

Structural dynamics and transcriptomic analysis of *Dehalococcoides mccartyi* within a TCE-Dechlorinating community in a completely mixed flow reactor

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CMFR

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PCE

Perchloroethene

SRA

Sequence read archive

SRT

Sludge retention time

TCE

Trichloroethene

TMM

Trimmed mean of M-values

ABSTRACT

A trichloroethene (TCE)-dechlorinating community (CANAS) maintained in a completely mixed flow reactor was established from a semi-batch enrichment culture (ANAS) and was monitored for 400 days at a low solids retention time (SRT) under electron acceptor limitation. Around 85% of TCE supplied to CANAS (0.13 mmol d^{-1}) was converted to ethene at a rate of 0.1 mmol d^{-1} , with detection of low production rates of vinyl chloride ($6.8 \times 10^{-3} \text{ mmol d}^{-1}$) and *cis*-dichloroethene ($2.3 \times 10^{-3} \text{ mmol d}^{-1}$). Two distinct *Dehalococcoides mccartyi* strains (ANAS1 and ANAS2) were stably maintained at $6.2 \pm 2.8 \times 10^8 \text{ cells mL}^{-1}$ and $5.8 \pm 1.2 \times 10^8 \text{ cells mL}^{-1}$, respectively. Electron balance analysis showed 107% electron recovery, in which 6.1% were involved in dechlorination. 16 S rRNA amplicon sequencing revealed a structural regime shift between ANAS and CANAS while maintaining robust TCE dechlorination due to similar relative abundances of *D. mccartyi* and functional redundancy among each functional guild supporting *D. mccartyi* activity. *D. mccartyi* transcriptomic analysis identified the genes encoding for ribosomal RNA and the reductive dehalogenases *tceA* and *vcrA* as the most expressed genes in CANAS, while *hup* and *vhu* were the most critical hydrogenases utilized by *D. mccartyi* in the community.

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1. Introduction

Perchloroethene (PCE) and trichloroethene (TCE) are common solvents detected in soils and groundwater due to historical improper storage, handling and disposal practices and accidental releases during industrial uses, such as dry cleaning and degreasing operations (Moran et al., 2007; McCarty, 2010). *In situ* bioremediation is a cost-effective and environmentally-friendly approach to

reach eco-toxicological safety endpoints at chlorinated ethene-contaminated sites (Stroo et al., 2010). Among all reported dehalogenating microorganisms (Maymo-Gatell et al., 1997; Holliger et al., 1998; Löffler et al., 2000; Adrian et al., 2000; Suyama et al., 2001; Sung et al., 2006; Hug et al., 2013; Dolinova et al., 2017), *Dehalococcoides mccartyi* is the only known species that metabolically reductively dechlorinates PCE and TCE to non-toxic ethene. Many studies on *D. mccartyi*-containing microbial communities have been carried out in the past decades (Carr et al., 2000; Cupples et al., 2004; Freeborn et al., 2005; Yu et al., 2005; Daprato et al., 2007; Duhamel et al., 2007; Ziv-El et al., 2012), with the identification of optimal growth conditions supporting *D. mccartyi* and functional robustness determinants of dechlorinating activity. However, the microbial community assembly processes and inter-species interaction networks that promote successful bioremediation are difficult to predict within the flow-dominated physico-chemical conditions occurring in the subsurface (Kinnunen et al., 2016).

Dechlorinating microbial communities have mainly been studied in batch reactors characterized by high initial concentrations of substrates, nutrients and electron acceptors and dynamic conditions as dechlorination proceeds. The knowledge acquired from such feast-and-famine laboratory incubations has been extremely useful for understanding the dynamics of dechlorinating communities but is difficult to extrapolate to flowing groundwater plumes which are continuous, oligotrophic fluidic ecosystems with site-specific hydraulic retention times and relatively constant and low concentrations of contaminants, substrates and nutrients (Stroo et al., 2010). Continuous-flow bioreactors generate conditions that are more environmentally and ecologically relevant to groundwater plumes than batch systems. In addition, continuous culture systems can be highly reproducible platforms for dynamic perturbation under rigorously defined and controlled conditions relevant to systems-level omics and electron flow studies.

Structural and functional dynamics of dechlorinating communities have been investigated in continuous-flow biofilm and packed column systems (Carr et al., 1998; Azizian et al. 2008, 2016; Chung et al., 2008; Maphosa et al., 2010; Sabalowsky et al., 2010; Mirza et al. 2016, 2017; Delgado et al., 2017). Continuous-flow suspended growth systems have also been used to study the impacts of solids retention times (SRTs) and other critical parameters on dechlorination activity of synthetic consortia and more complex communities (Gerritse et al., 1997; Carr et al., 2000; Drzyzga et al., 2001; Zheng et al., 2001; Sabalowsky et al., 2010). Yang and McCarty (1998) also reported stable production of ethene by a methanogenic PCE-dechlorinating enrichment culture maintained in a semi-continuous stirred-tank reactor fed with PCE and benzoate. These studies provided important information on aggregate properties of the system (e.g., functional endpoints, total biomass concentrations, process rates and optimization). What has yet to be reported are quantitative assessments of the dynamics of the dechlorinating populations (e.g., microdiversity and physiological responses of *D. mccartyi*) and functional guilds supporting them. Here, a functional guild is defined as a group of taxonomically-distinct microorganisms performing the same function (e.g., syntrophic, fermentative bacteria that catalyze the oxidation of lactate) (Wittebolle et al., 2009). Functional redundancy among a functional guild involves the presence of multiple functionally-equivalent taxa that enables functional robustness through compensation by metabolic equivalency and response asynchrony. In addition, several studies have reported successful and sustainable PCE/TCE dechlorination to ethene in completely mixed flow reactors (CMFRs, i.e. chemostat) with associated population dynamics (Table 1) (Berggren et al., 2013; Delgado et al., 2014; Marshall et al., 2014; Mayer-Blackwell et al. 2016, 2017).

In this study, a TCE-dechlorinating community (CANAS) sustained in a CMFR was established from a long-term dechlorinating enrichment culture (ANAS) which has been functionally maintained in a semi-batch reactor for more than 15 years (Richardson et al., 2002; Lee et al., 2006). The microbial community structure of ANAS and its responses to different environmental conditions have been well characterized (Freeborn et al., 2005; Lee et al., 2011; West et al., 2013). The main objective of this work is to study a continuous-flow mixed culture (CANAS) in the presence of sustained selective pressure (i.e., constant supply of chlorinated ethenes) and low concentrations of electron donor, as frequently observed in contaminated oligotrophic groundwater plumes (Stroo et al., 2010). 16 S rRNA gene amplicon sequencing was carried out to assess the effects on the microbial community structure induced by kinetic mode changes, from feast-and-famine conditions (ANAS) to the continuous feeding of low nutrient concentrations (CANAS). A *D. mccartyi* genus-wide microarray was applied to study the transcriptional dynamics of *D. mccartyi* populations present in CANAS.

2. Materials and methods

2.1. Inoculum culture

A 3.0 L CMFR was inoculated with dechlorinating enrichment culture ANAS at 5% v/v. ANAS was originally enriched from contaminated soil obtained from Alameda Naval Air Station (CA) and has been functionally stable for more than 15 years in a continuously stirred semi-batch reactor. Every two weeks, 25 mM (or 2.8 g L^{-1}) lactate was supplied as both electron donor and carbon source and 0.1 mM (or $13 \mu\text{g L}^{-1}$) TCE was supplied as the terminal electron acceptor. The growth and maintenance procedures of ANAS have been previously described (Lee et al., 2011; West et al., 2013). Two distinct *D. mccartyi* strains were identified and isolated from ANAS (strain ANAS1 which contains the TCE reductive dehalogenase gene *tceA*, and strain ANAS2 which contains the VC reductive dehalogenase gene *vcrA*) (Lee et al., 2011). ANAS has been extensively studied (Richardson et al., 2002; Lee et al., 2006; West et al. 2008, 2013) and contains 1056 bacterial and archaeal taxa on the basis of DNA PhyloChip analysis (Brisson et al., 2012).

2.2. CMFR operation and sampling

The CANAS culture was maintained in the 3.0 L CMFR with no headspace represented in Fig. 1 (New Brunswick Eppendorf, Enfield, CT) fitted with Teflon caps and viton tubing (Masterflex, Cole-Parmer Instrument Co., Vernon Hills, IL) (Supporting material, Fig. S1). Pure TCE was stored in a 1.0 mL gastight syringe with a PTFE plunger tip (Hamilton, Reno, NV) and was continuously fed into the glass reactor using a syringe pump (NE-300 programmable syringe pump, New Era Pump Systems, Farmingdale, NY) at a flow rate of $12 \mu\text{L d}^{-1}$ ($0.133 \text{ mmol d}^{-1}$). The composition of the influent (flow rate = 75 mL d^{-1}) was modified from the defined medium used for the growth of *D. mccartyi* strain 195 (He et al., 2007) by including 10 mM lactate as the electron donor/carbon source and by increasing the concentration of L-cysteine and sodium sulfide (Na_2S) reducing agent to 0.4 mM of each to ensure anoxic conditions inside the influent tubing and reactor (Supplemental material, Table S1) (Johnson et al., 2008). Lactate was supplied to the reactor at a rate of $750 \mu\text{mol d}^{-1}$, i.e., 3 meeq d^{-1} taking into account 4 eeq per mole of lactate on the basis of H_2 production (Supporting material, equation S1) which is in excess of electron equivalents required for dechlorination of TCE to ethene ($133 \mu\text{mol d}^{-1}$, i.e., 0.8 meeq d^{-1}).

The liquid flow rate of 0.075 L d^{-1} resulted in a solids retention time (SRT) of 40 days (dilution rate of 0.025 d^{-1}), theoretically equal

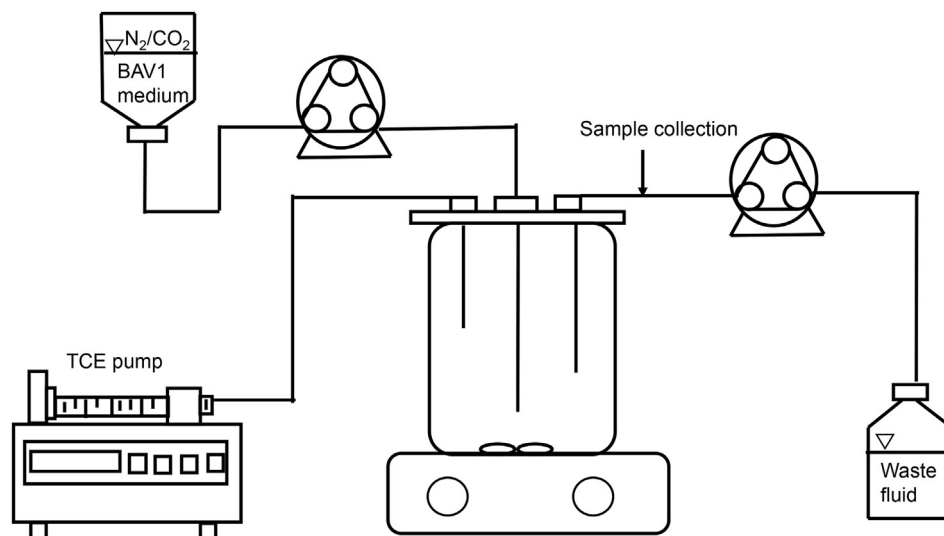


Fig. 1. Diagram of the CMFR system used for cultivation of CANAS.

to the hydraulic retention time in the CMFR. Influent pH was adjusted to 7.0–7.3 using 30 mM sodium bicarbonate and 10 mM TES buffer. The CMFR was continuously stirred with a magnetic stir bar (~10 rpm) at room temperature ($23 \pm 2^\circ\text{C}$) to ensure rapid dissolution of TCE and to minimize mass-transfer limitations (detailed information of operation is summarized in Supporting material, Table S2 and Fig. S2). 1.5 mL culture samples were collected frequently throughout the experimental period for quantification of metabolites and cell numbers. Cell pellets were harvested after centrifugation at 14,000 rpm (4°C) for 10 min. After the CMFR maintained steady-state for five SRTs (i.e., stable levels of chlorinated ethenes, ethene, methane, hydrogen, lactate, volatile fatty acids, and specific biomarkers for 200 days), duplicate culture samples were collected during three consecutive SRTs (at stages I, II, III) to generate biological triplicates for subsequent DNA/RNA studies. Briefly, 100 mL culture samples were collected in duplicate at the sample port and filtered on a $0.2\ \mu\text{m}$ autoclaved GVWP filter (Durapore membrane, Millipore, Billerica, MA). Each filter was immediately placed in a 2 mL attached screw cap micro-centrifuge tube (Corning, Lowell, MA), frozen with liquid nitrogen and stored at -80°C until further processing. The CMFR design, configuration and maintenance strategies, as well as routine sample collection and preparation procedures, are summarized in the supporting material.

2.3. Chemical and molecular methods

Chloroethenes, ethene and methane were measured by injecting 100 μL headspace sample to a FID-gas chromatograph (Agilent Technologies, Santa Clara, CA), and 300 μL headspace was injected into a RGD-gas chromatograph (Trace Analytical, Menlo Park, CA) for hydrogen quantification as previously described (Freeborn et al., 2005; Lee et al., 2006). Organic acids, including lactate, acetate and propionate were analyzed in 2 mL liquid samples by HPLC equipped with a photodiode array detector set at 210 nm (Waters 2996 photodiode array detector, Milford, MA) as previously described (Freeborn et al., 2005).

Quantification of *Bacteria*, *Archaea* and specific microbial populations or functional groups present in CANAS was performed at regular interval times using real-time quantitative PCR (qPCR) targeting *tceA* and *vcrA* genes and different regions of 16 S rRNA genes as previously reported (Men et al., 2012; West et al., 2013)

with primers summarized in Table S3. Cell lysis was carried out by mechanical and chemical disruption and nucleic acids were purified using a previously described phenol extraction (West et al., 2008). DNA and RNA were separated using AllPrep DNA/RNA Mini Kit (Qiagen, Valencia, CA) according to the manufacturer's instruction. RNA was eluted from the spin column with 100 μL of nuclease-free water and was further purified by using Turbo DNA-free kit (Ambion, Austin, TX) to remove additional DNA contamination according to the manufacturer's instructions. The quality of RNA samples was checked by electrophoresis, and the concentration of RNA samples was quantified using a nanophotometer P-300 (Implen, Westlake Village, CA, USA). The ratio of A260/A280 for all samples was between 1.80 and 2.0. Purified RNA was stored in nuclease-free water at -80°C prior to further analysis.

2.4. Microbial community structure analysis using targeted sequencing of 16 S rRNA genes

Sequencing of the V4 hypervariable region amplicons was performed on a HiSeq 2000 in a 2×150 bp configuration. Due to quality issues, single reads instead of assembled read pairs were kept for downstream analyses. Sequences were analyzed through our internal rRNA short amplicon analysis pipeline as previously described (Tremblay et al., 2015; Yergeau et al., 2015). Briefly, reads were quality-filtered and quality-trimmed, and clustered at 97% similarity. Resulting OTUs were assigned to a taxonomic lineage using the RDP classifier with a Greengenes (v13.5) training set. OTU tables were generated, filtered to exclude eukaryotes and chloroplasts and normalized using the trimmed mean of M-values (TMM) method in the edgeR package (Robinson et al., 2010; McMurdie et al., 2014). TMM-normalized OTU tables were used for assessing relative abundances of OTUs for every taxonomic level and computing beta-diversity metrics. Repeated rarefactions at 5000 reads (performing 10 iterations at each sampling depth) were generated from the raw (un-normalized) OTU table to generate alpha diversity metrics. Taxonomic and phylogenetic alpha and beta diversity metrics were computed using Qiime and downstream analyses were done with in-house Perl and R scripts. OTU tables and further details on 16 S amplicon sequencing are included in supplemental material.

2.5. Nucleotide sequence accession numbers

Raw sequence reads of the 16 S rRNA gene amplicon data for this project are available through the NCBI Sequence Read Archive (SRA) (study accession: SRP090533; Bioproject PRJNA309107).

2.6. Transcriptomic analysis

The samples selected for transcriptomic analysis were 1) ANAS-0h (before a new feeding cycle when the culture was in starvation condition; 2) ANAS-20 h (the culture was metabolically active after 20 h of feeding, all dechlorination metabolites were present); 3) CANAS (biological triplicate samples were collected from stage I, II and III at steady state) (Fig. 2). Microarray sample preparation and

application procedures were previously described (West et al., 2013). Briefly, 1 µg of community genomic DNA (gDNA) was used for DNA analysis and 10 µg of community total RNA was used as starting material for RNA analysis as previously described (West et al., 2008). All probe sets that were detected as “present” by DNA microarray analysis were used in the RNA analysis. DNA for any particular ORF was deemed “present” if each replicate probe set for that ORF had signal intensity greater than 140 and a P value less than 0.05 in the DNA chips. RNA for any particular ORF was considered “present” if the average signal intensity of the probe sets for that ORF was greater than 120. The criteria applied in this study are the same as previously described for the ANAS transcriptomic study (West et al., 2013).

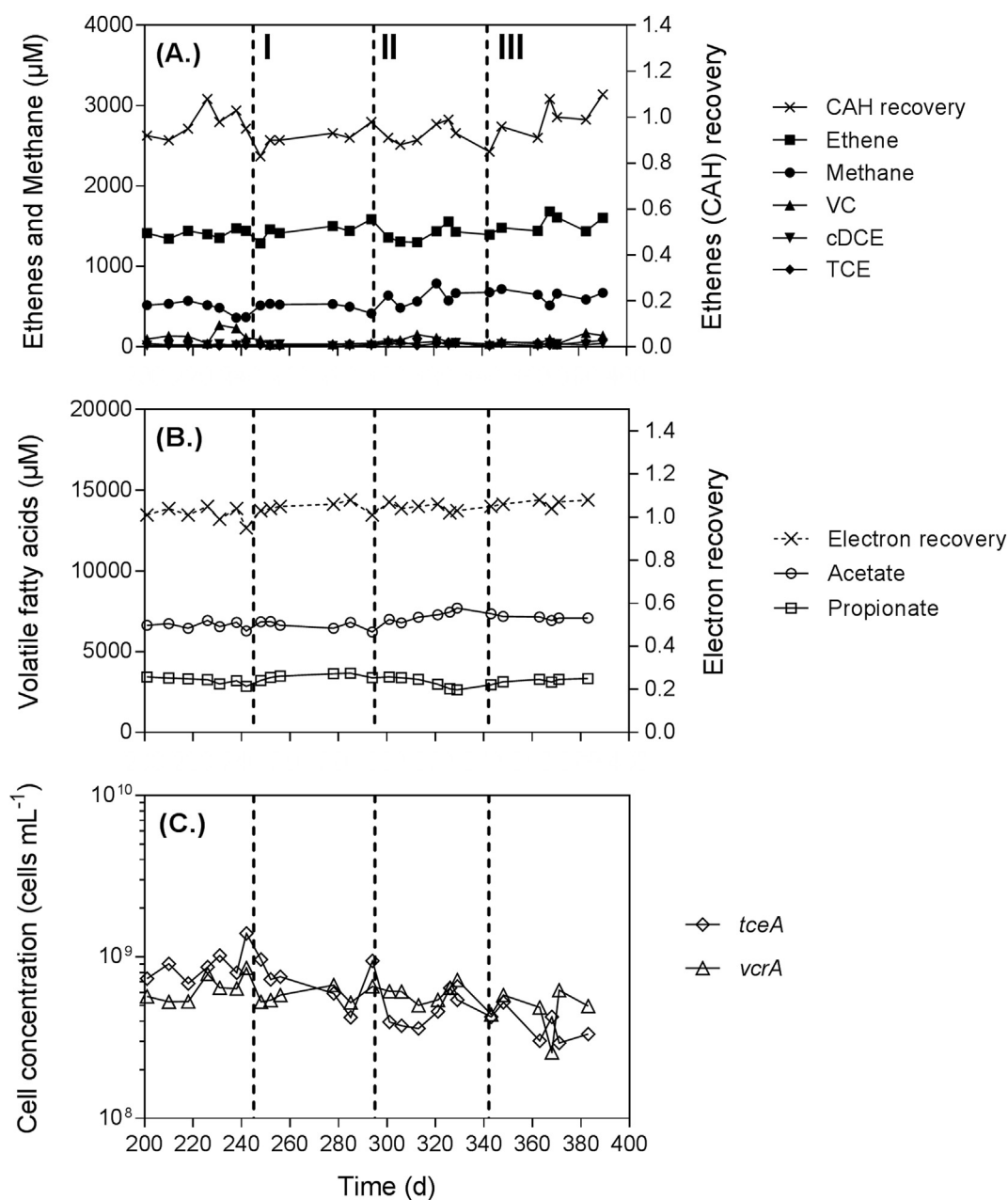


Fig. 2. Time course of (A.) TCE dechlorination, production of TCE metabolites and methane concentrations measured in the effluent samples and chlorinated aliphatic hydrocarbons (CAHs) recovery (× 100%); (B.) Volatile fatty acid concentrations (lactate was below the detection limit) and electron balance (see Table 2); and (C.) *D. mccartyi* strains ANAS1 (*tceA*) and ANAS2 (*vcrA*) cell concentrations at steady-state in CANAS during the monitored experimental period (stages I, II and III).

3. Results and discussion

3.1. Steady state performance of the community

The 40-day SRT selected for this study was lower than that of ANAS (Table S1) and was based on two objectives: To avoid washing out the slow growing microorganisms (e.g., syntrophs) and to maintain a sufficient steady-state volumetric biomass productivity (cells L⁻¹ d⁻¹) for molecular analyses. The basic maintenance parameters for CANAS are compared with previous CMFR PCE/TCE dechlorination to ethene studies in Table 1. The donor to acceptor ratio in the influent supplied to CANAS was about 19:1 (based on 12 equivalents of available electrons per mole of lactate and 6 eq per mole of TCE reduced), ensuring TCE to be the limiting reactant for dechlorination (as observed in TCE-contaminated plumes) with the electron donor concentration in excess. Samples were collected at steady-state for both microbial community structure and transcriptomic analyses during the experimental period from day 201 to day 400 (Fig. 2). During this time, around 92% of TCE in the influent (1.8 mM (0.13 mmol TCE d⁻¹/0.075 L d⁻¹ influent)) was converted to 1.5 ± 0.10 mM ethene, 0.09 ± 0.06 mM vinyl chloride (VC) and 0.03 ± 0.01 mM *cis*-DCE (Fig. 2A) based on effluent sample measurement. The chlorinated solvent recovery rate ranged from 83 to 108% during the experimental period (Fig. 2A). The dechlorination rate of the reactor was 120 μmol Cl⁻ L⁻¹ d⁻¹. Methane concentration in the reactor was maintained at a low level of 0.56 ± 0.10 mM (with a production rate of 14 ± 2.5 μmol L⁻¹ d⁻¹).

ANAS, the inoculum of CANAS, consistently exhibited faster growth of *D. mccartyi* strain ANAS1 (with one copy of *tceA*) than ANAS2 (with one copy of *vcrA*), resulting in an ANAS1:ANAS2 ratio of 4:1 (Lee et al., 2011). In CANAS, this ratio was stably maintained near 1:1 (6.2 ± 2.8 × 10⁸ cell mL⁻¹ vs. 5.8 ± 1.2 × 10⁸ cell mL⁻¹) (Fig. 2C), demonstrating that sustained exposure to low TCE concentrations (electron acceptor limitation) and constant supplies of electron donor, nutrients and growth factors can shift the relative abundances of *D. mccartyi* populations as has previously been shown under electron donor limitation (Mayer-Blackwell et al., 2017). The abundance of *D. mccartyi* strain ANAS2, which is responsible for VC reduction in the microbial community, could be increased in the completely mixed flow reactor and thus could facilitate more complete dechlorination.

Complete fermentation of 10 mM lactate in the influent was observed with the accumulation of propionate and acetate in the reactor effluent at concentrations of 3.1 ± 0.4 mM and 6.7 ± 0.7 mM, respectively (Fig. 2B). H₂ concentrations were maintained at around 0.2 μM (data not shown), which was greater than the reported minimum H₂ threshold (2 nM) to support dechlorination (Yang et al., 1998). Recovery of electron equivalents was 95–108% throughout the steady-state period (Table 2), a result similar to previous bioreactor studies (Drzyzga et al., 2001). Only 6.1% of the electrons were consumed for reductive dechlorination while 75.2% accumulated as propionate and acetate, and 2.4% as methane and trace H₂. Biomass production accounts for a large portion of electron flow in this reactor (15.8%), compared to previous studies (7.5–9.2%) (Azizian et al., 2010; Berggren et al., 2013). Biomass calculations assumed an elemental microbial cell composition of C₅H₇O₂N, an average dry cell weight of 1.1 × 10⁻¹⁴ g cell⁻¹ (with the assumption: the average cell size is 0.2 μm in diameter, 2 μm in length, 20% of the cell is in the solid form, 90% of the solid is in organic form, and an average of two 16 S rRNA copies are present in each bacteria cell), and total cell concentrations of *Bacteria* (2.3 ± 0.8 × 10¹⁰ 16 S rRNA copies mL⁻¹) and *Archaea* (0.7 ± 0.3 × 10⁷ 16 S rRNA copies mL⁻¹) based on average qPCR of 16 S rRNA copy numbers (Fig. S3) (Duhamel et al., 2007). The ratios of *D. mccartyi* 16 S rRNA copies to total 16 S rRNA copies (including

Bacteria and *Archaea*) were 4.2 ± 0.4%, 5.8 ± 0.3%, and 5.3 ± 0.4% for CANAS at stages I, II and III and 4.1 ± 0.2% and 4.7 ± 0.1% for ANAS during active and inactive dechlorination (Fig S3 B) (West et al., 2013).

To our knowledge, only one other study has reported cell densities and production rates of *D. mccartyi* in a CMFR with sustainable dechlorination to ethene (Table 1) (Delgado et al., 2014). Here, 85% of TCE in the influent was converted to ethene with a dechlorination rate of 120 μmol Cl⁻ L⁻¹ d⁻¹ which is lower than the highest dechlorination rate reported in a chemostat (130 μmol Cl⁻ L⁻¹ d⁻¹; SRT = 3 d) (Delgado et al., 2014) but higher than a previous study with a 55-day SRT exhibiting a dechlorination rate of 80 μmol Cl⁻ L⁻¹ d⁻¹ (Table 1) (Berggren et al., 2013). The relatively high *D. mccartyi* cell concentration (1.2 ± 0.3 × 10⁹ cells mL⁻¹) (Fig. S3A) observed in CANAS is comparable to the previously reported maximum *D. mccartyi* cell concentrations obtained in laboratory studies (Vainberg et al., 2009; Delgado et al., 2014). The *D. mccartyi* production rate in CANAS (3 × 10¹⁰ cells L⁻¹ d⁻¹, SRT 40 days) was about 10% of that observed in the 3-day SRT chemostat (Delgado et al., 2014) (Table 1), which is explained by the dilution rate (SRT⁻¹) of CANAS that is much lower than the reported maximum specific growth rates of *D. mccartyi* (0.21–0.4 d⁻¹) (Cupples et al. 2003, 2004; Johnson et al., 2008). The cell yield of *D. mccartyi* relative to TCE dechlorination in CANAS reached 2.5 ± 0.7 × 10⁸ cells per μmol Cl⁻ released (Table 1), which is in the range of previously reported cell yields of *D. mccartyi* isolates (He et al., 2007; Cheng et al., 2009; Men et al., 2012; Yan et al. 2012, 2013; Mao et al., 2015; Cooper et al., 2015), but lower than the yield reported in the 3-day SRT chemostat (5.3 × 10⁸ cells per μmol Cl⁻ released) (Delgado et al., 2014) likely due to enhanced endogenous cell decay within CANAS and the ability of *D. mccartyi* to decouple dechlorination from growth for maintenance activities (Cupples et al. 2003, 2004; Johnson et al., 2008).

3.2. Microbial community structure analysis

No significant difference in alpha diversity was observed between CANAS and ANAS using the Faith's phylogenetic diversity index (PD = 21.6 ± 3.1 and 23.7 ± 1.7, respectively ($p = 0.3709$; unpaired, two-tailed *t*-test with Welch's correction)) or the non-phylogenetic Shannon's index (3.8 ± 1.4 for CANAS and 6.0 ± 0.2 for ANAS, $p = 0.1163$) and Chao 1 estimators (551.9 ± 166.1 for CANAS and 717.9 ± 22.4 for ANAS, $p = 0.2241$). However, the composition and the relative abundance of OTUs identified in ANAS and CANAS demonstrated a microbial community structural shift along with the maintenance of TCE dechlorination performance. *Archaea* and *Bacteria* in CANAS represented 0.44% ± 0.19 and 99.56% ± 0.19 of the community respectively, with a significant difference in relative abundances compared to ANAS (4.86 ± 0.95 ($p = 0.0014$) and 95.1 ± 0.94 ($p = 0.0014$)). The lower relative abundance of *Archaea* in CANAS is consistent with the decreased methanogenic activity observed in the CMFR (Fig. 2A; 0.04 mol methane/mol lactate for CANAS and 0.23 mol methane/mole lactate for ANAS). At the phylum and class levels, CANAS is dominated by members of *Proteobacteria* (77% ± 9.7 of which 91% ± 8.8 belong to the class *Gammaproteobacteria*) and *Firmicutes* (16% ± 6.0, 99.9% ± 0.02 of which belong to the class *Clostridia*) (Figs. S4 and S5). ANAS is characterized by a more even community with *Proteobacteria* (49% ± 4.7) identified as the prominent phylum but with a dominance of the class *Deltaproteobacteria* (99.8% ± 0.03) (Figs. S4 and S5). The Gini coefficients (*G*) calculated on the basis of the taxonomy tables (phylum (L2) and class (L3) levels) demonstrated a lower evenness in CANAS than in ANAS ($G_{L2} = 0.94 ± 0.01$, $G_{L3} = 0.96 ± 0.01$ and $G_{L2} = 0.86 ± 0.02$, $G_{L3} = 0.94 ± 0.01$, respectively) (Fig. S6). At the taxonomic family level, twelve families with

a relative abundance higher than 1% were maintained during the feast and famine feeding cycle of ANAS (i.e., *Desulfovibrionaceae*, *Clostridiaceae*, members from an unidentified family of *Clostridiales* (*Clostridiales_order*), *Porphyromonadaceae*, *Sphaerochaetaceae*, *Thermovirgaceae*, *Methanobacteriaceae*, *Spirochaetaceae*, *Anaerolineaceae*, *Dethiosulfovibrionaceae*, members from an unidentified family of *Bacteroidales* (*Bacteroidales_order*) and *Thermotogaceae*) (Fig. 3). In CANAS, only five dominant families persisted at a relative abundance higher than 1% (i.e., *Pseudomonadaceae*, *Desulfovibrionaceae*, *Clostridiaceae*, members from an unidentified family of *Clostridiales* (*Clostridiales_order*) and *Peptococcaceae*) (Fig. 3).

Compared to ANAS, the microbial community in CANAS shifted to a new composition with the identification of two dominant families that exhibited a relative abundance (to bacteria and archaea) lower than 0.3% in ANAS (*Pseudomonadaceae* (66% ± 16 of CANAS) and *Peptococcaceae* (2.1% ± 0.5 of CANAS)). The high abundance of *Pseudomonas* was confirmed by qPCR of 16 S rRNA to exclude gene tag sequencing biases (data not shown). Although *Pseudomonadaceae* is mostly associated with aerobic and facultative anaerobic bacteria, all members of *Peptococcaceae* of the order *Clostridiales* are obligate anaerobes commonly involved in syntrophic associations (Kleinstaub et al., 2012; Stackebrandt, 2014). The relative abundance of *Clostridiaceae* increased from 1.5 to 7.0% in CANAS from stage I to III which is similar to ANAS (8.2% ± 4.5). *Desulfovibrionaceae* was relatively stable in CANAS (1.9% ± 0.28), which is much lower than the relative abundance observed in ANAS (48% ± 4.7). *Methanobacteriaceae* was the most abundant archaeal

family detected in both CANAS and ANAS and accounted for $0.43\% \pm 0.19$ and $4.6\% \pm 0.91$ of the total community, respectively. In CANAS, $98\% \pm 2.3$ of archaeal OTUs belong to the *Methanobacteriaceae* ($96\% \pm 6.0$ of *Methanobacterium* sp.) of which most members are hydrogenotrophic methanogens (Oren, 2014). In ANAS, $95\% \pm 0.5$ of archaeal OTUs belong to the *Methanobacteriaceae* and $4.6\% \pm 0.5$ belong to the *Methanospirillaceae* which mainly catalyzes methanogenesis from H_2/CO_2 (Oren, 2014). The relative abundances of *Dehalococcoidaceae* in CANAS ($0.59\% \pm 0.26$ for stages I, II, III) were similar to ANAS ($0.69\% \pm 0.04$ for ANAS-0h, ANAS-20 h and ANAS-27 h) with the maintenance of robust dechlorination rates (i.e., $12 \mu\text{mol Cl}^{-1} \text{L}^{-1} \text{d}^{-1}$ for CANAS and $59 \mu\text{mol Cl}^{-1} \text{L}^{-1} \text{d}^{-1}$ for ANAS). In all conditions, the relative abundance of *Dehalococcoides* sp. detected by gene tag sequencing was lower than the percentage calculated from qPCR analysis (4–6%, Fig. S3).

The structural regime shift observed between CANAS and ANAS (i.e., change in the microbial community composition and structure (Bürgmann et al., 2011; Engel et al., 2014; Andersen et al. 2009; Scheffer and Carpenter, 2003)) was confirmed using hierarchical cluster and PCoA analyses. Hierarchical cluster analysis of the most abundant OTUs (at the class level (Fig. S7) and family level (Fig. 4)) identified in ANAS and CANAS indicates that samples segregate according to their reactor type. Pairwise (intra- and inter-cluster) distances analysis for ANAS and CANAS (Fig. S8 A) showed that intra-cluster comparisons (ANAS-ANAS and CANAS-CANAS) and inter-cluster distances (ANAS-CANAS) were significantly greater than intra-cluster distances ($p < 0.0001$, Fig. S8 A). In addition, the

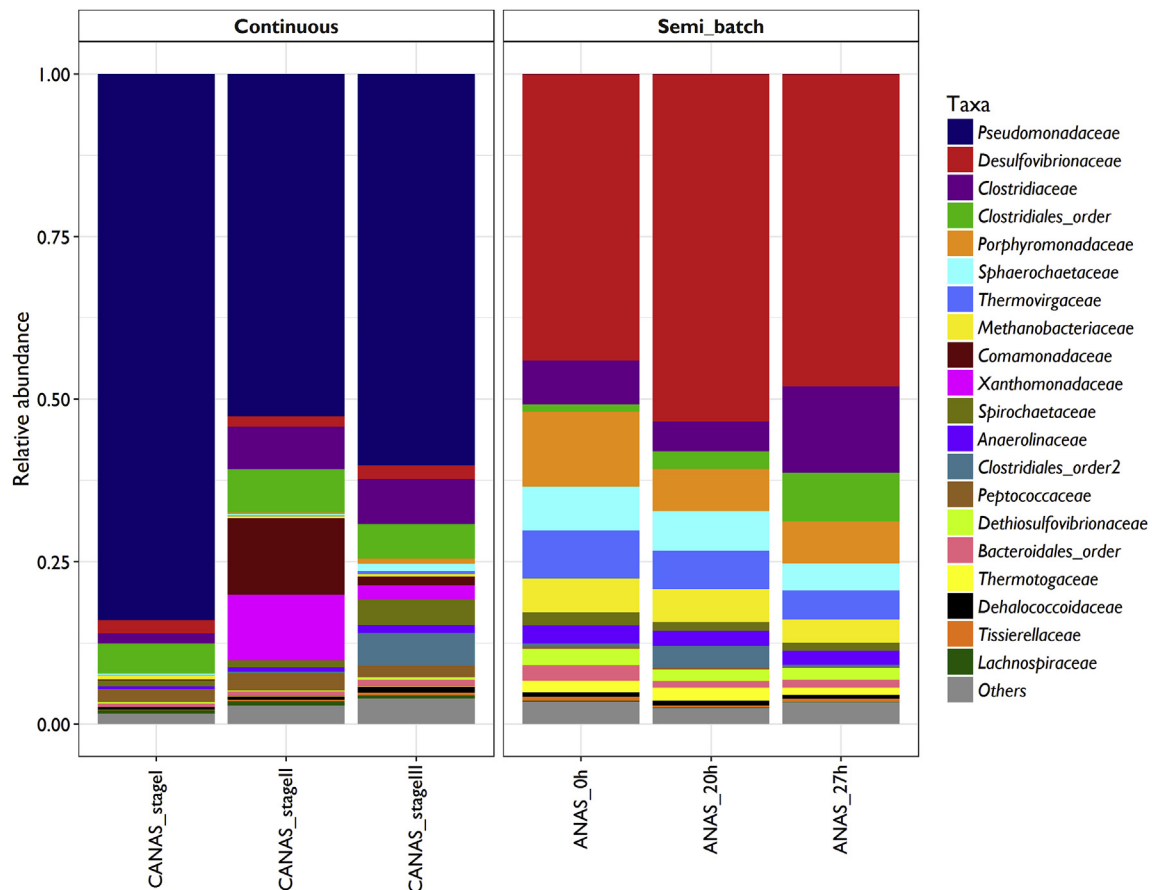
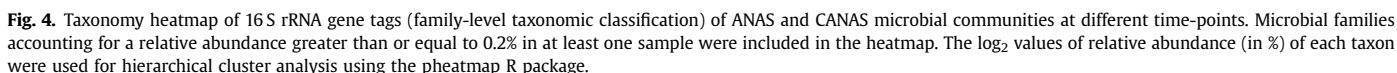


Fig. 3. Microbial community composition based on family-level taxonomic classification of 16 S rRNA gene tags for CANAS during three consecutive stages (I, II and III, roughly 50 days apart) and ANAS at three distinct time-points in a single feeding cycle (ANAS-0h, after a 14-day batch incubation (starved culture), ANAS-20 h, 20 h after lactate and TCE feeding (all lactate consumed) and ANAS-27 h, 27 h after lactate and TCE feeding (all TCE was converted to ethene)). The stacked bar charts present the nineteen most abundant taxa detected in ANAS and CANAS.



In this study, the microbial community structure of a TCE-dechlorinating enrichment culture, maintained functionally active for more than 15 years in a semi-batch reactor, significantly shifted after inoculation and maintenance in a CMFR. Despite the observed community structural shift, CANAS demonstrated a high functional robustness with the maintenance of efficient TCE dechlorination to

Using qPCR measurements normalized to the bacterial and archaeal 16S rRNA copies detected in the whole community.

D. mccartyi accounted for around 4–6% of the community in both ANAS and CANAS, as reported in other TCE-dechlorinating *D. mccartyi*-containing communities (Azizian et al., 2008; Behrens et al., 2008; West et al., 2013). In the EV-5L and VS-5L CMFRs (Table 1) (Mayer-Blackwell et al. 2016, 2017), the relative abundance of *D. mccartyi* was higher than 90% of the total bacterial community using targeted sequencing of the hypervariable V1–V2 or V2–V3 16 S rRNA regions. In the CMFR PM-2L (2-II) used to inoculate the CMFR PM-5L (Table 1), *Dehalococcoides* sp. accounted for around 35% of the bacterial community using 16 S rRNA-based qPCR assays (Berggren et al., 2013). In this study, we used the primer pair 515F-806R (Caporaso et al., 2011; Tremblay et al., 2015) for targeted amplicon sequencing which shows a single primer mismatch at the eleventh position from the 5'-end of the reverse primer 806R (position 10 from the 3' end; 5'-GGACTACHVG (G/A)GTWCTAAT-3') with the 16 S rRNA sequence of *D. mccartyi* strain 195. *In silico* analysis of the primer pair 515 F/806R with Primer-BLAST (Ye et al., 2012) revealed the same single base mismatch for all *D. mccartyi* 16 S rRNA sequences. This can explain the lower relative abundance values of *D. mccartyi* sp. in CANAS measured by 16S amplicon sequencing compared to qPCR assays. Aprill et al. (2015) (Aprill et al., 2015) reported the underestimation of SAR11 bacterioplankton due to the effect of a single internal primer-template mismatch at the position 8 from the 5' end of the reverse primer 806R. It is also possible that different sample preparation methods (cells were collected by filtration or by centrifugation) could lead to the discrepancy between the amplicon sequencing and the qPCR results.

Using qPCR measurements, the 16 S gene copies of methanogens in CANAS were below 30% of those measured in ANAS (Fig. S3A) (West et al., 2013). Similar results were obtained based on 16 S amplicon sequencing with a ratio of *Methanobacteriaceae* in CANAS vs. ANAS of 0.093 ± 0.045 , indicating that CANAS has about 10% of the abundance of *Methanobacteriaceae* compared to ANAS. H_2 levels measured in CANAS were maintained at about $0.2 \mu\text{M}$ which is higher than the required methanogenic threshold (Yang et al., 1998; Löffler et al., 1999). However, methane production was only about 30% compared to ethene production based on electron equivalents (Table 2). This contrasts with the ANAS bioreactor in which methane production was about 500% of ethene production (West et al., 2013). Since methanogenesis requires more reduced redox conditions ($E_H < -200 \text{ mV}$) than reductive dechlorination and oxygen reduction (Le Mer et al., 2001), methane production in CANAS might be limited by the increase of the redox potential as observed by the dominance of facultative anaerobic microorganisms.

3.3. Transcriptional analysis

Gene expression profiles of *D. mccartyi* in ANAS during active (ANAS-20 h) and inactive (ANAS-0 h) dechlorination were compared with those obtained in CANAS under sustained dechlorination at steady-state. In order to make valid comparisons of transcriptomic data between the two consortia, all expression levels were normalized to amended standard samples because *Dehalococcoides* species were present at similar relative abundances in both ANAS and CANAS (Fig. S3B). Unsurprisingly, the gene expression profiles of *D. mccartyi* in CANAS remained relatively constant in the three consecutive stages (Fig. 5), while the expression profile of ANAS varied significantly as expected in a semi-batch reactor. Most highly expressed genes in CANAS throughout the experimental period were genes encoding for ribosomal RNA (5 S, 16 S and 23 S rRNA) and reductive dehalogenases *tceA* and *vcrA*. Interestingly, the expression levels of *tceA* (DET0079) were significantly lower in CANAS ($p = 0.03$), while *vcrA* was

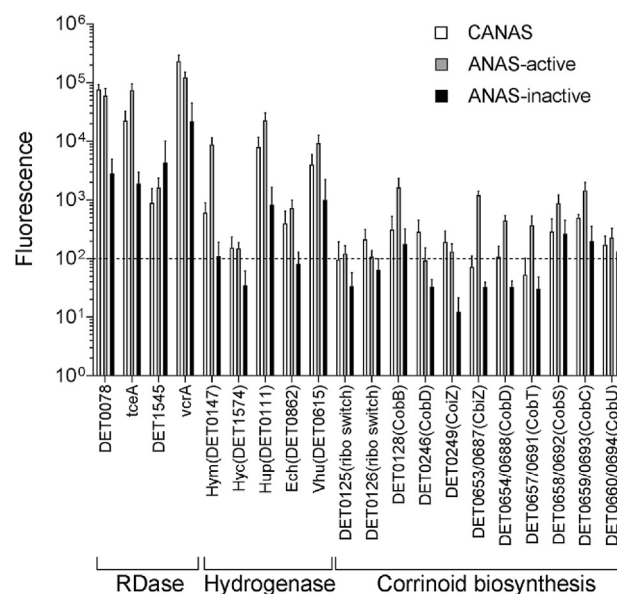


Fig. 5. Functional gene expression profiles of reductive dehalogenases (RDase) genes, oxidoreductase genes and corrinoid transport/biosynthesis genes in CANAS and ANAS during active dechlorination (20 h after feeding substrate, ANAS-active (ANAS-20 h)) and starvation (14 days after feeding substrate, ANAS-inactive (ANAS-0h)). Dashed line indicates significant fluorescence level (fluorescence signal > 100). Error bars represent the SD of biological triplicates.

significantly higher ($p = 0.03$) compared to ANAS during active dechlorination. The transcriptomic results agree with the higher cell ratio of ANAS2 to ANAS1 observed in CANAS compared to ANAS. The *tceA* anchor protein gene (DET0078), and two other RDase genes (DET0173 and DET 1545) in CANAS were similar to those detected in ANAS-active (Fig. 5). In contrast, the expression levels of putative hydrogenases (*hym*, *hup*, *vhu*, *hyc*, *ech*, *hyp*) and several oxidoreductase genes (*fdh*, *mod*, *nuo*, *fpoF*, *por*) were significantly lower compared to ANAS-active, but higher than expression levels detected in ANAS-starved (Fig. 5, supporting material Table S4). Specifically, the most highly expressed hydrogenases in CANAS were *hup* (DET0111), *hym* (DET0728), *vhu*, which were expressed to the same extent or slightly lower as detected in ANAS-active, but were significantly higher than ANAS-starved (Table S4).

Other oxidoreductases followed the same trend as the hydrogenase expression (Table S4), with the expression of the putative formate dehydrogenase (*fdh* DET 0185–0187) in CANAS at about 20% of that measured in ANAS-active, but 8 to 10 times higher than in ANAS-starved. A previous study had shown that the expression levels of DET 0187 in dechlorinating microbial communities were similar or higher than those of the hydrogenases under batch growth conditions (Morris et al., 2007). However, a recent transcriptomic study revealed that DET0187 was expressed at similar levels to the *Hup* subunit (DET 0112) in multiple *D. mccartyi* strains (Mansfeldt et al., 2014). The Fdh-like oxidoreductase (DET0187) was shown to be associated with the Fdh-like protein DET0112 from the *hup* operon (Mansfeldt et al., 2014) and a recent transcriptomic study of *D. mccartyi*-containing microbial communities indicated the expression pattern of the *Hup* and the Fdh-like oxidoreductase are strongly linked (Mansfeldt et al., 2016). Although the exact function of the Fdh-like oxidoreductase in *D. mccartyi* remains unknown, protein analysis of *D. mccartyi* strain CBDB1 suggested it is involved in the enzyme complexes that are critical to the electron transport chain of the *D. mccartyi* species (Kublik et al., 2016; Türkowsky et al., 2018). In our study, the expression levels of *fdh* (DET0186–DET 0187) in both ANAS and

CANAS were found to be at similar levels to the hydrogenases *hup*, *hym*, and *vhu* (Supplemental material Table S4), consistent with the previous findings (Mansfeldt et al. 2014, 2016; Heavner et al., 2018).

Previous studies have shown that *D. mccartyi* must rely on exogenous corrinoids for RDase activity (Magnuson et al., 1998; Banerjee and Ragsdale, 2003; Johnson et al., 2008; West et al., 2013; Yan et al., 2013; Men et al., 2015). Genes associated with corrinoid salvaging and transport were most highly expressed in ANAS-active with lower expression levels of both corrinoid transport and synthesis genes throughout the different monitored stages of CANAS supplemented with higher concentrations of vitamin B₁₂ (Fig. 5), consistent with a previous study of *Dehalococcoides mccartyi* 195 grown with excess or limiting vitamin B₁₂ (Johnson et al., 2008). The different expression patterns of corrinoid-related genes compared to genes for RDases indicate the expression of corrinoid-related genes may not be appropriate biomarkers for chlorinated ethene dechlorination activity. Previous studies have proposed hydrogenase genes (*hupL*) as biomarkers to indicate the activity of reductive dechlorination (Morris et al., 2007; Rowe et al., 2008; Berggren et al., 2013). In this study, we found similar positive correlation between *hupL* and *tceA* gene expression. Previous modeling studies have indicated that transcripts instead of DNA/proteins/VSS may serve as better biomarkers to predict reductive dechlorination performance (Baelum et al., 2013; Heavner et al., 2013), however, the consistent high levels of expression demonstrated for active and starving conditions of ANAS suggest that this approach is problematic.

4. Conclusion

Dechlorinating microbial communities have been extensively studied in batch reactors to reveal their dynamics as dechlorination proceeds and to construct kinetic models of reductive dechlorination pathways of chlorinated ethenes. However, it is difficult to extrapolate the knowledge acquired from such transient and fluctuating systems to flowing groundwater plumes with continuous and sustained supply of low concentrations of contaminants, nutrients and substrates. It is critical to understand the microbial community structure and dynamics in such continuous flow conditions, in order to validate the applications of molecular biomarkers or mathematical models during the bioremediation practices. In this study, we developed a completely mixed flow reactor containing the TCE-dechlorinating community (CANAS) from a long-term dechlorinating enrichment culture (ANAS) maintained in a semi-batch reactor. Based on the reactor performance assessment, microbial community structure analysis and the transcriptome dynamic analysis, the findings present in this study are as follows:

- The reductive dechlorinating microbial community CANAS developed in the completely mixed flow reactor showed effective TCE reduction at the selected SRT (40 days) with the major end-product of ethene (~85%) and a very low production of other intermediates (VC, *cis*-DCE) at the steady-state condition.
- The microbial community structure shifted significantly from semi-continuous flow condition to completely mixed flow condition while robust reductive dechlorination performance was stably maintained, suggesting that ANAS1 and ANAS2 could take advantage of the functional redundancy of microbial functional guilds that support their activities.
- The ratio of *D. mccartyi* strains ANAS1 to ANAS2 was stably maintained near 1:1 in the CANAS compared with 4:1 in the ANAS, demonstrating that sustained exposure to low TCE concentrations can shift the relative abundance of *D. mccartyi* populations.

- This is the first study to examine global gene expression of *Dehalococcoides* species under continuous flow conditions. The most highly expressed genes encoded for ribosomal RNA and reductive dehalogenases *tceA* and *vcrA*, while the expression of putative hydrogenases and several oxidoreductases genes were maintained at the levels between the ANAS-active and ANAS-inactive conditions, suggesting that we need to be careful when selecting biomarkers to indicate reductive dechlorination activity.

Declaration of interests

☑ The authors declare that they have no known competing financial interests or personal relationships that could have appeared to influence the work reported in this paper.

☐ The authors declare the following financial interests/personal relationships which may be considered as potential competing interests.

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Appendix A. Supplementary data

Supplementary data to this article can be found online at <https://doi.org/10.1016/j.watres.2019.04.038>.

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