or logistically challenging environments, where carrying out extensive sampling may not be feasible. Furthermore, this method is the first to use the SBSE technique to extract PFCAs from water without using solvents for desorption prior GC-MS analysis as thermal desorption unit is coupled with GC-MS. Solventless desorption simplifies the process, cuts down the steps, and lowers the chance of errors or contamination.

However, despite these strengths, the method does have its limitations. The matrix effects observed in wastewater samples indicate that complex sample matrices can influence the recovery and detection of PFCAs. Although the standard addition method and matrix-matched calibrations can be employed to counter these effects, they do add complexity and may increase the time

required for sample preparation and analysis. Overall, the SBSE-TD-GC-MS method represents a significant step forward for the efficient analysis of PFCAs in water samples.

3.5 CONCLUSIONS

The research successfully developed and validated a sophisticated SBSE-TD-GC-MS analytical method, characterized by simplicity, sensitivity, and robustness, for the extraction and quantification of PFCAs in aquatic environments. Through rigorous testing, the method demonstrated exemplary linearity across a concentration range of 75–2000 ng/L, repeatability with a %RSD within 14%, and a consistent recovery rate of 47-97%. The LODs ranged from 21.2 to 74.0 ng/L. These attributes emphasize the method's reliability as an alternative to the conventional methods for PFCAs analysis. Despite its heightened sensitivity and specificity, it is notable that none of the targeted PFCAs were identified in the real water samples analyzed. The environmental sustainability of this method is evident in its minimal requirement for sample volume (20 mL) and organic solvent (1 mL of methanol), marking it as a green analytical technique. This study also sets a precedent by being the first to report the use of SBSE without necessitating organic solvent for desorption. It surpasses traditional methods such as SPE, LLE, and SPME in terms of efficiency and cost-effectiveness, and its compatibility with commercially available stir bars makes it more user-friendly than previous SBSE methods associated with LC-MS/MS.

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Chapter 4: Green Analytical Method for Determination of Perfluorocarboxylic Acids (PFCAs) in Water by Stir Bar Sorptive Extraction Coupled with GC-MS

4.1 ABSTRACT:

Fluorotelomer Alcohols (FTOHs) and Perfluorocarboxylic Acids (PFCAs) are major group members of the Per- and Polyfluoroalkyl substances (PFAS) family. They are known for their versatile industrial and consumer applications and potential environmental and health risks. This study presents a comprehensive exploration into the presence and transport behavior of FTOHs and PFCAs in biosolid samples collected from wastewater treatment plants (WWTPs) in El Paso, Texas. We optimized an ultrasonic extraction method for efficient recovery of FTOHs and PFCAs compounds from biosolids, subsequently Stir Bar Sorptive Extraction (SBSE) and Thermal Desorption-Gas Chromatography-Mass Spectrometry (TD-GC-MS) analysis. The results revealed specific concentrations of FTOHs compounds in biosolid samples from the different WWTPs. 6:2 FTOH was not detected in any of the samples, while 8:2 FTOH was found in three WWTPs at varying concentrations: 100.30 ng/g in WWTP-1, 62.87 ng/g in WWTP-2, and 56.41 ng/g in WWTP-4. Additionally, 10:2 FTOH was detected in WWTP-1 at a concentration of 68.07 ng/g. Interestingly, despite the sensitive analytical approach employed, none of the targeted PFCAs were detected in any of the biosolid samples. These findings provide important insights into the distribution and prevalence of specific FTOHs in biosolids from WWTPs, that highlight the inherent variability in their occurrence. Since FTOHs compounds are potential precursors to PFCAs, this study contributes to the ongoing endeavor of monitoring and managing PFAS in water matrices.

Keywords: Fluorotelomer alcohol (FTOHs), Perfluorocarboxylic Acids (PFCAs), Stir bar sorptive extraction (SBSE), Method Development, GC-MS, Biosolids.

4.2 INTRODUCTION

Per- and Polyfluoroalkyl substances (PFAS) represent a vast family of over 4700 highly fluorinated compounds renowned for their remarkable properties, including water and oil repellency, thermal stability, resistance to biochemical degradation, bioaccumulative tendencies, and toxicological effects [136,203,204]. These versatile chemicals find their way into everyday consumer products like nonstick cookware, clothing, leather goods, upholstery, carpets, and more [102,203,205]. PFAS also play a crucial role in industrial applications, serving as wetting agents, additives, coatings, emulsifiers, paints, waxes, and polishes, while their presence in fire-fighting foams underscores their importance in safety applications [193,206–209]. These distinctive properties arise from their unique molecular structure, featuring a perfluorinated carbon chain that is both hydrophobic and oleophobic, coupled with hydrophilic charged functional groups for example carboxylic or sulfonic acid [189,203]. Fluorotelomer alcohols (FTOHs) and perfluorocarboxylic acids (PFCAs) are two major groups of PFAS with various uses, such as in cleaning products, fire-fighting foam, and making materials like fluoropolymers [102,189,193,204,210,211]. These compounds have become common pollutants in water, air, soil, food, and living organisms [91,96,136,212,213]. Studies have also shown that FTOHs can break down into other persistent, bioaccumulative, and toxic perfluorinated compounds, especially perfluorocarboxylic acids (PFCA) in water by various biotransformation mechanisms [7,214]. Therefore, FTOHs could be considered an indirect source of PFCAs in the environment. Furthermore, exposure to high levels of FTOHs and PFCAs has been linked to health problems, including reproductive issues, liver and kidney damage, immune system effects, and thyroid disruptions [215–218].

FTOHs and PFCAs were found in various water sources, including surface waters, underground aquifers, oceans, tap water, bottled water, and wastewater treatment plants (WWTPs)

[183,184,210,219–222]. In WWTPs, sewage sludge is a significant reservoir of PFCAs and their precursors [223–225]. Biosolids, which are the nutrient-rich solid residues derived from municipal sewage sludge, are often used as organic-rich fertilizers in agriculture due to their nutrient content. Land application is the favored method for biosolid use and disposal, returning nutrients and organic matter to the land at minimal costs [218,226–230]. However, the use and disposal of wastewater biosolids are not currently regulated for PFAS, making land application a potential major pathway for PFAS release into the environment. Additionally, laboratory studies have provided strong evidence for microbial biodegradation processes that transform FTOHs into PFCAs [230–232]. In particular, the biotransformation of 6:2 FTOH into perfluorohexanoic acid (PFHxA) and perfluoropentanoic acid (PFPeA) has been well-documented in diluted activated sludge [232]. Several research studies have investigated the levels of FTOHs and PFCAs in biosolids. For instance, Yoo research group [231] conducted an analysis of FTOHs concentrations in biosolid-amended soils, revealing elevated levels of 8:2 FTOH (ranging from 5 to 73 ng/g) and 10:2 FTOH (below 5.6 to 166 ng/g) in these soils. Moreover, another research group by Wang, [230] reported that FTOHs can undergo aerobic biotransformation in both soil and activated sludge, leading to the formation of PFHxA and PFOA. Additionally, Washington et al.,[225] observed the accumulation of perfluoroalkyl acids (PFAAs) and FTOHs in soils resulting from the land use of biosolids originating from industrial sources. These findings highlight the importance of monitoring and studying FTOHs and PFCAs in biosolids, given their potential presence at trace levels and their potential for transport into the environment through land application. Consequently, the development of a straightforward, sensitive, and robust analytical method is crucial for the accurate identification and quantification of FTOHs and PFCAs in biosolid matrices.

In analyzing FTOHs and PFCAs in biosolid samples, researchers commonly utilize both liquid chromatography (LC) and gas chromatography (GC) methods for qualitative and quantitative determination of these compounds [216,219,225,233–235]. Both methods require robust pre-treatment processes due to the need to detect these compounds at ultra-trace levels. Solid-phase extraction (SPE) is mostly used for the extraction process for these compounds [118,119,122]. But, in recent years, microextraction techniques have emerged as a modern and efficient alternative to traditional extraction methods like liquid-liquid extraction (LLE) and solid-phase extraction (SPE) [202,236]. Stir Bar Sorptive Extraction (SBSE) has gained prominence in this context, primarily due to its environmentally friendly approach and cost-effectiveness [197,237–239]. SBSE excels in handling large sample volumes with minimal labor involvement and aligns well with the principles of green chemistry. Notably, SBSE offers a higher absorbent volume compared to Solid-Phase Microextraction (SPME), which significantly enhances sensitivity, enabling the detection of compounds at sub-ng/L concentrations [196,239]. This technique's robust adsorption capacity and high extraction efficiency make it particularly suitable for the extraction of FTOHs and PFCAs from biosolid samples. Yet, this SBSE technique in line with thermal desorption-gas chromatography-mass spectrometry (TD-GC-MS) is a relatively unexplored area for PFCAs and FTOHs quantification.

This study aimed to investigate the presence of FTOHs and PFCAs in municipal wastewater treatment plant (WWTP) biosolid samples. To achieve this, we incorporated an ultrasonic pre-extraction method for the efficient extraction of these target compounds from biosolid samples. Subsequently, the extracts were subjected to our previously established Stir Bar Sorptive Extraction (SBSE) techniques for quantifying FTOHs and PFCAs in water samples using TD-GC-MS. The developed method was applied to analyze FTOHs and PFCAs in biosolid

samples, representing a pioneering use of SBSE for efficient extraction from this matrix based on existing literature. To quantify these target compounds in biosolid samples, a solvent-free desorption step was conducted using a thermal desorption unit before GC-MS analysis. This research contributes to an improved understanding of the transport and release of FTOHs and PFCAs from municipal WWTPs into the environment.

4.3 EXPERIMENTAL

4.3.1 Standards and reagents

The analytical standard of PFCAs and FTOHs, as listed in **Table 4.1**, was purchased from Fisher Scientific, USA. Standard stock solutions of 1000 µg/mL of FTOHs were prepared in acetonitrile. HPLC-grade acetonitrile, hexane, and methanol were purchased from J.T.Baker®, USA. Mirex (Fisher Scientific, USA) was used as the internal standard, and 1000 µg/mL and 10 µg/mL Mirex stock solutions were prepared in acetonitrile. Deionized (DI) water, purchased from J.T.Baker®, USA, was used in dilutions and sample preparations.

Table 4.1: List of the targeted PFCAs and FTOHs.

	Compound Name	Acronym	Molecular Formula	Molecular Weight	CAS No.
1	Perfluoroheptanoic acid	**PFHpA**	$C_7HF_{13}O_2$	364.06	375-85-9
2	Perfluorooctanoic acid	**PFOA**	$C_8HF_{15}O_2$	414.07	335-67-1
3	Perfluorononanoic acid	**PFNA**	$C_9HF_{17}O_2$	464.08	375-95-1
4	Perfluorodecanoic acid	**PFDA**	$C_{10}HF_{19}O_2$	514.08	335-76-2
5	Perfluoroundecanoic acid	**PFUnA**	$C_{11}HF_{21}O_2$	564.09	2058-94-8
6	Perfluorododecanoic acid	**PFDoA**	$C_{12}HF_{23}O_2$	614.10	307-55-1
7	Perfluorotetradecanoic acid	**PFTeDA**	$C_{14}HF_{27}O_2$	714.11	376-06-7
8	Perfluorohexadecanoic acid	**PFHxDA**	$C_{16}HF_{31}O_2$	814.13	67905-19-5
9	Perfluorooctadecanoic acid	**PFODA**	$C_{18}HF_{35}O_2$	914.10	16517-11-6
10	1H,1H,2H,2H-Perfluorooctan-1-ol	**6:2 FTOH**	$C_8H_5F_{13}O$	364.10	647-42-7

11	1H,1H,2H,2H-Perfluorodecan-1-ol	**8:2 FTOH**	$C_{10}H_5F_{17}O$	464.12	678-39-7
12	1H,1H,2H,2H-Perfluorododecan-1-ol	**10:2 FTOH**	$C_{12}H_5F_{21}O$	564.13	865-86-1

4.3.2 Sample Collection

In this study, biosolid samples from wastewater treatment plants were obtained from El Paso Water in El Paso, Texas, USA, in September 2023. Biosolid samples were taken from four municipal wastewater treatment plants, denoted as WWTP-1, WWTP-2, WWTP-3, and WWTP-4. All the wastewater treatment plants are located in El Paso, Texas, USA. The samples were collected in 500 mL polypropylene bottles, ensuring no headspace remained, and were then promptly stored at 4 °C. All samples were analyzed for FTOHs and PFCAs within 7 days from their collection date.

4.3.3 Sample preparation:

4.3.3.1 Ultrasonic extraction

Initially, the biosolid samples underwent a deionized water wash to remove any residual wastewater. This washing process was carried out through vacuum filtration. Then, the biosolid samples were left to air dry. After that, the dry biosolid samples were ground using mortar and pestle. For pre-extraction method development, 1 gram of dry biosolid sample was measured and placed into a 15-milliliter polypropylene (PP) tube. Subsequently, 500 µL of 1 µg/mL mixture containing three target FTOHs standards were introduced into the tube. The PP tube was vortexed for one minute to ensure a thorough and uniform blending of the standards with the biosolid. Following this, the samples were allowed to rest overnight in a refrigerator at 4 °C. Afterward, 10 mL of extraction solvent was added, and the mixture was once again vortexed for one minute. Various solvents were tested to assess their effects, including methanol (MeOH), acetonitrile (ACN), acetone (ACE), a 1:1 (v/v) mixture of methanol and acetonitrile (MeOH/ACN), a 1:1

mixture of methanol and acetone (MeOH/ACE), and a 1:1 mixture of acetonitrile and acetone (ACN/ACE). Ultrasonic extraction was performed using a Fisher Scientific FS 30H ultrasonic bath. The vial containing the 10 mL of organic solvent with the biosolid sample was subjected to ultrasonication for varying durations, including 30 minutes, 60 minutes, and 90 minutes, to optimize the extraction process. Following ultrasonic extraction, centrifugation was carried out at 4000 rpm for 20 minutes and the temperature was set at 4 °C using the Sorvall Legend X1R centrifuge from Thermo Scientific, USA. Afterward, the liquid supernatant was transferred into a new 15 mL PP tube for use in the subsequent SBSE process. **Figure 4.1** provides a detailed workflow of this pre-extraction procedure.

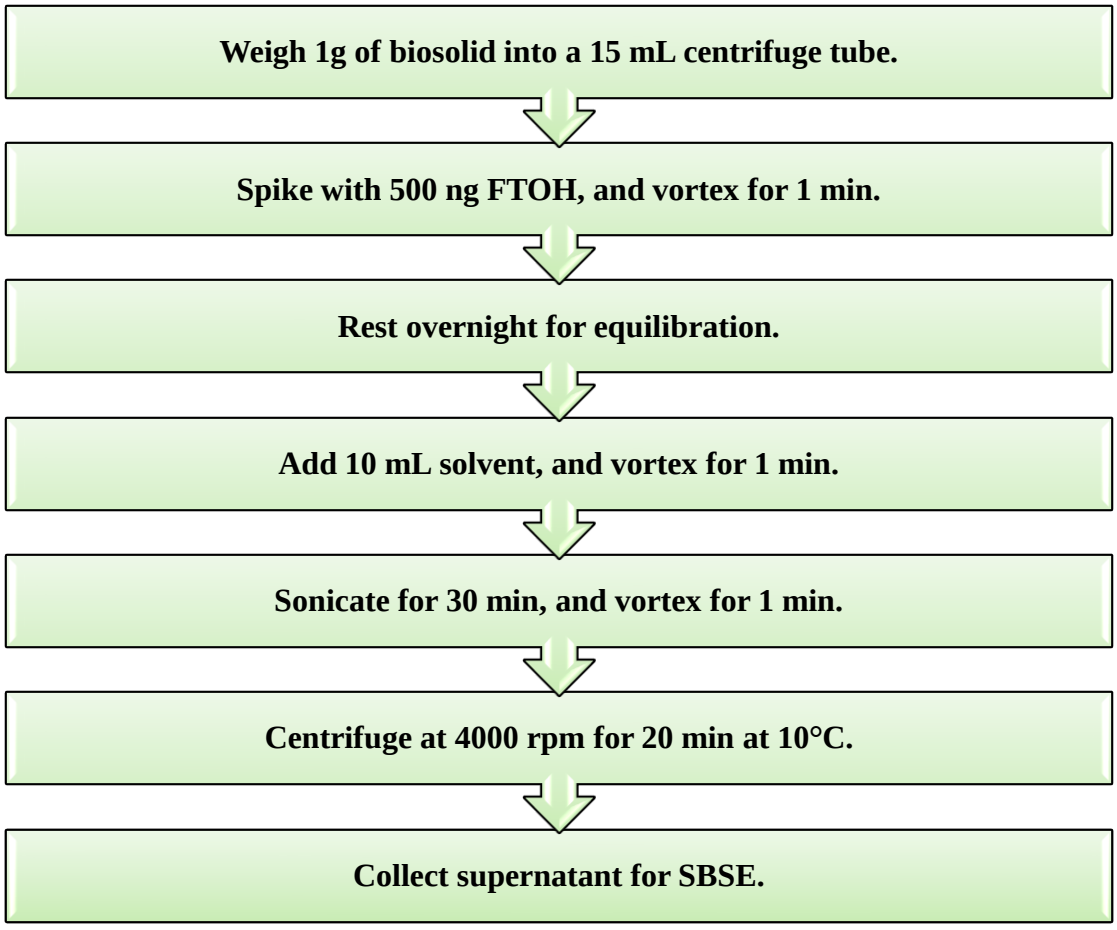

Figure 4.1: Workflow for the ultrasonic extraction from biosolid samples. FTOHs are used as the model PFAS group.

4.3.3.2 Stir Bar Sorptive Extraction (SBSE)

We employed our previously established Stir Bar Sorptive Extraction (SBSE) technique as a preliminary step for extraction and concentration before conducting Thermal Desorption-Gas Chromatography-Mass Spectrometry (TD-GC-MS) analysis. To outline the procedure briefly, we initiated the process in a 20 mL amber vial, where 3 mL of the prior extract was combined with 17 mL of deionized (D.I.) water. Additionally, 20 µL of Mirex, at a concentration of 10 µg/mL, was introduced as the internal standard. We then inserted a commercially available stir bar coated with polydimethylsiloxane (Twister™, 10 mm × 1 mm, Gerstel, USA) into the vial. This solution was subsequently stirred following the previously established SBSE method for FTOHs and PFCAs, as described in Chapter 3 and Chapter 4 of the experimental section. After stirring, the stir bar was carefully removed, rinsed with D.I. water to eliminate any residual sample, and dried using a lint-free tissue. Subsequently, the stir bar was placed into a thermal desorption tube (TDT) for the final TD-GC-MS analysis.

4.3.4 Thermal desorption-gas chromatography-mass spectrometry (TD-GC- MS) analysis

The TD-GC-MS analysis was conducted following our previously established method specifically for FTOHs (detailed in Chapter 3) and PFCAs (as outlined in Chapter 4). A summary of both methods is represented in **Table 4.2**.

Table 4.2: TD-GC-MS method parameters for FTOHs and PFCAs analysis.

Method Parameter	**FTOHs Analysis**	**PFCAs Analysis**
Instrumentation	TDU (Gerstel, MD, USA), 8890-GC, 5977B MS (Agilent Technologies, CA, USA)	
GC-MS Column	HP-5MS UI column (30 m × 0.25 mm i.d., 0.25 µm film thickness, Agilent, CA, USA)	
Carrier Gas	Ultra-high purity helium, 1.2 mL/min	

Method Parameter	FTOHs Analysis	PFCAs Analysis
Thermal Desorption Program	Initial Temperature: 50°C for 0.5 min; Temperature Ramp: Increase to 280°C at 60°C/min, Hold at 280°C for 7.0 min	Initial Temperature: 40°C for 0.5 min; Temperature Ramp: Increase to 280°C at 60°C/min, Hold at 280°C for 5.0 min
TD Transfer Line Temp.	290°C	290°C
Cold Injection System (CIS)	CIS4/TDU baffled liner (Gerstel, USA) at -40°C during desorption, then heated to 300°C at 12°C/sec and held for 5 min in splitless mode	CIS4/TDU baffled liner (Gerstel, USA) at -40°C during desorption, then heated to 300°C at 12°C/sec and held for 5 min in splitless mode
GC Oven Temp. Program	Initial Temperature: 35°C for 3 min, Temperature Ramp: Increase at 5°C/min to 100°C, hold for 2 min, Temperature Ramp: Increase at 25°C/min to 300°C, hold for 3 min, Total Run Time: 29 min	Initial Temperature: 40°C for 2 min, Temperature Ramp: Increase at 10°C/min to 200°C, hold for 2 min, Temperature Ramp: Increase to 300°C at 25°C/min, hold for 3 min, Total Run Time: 27 min
MSD Transfer Line Temp.	280°C	280°C
Ionization Energy	Electron Ionization (EI) at 70 eV	Electron Ionization (EI) at 70 eV
Solvent Delay	3 min	1 min
Mass Spectrometry Mode	Selected Ion Monitoring (SIM)	Selected Ion Monitoring (SIM)
SIM Ion (m/z)	69, 95, 131, 169, 181, 272	69, 131, 169, 181, 272

4.3.5 Analysis of FTOHs and PFCAs in biosolid samples

For the determination of FTOHs and PFCAs in biosolid samples, an optimized pre-extraction method was carried out before SBSE-TD-GC-MS analysis. Initially, the biosolid samples underwent a deionized water wash to remove any residual wastewater. This washing process was carried out through vacuum filtration. Then, the biosolid samples were left to air dry. After that, the dry biosolid samples were ground using mortar and pestle. For pre-extraction, 500

mg of dry biosolid sample was measured and placed into a 15-milliliter polypropylene (PP) tube. Then, 10 mL acetonitrile as extraction solvent was added, and the mixture was vortexed for one minute. Ultrasonic extraction was performed for 30 min. Following ultrasonic extraction, centrifugation was carried out at 4000 rpm for 20 minutes and 4 °C using the Sorvall Legend X1R centrifuge from Thermo Scientific, USA. Afterward, the liquid supernatant was transferred into a new 15 mL PP tube for use in the subsequent SBSE process for FTOHs and PFCAs analysis. Three mL of extract were used for FTOHs determination as described in Chapter 3; while 1 mL extract was used for derivatization and extraction of PFCA by SBSE applying the developed method outlined in Chapter experimental section.

4.4 RESULTS AND DISCUSSIONS

4.4.1 Optimization of Pre-treatment of Biosolids by Ultrasonic Extraction

4.4.1.1 Extraction Solvent Optimization

We conducted ultrasonic extraction to extract FTOHs and PFCAs from biosolid samples. In the process of optimizing the ultrasonic method, we focused on the influence of different solvents used during the extraction procedures. The optimization was guided by the outcomes derived from the extraction recovery data of the three studied FTOHs compounds. Various solvents, including methanol, acetonitrile, acetone, and acetonitrile:acetone 1:1 (v/v), were assessed. The extraction efficiency for 6:2 FTOH was not significantly different ($P < 0.05$) when using either acetonitrile or a 1:1 (v/v) mixture of acetonitrile and acetone **(Figure 4.2)**. However, when it came to 8:2 FTOH and 10:2 FTOH, acetonitrile consistently produced significantly higher ($P < 0.05$) response compared to the other solvent compositions tested. This finding aligns with a study by Chen research group [211] who also reported optimal extraction recovery results when using acetonitrile for extracting FTOHs from wastewater sludge samples. As a result of these

investigations, we selected acetonitrile as the preferred extraction solvent during ultrasonication for extracting FTOHs and PFCAs from biosolid samples.

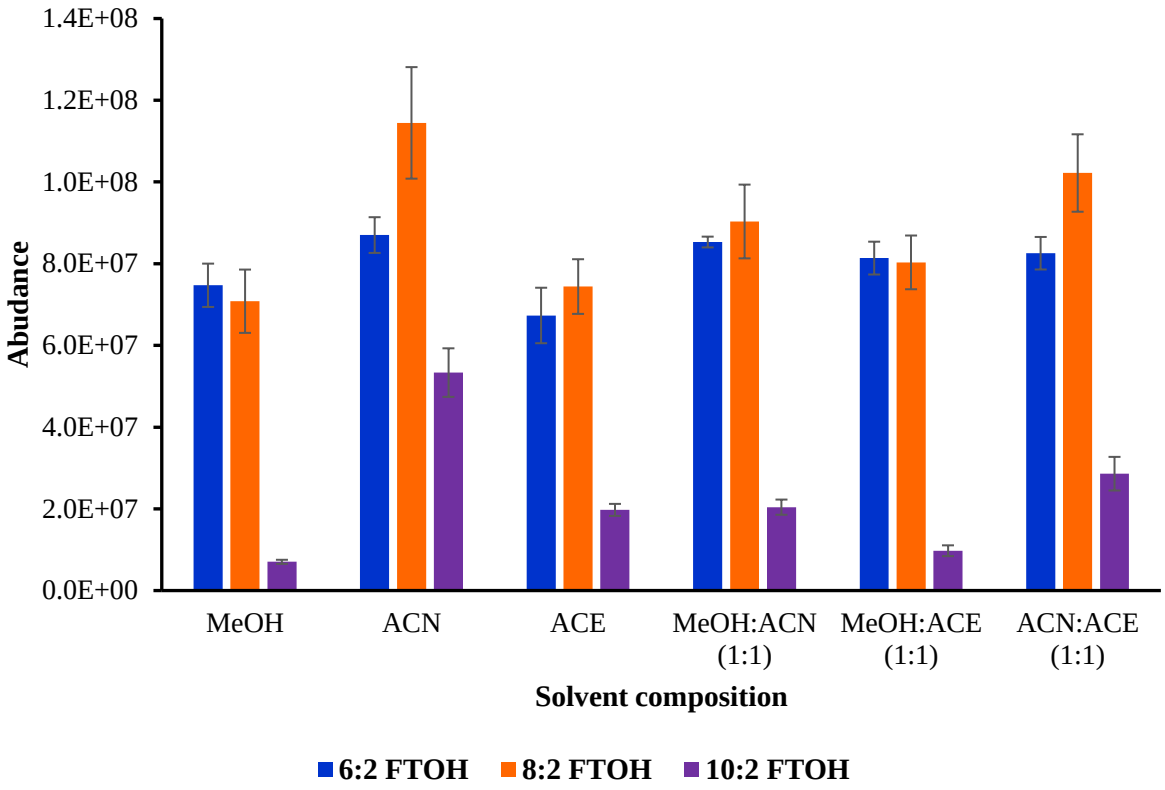

Figure 4.2: Optimization of extraction solvent: instrument response (i.e., extraction recovery) of FTOHs with various extraction solvent compositions. Standard deviations of duplicate measurements are represented as error bars in this bar chart. Here, MeOH- methanol, ACN- acetonitrile, and ACE- acetone.

4.4.1.2 Sonication Time Optimization

Two solvent systems: acetonitrile and a 1:1 (v/v) mixture of acetonitrile and acetone (ACN:ACE, 1:1) were used in this optimization. To optimize the sonication time, we considered three different durations for the extraction process: 30 minutes, 60 minutes, and 90 minutes. **Figure 4.3** displays the results of the extraction recovery of FTOHs at these three sonication time intervals. For 8:2 FTOH, consistent response levels were observed when using acetonitrile across all three sonication time points. In the case of 6:2 FTOH, a significantly higher ($P < 0.05$) response was

noted at the 60-minute sonication time for both solvent systems. Meanwhile, 10:2 FTOH exhibited significantly higher ($P < 0.05$) response at the 30-minute sonication time. Overall, the 30-minute sonication time provided a comparable response for all three target compounds and offered a time-saving advantage for ultrasonic extraction. Therefore, we selected a 30-minute sonication period with acetonitrile as the solvent for ultrasonic extraction for the subsequent experiment.

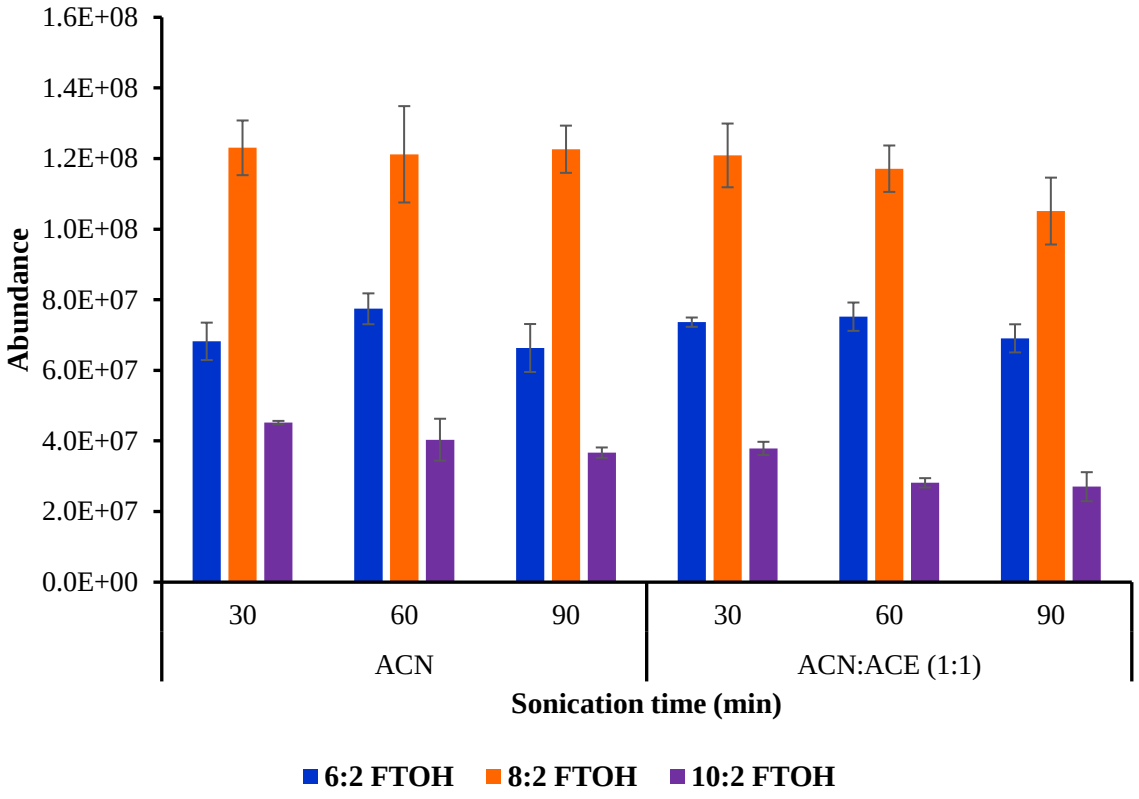

Figure 4.3: Optimization of sonication time: Instrument response (i.e., extraction recovery) of FTOHs with various sonication times. Error bars in this bar chart represent the standard deviations of duplicate measurements. Here, ACN- acetonitrile and ACE- acetone.

4.4.1.3 Effect of Sample Load on Ultrasonic Extraction

The influence of sample load was assessed at three different amounts: 100 mg, 500 mg, and 1000 mg of biosolids. The results of the extraction recovery are presented in **Figure 4.4** as a percentage recovery. From **Figure 4.4**, it became evident that both the 100 mg and 500 mg sample

loads exhibited higher extraction recovery rates compared to the 1000 mg biosolid sample load for all three target compounds. Among the three options, the 500 mg sample load demonstrated the most favorable performance in terms of extraction recovery for all three FTOHs compounds. It's noteworthy that Chen and colleagues [211] also used a 500 mg sample amount when analyzing FTOHs in wastewater sludge samples, as reported in the literature. Given the extraction recovery results, we have chosen 500 mg as the optimal sample quantity for the ultrasonic extraction process.

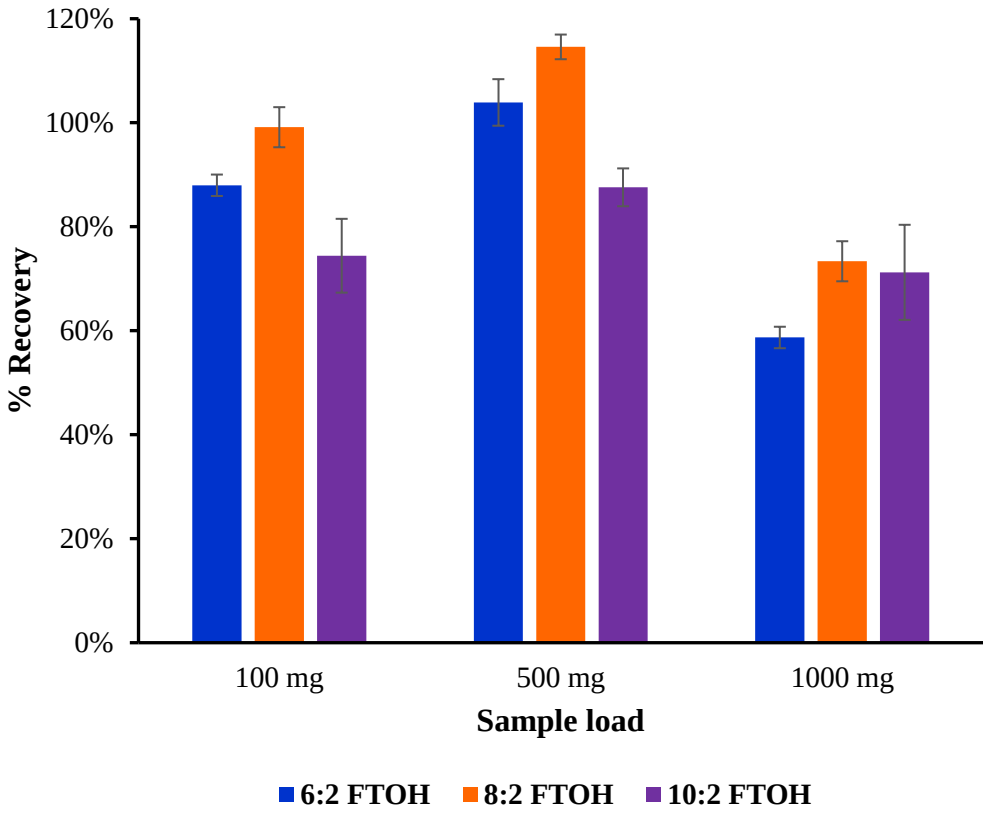

Figure 4.4: Effect of sample load on ultrasonic extraction: % recovery of FTOHs with 3 sample amounts: 100 mg, 500 mg, and 1000 mg. Error bars in this bar chart represent the standard deviations of duplicate measurements.

4.4.2 OCCURRENCE AND PROFILES OF FTOHS AND PFCAS IN BIOSOLID SAMPLES

The results presented in **Table 4.3** provide insightful information about the presence of FTOHs in biosolid samples from different wastewater treatment plants (WWTPs). Specifically, the analysis focused on three types of FTOHs: 6:2 FTOH, 8:2 FTOH, and 10:2 FTOH. It was observed that 6:2 FTOH was not detected in any of the samples across all four WWTPs, indicating its absence in these biosolid samples. In contrast, 8:2 FTOH was found in three of the four WWTP samples, with varying concentrations: 100.30 ng/g in WWTP-1, 62.87 ng/g in WWTP-2, and 56.41 ng/g in WWTP-4. These findings align with prior research conducted by two research group, Chen [211] and Ellington [225] who found 8:2 FTOH in sewage sludges from wastewater treatment plants. This variation suggests differing levels of this compound's presence in the wastewater treatment processed by each plant. Notably, 10:2 FTOH was detected in only one sample (WWTP-1) at a concentration of 68.07 ng/g, highlighting its relatively less frequent occurrence compared to 8:2 FTOH in biosolid samples. These findings suggest that the presence of these FTOHs compounds in biosolid samples varies among WWTPs, which supports the results from previous research outcomes [210,211,213]. These FTOHs are the source of PFCAs contamination in waster matrices as they bio-transform into PFCAs [227,230,240]. Also, the findings are significant as they provide a record of the distribution and prevalence of specific FTOHs in wastewater treatment plants.

Table 4.3: Results of determination of target FTOHs in biosolid samples; each sample ran in duplicates.

Biosolid Samples	Detected (ng/g)/ Not detected (N.D.)		
	6:2 FTOH	**8:2 FTOH**	**10:2 FTOH**
WWTP-1	N.D.	100.30	68.07
WWTP-2	N.D.	62.87	N.D.
WWTP-3	N.D.	N.D.	N.D.
WWTP-4	N.D.	56.41	N.D.

In addition to the determination of FTOHs compounds, our comprehensive investigation extended to the analysis of PFCAs within biosolid samples collected from the aforementioned WWTPs. Despite utilizing a highly sensitive method optimized for the extraction and detection of PFCAs, our findings showed the absence of any of the targeted PFCAs in the biosolid samples across all WWTP sources. This intriguing outcome indicates a difference between the presence of FTOHs compounds and PFCAs in biosolid samples.

4.5 CONCLUSIONS

This comprehensive study provided valuable insights into the occurrence and transport of the FTOHs and PFCAs in wastewater biosolids collected from WWTPs. Our analytical approach involved the optimization of ultrasonic extraction methods to extract FTOHs and PFCAs from biosolid samples efficiently prior to SBSE-TD-GC-MS analysis; those methods were developed in our previous study. The research found variations in the occurrence of specific FTOHs compounds across multiple WWTPs in El Paso, Texas, with 6:2 FTOH absent in all samples, while 8:2 FTOH exhibited variable concentrations in three of the four WWTPs. Importantly, the study also highlighted the complete absence of any of the targeted PFCAs in the biosolid samples despite employing a highly sensitive analytical approach. Overall, this research contributes valuable data to ongoing efforts to monitor and manage emerging contaminants in wastewater treatment systems.

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Chapter 5: Conclusions and Recommendations for Future Work

5.1 CONCLUSIONS

This dissertation aimed to investigate the occurrence and transport of two predominant groups of PFAS: FTOHs and PFCAs in wastewater and biosolids. In this study, we developed and validated a simple, low-cost, no clean-up, and sensitive method for the determination of FTOHs and PFCAs in wastewater and biosolids samples by applying 'green chemistry' based extraction, i.e., stir bar sorptive extraction (SBSE) coupled with thermal desorption-gas chromatography-mass spectrometry (TD-GC-MS). We applied this newly developed method to investigate FTOHs and PFCAs in four distinct wastewater treatment plants (WWTPs) within our region.

In this study, our developed SBSE-TD-GC-MS provides sensitive techniques for analyzing FTOHs in water. Using three FTOHs (6:2, 8:2, and 10:2) as model compounds, our method demonstrated low detection limits ranging from 2.16 ng/L to 16.7 ng/L, good linearity (25–500 ng/L), repeatability (%RSD below 10%), and recoveries (55%–111%). These characteristics establish the method as a dependable alternative for monitoring FTOHs in aquatic environments. By applying this optimized method, we detected two FTOHs, namely 6:2 FTOH and 8:2 FTOH, in two wastewater treatment plants at levels as low as parts per trillion (ppt). We also successfully developed another analytical method for PFCAs analysis using the SBSE-TD-GC-MS approach. This method allowed for the extraction and quantification of PFCAs in aqueous samples and demonstrated good linearity across a concentration range of 75–2000 ng/L, repeatability (%RSD within 14%), and recovery rates of 47-97%. The method's limits of detection (LODs) ranged from 21.2 to 74.0 ng/L for all targeted PFCAs considered during method development. These attributes emphasize the method's reliability as an alternative to conventional PFCAs analysis techniques. Our comprehensive study yielded valuable insights into the presence and distribution of FTOHs

and PFCAs in wastewater biosolids collected from WWTPs. We optimized ultrasonic extraction methods for efficiently extracting FTOHs and PFCAs from biosolid samples before SBSE-TD-GC-MS analysis. The study revealed variations in the occurrence of specific FTOH compounds across multiple WWTPs in El Paso, Texas, with 6:2 FTOH absent in all samples and 8:2 FTOH showing variable concentrations in three of the four WWTPs. Particularly, none of the targeted PFCAs were detected in the wastewater and biosolid samples despite the use of a highly sensitive analytical approach.

Overall, SBSE proved to be a sensitive and environmentally friendly analytical technique, requiring a small sample volume (20 mL) and minimal organic solvent (1 mL methanol). Its simplicity and compatibility with the sensitive GC-MS system enable the detection of FTOHs and PFCAs at low concentrations in water samples and surpassing traditional methods in terms of efficiency and cost-effectiveness. This study also sets a precedent by being the first to report the use of SBSE coupled with GC-MS for FTOHs and PFCAs determination in water matrices. Largely, this research provides valuable data contributing to ongoing efforts to monitor and manage emerging contaminants in wastewater treatment systems.

5.2 RECOMMENDATIONS FOR FUTURE WORK

In the field of environmental analysis, the detection and quantification of PFAS in water matrices are of utmost importance due to the persistent nature and potential risks of these substances. Future research directions in this area may include:

I. **Extended Monitoring and Sampling:** Expanding the monitoring efforts to include a wider geographical area such as WWTPs of these nearby cities (Juárez, Mexico and Las Cruces, New Mexico) could provide a more comprehensive understanding of the regional

and even national distribution of FTOHs and PFCAs. This broader data collection could help identify potential hotspots and trends in PFAS contamination.

II. **Diverse Field Application and Monitoring:** Implementing the developed method by conducting long-term monitoring campaigns at various water sources, such as lakes, rivers, and groundwater, can help track PFAS trends, sources, and seasonal variations.

III. **Incorporating More PFAS Variants:** Expanding the analysis to include a wider range of PFAS variants, beyond FTOHs and PFCAs, can provide insights into the presence and behavior of other emerging PFAS compounds.

IV. **Method Refinement:** Continuous refinement and optimization of the analytical method for PFAS detection in water matrices is essential. Researchers can explore variations in extraction techniques, chromatographic separation, and mass spectrometry detection to further improve sensitivity, accuracy, and efficiency.

V. **Community and Stakeholder Engagement:** Engaging with local communities and stakeholders to raise awareness about PFAS contamination in water sources, its potential health risks, and mitigation strategies is important.

In summary, this dissertation represents a significant step forward and contributes valuable data to the ongoing efforts to monitor and manage emerging contaminants in wastewater treatment systems. Through the development and validation of a cost-effective, environmentally friendly, and sensitive analytical method, we have provided a reliable alternative for monitoring these contaminants in aquatic matrices. This study offers a foundation for future studies in this field.

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Vita

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