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An estimation of chemical compound concentrations used in onshore gas production, a review of their degradation, and associated policy frameworks in South Australia

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Important disclaimer

This report is in partial fulfilment of the GISERA project 'Chemical compounds used in onshore gas production in the SE of South Australia: microbial degradation, microbial community impact and indicator taxa', project number W15. The completion of this report fulfils Task 1 of this project. Task 1 sets out to provide a literature and policy review. The remaining tasks consist of sample collection (Task 2), experimental microbiology work (Tasks 3-4), DNA profiling (Task 5), and final reporting (Task 6).

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Acknowledgments

We thank CSIRO's Gas Industry Social and Environmental Research Alliance (GISERA), which is a collaboration between CSIRO, Commonwealth and state governments and industry established to undertake publicly-reported independent research. The purpose of GISERA is for CSIRO to provide quality assured scientific research and information to communities living in gas development regions focusing on social and environmental topics including: groundwater and surface water, biodiversity, land management, the marine environment, human health impacts and socioeconomic impacts. The governance structure for GISERA is designed to provide for and protect research independence and transparency of research outputs. Visit gisera.csiro.au for more information about GISERA's governance structure, projects and research findings.

Executive summary

This report is part of a wider GISERA (Gas Industry Social and Environmental Research Alliance) project entitled 'Chemical compounds used in onshore gas production in the SE of South Australia: microbial degradation, microbial community impact and indicator taxa', project number W15. This wider project seeks to understand whether chemical compounds used in the production of onshore gas are degraded by microbes in relevant southeast South Australian soils and subsurface aquifers, in both oxic and anoxic conditions. Specifically, this project will examine the microbial degradation of approximately 28, previously identified hazardous chemical compounds associated with onshore gas activities. This report sets the scene for this study by estimating compound concentrations of the 28 selected chemicals, reviewing past findings on their degradation propensities and listing associated policy frameworks.

While current onshore gas technologies enhance the production efficiency of numerous hydrocarbon reservoirs, community concerns have been raised about potential risks of chemical contamination from such activities to water and soil quality. While the chemicals used in such activities typically comprise only a small fraction of drilling and stimulation fluids, they perform important functions in the exploitation process by acting as viscosity modifiers, friction reducers, surfactants, antiscalants, and biocides. In onshore gas production activities, water, proppants, and chemicals are mixed onsite to produce drilling/stimulation fluids. Estimates of water use for such fluids can vary and depend on multiple factors including well type, number of production wells, number of stimulation stages and designs, and the extent of naturally occurring fractures. A range of naturally occurring abiotic and biotic processes have the capacity to attenuate such chemical compounds in the environment. Such processes are therefore important to identify when assessing potential impacts in areas where onshore gas activities are about to be undertaken.

Typical chemical concentrations in drilling/stimulation fluids were ascertained by investigating online databases, reports and journal publications. Since the chemical composition of conventional drilling fluids and hydraulic fracturing fluids can be broadly similar, the majority of concentration data in this study was obtained from FracFocus (http://fracfocus.org/), the national (USA) hydraulic fracturing chemical registry. The following chemical compounds were investigated: Bronopol; Methylchloroisothiazolinone; Polyacrylamide / polyacrylate copolymer; Acrylamide; Xanthan gum; Polyoxypropylene diamine; Hexahydro-1,3,5-tris(2-hydroxyethyl)-sym-triazine; Glyoxal; 2-Aminoethanol; Limonene; 2-Methylphenol; Naphthalene; Acetic Acid; Alcohols, C_{6}-$_{12}$ ethoxylated; Alkanes, C_{12}-$_{26}$ branched and linear; Benzisothiazolinone; 2-Butoxyethanol; Diethylene glycol ethyl ether; Ethanol; Ethylene glycol; Glutaraldehyde; Isopropanol; Methanol; Methylisothiazolinone; Pigment Red 5; Triethanolamine; Propylene glycol; and 2-Ethylhexanol.

For each of these compounds both abiotic and biotic degradation processes were identified. Abiotic process involved photolysis and hydrolysis, while biotic process involved the microbially-induced breakdown (biodegradation) of compounds by bacterial and/or fungal species. In many cases both

oxic and anoxic biodegradation processes were identified and degradation (typically measured in half-life) could proceed within days to weeks.

Australian state and territory governments are mainly responsible for the legislative framework, licensing and decision making processes governing both conventional and unconventional gas exploitation. Federal legislative powers regarding oil and gas regulation are limited to the corporation's power, interstate, as well as overseas trade. An exception is *The Environment Protection and Biodiversity Conservation Act 1999*, which provides a legal framework to protect and manage impacts upon matters of national environmental significance. In addition, the *Industrial Chemicals (Notification and Assessment) Act 1989* requires industrial chemicals used in drilling and hydraulic fracturing to be listed on the national Australian Inventory of Chemical Substances. In South Australia, onshore geothermal, petroleum, and gas exploration, development, and storage is administered by the Energy Resources Division under the *Petroleum and Geothermal Energy Act 2000* and associated regulations. As of late 2018, the Upper House of the South Australian Parliament passed a 10 year fraccing moratorium in the South East of South Australia into law.

1 Introduction

Fluids used in onshore gas activities enhance the production efficiency of numerous gas reservoirs. The associated drilling/stimulation fluids usually consist of a mixture of water and various chemical additives (e.g., Barati and Liang, 2014; Flores 2014; Schinteie et al., 2015; Barbati et al., 2016). The composition of these fluids varies, depending on site-specific conditions and is usually tailored to project needs (e.g., Barati and Liang, 2014; Flores 2014; Schinteie et al., 2015; Barbati et al., 2016; Mumford et al., 2018). Numerous risks associated with these chemical-laden fluids have been identified and formed the focus of numerous studies and reviews (e.g., US-EPA, 2004; Schinteie et al., 2015; Elliot et al., 2017), which have identified potential environmental and human health impacts. Therefore, community concerns have been raised about potential risks of chemical contamination from onshore gas activities to water and soil quality.

Degradation refers to both abiotic and biological processes whereby a chemical substance is broken down into smaller molecules (e.g., Peters et al., 2005; Pandey et al., 2016; Schinteie et al., 2018). It is an important process to consider when assessing the fate of chemical compounds released into the natural environment because it changes both the nature and concentration of these substances. Abiotic degradation includes processes such as hydrolysis and photolysis in the atmosphere. Biodegradation, by contrast, is the process by which substances are broken down by living organisms. Ultimately, degradation acts as a mass conversion process that results in a decline in the total mass of chemical compounds (e.g., Peters et al., 2005; Pandey et al., 2016; Schinteie et al., 2018). Degradation is typically measured by the half-life of particular compound; a greater half-life denoting a greater persistence of the compound in the environment (e.g., Sinkkonen and Paasivirta, 2000).

This report is part of a wider GISERA (Gas Industry Social and Environmental Research Alliance) project entitled 'Chemical compounds used in onshore gas production in the SE of South Australia: microbial degradation, microbial community impact and indicator taxa', project number W15. This wider project seeks to understand whether chemical compounds used in the production of onshore gas are degraded by microbes in relevant southeast South Australian soils and subsurface aquifers, in both oxic and anoxic conditions. In addition, the project seeks to examine the impact of these compounds on microbial communities. Specifically, this project will examine the microbial degradation of approximately 28, previously identified hazardous chemical compounds associated with onshore gas activities. These chemicals were selected since they are globally used in onshore gas activities. Since we cannot predict what specific chemicals companies will use in the future in South Australia, the common and worldwide usage of these 28 selected chemicals provide us with a predictive data set. Degradation will be investigated under a mixed oxic and anoxic matrix (soil), as well as in an anoxic water matrix (subsurface aquifer). This report sets the scene for this study by estimating compound concentrations of the 28 selected chemicals (Table 1), reviewing past findings on their degradation propensities and listing associated policy frameworks.

Table 1 Onshore gas production chemical compounds of interest examined in this review

Compound	Use in onshore gas activities
Bronopol	Biocide
Methylchloroisothiazolinone	Antimicrobial preservative
Polyacrylamide / polyacrylate copolymer	Friction reducer
Acrylamide	Friction reducer
Xanthan gum	Viscosity management
Polyoxypropylene diamine	Pipework/Epoxy resins
Hexahydro- 1,3,5-tris(2-hydroxyethyl)-sym-triazine	Biocide
Glyoxal	Drilling additive
2-Aminoethanol	Drilling additive
Limonene	Surfactant
2-Methylphenol (o-cresol)	Biocide
Naphthalene	Corrosion inhibitor
Acetic acid	Buffer, stabiliser, solvent
Alcohols, C6-12, ethoxylated	Surfactant
Alkanes, C12-26 branched and linear	Surfactant
Benzisothiazolinone	Biocide
2-Butoxyethanol	Surfactant
Diethylene glycol ethyl ether	Solvent
Ethanol	Surfactant
Ethylene glycol	Viscosity management
Glutaraldehyde	Biocide
Isopropanol	Surfactant
Methanol	Surfactant
Methylisothiazolinone	Biocide
Pigment Red 5	Viscosity management
Triethanolamine	Viscosity management

Propylene glycol	Viscosity management
2-Ethylhexanol	Surfactant

2 Preparation of drilling and stimulation fluids

Drilling fluid is essentially a mud that is composed of a mixture of clay with either:

1) water (water-based drilling mud)

2) oil (oil-based drilling mud)

3) synthetic organic matter (synthetic-based drilling mud; Hyne, 2012).

Various chemical compounds (called additives) can be mixed with the mud for various effects (e.g. thinners, foaming agents, flocculants, emulsifiers, biocides, pH control). The clay and additives are brought onto the drillsite in dry sacks and stored in the mud house. They are added to the mud through a hopper into the mud tanks (Hyne, 2012). The drilling mud and associated additives passes through numerous areas in drilling site and are listed below and depicted schematically in Figure 1.

Drilling mud is stored in mud pits/tanks on the ground near the drilling rig. The mud is kept mixed in these tanks via a mud agitator (rotating paddles) or a mud gun (high pressure jet). During drilling, mud is pumped from these storage tanks via a mud hose into the hollow, rotating drill string through to the bottom of the well. There, drilling mud will pick up cut rock chips (cuttings) and transport it up the concentric well space (annulus) between the drill string and the well walls (e.g. casing). Upon return to the surface, drill mud will move through the shale shaker (to separate coarse well cuttings from the mud) and through desanders and desilters (to remove finer sedimentary particles). The mud subsequently flow back into the mud tanks and eventually recirculated down the well. Discarded mud can be held for reuse in earthen pits called reserve pits and are located adjacent to the mud tanks (Hyne, 2012).

Stimulation fluids are typically prepared in a different fashion (Figure 2). Water, proppants, and chemicals are mixed onsite to produce the fluid. The preparation of this fluid requires onsite storage, mixing and pumping (e.g. Landis, 2015). Specialized feeding and mixing equipment is required with mixing generally performed mechanically on a truck-mounted blender. The mixing process is electronically monitored and controlled by an operator in a separate truck with numerous hoses and pipes required to transfer hydraulic fracturing fluid components from storage units to the mixing equipment and finally to the wellhead (e.g. Landis, 2015).

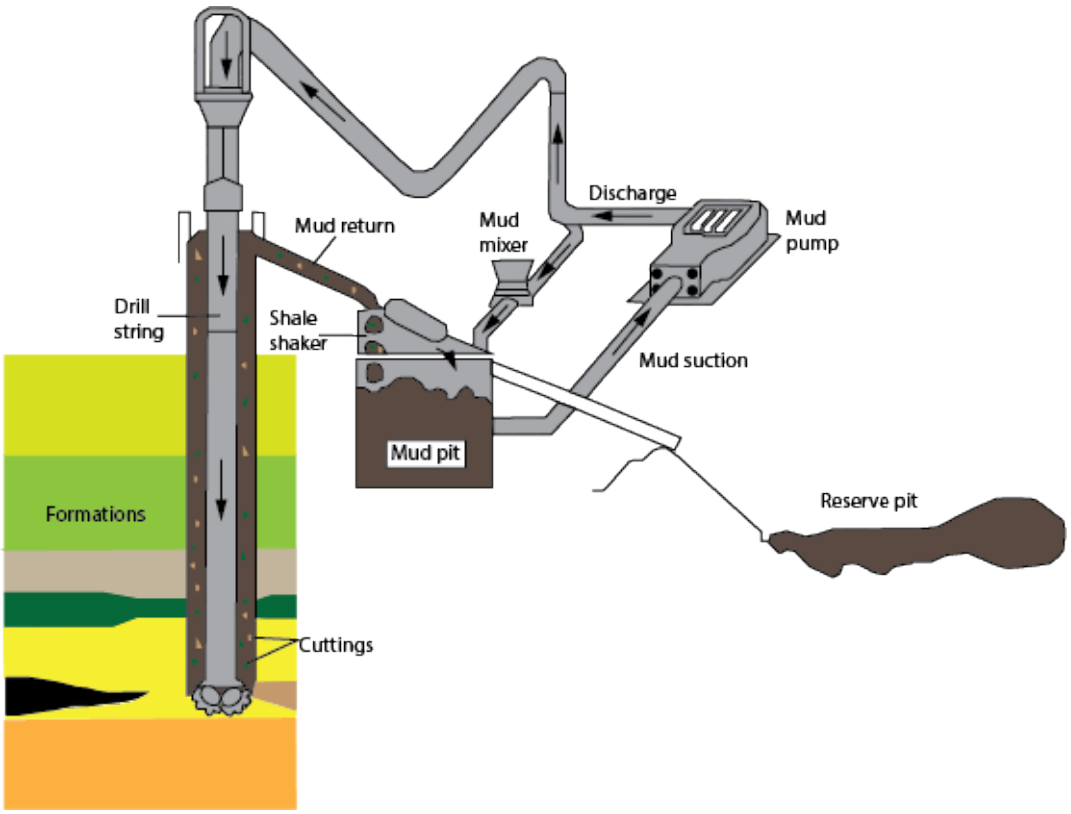


Figure 1 Schematic flow diagram showing a typical drilling fluid circulation system. Adapted with modification from Aboulrous et al. (2016).

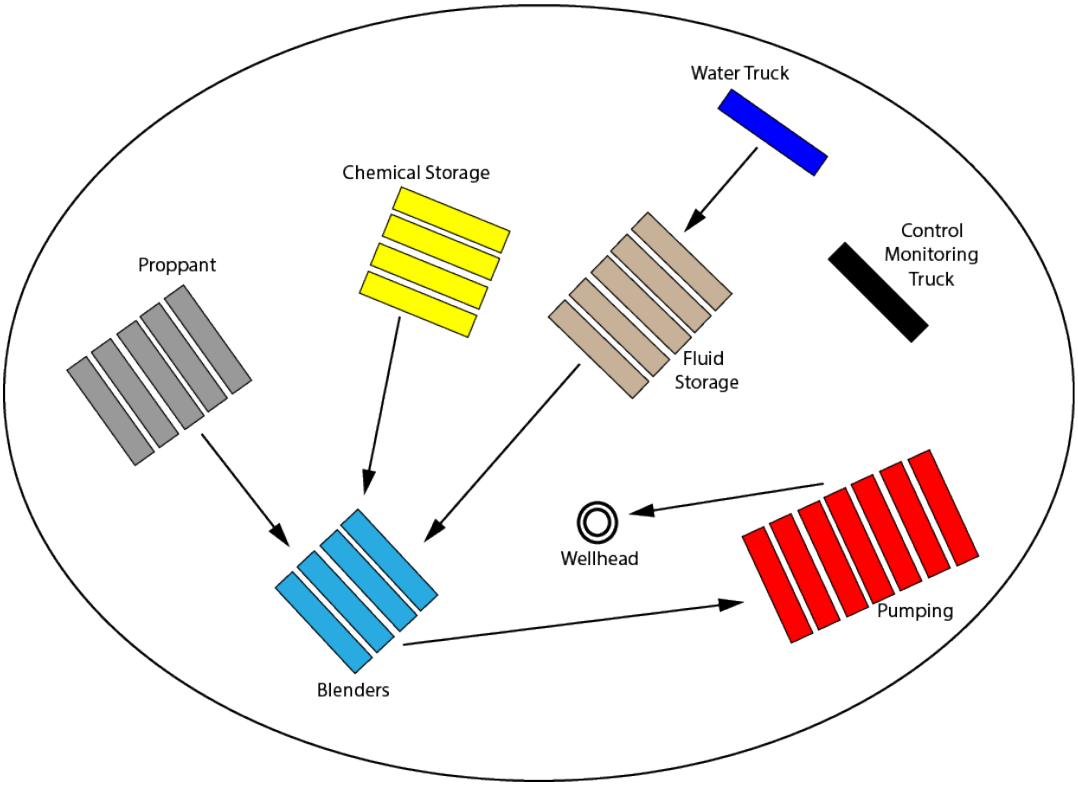


Figure 2 Schematic flow diagram showing the chemical mixing process at a well site to create stimulation fluid prior to injection. Each different part of the process is hosted in containers that are typically truck mounted. Adapted with modifications from Landis (2015).

3 Estimated concentrations and degradation processes of selected chemical compounds found in drilling/stimulation fluids

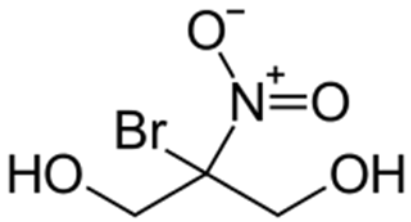
In this study, the degradation of 28 previously identified hazardous chemical compounds found in fluids used in onshore gas activities will be investigated. 'Degradation' refer to biological (mostly microbial), chemical, and physical (e.g., light, heat) processes that break down chemical substances into simpler components. In this review, we describe findings of previous degradation studies as well as focus on the typical concentrations of these 28 chemicals as recorded in various records, including online databases, reports and journal publications. Hence, this report sets the scene and provides information and directions for future experimental work that is part of the wider project. The majority of concentration data for this review is obtained from FracFocus (http://fracfocus.org/), the national (USA) hydraulic fracturing chemical registry. This registry is managed by the US Ground Water Protection Council and Interstate Oil and Gas Compact Commission and aims to provide the public access to reported chemicals used for hydraulic fracturing within their area. We use stimulation fluids as a guide in estimating the typical concentrations of the 28 selected chemicals used in onshore gas activity. In this study, we randomly selected North American wells (due to availability) where a chemical compound of interest was used and these are listed in the tables below. Note the following table headings:

- Maximum Concentration in Additive (% mass). This heading refers to the amount of ingredient within the additive (Trade Name) as a percent of the total mass of the additive. Since the % mass of the additive may be expressed in its maximum concentration, the total % mass of ingredient percentage may exceed 100% (see http://fracfocus.org/ for more information).
- Maximum Concentration in HF Fluid (% mass). This heading refers to the amount of ingredient as a percent of the total mass of the hydraulic fracturing (HF) fluid including carrier fluid and additives. Note that the total may not equal 100% due to the redaction of proprietary components in accordance with the Trade Secrets provisions of the Occupational Safety and Health Administration act (see http://fracfocus.org/ for more information).

Note that 'additives' refer to the chemicals used in HF fluids. Ingredients recorded as 100% are single-ingredient additives. Typically, additive chemicals make up

about 1 per cent of the HF fluid. The remainder of the fluid is composed of water and proppants (typically sand). These chemical data listed below in the various tables act as indications of typical compound concentrations in hydraulic fracturing fluids. Keeping in mind that the hydraulic fracturing compositions used can vary due to a range of geological and engineering factors, the concentrations listed below nevertheless provide a rough indication.

4.1 Bronopol ($C_3H_6BrNO_4$; 2-bromo-2-nitropropane-1,3- diol; CAS # 52-51-7)



Function: Biocide

Degradation: This compound is an electrophilic biocide that reacts with electron-rich thiol groups in glutathione or in cysteines in membrane proteins of bacterial cell walls (Sharma et al., 2017). By reacting with oxygen and thiols, it leads to the production of a reactive oxygen species called a superoxide (the anionic form of O_2; Williams and Mcginley, 2010).

A major degradation pathway common to electrophilic biocides is hydrolysis, which is characterized by the addition of a water molecule resulting in two smaller fragment molecules. Hydrolysis is strongly affected by the pH of the surrounding environment and, as is the case with bronopol, hydrolysis products can be more toxic and/or persistent than their parent compounds (e.g. Kahrilas et al., 2015). Bronopol has been shown to hydrolyse into formaldehyde, nitrosamines, and other molecules within a period of three hours at 60°C and a pH of 8 (Swenberg et al., 1980; Dunnett and Telling, 1984; Challis and Yousaf, 1990; Loeppky, 1994). While the occurrence of bronopol is considered short-lived in the environment, the degradation products are both toxic and more persistent (Douglass et al., 1978; Deutschle et al., 2006).

Due to the presence of chromophores, bronopol has been shown to undergo direct photolysis reactions when exposed to ultraviolet (UV) light, producing tris(2-hydroxymethyl-2-nitropropane-1,3-diol; EPA-USA. 1995). Additionally, Kahrilas et al. (2015) noted that all biocides can be degraded

by the action of reactive species from indirect photodegradation, although no experimental data are available for this process.

Biodegradation of bronopol is not expected to be an important fate process in the natural environment. Using a mixed culture sewage sludge test, this compound was not observed to be biodegraded (Wotzka et al., 1994).

It has been demonstrated that bronopol hydrolyses in aqueous media, resulting in non-reproducible analytical data (Moore and Stretton, 1981; Lian, 1997; Wang et al., 2002). Wang et al. (2002) overcame this problem by using methanol for sample preparation with bronopol standards being reported as stable in excess of 1 month at ambient temperature.

Concentrations:

Maximum concentration range in HF fluid (% mass): 0.00162-0.00343

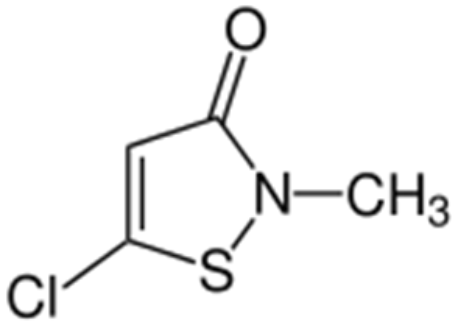
Average concentration range in HF fluid (% mass): 0.00222

Table 2 Bronopol concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Gage Com 001 (New Mexico)	BE-6 MICROBIOCIDE	Halliburton	Biocide	100	0.00343
Price 001E (New Mexico)	BE-6 MICROBIOCIDE	Halliburton	Biocide	100	0.00179
2S-12 (Alaska)	BE-6 MICROBIOCIDE	Halliburton	Biocide	100	0.00162
Larsen 1-12-1 (Colorado)	BE-6 MICROBIOCIDE	Halliburton	Biocide	100	0.00249
Cundiff Federal GU A 2 (Colorado)	BE-6 MICROBIOCIDE	Halliburton	Biocide	100	0.00252
2S-09 (Alaska)	BE-6 MICROBIOCIDE	Halliburton	Biocide	100	0.00185

Roberts Gas Com B #001 (New Mexico)	BE-6 MICROBIOCIDE	Halliburton	Biocide	100	0.00232
CD5-99 (Alaska)	BE-6 MICROBIOCIDE	Halliburton	Biocide	100	0.00174

4.2 Methylchloroisothiazolinone (C_4H_4ClNOS; 5-Chloro-2-methyl-1,2-thiazol-3(2*H*)-one; CAS # 26172-55-4)



Function: Biocide/antimicrobial preservative

Degradation: In the atmosphere, vapour-phase methylchloroisothiazolinone will be degraded by reaction with photochemically-produced hydroxyl radicals, exhibiting a half-life of 18 hours (TOXNET, 2015a). This compound is also susceptible to direct photolysis by sunlight with a half-life of 158 hours at surface water conditions (TOXNET, 2015a). While free methylisothiazolinone is unstable, greater stability is afforded by the formation of adducts with calcium or magnesium salts (Chan et al., 1983; Krzeminski et al., 1975a).

Rapid biodegradation of methylchloroisothiazolinone has been observed in both aquatic and terrestrial environments (Burnett et al., 2010). For example, the half-life of methylisothiazolinone in an aerobic microcosm using river water and sediment was observed to be 9 days (Jacobson and Williams, 2000). The major biodegradative pathway in the environment for the calcium chloride salt of methylchloroisothiazolinon was identified by Krzeminski et al. (1975b). Principally, degradation involves involved calcium chloride dissociation, ring opening, loss of chlorine and sulfur, and subsequent formation of *N*-methylmalonamic acid. Further degradation then proceeds through malonamic, malonic, acetic, and formic acids to carbon dioxide. Other tentatively identified products consisted of 5-chloro-2-methyl-4-isothiazolin-1oxide, *N*-methylglyoxylamide, ethylene glycol, and urea.

Biodegradation of methylchloroisothiazolinone has been observed to occur largely in aerobic environments. For example, biodegradation of this compound was measured by Voets et al. (1976) in synthetic sewage and in a mineral solution under both aerobic and anaerobic conditions. While

degradation of between 80 to 100% was observed in the organic medium under aerobic conditions, no degradation was noted under anaerobic conditions.

Concentrations:

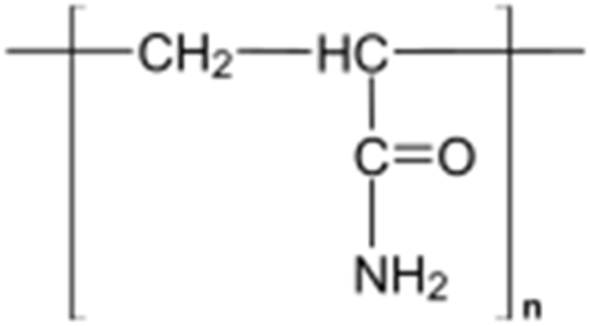
Maximum concentration range in HF fluid (% mass): 0.00001-0.00048

Average concentration range in HF fluid (% mass): 0.0001575

Table 3 Methylchloroisothiazolinone concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Bailey 9-24H2 (North Dakota)	SANIFRAC 8123	CWS	Microbiocide	0.10000	0.00002
Wilbur USA 31-2TFH (North Dakota)	SANIFRAC 8123	CWS	Microbiocide	0.10000	0.00008
McGrede 3 (Texas)	M-275	Baker Petrolite	Microbiocide	10	0.00041
Chauncey USA 31-2H (North Dakota)	SANIFRAC 8123	CWS	Microbiocide	0.10000	0.00001
Winona USA 21-2TFH-2B (North Dakota)	SANIFRAC 8123	CWS	Microbiocide	0.10000	0.00008
Harrell Gas Unit 1 Well 2 (Texas)	M-275	Baker Petrolite	Microbiocide	10	0.00048
Bailey 10-24H (North Dakota)	SANIFRAC 8123	CWS	Microbiocide	0.10000	0.00001
S-200A (Alaska)	M275	Schlumberger	Bactericide	0.00156	0.00017

4.3 Polyacrylamide / polyacrylate copolymer ($(C_3H_5NO)_n$; poly(2-propenamide); CAS # 9003-05-8)



Function: Friction reducer

Degradation: Experimental evidence exists for the photochemical degradation of polyacrylamide with increasing dosages of O_3, H_2O_2, and UV radiation (Ren et al., 2006). However, it is not clear how these experimental results could translate to environmental conditions. Mechanical degradation of polyacrylamide has also been reported due to the high shear and elongational rates under turbulent flow through small pores and fractures within porous media in shale formations (Xiong et al., 2018, and references therein).

Biodegradation is another pathway that results in the breakdown of polyacrylamide due to the utilization of the amide group as a nitrogen source and/or the carbon backbone as a carbon source (Nakamiya and Kinoshita, 1995; Wen et al., 2010). Extracellular amidases hydrolyze the amide group and these have been found to be expressed by a range of microorganisms, including *Enterobacter aerogenes*, *Helicobacter pylori*, *Acinetobacter* sp., *Azomonas* sp., *Bacillus* sp., *Chlostridium* sp., *Pseudomonas* sp., and *Rhodococcus* sp. (Xiong et al., 2018, and references therein). The associated degradation process can be enacted relatively quickly. For example, Yu et al. (2015) expressed aliphatic amidase from *Pseudomonas putida* in the presence of polyacrylamide and reported 46% degradation of 1000 mg/L polyacrylamide after 7 days at 39°C. The microbial utilization of the polyacrylamide carbon backbone as a sole carbon source, by contrast, is significantly more difficult due to the lack of appropriate enzymes (Kay-Shoemake et al., 1998).

Polyacrylamide can also be degraded under anaerobic conditions. For example, Dai et al. (2014) have shown that anaerobes can hydrolyse polyacrylamide complexed with tyrosine-rich proteins. Furthermore, cationic polyacrylamide has been shown to be converted to methane via methanogenesis as well as depolymerized to acrylamide and acrylic acid (Wang et al. 2018). After a 22-day digestion of sludge containing 12 g/kg total suspended solids equivalent to 240 mg/L polyacrylamide, acrylamide concentrations as high as 15 mg/L were identified (Wang et al. 2018).

Concentrations:

Note: chemicals for the concentrations below are labelled as “anionic polyacrylamide”. They do not have a CAS number and are regarded as “proprietary”.

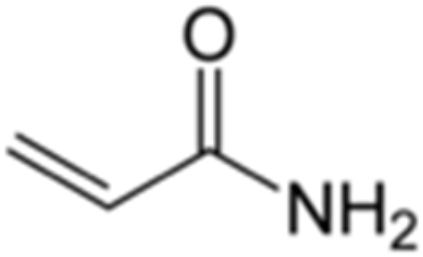
Maximum concentration range in HF fluid (% mass): 0.00433-0.05528

Average concentration range in HF fluid (% mass): 0.02257

Table 4– Polyacrylamide concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
AA Post Unit 1H (West Virginia)	WFRA-405	U.S. Well Services	Friction Reducer	40	0.01828
McQuarters (North Dakota)				100	0.00433
McCoy 14-33 (Colorado)				100	0.05528
NICHOLSON BEN 8-16 5-8H5 (Arkansas)	SFR-470	SWN Well Services	Friction Reducer	40	0.01524
NBU 1022-5J1CS (Utah)				60	0.01972

4.4 Acrylamide (C_3H_5NO; Prop-2-enamide; CAS # 79-06-1)



Function: Friction reducer

Degradation: Vapour-phase acrylamide can be degraded by reaction with photochemically-produced hydroxyl radicals, exhibiting a half-life of 6.6 hours (Croll et al., 1974).

Acrylamide has been shown to partially biodegrade in water (Howard, 1989) and on land (Charoenpanich, 2013) within days to a few weeks. For example, 79 to 80% degradation occurred in 6 days in surface soils that were moistened to field capacity (Lande et al., 1979). The biodegradation of this compound is influenced by several physicochemical factors including pH, temperature, concentration, the nature of the microbial strain and microbial growth time (Nawaz et al., 1998; Chandrashekar et al., 2014; Guezennec et al., 2014).

A diverse range of microbial isolates responsible for acrylamide biodegradation have been studied with most belonging to *Bacillus* (Shukor et al., 2009), *Pseudomonas* (Prabu and Thatheyus, 2007; Chandrashekar et al., 2014), and *Rhodococcus* (Nawaz et al., 1998) genera. Other genera/species

shown to possess the ability to degrade acrylamide are *Enterobacter* (Buranasilp and Charoenpanich, 2011), *Xanthomonas maltophilia* (Nawaz et al., 1993), *Ralstonia eutropha* (Cha and Chambliss, 2011), *Geobacillus thermoglucosidasius* (Cha and Chambliss, 2013), *Variovorax boronicumulans* (Jebasingh et al., 2013; Liu et al., 2013), *Stenotrophomonas acidaminiphila* (Lakshmikandan et al., 2014), and most recently, *Arthrobacter* sp. (Bedade and Singhal, 2017).

The acrylamidase enzyme in these aforementioned microbes hydrolyses acrylamide to acrylic acids and free ammonia (Bedade et al., 2018). Numerous aerobic microorganisms have been found to utilize acrylamide as their sole source of carbon and energy (e.g., *Pseudomonas* sp. and *Xanthomonas maltophilia*; Charoenpanich, 2013). However, under anaerobic conditions, acrylamide degradation has been rarely described. *Rhodopseudomonas palustris* has been found able to use acrylamide under photoheterotrophic conditions but grew poorly under anaerobic dark or aerobic conditions (Charoenpanich, 2013). More recently, rapid anaerobic degradation of acrylamide in sludge and water under environmental conditions was reported (Guezennec et al., 2015).

Concentrations:

Maximum concentration range in HF fluid (% mass): 0.00138-0.03430

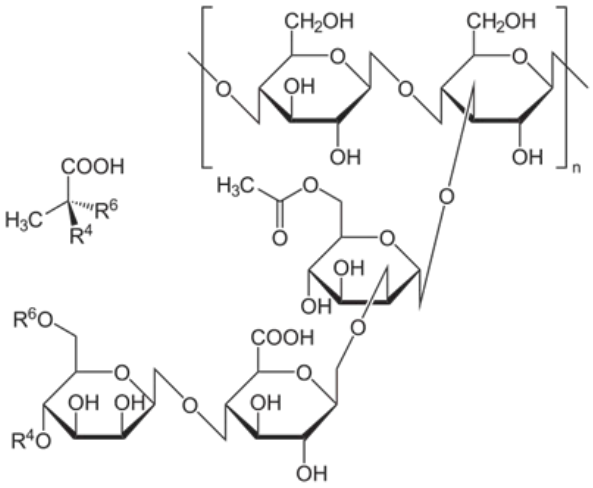
Average concentration range in HF fluid (% mass): 0.01548

Table 5 Acrylamide concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Description	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Solberg 31-2WH (North Dakota)	Econo-FR400	RockPile Energy	Acrylamide and Acrylic Acid	30	0.00672
Rufus Garrett A3 (Texas)	FRW-15A	BHI	Copolymer of Acrylamide and Sodium Acrylate	40	0.01257
University Amanda #3 (Texas)	WFR-4	Universal Pressure Pumping, Inc.	Copolymer of acrylamide and sodium acrylate	34	0.00449
University 31-2H (Texas)			Copolymer of acrylamide and sodium acrylate	33	0.02812
UL MS HZ BLK 6 Unit 4106 (Texas)			Copolymer of acrylamide and sodium acrylate	33	0.02615
RK-UTL 3031B-17 1H (Texas)			Copolymer of acrylamide and sodium acrylate	30	0.03010
University 41-01-DH (Texas)	Trade Secret		Acrylamide Polymer	30	0.01816

University 3-35 #104HB (Texas)			Acrylamide/ammonium acrylate copolymer (26100-47-0)	0.12812	0.01276
University 52-7 #1HC (Texas)	WFR-4	Universal Pressure Pumping, Inc.	Copolymer of acrylamide and sodium acrylate	34	0.03430
University FN 3731	FR-601	SNF	Copolymer of acrylamide and sodium acrylate	10	0.00553
FULLERTON CLEARFORK 2341H (Texas)			Acrylamide/ammonium acrylate copolymer (26100-47-0)	0.04295	0.00548
University 43-15 # 7H (Texas)			Acrylamide (79-06-1)		0.00138

4.5 Xanthan gum ($C_{35}H_{49}O_{29}$ (monomer); CAS # 11138-66-2)



Function: Viscosity management

Degradation: Disagreement exists regarding the resistance of xanthan gum against biodegradation with some (Cadmus et al., 1982; Hou et al., 1986; Qian et al., 2007; Li et al., 2008) regarding it as a highly stable biopolymer only to be completely degraded by a few microorganisms, while others (Bragg et al., 1983; McInerney et al., 2005) noted it to be more ubiquitously biodegraded, particularly in oil fields. Regardless, only few enzyme systems have been reported that hydrolyse xanthan gum (Cadmus et al., 1989; Ahlgren, 1993; Ruijssenaars et al., 1999; Lui et al., 2005). Initiation of xanthan gum degradation is conducted by secreted enzymes, depolymerizing the polysaccharide. The key enzyme is xanthanase (endo-β-d-glucanase), catalyzing the hydrolysis of the cellulosic backbone and thereby reducing the viscosity of xanthan solution (Sutherland, 1987; Hashimoto et al., 1998; Chen et al., 2013). Other enzymes include xanthan lyase, which eliminates

the terminal mannose residue from the side chains of the biopolymer (Ahlgren, 1991; Hasimoto et al., 1998), cellulases which can partially degrade the biopolymer in an unordered fashion (Rinaudo and Milas, 1980), and even salt tolerant, heat-stable xanthanase, functional in brines up to 65°C (Cadmus et al., 1989). Such enzymes were shown not to be inhibited by anoxic conditions and various chemicals and biocides used in enhanced oil recovery operations (Hou et al., 1986). Cultures producing extracellular hydrolytic enzymes degrading xanthan include a salt tolerant *Bacillus* sp. K11 (Cadmus et al., 1982), Bacillus sp., a Corynobacterium sp. (Sutherland, 1987), *Bacillus* sp. GL1 (Hashimoto et al., 1998), *Paenibacillus alginolyticus* XL-1 (Ruijssenaars et al., 1999), *Cellulomonas* sp. LX (Liu et al., 2005), *Microbacterium* sp. strain XT11 (Qian et al., 2007), and *Enterobacter* sp. nov. LB37 (Chen et al., 2013).

Concentrations:

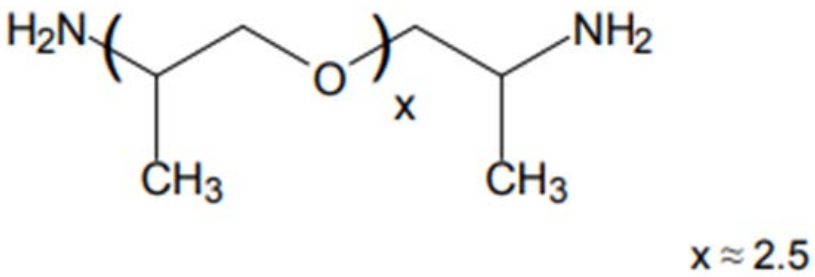
Maximum concentration range in HF fluid (% mass): 0.00001-0.01261

Average concentration range in HF fluid (% mass): 0.005485

Table 6 Xanthan gum concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Nelson A 7H (Texas)		Schlumberger		0.10312	0.00846
ZLS 7H (Texas)		Schlumberger		0.05615	0.00777
Ruppert A 4H (Texas)		Schlumberger		0.10106	0.00859
Bodogginz Unit A4 (Texas)		Schlumberger		0.05840	0.01012
Hammel 44-8TFH (North Dakota)		Schlumberger		0.03726	0.00699
Buehring Unit AC 2H (Texas)		Schlumberger		0.10017	0.01261
Weikel A 6H (Texas)				0.20000	0.00019
North Dollarhide Unit 510H (Texas)				1.00000	0.00010
Cloyce Clark #19-4 (Louisiana)				0.20000	0.00001
Mary Ann 1407 2-8WH (Oklahoma)				0.20000	0.00001

4.6 Polyoxypropylene diamine (Jeffamine D-230; CAS # 9046-10-0)

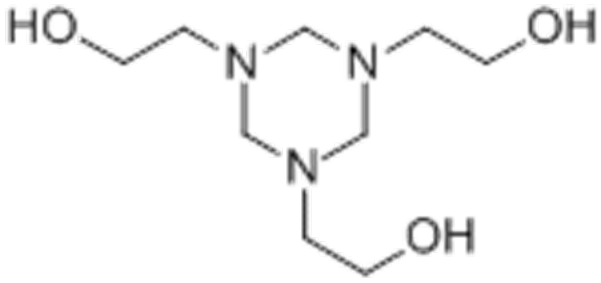


Function: Pipework/Epoxy resins

Degradation: Very little information on the degradability of this compound has been found in the literature. Dow Chemical Company (2015a) noted that this compound is not readily biodegradable according to OECD/EEC guidelines. Experiments conducted in accordance to OECD Test Guideline 301B resulted in 0% biodegradation of this compound over an exposure time of 28 days (Dow Chemical Company, 2015a).

Concentrations: Could not find concentration levels for this compound. However, clay stabilizers typically occupy 0.05 % of hydraulic fracturing fluid (NYSDEC, 2009).

4.7 Hexahydro- 1,3,5-tris(2-hydroxyethyl)-sym-triazine ($C_9H_{21}N_3O_3$; CAS # 4719-04-4)



Function: Biocide

Degradation: The rate of hydrolysis of this compound was shown to be strongly dependent on the pH of the aqueous solution (Bakke et al. 2001). Little information was found on the biodegradability of this compound in the literature. Available data (Angus Chemical Co., 1997) indicate that the compound at 50 mg/L concentration or less should readily biodegrade.

Concentrations:

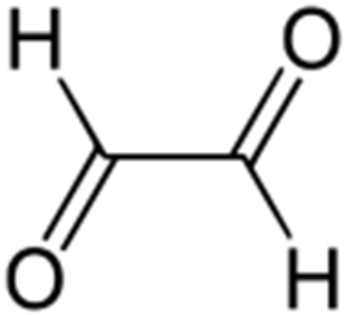
Maximum concentration range in HF fluid (% mass): 0.01973-0.07938

Average concentration range in HF fluid (% mass): 0.04294

Table 7 Hexahydro- 1,3,5-tris(2-hydroxyethyl)-sym-triazine concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Barstow 11-7 (Texas)	ICI-1002	FTSI	Hydrogen Sulfide Scavenger	80	0.07938
STO SEC 2 #212 (Texas)	ICI-1002	FTSI	Hydrogen Sulfide Scavenger	80	0.01973
Sugg Farmer 13-4 (Texas)	ICI-1002	FTSI	Hydrogen Sulfide Scavenger	80	0.02971

4.8 Glyoxal ($C_2H_2O_2$; Oxaldehyde; CAS # 107-22-2)



Function: Crosslinker/ Drilling additive

Degradation: Glyoxal is rapidly converted by abiotic processes, such as photochemically produced hydroxyl radicals with a lifetime of 1.1 days (Atkinson, 2000). The compound is also readily biodegraded and transformed by bacteria and fungi. For example, glyoxal was readily biodegradable by 65% of biochemical oxygen demand (BOD) at an incubation for 14 days (MITI, 1992). Similarly, in a different study, >70% was eliminated in 7 days (Hoechst AG, 1991). Conway et al. (1983) and Gerike and Gode (1990) established an inhibition limit of glyoxal biodegradation of 500 mg/litre.

Numerous microbial enzymes can catalyse the transformation of glyoxal to common intermediates. Efficient transformation can be achieved from glyoxal to glycolaldehyde by glyoxal reductase from *Bacillus subtilis* (Sakai et al., 2001) or oxidized by enzymes such as the fungal glyoxal oxidase (Kersten, 1990) and its bacterial counterpart (Whittaker et al., 1999) to yield glyoxylic acid, while the glyoxalase system (Cooper, 1984) should yield glycolate. Glyoxal has also been shown to react

with arginine, leading to the formation of 1-(4-amino-4-carboxybutyl)-2-imino-5-oxo-imidazolidine (Schwarzenbolz et al., 1997). Therefore, arginine residues in proteins can act as scavengers for glyoxal. It has also been shown that glyoxal can oxidize amino acids such as phenylalanine to numerous products such as Strecker aldehydes and O- and N-heterocycles (Adamiec et al., 2001), as well as amides from amino acids (Glomb and Lang, 2001; Glomb and Pfahler, 2001).

Concentrations:

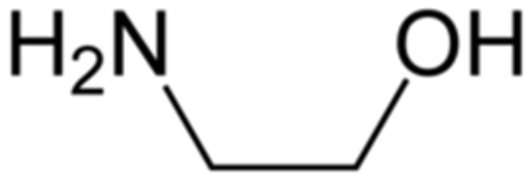
Maximum concentration range in HF fluid (% mass): 0.00783-0.11040

Average concentration range in HF fluid (% mass): 0.04101

Table 8 Glyoxal concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Shinnery 1 Federal 1 (New Mexico)	XLW-24	Baker Hughes	Crosslinker	10	0.01089
C-211 (California)	XLW-56, 330 gal tote	Baker Hughes	Crosslinker	30	0.09706
Txl South Unit 5721 (Texas)	XLW-24	Baker Hughes	Crosslinker	10	0.00783
Woodley F L 92 (Texas)	XLW-24	Baker Hughes	Crosslinker	10	0.00854
Drinkard Northeast Unit 851 (New Mexico)	XLW-24, tote	Baker Hughes	Crosslinker	10	0.01135
363-M-18 (California)	XLW-56, 330 gal tote	Baker Hughes	Crosslinker	30	0.11040

4.9 2-Aminoethanol (C_2H_7NO; 2-Aminoethan-1-ol; CAS # 141-43-5)



Function: Drilling additive

Degradation: In the atmosphere, 2-aminoethanol is expected to exist almost entirely in the vapour phase (Fisher Scientific, 2008). Reactions through photochemically generated hydroxyl radicals is predicted to be the main removal mechanism with a half-life of 4 hours (Fisher Scientific, 2008).

The biodegradability of 2-aminoethanol under both aerobic and anaerobic conditions by several bacteria with different metabolic pathways has been previously demonstrated (e.g., Narrod and Jakoby, 1964; Scarlett and Turner, 1976; Frings et al. 1994; Knapp et al., 1996; Ndegwa et al., 2004). This compound has been shown to biodegrade in both water and soil (e.g., Chong 1994; Emtiazi and Knapp 1994) and soil (e.g., Lee and Portier, 1999; Ndegwa et al., 2004) under aerobic and anaerobic conditions, creating a large reservoir of ammonium, ethanol, and acetic acid. For example, in soil, 2-aminoethanol is biodegraded through hydrolysis to ammonium and acetaldehyde, which, under aerobic conditions, oxidized to nitrite and then nitrate, and to ethanol and acetic acid, respectively (Ndegwa et al., 2004). While previous work on bio-slurries indicated that 2-aminoethanol concentrations exceeding 1500 mg/kg inhibited in-situ biodegradation (Sorensen et al., 1997), this was later shown by soil studies not to be the case (Ndegwa et al., 2004). Sorption of this compound onto clay minerals may limit biodegradation with studies showing that 2-aminoethanol biodegrading faster in the water than in soil (Mrklas et al., 2004). Expected half-life of 2-aminoethanol is on the order of days to weeks (Fisher Scientific, 2008).

Concentrations:

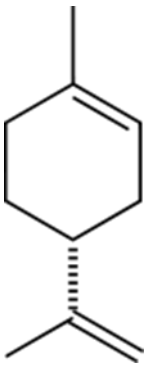
Maximum concentration range in HF fluid (% mass): 0.00065-0.00378

Average concentration range in HF fluid (% mass): 0.00128

Table 9 2-Aminoethanol concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Hershey 32-29-1H (Oklahoma)	GN-532	EES		50	0.00121
McGuffin 2-19H (Oklahoma)	SSX-1	EES	Solid H2S Scavenger	50	0.00065
Irwin 2-4H (Oklahoma)	SSX-1	EES	Solid H2S Scavenger	50	0.00071
TEDDY 013625 #7H (Oklahoma)				5	0.00378
Schenk 3-17 HXL (Oklahoma)	SSX-1	EES	Solid H2S Scavenger	50	0.00069
Schenk 2-17 HXL (Oklahoma)	SSX-1	EES	Solid H2S Scavenger	50	0.00065

4.10 Limonene ($C_{10}H_{16}$; 1-Methyl-4-(prop-1-en-2-yl)cyclohex-1-ene; CAS # 138-86-3)



Function: Surfactant

Degradation: Hydrolysis of limonene is not expected due to a lack of functional groups for hydrolysis and the cyclohexene ring and ethylene groups being resistant to this degradation process (US-EPA, 1994). However, limonene is expected to rapidly undergo reactions with photochemically produced hydroxyl radicals, as well as with ozone, and nitrate radicals. Calculated lifetimes for the reaction of *d*-limonene with photochemically produced hydroxyl radicals range from 0.3 to 2 hours (e.g., Winer et al., 1976; Atkinson and Carter, 1984; Atkinson et al., 1984; Atkinson, 1990).

Aerobic biodegradation of limonene has been previously reported by some microorganisms such as *Penicillium digitatum*, *Corynespora cassiicola*, *Diplodia gossypina*, and a soil strain of *Pseudomonas* sp. (PL strain) (Dhavalikar and Bhattacharayya, 1966; Shulka and Bhattacharayya, 1968; Abraham et al., 1985). Using the standard OECD 301 C Modified MITI Test (I), limonene was found to be readily biodegradable with 41–98% degradation by biochemical oxygen demand in 14 days (MITI, 1992). In another study simulating aerobic sewage treatment using the OECD 303 A test, limonene disappeared almost completely (>93.8%) during 14 days of incubation (Schwartz et al., 1990). However, it is speculated that the limonene in that study also partly disappeared due to volatilization and that the extent of biodegradation could not be determined (Filipsson et al., 1998).

While anaerobic biodegradation of limonene was previously not thought to occur, more recent studies have shown otherwise. Rotaru (2009) demonstrated in a methanogenic enrichment culture limonene consumption with proportional formation of methane. A broad diversity of microorganism was noted to be involved in this anaerobic process.

Concentrations:

Note: All compounds in the table below are d-Limonene (CAS # 94266-47-4)

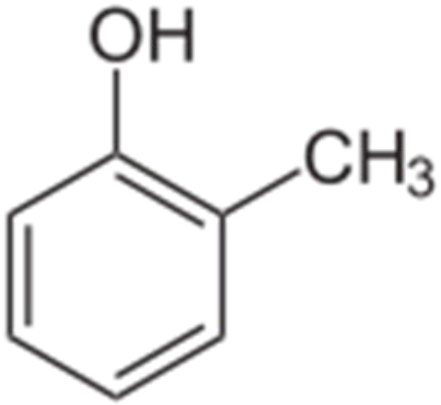
Maximum concentration range in HF fluid (% mass): 0.00001-0.00097

Average concentration range in HF fluid (% mass): 0.000416

Table 10 Limonene concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
University Amanda #3 (Texas)				1	0.00097
DALE UNIT # 1H (Texas)	S-602	UPPI	Surfactant	1	0.00021
ST 40-6 (Texas)	S-602	Universal	Surfactant	1	0.00088
Davidson 22F-18 (Texas)	WT-603	Frac Specialists	Wetting Agent	0.01000	0.00001
Limpia 14A #10 (Texas)	WT-603	Frac Specialists	Wetting Agent	0.01000	0.00001

4.11 2-Methylphenol (C_7H_8O; 2-Methylphenol; CAS # 95-48-7)



Function: Biocide

Degradation: Methylphenols belong to a group of phenolic compounds that are also collectively known as cresols. Such compounds have been shown to rapidly degrade in air with daytime removal dominated by reaction with hydroxyl radicals, while during the night it is dominated by reaction with nitrate radicals (ATSDR, 2008). Using an average tropospheric hydroxyl radical concentration of 5 x 10^5 molecules cm^3, the atmospheric half-live for 2-methylphenol was calculated to be 9.63 hours (Atkinson 1985).

Microbial degradation also play a significant role in the removal of phenol and cresols and are degradable under both oxic and anoxic conditions (Krastanov et al., 2013). Under oxic conditions, these compounds can be hydroxylated to catechol via catalysis with phenol hydroxylase using molecular oxygen as a co-substrate (e.g. Leahy et al., 2003). Under anoxic conditions, by contrast, phenol is initially carboxylated to 4-hydroxybenzoate prior to aromatic ring reduction (e.g. Schühle and Fuchs, 2004).

In biodegradation screening and sewage treatment plant simulation studies, cresol compounds were found to degrade rapidly with half-lives between <24 hours and <7 days (ATSDR, 2008, and references therein). In surface soils, by contrast, half-lives from 46 days to about 1 year were noted (Dobbins and Pfaender, 1988).

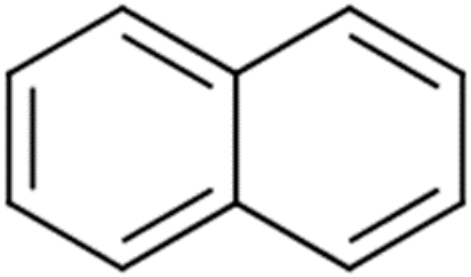
Phenolic compounds can be biodegraded by a range of different microorganisms, with *Pseudomonas* members being commonly involved (e.g. Ahmed et al., 1995; Bastos et al., 2000; Chung et al., 2003). Microbial degradation of phenol compounds at concentrations less than 1200 mg/L have been reported (e.g. Prieto et al., 2002).

Concentrations: Reported concentrations (µg/L) in produced water for two shale formations, presented as: average (minimum-maximum) or median (minimum maximum).

Barnett (TX): 28.3 (5.8 – 76)

Marcellus (MD, NY, OH, PA, VA, WY): 13 (11 – 15) (EPA-USA, 2016)

4.12 Naphthalene ($C_{10}H_8$; Bicyclo[4.4.0]deca-1,3,5,7,9-pentaene; CAS # 91-20-3)



Function: Corrosion inhibitor

Degradation: Polycyclic aromatic hydrocarbons (PAHs), like naphthalene, can undergo photo-oxidation, chemical oxidation and become partly lost by volatilization (Cerniglia, 1992).

Since naphthalene represents the simplest polycylic aromatic hydrocarbon (PAH), it is often used as a model substrate for studies on the metabolism of PAHs (Mallick et al., 2011, and references therein). It was found that the rate of naphthalene biodegradation in soil can be enhanced by the addition of inorganic nutrients and inoculum and that treatment under combined biostimulation and bioaugmentation exhibited the highest degree of biodegradation with the lowest measured half-life time of 10.8 days (Agarry and Oghenejoboh, 2015). Various bacteria have been observed to

degrade naphthalene with numerous unique metabolic pathways documented (e.g., Cerniglia, 1992; Peng et al., 2008; Seo et al., 2009; Mallick et al., 2011). Aerobically, the oxygen serves as the final electron acceptor as well as a co-substrate for the hydroxylation and oxygenolytic aromatic ring cleavage (Ghosal et al., 2016). During anaerobic catabolism, by contrast, reductive reactions are used (Foght, 2008; Carmona et al., 2009). Investigations into anaerobic biodegradation of PAHs have occurred only relatively recently, with only a limited number of preliminary studies existing that demonstrate the anaerobic degradation of PAHs including naphthalene (Foght, 2008; Carmona et al., 2009; Mallick et al., 2011). Furthermore, very little is known on anaerobic degradation of PAHs under sulfate- and nitrate- reducing conditions and this area has been described as an emerging field (Ghosal et al., 2016).

Naphthalene degrading bacteria have been ubiquitously detected in nature (e.g., Cerniglia, 1992; Peng et al., 2008; Seo et al., 2009; Mallick et al., 2011) and the naphthalene catabolic genes present in the plasmid NAH7 in *Pseudomonasputida* G7 are well characterized (Simon et al., 1993). Such genes have also been identified in *Ralstonia sp.* U2, *Comamonas testeroni* strain GZ42, sphingomonads, *Rhodococcus* strains, *Marinobacter hydrocarbonoclasticus*, and various other *Pseudomonas* strains like *P. putida* G7, NCIB 9816-4, ND6, and *P. stutzeri* AN10 (Ghosal et al., 2016, and references therein).

In addition, microalgae (e.g., Cerniglia et al., 1979) and numerous fungal species have also been reported to metabolize different PAHs (e.g., Cerniglia, 1992). Most fungi cannot use PAHs as sole sources of carbon and energy they may co-metabolize PAHs to a wide variety of oxidized products and sometimes to CO_2 (Ghosal et al., 2016; Cerniglia and Sutherland, 2010). While bacterial PAHs degradation mainly involves dioxygenase enzymes and partially monooxygenase mediated reactions, fungal PAHs degradation mainly involves monooxygenase enzymes (Cerniglia and Sutherland, 2010, and references therein).

Concentrations:

Maximum concentration range in HF fluid (% mass): 0.00003-0.00386

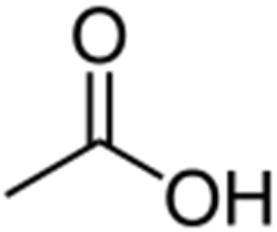
Average concentration range in HF fluid (% mass): 0.000926

Table 11 Naphthalene concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Wright 4-33 #1H (North Dakota)	LoSurf-300D	Halliburton	Surfactant	5	0.00386
RK-UTL 3031B-17 1H (Texas)				5	0.00014
UTL 2932-17 1H (Texas)				5	0.00032

FULLERTON CLEARFORK 2341H (Texas)				0.00216	0.00028
Larson A23-672 (Colorado)				5	0.00003

4.13 Acetic Acid ($C_2H_4O_2$; Ethanoic acid; CAS # 64-19-7)



Function: Buffer, stabiliser, solvent

Degradation: Vapour phase acetic acid in the atmosphere can react with photochemically produced hydroxyl radicals, resulting in an estimated half-life of 26.7 days (Science Company, 2009). In water or soil, acetic acid has been found to biodegrade readily. In water, biodegradation has been measured as 63 to 81% BOD (Anchem, 2016). Similarly, 75% degradation was reported in 14 days using garden soil as an inoculum (Kool, 1984).

Concentrations:

Maximum concentration range in HF fluid (% mass): 0.00003-0.01485

Average concentration range in HF fluid (% mass): 0.00182

Table 12 Acetic acid concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Vinson Fee F5 (Texas)	Ferriplex 66	Chemplex	Iron Control	50	0.00052
University Amanda #3 (Texas)				82	0.00220
University 31-2H (Texas)				80	0.00039

UL MS HZ BLK 6 Unit 4106 (Texas)				5	0.00011
RK-UTL 3031B-17 1H (Texas)				1	0.00003
University 41-01-DH (Texas)				5	0.00013
UTL 2932-17 1H (Texas)				1	0.00006
University 3-35 #104HB (Texas)				0.00780	0.00078
University 52-7 #1HC (Texas)				82	0.00069
University FN 3731 (Texas)	BF-125	Cudd	Buffers / Ph Control	6.4	0.00029
University Red 5031B (Texas)	IC5-Iron Control	Solnexus	Iron Control	60	0.00182
FULLERTON CLEARFORK 2341H (Texas)				0.11630	0.01485

4.14 Alcohols, C_{6-12} ethoxylated (CAS # 68439-45-2)

Function: Surfactant

Degradation: Linear alcohol ethoxylate homologs are identified by using the convention C_xE_y, where x is the total carbon number in the alkyl chain and y is the number of ethoxylate groups (e.g., Itrich and Federle, 2004). Collectively, alcohol ethoxylates experience rapid biodegradation under both laboratory and field conditions (e.g., Talmage 1994). The biodegradation of these compounds is ultimately favoured by linearity of the hydrophobic chain and the shortness of the ethoxylate chain (Swisher, 1987). For example, Larson and Games (1981) noted that for the $C_{12}E_9$ homolog, the alkyl chain was converted to CO_2 more than twice as fast as the ethoxylate moiety. Three different biodegradation pathways have been identified, with the most common one proceeding by cleavage of the ether bond that connects the hydrophobic chain to the ethoxylate chain (Swisher, 1987). Another pathway is initiated by omega oxidation of the terminal carbon atom of the alkyl chain followed by inward biodegradation involving beta oxidation. Alternatively, biodegradation can be

initiated hydrolytic or oxidative attack of the hydrophilic portion (Swisher, 1987). Experimental conditions and the complexity of the hydrophobic portion appears to affect the mode of attack (Marcomini, 1999). While past research has demonstrated the occurrence of alcohol ethoxylate biodegradation under aerobic conditions, more recent work by Traverso-Soto et al. (2016) also observed degradation under anaerobic conditions.

Concentrations:

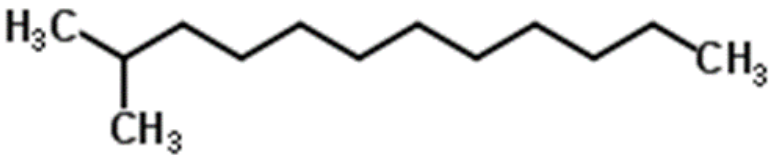
Maximum concentration range in HF fluid (% mass): 0.00252-0.02542

Average concentration range in HF fluid (% mass): 0.013845

Table 13 Alcohols, C_{6-12} ethoxylated concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Jerry 2-8H (North Dakota)	Surf 601	ChemRock	Surfactant	30	0.01641
Edmondson A U29-1H - 301087 (Texas)	SURF 601	Keane	Surfactant	30	0.02542
Jerry 6-8H1 (North Dakota)	Surf 601	ChemRock	Surfactant	30	0.00252
Poplar Gap 52 (Virginia)	Flomax 50	Universal Well Services, Inc.	Surfactant	15	0.00359
Angelina SN 33-28/Well No. 201H (Texas)	Surf 601	Chem Rock	Surfactant and Flow Back Aid	30	0.01902
HERNDON 102H (Texas)	PTLST601	PTL	SURFACTANT	30	0.01611

4.15 Alkanes, C_{12-26} branched and linear (CAS # 90622-53-0)



Function: Surfactant

Degradation: In the atmosphere, alkanes have been observed to completely photodegrade within less than 28 days (Mathys, 2017). Alkane biodegradation has been reported under both aerobic conditions, with oxygen serving as the electron acceptor, and under anaerobic conditions with sulfate and nitrite accepting electrons in order to complete the process (Ji et al., 2013). The linear *n*-alkanes degrade faster than their branched counterparts. Using the OECD 301B and OECD 301F guidelines, > 80% alkanes have been biodegraded within 28 days (Mathys, 2017). Different enzymatic systems are involved in the biodegradation of alkanes. Aerobically, alkane degradation reaction is initiated by oxygenases that introduce oxygen atom(s) into alkane substrates (e.g., Coon, 2005; Li et al., 2008; Ji et al., 2013). Alkane oxidation pathways have been observed in different bacteria such *Pseudomonas aeruginosa* (Forney and Markovetz, 1970), *Gordonia* sp. strain TY-5 (Kotani et al., 2003), and *Acinetobacter* sp. strain HO1-N (Finnerty, 1988; Maeng et al., 1996). Anaerobically, two mechanisms of alkane degradation have been observed. One involves the fumarate addition pathway and the other the carboxylation pathway (Ji et al., 2013). Microorganisms that were studied under such conditions include the sulfate-reducing bacterial strain Pnd3 (Aeckersberg et al., 1998), strain AK-01 (So and Young, 1999), strain Hxd3 (So et al., 2003); strain CV2803T (Cravo-Laureau et al., 2005), and the denitrifying bacterial strain HxN1 (Rabus et al., 2001). Furthermore, anaerobic biodegradation of *n*-alkanes with bacterial enrichment culture has also been demonstrated (Kropp et al., 2000; Callaghan et al., 2006, 2009).

Concentrations: Note: Could not find concentrations for C12-26. However, was able to find ranges for C_{10-24} (branched and linear; CAS # 848301-67-7). These are tabulated below.

Maximum concentration range in HF fluid (% mass): 0.00011-0.05244

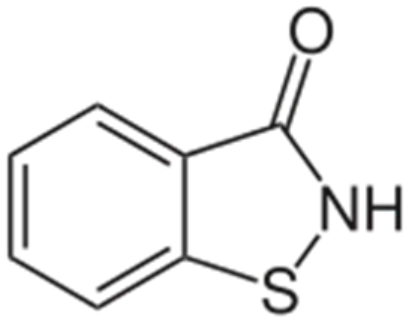
Average concentration range in HF fluid (% mass): 0.02204

Table 14 Alkanes, C_{10-24} branched and linear concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
GORILLA 20 16H (Texas)	SV-1	C&J Well Services	Paraffin & Scale Additives	90	0.00011
Clark 33-2 #1 (Alabama)	SV-1	C&J Well Services	Paraffin & Scale Additives	90	0.02827
MUSTANG 3411 (Texas)	SV-1	C&J Well Services	Paraffin & Scale Additives	90	0.04983
MABEE 140B #4803 (Texas)	SV-1	C&J Well Services	Paraffin & Scale Additives	90	0.05244
Statler 10H (West Virginia)	SV-1	C&J Well Services	Paraffin & Scale	90	0.00049

			Additives		
ELAND STATE 14 15H (Texas)	SV-1	C&J Well Services	Paraffin & Scale Additives	90	0.00153
JACKAL STATE 28 13H (Texas)	SV-1	C&J Well Services	Paraffin & Scale Additives	90	0.02162

4.16 Benzisothiazolinone ($C_7H_5NO_S$; 1,2-benzisothiazol-3(2H)-one; CAS # 2634-33-5)



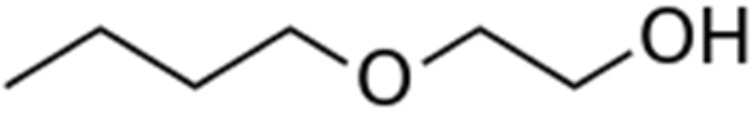
Function: Biocide

Degradation: In the atmosphere, vapour-phase benzisothiazolinone will be degraded by reaction with photochemically-produced hydroxyl radicals, exhibiting a half-life of 23 days (NCBI, 2005). The compound is susceptible to photolysis under aqueous conditions and may photodegrade (NCBI, 2005).

Benzisothiazolinone has been reported to readily biodegrade in aerobic soil with a half-life of less than 24 hours in one sandy loam soil (US-EPA/OPPTS, 2005). Likewise, rapid biodegradation of this compound was observed in a batch adsorption study with secondary sewage sludge (Wick et al., 2011). However, using the Japanese MITI test and a 100 ppm benzisothiazolinone concentration, 0% of theoretical BOD was measured using activated sludge (NITE, 2015). It can be speculated whether the high benzisothiazolinone concentration of 100 ppm may have been toxic to the microorganisms (NCBI, 2005).

Concentrations: Concentrations for this compound are reported as <0.00000 for a range of wells

4.17 2-Butoxyethanol ($C_6H_{14}O_2$; 2-Butoxyethan-1-ol; CAS # 111-76-2)



Function: Surfactant

Degradation: Since 2-butoxyethanol have low potential for bioconcentration in aquatic organisms, and do not adsorb to suspended solids and sediments, they are expected to biodegrade rapidly in aquatic environment (Lyman, 1990). However, recent work by Rogers et al. (2017) demonstrated that this compound can be both mobile and persistent in groundwater under a range of redox conditions. This observation is consistent with detections of this compound in groundwater field samples (Rodgers et al., 2017, and references therein).

Concentrations:

Maximum concentration range in HF fluid (% mass): 0.00011-0.00751

Average concentration range in HF fluid (% mass): 0.00236

Table 15 2-Butoxyethanol concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Goodall USA #11-29H (N.A.)	Inflo 250W	BHI	Surfactants	20	0.00751
University 31-2H (Texas)				10	0.00014
UL MS HZ BLK 6 Unit 4106 (Texas)				10	0.00011
University 41-01-DH (Texas)				10	0.00472
Wapiti 12-17 #4H (Texas)				15	0.00100
University 43-15 # 7H (Texas)	WAI-251LC	WFT	Inhibitor	10	0.00067

4.18 Diethylene glycol ethyl ether ($C_6H_{14}O_3$; 2-(2-Ethoxyethoxy)ethanol; CAS # 111-90-0)

Function: Solvent

Degradation: Vapour-phase diethylene glycol monoethyl ether degrades in the atmosphere by reacting with photochemically-produced hydroxyl radicals. The half-life for this reaction in air is estimated to be 6.7 hours (Meylan and Howard, 1993; Aschmann et al., 2001; TOXNET, 2007). Diethylene glycol ethyl ether is not expected to undergo hydrolysis or direct photolysis in the environment since alcohols and ethers are generally resistant to hydrolysis and do not absorb UV light at environmentally significant wavelengths (Silverstein and Bassler, 1963; Lyman et al., 1990). While diethylene glycol ethyl ether in soil is expected to have very high mobility based upon an estimated K_{oc} of 12 (Lyman et al., 1990), volatilization from moist soil surfaces is not expected to be important due to an estimated Henry's Law constant of 2.2 x 10^{-8} atm-cu m/mole (Swann et al., 1983).

Aerobically, biological screening studies indicate that diethylene glycol ethyl ether should biodegrade rapidly in soil and water following an acclimation period (e.g., Bogan and Sawyer, 1955; Price et al., 1974; Bridie et al., 1979; Zahn and Wellens, 1980; Dow Chemical Company, 1981; TOXNET, 2007). For example, a 5 day BOD test at 20°C showed a 34.3% loss after 16 days of acclimation (Bogan and Sawyer, 1955). Incubation of diethylene glycol ethyl ether without an acclimation period for 5, 10, and 20 days resulted in BOD values, measured in % theoretical, of 5, 31, and 48, respectively (Dow Chemical Company, 1981). Inoculated with wastewater, this compound was bio-oxidized 17, 71, 75, and 87% following 5, 10, 15, and 20 days incubation, respectively (Price et al., 1974). In seawater, diethylene glycol ethyl ether was bio-oxidized 11, 44, 57, and 70% in 5, 10, 15, and 20 days, respectively (Price et al., 1974). >90% loss of 400 ppm diethylene glycol ethyl ether (according to the Zahn-Wellens screening method) occurred in 28 days (Zahn and Wellens, 1980). A study using activated sludge gave a degradation rate for diethylene glycol monoethyl ether of 0.18/hour (Cowan and Kwon, 1999).

Concentrations:

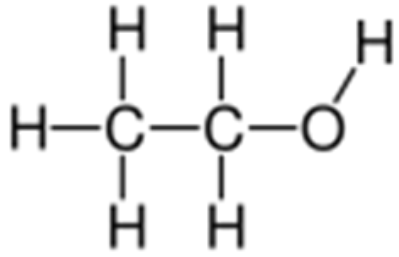
Maximum concentration range in HF fluid (% mass): 0.00002-0.00003

Average concentration range in HF fluid (% mass): 0.000025

Table 16 Diethylene glycol ethyl ether concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Description	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
UL MS HZ BLK 6 Unit 4106 (Texas)			Diethylene glycol (111-46-6)	1	0.00002
University 41-01-DH (Texas)			Diethylene glycol (111-46-6)	1	0.00003

4.19 Ethanol (C_2H_6O; CAS # 64-17-5)



Function: Surfactant

Degradation: Biodegradation is regarded as the main method of removal of ethanol from water, since it is stable to hydrolysis and reaction with hydroxyl radicals is unlikely to be a significant process (Anbar, 1967). The atmospheric half-life of ethanol affected by photodegradation is estimated to be 2.99 days (Dow Chemical Company, 2015b). Ethanol is considered to be readily biodegradable as it passes OECD test(s) for ready biodegradability. Tests conducted according to OECD Test Guideline 301D or equivalent demonstrated >70% biodegradation over a 5 day exposure period (Dow Chemical Company, 2015b). Similarly, under aerobic conditions using adapted wastewater from domestic sewage, degradation was observed to be 74% after 5 days rising to 95% by day 15 (Price, 1974). Tests conducted under the MITI protocol demonstrated degradation of 89% ethanol after 14 days and >70% after 10 days (OECD SIDS, 2004) and >90% within 10 days (Birch, 1991). In anaerobic conditions, the rate of biodegradation was calculated to be 17.9 ppm ethanol/day with a total methane recovery of 91% of the theoretical limit (Suflita, 1993).

Concentrations:

Maximum concentration range in HF fluid (% mass): 0.00002-0.04638

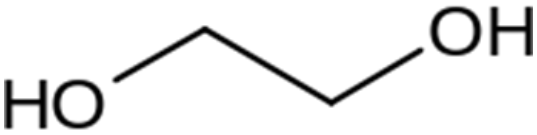
Average concentration range in HF fluid (% mass): 0.00688

Table 17 Ethanol concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Wright 4-33 #1H (North Dakota)	LoSurf-300D	Halliburton	Surfactant	60	0.04638
Vinson Fee F5 (Texas)	*Antimicrobial 220*	Frac-Chem	Biocide	3	0.00045
JEFFRESS 3H (Texas)	Bactron K-87 Microbiocide	CHAMPION TECHNOLOGIES INC	Anti-Bacterial Agent	1	0.00022

JEFFRESS 3H (Texas)	MC B 8642 WS	MULTI-CHEM GROUP LLC	Anti-Bacterial Agent	1	0.00012
University Amanda #3 (Texas)	BIO-15G	Universal Pressure Pumping, Inc.	BIOCIDES	3	0.00068
UL 21 Bighorn 1H (Texas)				1	0.00029
FULLERTON CLEARFORK 2341H (Texas)				0.00013	0.00002

4.20 Ethylene glycol ($C_2H_6O_2$; Ethane-1,2-diol; CAS # 107-21-1)



Function: Viscosity management

Degradation: Ethylene glycol is not expected to undergo photodegradation (Freitag et al., 1985) and hydrolysis (Lyman et al., 1982) but will undergo breakdown with hydroxyl radicals (Howard et al., 1991; Nielsen et al., 1993). Estimated half-life of this compound in the atmosphere for reaction with hydroxyl radicals can range from 1 day (Nielsen et al., 1993) to 3.5 days (Howard et al., 1991).

Standard biodegradation tests indicate that ethylene glycol is readily biodegradable and several strains of microorganisms capable of utilizing this compound as a carbon source have been identified (Dobson, 2000). For example, Haines and Alexander (1975) identified a soil bacterium (*Pseudomonas aeruginosa*) capable of degrading ethylene glycol. Watson and Jones (1977) identified *Acinetobacter* and *Pseudomonas* strains that degraded ethylene glycol. Under strongly aerobic conditions, *Flavobacterium* sp. was also observed to convert ethylene glycol to glycolate and eventually carbon dioxide (Willetts, 1981).

Biodegradation of ethylene glycol can occur under both aerobic and anaerobic conditions with some studies suggesting a lag phase before degradation, while other studies not reporting this to occur. As noted by Staples et al. (2001), the extent of ethylene glycol primary degradation under aerobic conditions depends on the microbes used, test duration, nutrient composition and the specific methods (i.e. protocol tests) used.

The biodegradation of ethylene glycol under aerobic conditions can occur relatively rapidly under range of different environmental conditions. Means and Anderson (1981) measured biodegradation of this compound in five different tests using various aqueous media. Ethylene glycol was readily degraded in all tests with a lag period of up to 3 days and degradation to 10% or less of the starting

concentration reported in all tests after between 1 and 21 days. Pitter (1976), by contrast, reported 96.8% removal of ethylene glycol within 120 h using adapted activated sewage sludge. Similarly, Zahn and Wellens (1980) reported >90% degradation after 4 days incubation of ethylene glycol in a batch biodegradability study with no observed lag period. Bridie et al. (1979) observed 63% degradation of this compound as BOD after 5 days.

Price et al. (1974), who assessed the biodegradation of ethylene glycol in both fresh and salt water over a 20-day incubation period, observed biodegradation of ethylene glycol over several weeks. In fresh water, the authors observed 34% degradation after 5 days, rising to 86% by day 10 and 100% by day 20. Degradation rates were noted to be less in salt water with 20% degradation after 5 days and 77% after 20 days. In a different study, Boatman et al. (1986) used acclimated sewage sludge as inoculum and ethylene glycol at a concentration of ~20 mg carbon/litre. They reported an estimated lag period of 8–10 days since no significant biodegradation occur until day 14 of the test. However, by day 21, 71% of the ethylene glycol was reported to be degraded.

McGahey and Bouwer (1992) observed ethylene glycol degradation using natural groundwater and soil inocula. An initial glycol concentration of 111 mg/litre was degraded in groundwater with a rate constant of 0.76/day at 25 °C, a lag period < 3 days, and an estimated half-life of 22 h. Ethylene glycol degradation was also assessed in sandy loam soil and sandy silt soil at rates of 1.01 and 2.90/day, respectively (McGahey and Bouwer, 1992). An increase in the ethylene glycol concentration to 10 000 mg/litre in the sandy loam resulted in a minimal degradation of the glycol. A reduction in the temperature of the sandy silt inoculum from 25°C to 10°C resulted in a concomitant increase in the half-life of ethylene glycol from 6 to 14 h (McGahey and Bouwer, 1992).

Under anaerobic conditions, ethylene glycol was also observed to be biodegraded. For example, non-adapted strains of *Acetobacter* could degrade ethylene glycol under anaerobic conditions at concentrations between 5 and 15 g/litre using the compound as sole carbon source (Kaushal and Walker, 1951; Hrotmatka and Polesofsky, 1962). Gaston and Stadtman (1963) isolated the anaerobic bacterium *Clostridium glycolicum* from pond ooze and adapted to ethylene glycol could degrade 5.3 or 6.7 g ethylene glycol/litre under anaerobic conditions.

Concentrations:

Maximum concentration range in HF fluid (% mass): 0.00205-0.06840

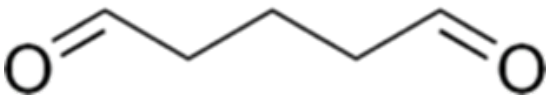
Average concentration range in HF fluid (% mass): 0.01405

Table 18 Ethylene glycol concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Solberg 31-2WH (North Dakota)	Ecopol-EC101	RockPile Energy	Crosslinker	55	0.01927
Wright 4-33	WSI 3607	JACAM	Scale Inhibitor	100	0.06840

#1H (North Dakota)					
Wright 4-33 #1H (North Dakota)	WOS 1N	JACAM	Oxygen Scavenger	100	0.00205
JEFFRESS 3H (Texas)	MC S-2510T (WS)	MULTI-CHE M GROUP LLC	Scale Inhibitor	60	0.00581
Zogol 2H (West Virginia)	EC 6486A	Universal	Scale Inhibitor	30	0.007
University Amanda #3 (Texas)	CX-9	Universal Pressure Pumping, Inc.	CROSSLINKERS AND DELAYERS	10	0.00980
University 31-2H (Texas)				13	0.00248
UL MS HZ BLK 6 Unit 4106 (Texas)				13	0.00499
University 41-01-DH (Texas)				10	0.00307
Wapiti 12-17 #4H (Texas)				40	0.00267
University 3-35 #104HB (Texas)				0.27025	0.02692
University FN 3731 (Texas)	XL-335	ASK	Crosslinkers	25	0.01619

4.21 Glutaraldehyde ($C_5H_8O_2$; 1,5-Pentanedial; CAS # 111-30-8)



Function: Biocide

Degradation: Glutaraldehyde degradation includes abiotic hydrolysis and photolysis, as well as metabolism by aerobic and anaerobic microorganisms (Leung, 2001). A filter-sterilized aqueous solution of glutaraldehyde was observed to be stable in the dark at 20°C for 28 days. At a higher temperature (50°C), by contrast, some degradation of ~8% was observed after 14 days (SLI, 1994). Glutaraldehyde was also shown to degrade slowly at pH 5 and 7 as a result of hydrolysis in a dark, sterile aqueous solution during 31 days with extrapolated half-lives of 508 and 102 days, respectively. However, at pH 9, appreciable degradation was observed with a half-life of 46 days (PTRL, 1992a). The photodegradation of glutaraldehyde was also examined in a sterile aqueous

solutions at pH 5 (PTRL, 1992b). Glutaraldehyde degradation was shown to be slow under these experimental conditions with a half-live 196 days. Based on these studies, Leung (2001) concluded that aqueous solutions of glutaraldehyde are stable at acidic to neutral pH, room temperature and sunlight. It appears unstable at alkaline pH and elevated temperatures.

Glutaraldehyde is demonstrated to have a moderate to high rate of aerobic biodegradation - the criteria to classify glutaraldehyde as 'readily biodegradable' (WIL, 2000; Leung, 2001). A variety of screening test methods have been used to examine the aerobic biodegradability of glutaraldehyde. Details of the test conditions and the results are summarized in Leung (2001), who showed that variable glutaraldehyde biodegradation rates exist and depend on the screening test methods used. For example, high biodegradation rates were observed in the OECD 301A test (83% in 5 days; WIL, 2000), while low rates were observed in the OECD 301B test (0% in 5 days; WIL, 1996). It appears that such results were influenced by the test concentration with higher biodegradability observed when glutaraldehyde concentrations were low than when the concentrations were high (Leung, 2001). In addition, glutaraldehyde reacts with ammonium ions in the test media to yield a product that is more recalcitrant to biodegradation (UCC, 1995).

Concentrations:

Maximum concentration range in HF fluid (% mass): 0.00048-0.00839

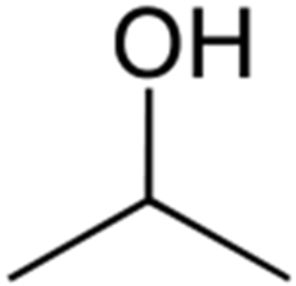
Average concentration range in HF fluid (% mass): 0.00484

Table 19 Glutaraldehyde concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Wright 4-33 #1H (North Dakota)	Biocide 5000	JACAM	Antibacterial	50	0.00277
Vinson Fee F5 (Texas)	Antimicrobial 220	Frac-Chem	Biocide	14	0.00212
JEFFRESS 3H (Texas)	Bactron K-87 Microbiocide	CHAMPION TECHNOLOGIES INC	Anti-Bacterial Agent	30	0.00666
JEFFRESS 3H (Texas)	MC B 8642 WS	MULTI-CHE M GROUP LLC	Anti-Bacterial Agent	60	0.00721
Rufus Garrett A3 (Texas)	X-Cide 150	BHI	Biocide	60	0.00839
University 3-35 #104HB (Texas)				0.07948	0.00792

Wapiti 12-17 #4H (Texas)				1	0.00048
University Amanda #3 (Texas)	BIO-15G	Universal Pressure Pumping, Inc.	BIOCIDES	14	0.00318

4.22 Isopropanol (C_3H_8O; Propan-2-ol; CAS # 67-63-0)



Function: Surfactant

Degradation: Isopropanol may be degraded photochemically and through volatilization and biodegradation processes. This compound is expected to have very high mobility in soils and therefore not expected to adsorb to suspended solids and sediment (Pesticide Research Institute, 2014). Furthermore, since the Henry's Law constant for isopropanol is 8.1 x 10^{-6} atm · m^3/mol, volatilization from moist soil and water surfaces is likely to be an important fate process. Based on its relatively high vapour pressure, isopropanol may also volatilize from dry soil surfaces and water bodies (Pesticide Research Institute, 2014), with calculated volatilization half-lives for a model river and lake being 86 hours and 29 days, respectively (HSDB, 2012).

Due to its relatively high vapour pressure of 45 mm Hg at 25 °C, isopropanol will exist as a vapour in the atmosphere, where it is subject to relatively rapid oxidation predominantly by photochemically-produced hydroxyl radicals (Pesticide Research Institute, 2014). Half-lives of nine hours to five days have been determined for hydroxyl radical-mediated photodegradation (Pesticide Research Institute, 2014).

Isopropanol is rapidly biodegraded in both aerobic and anaerobic conditions and reported to occur in pure or mixed microbial cultures (e.g., Bustard et al., 2000; Mohammad et al., 2006; Geng et al., 2015). Forster (1940) first studied the degradation of isopropanol in the bacterium *Rubrivivax gelationosus*, which utilize isopropanol as the sole carbon source. Subsequently, Siegel (1950) demonstrated isopropanol degradation through a dehydrogenation reaction by various bacterial strains which utilize isopropanol as a unique carbon and energy source. Later, it was shown that some of these organisms can grow in the presence of relatively high isopropanol concentrations of up to 38 g (Mohammad et al., 2006). Biodegradation of isopropanol appears to be rapid, with the aerobic soil half-life for this compound ranging from one to seven days (Howard et al., 1991). Furthermore, an estimated Bioconcentration Factor of 3 suggests that there is low potential for bioaccumulation of isopropanol in aquatic organisms (HSDB, 2012). Therefore, it has been

concluded that isopropanol meets the criteria for being considered readily biodegradable (HSDB, 2012; UNEP, 1997).

While Fox and Ketha (1996) demonstrated anaerobic biodegradation of isopropanol, this process was conducted through co-metabolism of glucose. Bustard et al. (2000), by contrast, showed that isopropanol may be utilized aerobically by a mixed consortium as the sole carbon source without extra nutrient supplementation.

Concentrations:

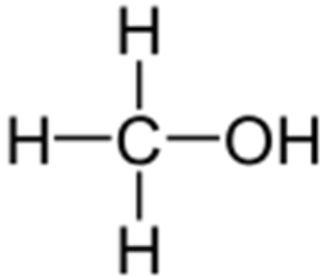
Maximum concentration range in HF fluid (% mass): 0.00007-0.04962

Average concentration range in HF fluid (% mass): 0.012909

Table 20 Isopropanol concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Solberg 31-2WH (North Dakota)	AC-100	RockPile Energy	Corrosion inhibitor	1	0.00007
University 31-2H (Texas)				60	0.00245
UL MS HZ BLK 6 Unit 4106 (Texas)				60	0.00128
RK-UTL 3031B-17 1H (Texas)				100	0.01919
University 41-01-DH (Texas)				60	0.00158
University 43-15 9H (Texas)	AI-265	Reef	Acid Inhibitor	15	0.00061
UL 21 Bighorn 1H (Texas)				30	0.04007
University FN 3731 (Texas)	StimOil FBA M	CESI Chemical	Surfactants	30	0.01388
University Red 5031B (Texas)	STIMENX-StimOil ENX	Flotek	Surfactant	30	0.04962
University 43-15 # 7H (Texas)	WAI-251LC	WFT	Inhibitor	5	0.00034

4.23 Methanol (CH_3OH; CAS # 67-56-1)



Function: Surfactant

Degradation: Volatilization and biodegradation are thought to be major removal mechanisms of methanol (Katsumata and Kastenberg, 1996). However, conflicting reports occur on the volatilization of methanol. While Katsumata and Kastenberg (1996) regarded volatilization as a major player in the removal of methanol from surface waters, Malcolm Pirnie Inc, (1999) held a contrarian view. Such controversy arises since volatilization estimates in surface waters are complex and involve numerous parameters such as wind and current speed, water depth and surface temperatures and the estimation of Henry's Law (e.g., Lyman et al., 1982).

Methylotrophic bacteria capable of biodegrading methanol are detected in soil, plant materials, fresh and marine waters, as well as air although few are obligate methylotrophs (Goldberg and Rockem, 1991). The methanol degradation pathway involves oxidation to form formaldehyde, which can then be used in an assimilatory fashion to synthetize cell material or it can continue to be oxidized to CO_2 in a dissimilatory way (Vestal, 1984). Most of these methylotrophic bacteria utilize either the ribulose monophosphate (RuMP) pathway or the serine pathway with most obligate methylotrophs degrading methanol more efficiently through RuMP (Goldberg et al., 1976, 1991). For example, some bacteria from genera *Hyphomicrobium* and *Pseudomonas* that grow on methanol are also able to utilize a large number of higher molecular weight carbon compounds (Brock et al., 1984).

Methanol can be biodegraded under aerobic and anaerobic conditions. For example, White (1986) conducted microcosm studies on the biodegradation potential of methanol as the sole substrate, indicating that complete utilization of concentrations as high as 1000 mg/L occurred in a period less than a year. Goldsmith (1985) also conducted microcosm studies on the degradation of methanol and found that this compound was readily biodegraded at concentrations ranging from 80 to 500 mg/L under anoxic conditions.

Concentrations:

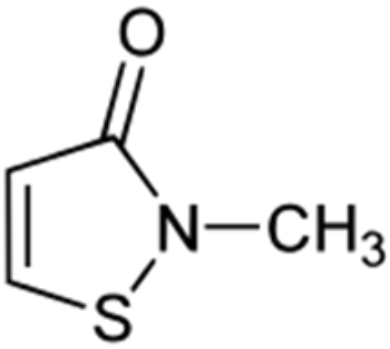
Maximum concentration range in HF fluid (% mass): 0.00007-0.05947

Average concentration range in HF fluid (% mass): 0.00638

Table 21 Methanol concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
University Red 5031B (Texas)	CI200-Corrosion Inhibitor	Economy Polymers	Corrosion Inhibitor	60	0.00054
University Red 5031B (Texas)	NE601-NE/Surfactant	Solnexus	Surfactant	70	0.00041
University FN 3731 (Texas)	I-122	Chemplex L.C.	Acid Corrosion Inhibitors	70	0.00083
University FN 3731 (Texas)	NE-227	CESI	Non and De-emulsifiers	15	0.00719
Test Well 1 (Pennsylvania)	A999	PP CO	Acid Corrosion Inhibitor	40	0.00050
University 43-15 # 7H (Texas)	Inhibitor	WFT	Inhibitor	2.5	0.00007
UL COPELAND 2425-17 1H (Texas)	N.A.	N.A.	N.A.	60	0.00049
UTL 2932-17 1H (Texas)	N.A.	N.A.	N.A.	60	0.00047
UL MS HZ BLK 6 Unit 4106 (Texas)	N.A.	N.A.	N.A.	60	0.00128
University 43-15 9H (Texas)	AI-265	Reef	Acid Inhibitor	15	0.00061
University 43-15 9H (Texas)	Plexsurf 240E	Chemplex	Non Ionic Surfactant	20	0.01644
University Amanda #3 (Texas)	CIA - 5	Universal Pressure Pumping, Inc.	Acid Corrosion Inhibitors	40	0.00086
University Amanda #3 (Texas)	NE-1	Universal Pressure Pumping, Inc.	Non-Emulsifier and Deemulsifiers	15	0.00014
Solberg 31-2WH (North Dakota)	Ecopol-NE601	RockPile Energy	Non-emulsifying Agent	50	0.05947

4.24 Methylisothiazolinone ($C_4H_5NO_S$; 2-Methyl-1,2-thiazol-3(2H)-one; CAS # 2682-20-4)



Function: Biocide

Degradation: In the atmosphere, vapour-phase methylisothiazolinone will be degraded by reaction with photochemically-produced hydroxyl radicals, exhibiting a half-life of 13 hours (TOXNET, 2015b). However, this compound is not expected to be susceptible to direct photolysis by sunlight (US-EPA/OPPTS, 2014). While free methylisothiazolinone is unstable, greater stability is afforded by the formation of adducts with calcium or magnesium salts (Chan et al., 1983; Krzeminski et al., 1975a).

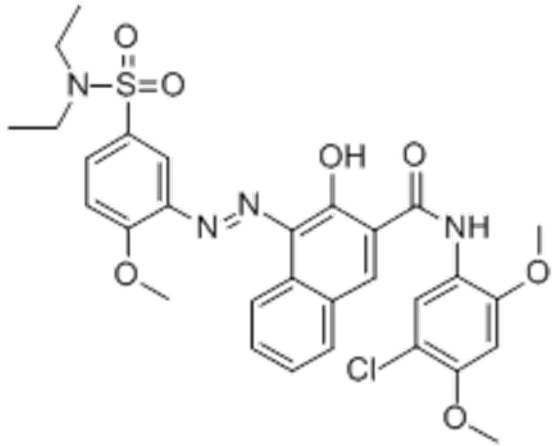
Rapid biodegradation of methylisothiazolinone has been observed in both aquatic and terrestrial environments (Burnett et al., 2010). For example, the half-life of methylisothiazolinone in an aerobic microcosm using river water and sediment was observed to be 9 days (Jacobson and Williams, 2000).

The major biodegradative pathway in the environment for the calcium chloride salt of methylisothiazolinone was identified by Krzeminski et al. (1975b). Principally, degradation involves calcium chloride dissociation, ring opening, loss of chlorine and sulfur, and subsequent formation of *N*-methylmalonamic acid. Further degradation then proceeds through malonamic, malonic, acetic, and formic acids to carbon dioxide. Other tentatively identified products consisted of 5-chloro-2-methyl-4-isothiazolin-1oxide, *N*-methylglyoxylamide, ethylene glycol, and urea.

Biodegradation of methylisothiazolinone has been observed to occur largely in aerobic environments. For example, biodegradation of this compound was measured by Voets et al. (1976) in synthetic sewage and in a mineral solution under both aerobic and anaerobic conditions. While degradation of between 80 to 100% was observed in the organic medium under aerobic conditions, no degradation was noted under anaerobic conditions.

Concentrations: Typical injection in stimulation fluid 0.0001% (AECOM Australia Pty Ltd, 2017)

4.25 Pigment Red 5 ($C_{30}H_{31}ClN_4O_7S$; (4E)-N-(5-chloro-2,4-dimethoxyphenyl)-4-[[5-(diethylsulfamoyl)-2-methoxyphenyl]hydrazinylidene]-3-oxonaphthalene-2-carboxamide; CAS # 6410-41-9)



Function: Viscosity management

Degradation: Due to their hydrophobicity, hydrolysis is unlikely to play any significant role in the environmental degradation of such azo pigments. For example, no hydrolysis was detected in a 56-day experiment on Pigment Yellow 83 (IUCLID, 1996). Furthermore, since the stability of such pigments to visible and UV light is very high, only slow degradation through photolysis may occur (Clarke and Anliker, 1980).

Due to their insolubility, azo pigments are essentially considered non-bioavailable (ETAD, 1989). Studies on Pigment Yellow 17, for example, demonstrated no evidence for the occurrence of anaerobic biodegradation of this pigment (ETAD et al., 1995). However, aerobic degradation by activated sludge may take place. For example, in a 15-day study were pigments were dispersed in, among other reagents, ethandiol, 40 and 81% of Pigment Yellow 83 and Pigment Yellow 12, respectively, were degraded (IUCLID, 1996). Furthermore, biodegradation though extracellular oxidases has been demonstrated by the white-rot fungus *Pycnoporus cinnabarinus,* which has been able to decolourise an effluent from a pigment plant by up to 90% in 3 days (Banat et al., 1996). Indeed, fungi play an important role in the degradation and decolourisation of synthetic textile dyes through enzymes as well as by the absorption, adsorption and accumulation of colourants from effluents (Singh, 2017).

Concentrations:

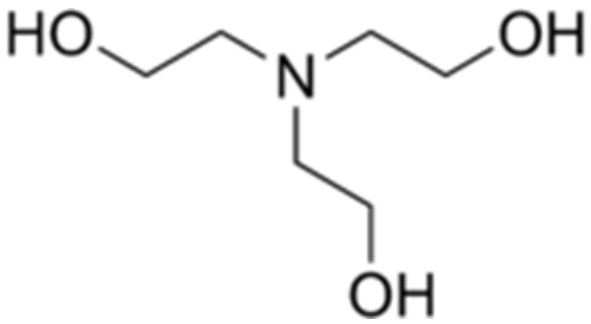
Maximum concentration range in HF fluid (% mass): 0.00001-0.00010

Average concentration range in HF fluid (% mass): 0.00006

Table 22 Pigment Red 5 concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
MASTIFF FEDERAL 3H (New Mexico)				1	0.00004
FC Leidy COM 1 (Colorado)				1	0.00008
Eata Fajita State 13H (New Mexico)				1	0.00001
High Flume 10-10 (Colorado)				1	0.00009
ADMIRAL FEDERAL COM 002H (New Mexico)				1	0.00002
Anderson C #1 (Colorado)				1	0.00010
SCREECH OWL FEDERAL 001H (New Mexico)				1	0.00005
Piccoli A 1 (Colorado)				1	0.00009

4.26 Triethanolamine ($C_6H_{15}NO_3$; 2,2',2''-Nitrilotri(ethan-1-ol); CAS # 102-71-6)



Function: Viscosity management

Degradation: Vapour-phase triethanolamine has been shown to degrade in the atmosphere via the reactions with photochemically-produced hydroxyl radicals, resulting in an estimated half-life in air of 3.5 hours (NCBI, 2018). Triethanolamine is not expected to be susceptible to direct photolysis by sunlight as it does not contain chromophores that absorb at wavelengths >290 nm (NCBI, 2018). Due to the lack of functional groups that hydrolyse under environmental conditions, triethanolamine is not expected to undergo hydrolysis in the environment (Lyman et al., 1990).

Biodegradation of triethanolamine has been previously assessed using the BOD screening test and various inherent biodegradability tests (e.g., West and Gonsior, 1996). Variations in reported results

appears to indicate that inoculum source and/or microbial acclimation may be key factors influencing biodegradation of this compound (West and Gonsior, 1996). For example, Bridie et al. (1979), using effluent from a biological sanitary waste treatment plant as an inoculum, observed triethanolamine degradation of 5% theoretical oxygen demand (ThOD; unadapted) and 28% ThOD (adapted) in a 5 day test. Lamb and Jenkins (1952) reported only 6.8% of the ThOD consumed after 20 day incubation using dilute sewage inoculum and 2.5 mg/L triethanolamine. However, this result contrasts with Price et al. (1974) who reported 66% of ThOD consumed after 20 days in a similar BOD test. Inherent biodegradability tests reported even higher percentages of biodegradation. For example, Gerike and Fisher (1980) reported 82% removal of the triethanolamine after only 8 days.

Triethanolamine has also been shown to biodegrade anaerobically. Speranza et al. (2006), for example, demonstrated triethanolamine conversion into acetate and ammonia by a strictly anaerobic, gram-positive *Acetobacterium* strain LuTria3.

Concentrations:

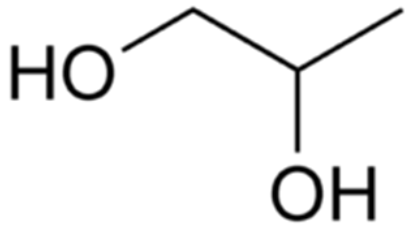
Maximum concentration range in HF fluid (% mass): 0.00085-0.08424

Average concentration range in HF fluid (% mass): 0.0372

Table 23 Triethanolamine concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
University F #4L (Texas)	BC-1	Rockwater Energy Solutions	Buffers / Ph Control	85	0.00085
Clark Wood Ranch K150 (Texas)	BC-1	Rockwater Energy Solutions	Buffers / Ph Control	85	0.01940
WILLARD UNIT 246A (Texas)	BC-1	Rockwater Energy Solutions	Buffers / Ph Control	85	0.04217
Lake 1-4 (Texas)	BA-620	P3	Breaker	65	0.07130
Good Bull #1 (Texas)	BC-1	Rockwater Energy Solutions	Buffers / Ph Control	85	0.00526
Jerry Thornhill, Jr. #3 (Mississippi)				65	0.08424

4.27 Propylene glycol ($C_3H_8O_2$; Propane-1,2-diol; CAS # 57-55-6)



Function: Viscosity management

Degradation: Propylene glycol has been shown to readily be biodegraded by common aerobic and anaerobic bacteria (Willetts, 1979; Klecka et al. 1993; Veltman et al., 1998; Shupack and Anderson 2000; Toscano et al., 2013). The rate and extent of propylene glycol biodegradation can be limited by factors such as temperature, biomass concentration, and availability of additional nutrients and of electron acceptors. For example, biodegradation of propylene glycol is temperature dependent with metabolic activity of propylene glycol degraders severely inhibited by low temperatures (Klecka et al., 1993; Revitt and Worrall, 2003; Jaesche et al., 2006). Increasing soil temperatures, by contrast, accelerate propylene glycol degradation with a concomitant depletion of locally available oxygen (French et al., 2001; Lissner et al., 2013). Consequently, the alternative electron acceptors Fe and Mn oxides are used, resulting in reduced Mn(II) and Fe(II) species in pore water (French et al. 2001; Jaesche et al. 2006; Lissner et al. 2013; Schotanus et al. 2014).

Several unrelated microorganisms are able to grow on propylene glycol as a sole source of carbon and energy (Toscano et al., 2013). Aerobically, propylene glycol is oxidised to lactaldehyde either by a diol oxidase, NAD^+-dependent alcohol dehydrogenases or pyrroloquinoline quinone (PQQ)-dependent diol dehydrogenases (*Xanthobacter autotrophicus,* Willetts, 1979; *Escherichia coli,* Sridhara et al. 1969; *Stenotrophomonas maltophilia,* Tachibana et al. 2008; *Methylobacterium extorquens AM1,* Bolbot and Anthony 1980*; Pseudomonas putida,* Toyama et al. 1995). Subsequently, the resulting propionaldehyde may be either reduced to *n*-propanol or oxidised to propionic acid (Toscano et al., 2013). Methanogenic microbial consortia from sewage sludges have been shown to conduct further anaerobic degradation of propionic acid and propanol (Veltman et al. 1998). Toscano et al. (2013) selected bacterial consortia from Norwegian soil samples that were able to grow on propylene glycol as sole carbon and energy source. Enrichment cultures showed that propylene glycol-degrading populations were mainly composed by *Pseudomonas* species, although *Bacteroidetes* were also found (Toscano et al., 2013).

Concentrations:

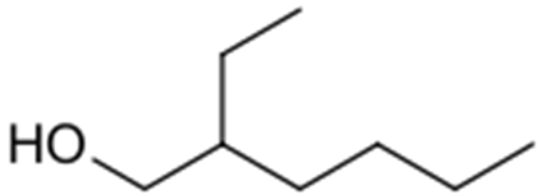
Maximum concentration range in HF fluid (% mass): 0.00003-0.02789

Average concentration range in HF fluid (% mass): 0.009676

Table 24 Propylene glycol concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
Constantan (Texas)	Gyptron T-490	Champion Technologies	Scale Inhibitor	10	0.00533
NBU 1022-4B3DS (Utah)	WFR-3B	NABORS	FRICTION REDUCER	25	0.00169
Teton 5-8-10MBH (North Dakota)					0.00089
SALE RANCH 20A 1H (Texas)					0.02789
Diana 4-34/27H *(Oklahoma)*				10	0.00003
MEL 2H-30 (Oklahoma)				1	0.00070
SHACKELFORD 3207H (Texas)					0.02767
Hicklin 2-28HA (Oklahoma)				10	0.00497
Ruth 28-33 6TFH (North Dakota)				1	0.00023
MCCLINTIC E 7H (Texas)					0.02736

4.28 2-Ethylhexanol ($C_8H_{18}O$; 2-Ethylhexan-1-ol; CAS # 104-76-7)



Function: Surfactant

Degradation: 2-Ethylhexanol dissolved in water will either volatilize to air or undergo biodegradation. The estimated half-life in surface water in a model river due to volatilization is 1.7 days (HSDB, 2004). In soil, this compound will likely volatilize from the surface or migrate to water (HSDB, 2004; Genium, 1999). It is not expected to adsorb to sediments or bioconcentrate (Genium, 1999). Vapour phase 2-ethylhexanol is degraded in the atmosphere by photochemically produced hydroxyl radicals and exhibit an atmospheric half-life of 1.2 days (HSDB, 2004; Genium, 1999).

Since 2-ethylhexanol exhibits limited solubility in water, this alcohol will eventually form droplets at a given concentration in excess of the solubility limit. Such droplets are not available for biodegradation and have little tendency to transfer to the gas phase since most of droplets are found in the bulk of the water (Nalli, 2005).

Degradation kinetics of 2-ethylhexanol in the absence of the biocide glutaraldehyde were measured by Rodgers et al. (2017). 2-Ethylhexanol was shown to more rapidly degrade than other compounds (2-propanol, ethylene glycol, propargyl alcohol, and 2-butoxyethanol), particularly under oxic conditions. Under anoxic conditions, there was limited removal (<10%) of this compound. It was also shown that biodegradation rates of 2-ethylhexanol were 2−5 times slower in the presence of the glutaraldehyde trimer (Rodgers et al., 2017).

Oxidation of 2-ethylhexanol creates the recalcitrant compound 2-ethylhexanoic acid (Nalli et al, 2002). This partial degradation has been observed with a range of different bacteria and fungi and the resistance of 2-ethylhexanoic acid to mineralization could be attributed to the ethyl substitution on the second carbon (Nalli, 2005; Nalli et al, 2006).

Concentrations:

Maximum concentration range in HF fluid (% mass): 0.00001-0.01236

Average concentration range in HF fluid (% mass): 0.00332

Table 25 2-Ethylhexanol concentrations as reported in various North American wells. Source: http://fracfocus.org/

Well Name (Location)	Chemical Trade Name	Supplier	Purpose	Maximum Concentration in Additive (% mass)	Maximum Concentration in HF Fluid (% mass)
University 43-15 # 7H (Texas)	WNE-363L	WFT	Surfactant	7	0.00637
Go Yeamans #1 (Colorado)	NE-9	EES	ANIONIC OIL SOLUBLE NON EMULSIFIER	16	0.01236
Munger Nix A LZS #66 (Texas)	WNE-363L	WFT	Surfactant	7	0.00050
Herring 1H (Texas)	NE-6	Sanjel	Non-emulsifier	10	0.00373
592721 (Pennsylvania)	NEFE-180	Stingray Pressure Pumping, LLC	Acid inhibitor	10	0.00009
Helling Trust 6494 41-22 11T2 (North Dakota)	DCF-2520	WST-OneCor	Demulsifier	10	0.00001
BOONE 213 UNIT 3H (Texas)	WNE-363L	WFT	Surfactant	7	0.00015

4 Policy and regulatory frameworks associated with the harnessing of onshore gas resources

In Australia, state and territory governments are mainly responsible for the legislative framework, licensing and decision making processes governing onshore gas exploitation (Commonwealth of Australia, 2014). Due to a lack of federal lands, Australia's national (or federal) government has jurisdiction only within its specifically enumerated constitutional powers, with all other jurisdiction under the regulatory ambit of the states and territories (Ingelson and Hunter, 2014, and references therein). Federal legislative powers regarding oil and gas regulation are limited to the corporation's power, interstate, as well as overseas trade (Ingelson and Hunter, 2014, and references therein).

An exception is The *Environment Protection and Biodiversity Conservation Act 1999*, which stands as the main piece of Commonwealth Government environmental legislation and provides a legal framework to protect and manage impacts upon matters of national environmental significance (Commonwealth of Australia, 2014). The triggering of this act not only assesses the potential impact on water resources but also all potential significant impacts on matters of national environmental significance (e.g., important wetlands, listed threatened species and ecological communities; e.g., APPEA, 2016). This act works on a self-referral basis whereby if proponents consider their project may trigger the EPBC Act provisions, they must submit a referral for a decision (APPEA, 2016). Once a project commences, ongoing compliance is required that include third party auditing, obtaining and securing biodiversity offsets, undertaking monitoring/investigations and, if required, take actions such as mitigation strategies or discontinuation of activities (APPEA, 2016).

In addition, the *Industrial Chemicals (Notification and Assessment) Act 1989* requires industrial chemicals used in drilling and hydraulic fracturing to be listed on the national Australian Inventory of Chemical Substances (Ingelson and Hunter, 2014, and references therein). This inventory is maintained by the National Industrial Chemicals Notification and Assessment Scheme (NICNAS) and all new chemicals, including those used for fraccing, must be assessed by this scheme (Ingelson and Hunter, 2014, and references therein). Furthermore, both federal and state/territory governments have also published harmonised frameworks to regulate the unconventional petroleum industry, such as the National Harmonised Regulatory Framework for Natural Gas from Coal Seams (SCER, 2013; Commonwealth of Australia, 2014). Before introducing new chemicals or using chemicals for a new purpose, it is a requirement to notify the chemical to NICNAS or satisfy the requirements for introduction without notification. Furthermore, importers and manufacturers of industrial chemicals for commercial purposes (e.g., for drilling or hydraulic fracturing activities) are required to register with NICNAS (APPEA, 2016).

Since this project is part of a wider project involving South Australia (SA), we only focus here on regulatory frameworks involving this state. Onshore geothermal, petroleum, and gas exploration, development, and storage in SA is administered by the Energy Resources Division under the *Petroleum and Geothermal Energy Act 2000* (formerly *Petroleum Act 2000*) and associated regulations (e.g., Commonwealth of Australia, 2014).

Below we list the main features of the *Petroleum and Geothermal Energy Act 2000*. These features are quoted directly from the following website of the SA government:

(http://www.energymining.sa.gov.au/petroleum/legislation_and_compliance/petroleum_and_geothermal_energy_act_and_Regulations) Accessed 28/12/2018

- Establishment of a co-regulatory regime focused on achieving environmental, public safety and resource management objectives, and reduced compliance costs
- Licence allocation and management mechanisms to facilitate competition in line with competition policy principles
- Rights of third party access to licenced pipelines (where not covered by the national access regime), to depleted reservoirs (for gas storage purposes), and to pipeline easements
- Greater security of tenure for licences through improved registration procedures
- Public consultation processes with regard to establishment of environmental objectives and for significant proposed activities (consistent with provisions of the Development Act 1993)
- Reduced risk to government for liabilities arising from the activities of the industry
- A fee structure designed to encourage the industry to adopt management systems to undertake activities
- Inclusion of an effective and expeditious regulatory and approvals framework applicable to geothermal and gas storage activities

Of particular note is Part 12 of the *Petroleum and Geothermal Energy Act 2000*, which provides requirements to protect the environment from potential and adverse impacts related to petroleum activities. These requirements include the need for licensees to prepare a Statement of Environmental Objectives (SEO), based on an Environmental Impact Report (EIR). Therefore, before commencing a project, an EIR must be prepared covering potential environmental threats and how they will be managed. As noted by APPEA (2016), consultation is essential in the preparation of an EIR and should be compliant with the principles outlined in the Ministerial Council on Mineral and Petroleum Resources endorsed document: *Principles for Engagement with Communities and Stakeholders*. Three different levels of assessment are provided for being 1) Low Impact Classification, 2) Medium Impact Classification, and 3) High Impact Classification. For Low Impact Classification, the Mineral Resources Division of the SA Department of State Development will undertake internal government consultation (e.g., with The Department for Water and The Department of Environment and Natural Resources; APPEA, 2016). For Medium Impact, the Energy Resources Division seeks community comment through a public consultation period, which is required to run for at least 30 business days (APPEA, 2016). For High Impact Classification, the proposal is referred to the Minister responsible for the *Development Act 1993* for an EIA under Part 8 of that Development Act. Furthermore, an approved Statement of Environmental Objectives (SEO) must also be in place for relevant activities (APPEA, 2016). Once a project is approved, ongoing activities include performance measurements against the criteria for each objective as outlined in the SEO. If the objectives cannot readily be measured through quantitative assessment, techniques such as Goal Attainment Scaling (GAS) are required to be adopted to measure performance (APPEA, 2016). In relation to water use, operations of licensed wells in relation to water extraction must be consistent with the provisions of the appropriate SEO (APPEA, 2016). The taking and use of water

resulting as a by-product of petroleum production needs to be licensed by purpose in an area specified by the licence and is subject to annual reporting of total volume used for that purpose by the SA Department of State Development (DSD; APPEA, 2016). The *Environment Protection Act 1993* also provides the water quality management framework and offences. Importantly, details regarding water quality management are provided in the *Environment Protection (Water Quality) Policy 2015* and these are expected to be addressed in the EIR and SEO (APPEA, 2016).

As of late 2018, the Upper House of the South Australian Parliament passed a 10 year fraccing moratorium in the South East of South Australia into law. The aim of this moratorium is to provide the region with certainty regarding the protection water resources and agricultural industries (https://www.sbs.com.au/news/sa-parly-passes-southeast-fracking-ban; https://www.abc.net.au/news/2018-09-05/liberals-to-legislate-se-fracking-moratorium/10203578 (accessed 18/01/2019).

5 Conclusions

This report estimated compound concentrations of 28 selected chemicals used in onshore gas activities, reviewed past findings on their degradation propensities and listed associated policy frameworks. The following conclusions were made:

- A range of naturally occurring abiotic and biotic processes have the capacity to attenuate chemical compounds in the environment. Such processes are important to identify when assessing potential impacts in areas where onshore gas activities are about to be introduced.
- Abiotic process involved photolysis and hydrolysis, while biotic process involved the microbially-induced breakdown (biodegradation) of compounds by bacterial and/or fungal species.
- In many cases both oxic and anoxic biodegradation processes were identified and degradation could proceed within days to weeks.
- With some exceptions, Australian state and territory governments are mainly responsible for the legislative framework, licensing and decision making processes governing onshore gas exploitation.

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