Biochimica et Biophysica Acta 1834 (2013) 247–271

Contents lists available at SciVerse ScienceDirect

Biochimica et Biophysica Acta

journal homepage: www.elsevier.com/locate/bbapap

Review

Recent advances in the structural mechanisms of DNA glycosylases

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ARTICLE INFO

Article history:
Received 27 June 2012
Received in revised form 24 September 2012
Accepted 5 October 2012
Available online 14 October 2012

Keywords:
Base excision repair
DNA glycosylase
Protein–DNA interactions
DNA oxidation
DNA alkylation
Active DNA demethylation

ABSTRACT

DNA glycosylases safeguard the genome by locating and excising a diverse array of aberrant nucleobases created from oxidation, alkylation, and deamination of DNA. Since the discovery 28 years ago that these enzymes employ a base flipping mechanism to trap their substrates, six different protein architectures have been identified to perform the same basic task. Work over the past several years has unraveled details for how the various DNA glycosylases survey DNA, detect damage within the duplex, select for the correct modification, and catalyze base excision. Here, we provide a broad overview of these latest advances in glycosylase mechanisms gleaned from structural enzymology, highlighting features common to all glycosylases as well as key differences that define their particular substrate specificities.

1. Introduction

The integrity of the chemical structure of DNA and its interactions with replication and transcription machinery is important for the faithful transmission and interpretation of genetic information. Oxidation, alkylation, and deamination of the nucleobases by a number of endogenous and exogenous agents create aberrant nucleobases (Fig. 1) that alter normal cell progression, cause mutations and genomic instability, and can lead to a number of diseases including cancer [reviewed in 1]. Many of these lesions are removed by the base excision repair (BER) pathway [2], which is initiated by a DNA glycosylase specialized for a particular type of chemical damage. Upon locating a particular lesion within the DNA, glycosylases catalyze the excision of the nucleobase from the phosphoribose backbone by cleaving the *N*-glycosidic bond, generating an apurinic/apyrimidinic (AP) site (Fig. 2). Monofunctional glycosylases catalyze only base excision, whereas bifunctional glycosylases also contain a lyase activity that cleaves the backbone immediately 3′ to the AP site. The resulting single-stranded and nicked AP sites are processed by AP endonuclease 1 (APE1), which hydrolyzes the phosphodiester bond 5′ to the AP site. This generates a 3′ hydroxyl substrate for replacement synthesis by DNA polymerase β, followed by sealing of the resulting nick by DNA ligase.

Since the glycosylases are the first line of defense against a vast array of DNA damage, they have been the subject of a large body of work to understand their mechanisms of action and cellular roles [3–12]. The first crystal structures of DNA glycosylases were reported in 1992 for bacteriophage T4 Endonuclease V (EndoV) and *Escherichia coli* (*E. coli*) Endonuclease III (EndoIII), which remove pyrimidine dimers and oxidized pyrimidines, respectively [13,14]. Soon thereafter, DNA or inhibitor-bound structures of EndoV and uracil DNA glycosylase (UDG) established that these enzymes use a base-flipping mechanism to gain access to modified nucleobases in DNA [15–19]. Subsequent studies established that glycosylases fall into one of six structural superfamilies (Fig. 3). Despite their divergent architectures, these proteins, with the exception of the ALK family (see Section 3.3) [12], have evolved the base-flipping strategy to correctly identify and orient their substrates for catalysis. Recognition of the target modification likely proceeds in several steps, in which the protein probes the stability of the base pairs through processive interrogation of the DNA duplex, followed by extrusion of the aberrant nucleobase into a specific active site pocket on the enzyme [9,20]. The enzyme-substrate complex is stabilized by nucleobase contacts within the active site and a pair of side chains that plug the gap in the DNA left by the extrahelical nucleotide and wedge into the DNA base stack on the opposite strand [3–12].

In this review, we focus on the most recent advances toward understanding the mechanisms by which each class of DNA glycosylase locates, selects, and removes its target lesions. A growing number of structures and mechanistic studies of glycosylases specific for oxidized nucleobases (Section 2), alkylation damage (Section 3), and cytosine deamination products (Section 4) have elucidated many of the structural determinants of substrate specificity and have provided new insights into catalysis of *N*-glycosidic bond cleavage. Some aspects of substrate selection and excision are common across different structural classes or substrate specificities, while others are specific to a given enzyme. Our goal in this review, therefore, is to provide a broad overview of the structural mechanisms for the entire

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1570-9639/$ – see front matter
http://dx.doi.org/10.1016/j.bbapap.2012.10.005

repertoire of DNA glycosylases in order to highlight key similarities and differences between each structural class. We note that the roles of DNA glycosylases in the cell and in the context of BER have been the subject of recent reviews, and thus we focus our discussion on the structural enzymology.

2. Oxidative damage

DNA bases undergo oxidative damage from chemical oxidants, free radicals and reactive oxygen species (ROS) produced from cellular respiration, inflammatory responses, and ionizing radiation [21–23]. Oxidized bases are often used as biomarkers for oxidative stress and cancer [22,24]. Guanines are especially susceptible to oxidation, leading to a number of lesions that are substrates for BER (Fig. 1A) [25]. Attack of a hydroxyl radical at the C8 position of guanine produces 7,8-dihydro-8-hydroxyguanine (8-OHG), which tautomerizes to 8-oxo-7,8-dihydroguanine (8oxoG), or the ring-opened 2,6-diamino-5-formamido-4-hydroxy-pyrimidine (FapyG), two of the most abundant oxidative DNA adducts [26,27]. 8oxoG is a particularly insidious lesion because of its dual coding potential by replicative polymerases, leading to G→T transversion mutations likely as a result of its ability to form both 8oxoG(syn)•A(anti) and 8oxoG(anti)•C(anti) base pairs [22,23,28–30]. Oxidation of guanine and 8oxoG also produces a variety of ring-opened purines in addition to FapyG, including hydantoin lesions, spiroiminodihydantoin (Sp), guanidinohydantoin (Gh), and its isomer iminoallantoin (Ia) (Fig. 1A) [31–33]. Fapy lesions inhibit DNA polymerases and are potentially mutagenic [34]. Hydantoin lesions have been suggested to lead to an increase in G→T and G→C transversions and stall the replication machinery [31,32,35,36]. In addition to purines, reaction of hydroxyl radicals at positions 5 or 6 of thymine produces 5,6-dihydroxy-5,6-dihydrothymine (thymine glycol, Tg), a cytotoxic lesion that distorts the DNA duplex and can inhibit replication [26,37]. Other potentially harmful pyrimidines include dihydrothymine (DHT), dihydrouracil (DHU), 5-hydroxyuracil (5-OHU), 5-hydroxycytosine (5-OHC), 5-hydroxymethyluracil (5hmU), and 5-formyluracil (5fU) [38–43].

DNA glycosylases that remove oxidative DNA damage can be categorized on the basis of their preferences for purine or pyrimidine lesions and their structural folds (Table 1). Oxidized purines, including 8oxoG and FapyG, are removed from DNA by 8oxoG DNA glycosylase (OGG1) in eukaryotes and MutM (also known as FapyG DNA glycosylase, Fpg) in bacteria [recently reviewed in 23]. Oxidized pyrimidines are removed by endonuclease III (EndoIII, or Nth) and endonuclease VIII (Endo VIII, or Nei), and their eukaryotic orthologs, NTH1 and NEIL1 (Nei-like1), respectively. Despite their different substrates, OGG1 and EndoIII/Nth adopt a common architecture characteristic of the Helix–hairpin–Helix (HhH) superfamily of DNA glycosylases [44]. MutM/Fpg and EndoVIII/Nei are also structurally similar, with helix-two turn-helix (H2TH) and antiparallel β-hairpin zinc finger motifs, and they share a common bifunctional catalytic mechanism involving both base excision and AP lyase activities [45–49].

2.1. 8oxoG repair

Eukaryotic OGG1 and bacterial MutM/Fpg preferentially catalyze removal of 8oxoG paired with C [50,51]. Both enzymes are bifunctional in

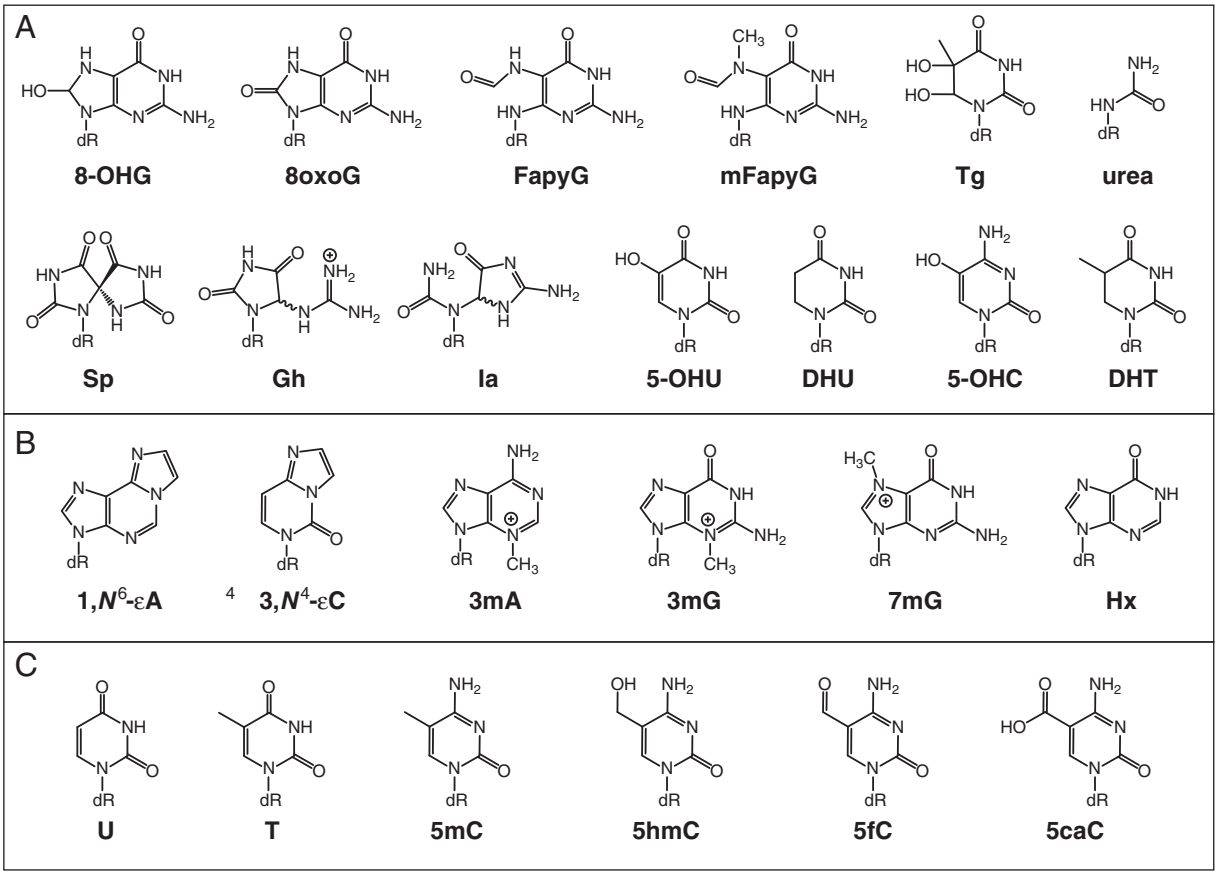

Fig. 1. Common DNA lesions referenced in this review. (A) Oxidized nucleobases. 8-OHG, 7,8-dihydro-8-hydroxyguanine; 8oxoG, 8-oxo-7,8-dihydroguanine; FapyG, 2,6-diamino-4-hydroxy-5-formamidopyrimidine; mFapyG, *N*7-methylFapyG; Tg, thymine glycol; Sp, spiroiminodihydantoin; Gh, guanidinohydantoin; Ia, iminoallantion; 5-OHU, 5-hydroxyuracil; DHU, dihydrouracil; 5-OHC, 5-hydroxycytosine; DHT, dihydrothymine. (B) Alkylated nucleobases. εA, 1,N^6-ethenoadenine; εC, 3,N^4-ethenocytosine; 3mA, *N*3-methyladenine; 3mG, *N*3-methylguanine; 7mG, *N*7-methylguanine; Hx, hypoxanthine. (C) Nucleobases repaired by the UDG/TDG family of DNA glycosylases. U, uracil; T, thymine; 5mC, 5-methylcytosine; 5hmC, 5-hydroxymethylcytosine; 5fC, 5-formylcytosine; 5caC, 5-carboxylcytosine.

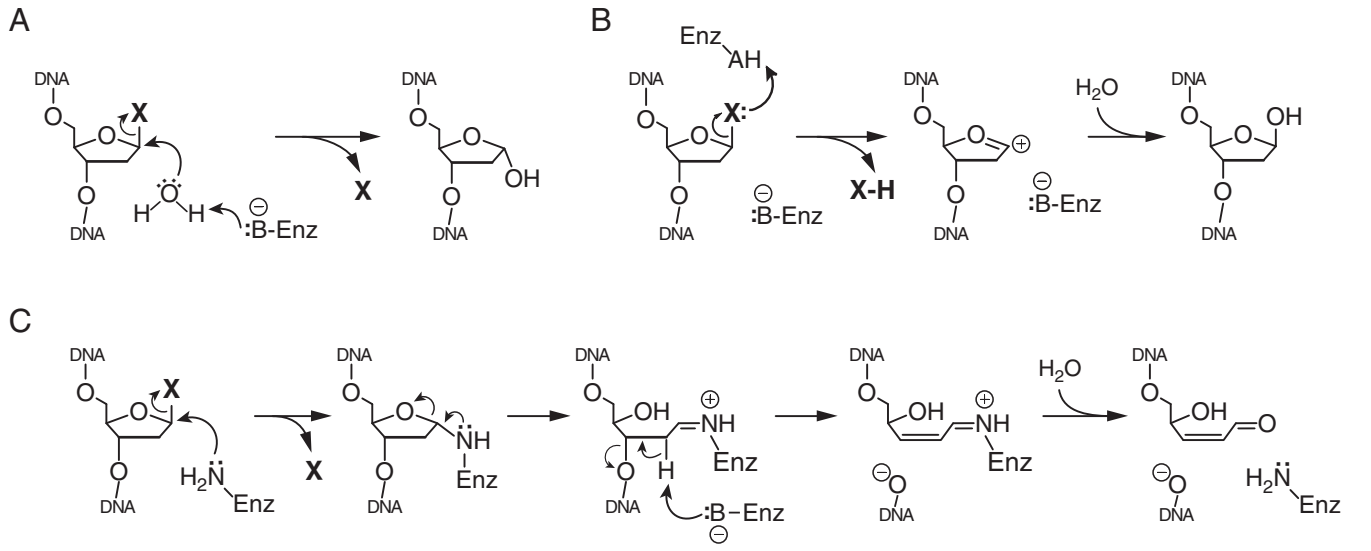

Fig. 2. Chemical reaction catalyzed by DNA glycosylases. (A,B) Monofunctional glycosylases cleave the *N*-glycosidic bond to liberate free nucleobase (X) from the phosphoribose backbone through either associative (A) or dissociative (B) mechanisms. (C) Bifunctional mechanism, in which both the *N*-glycosidic bond and the DNA backbone are cleaved.

that they contain both base excision and AP lyase activities, although a recent report suggests that human OGG1 (hOGG1) may function as a monofunctional glycosylase under physiological conditions (see Section 2.1.1) [44,52,53]. The OGG enzymes can be subdivided into three structural families (Fig. 4): (1) OGG1, including human OGG1 and the recently discovered *Clostridium acetobutylicum* (CaOGG) enzyme (Fig. 4A–C) [54–63], (2) archaeal OGG2 (Fig. 4D–F) [64,65], and (3) archaeal 8oxoG glycosylase (AGOG), represented by the *Pyrobaculum aerophilum* enzyme (Fig. 4G–H) [66]. Structural studies of the various OGG orthologs [67] and of MutM have elucidated the molecular details required for 8oxoG recognition and excision from two distinct protein architectures and in recent years have advanced our understanding of how DNA glycosylases in general scan unmodified DNA in search of damage (for an excellent review, see Ref. [4]).

2.1.1. OGG1

A battery of recent structures of hOGG1 in complex with DNA containing an 8oxoG•C base pair (Lesion Recognition Complex, LRC) or a normal G•C base pair (Interrogation Complex, IC) from the Verdine group has been invaluable in understanding how DNA glycosylases recognize and discriminate their substrates from normal DNA [52,68–70] (the K_m values of murine OGG1 (mOGG1) are 42.7 ± 14.6 nM for 8oxoG•C and 694 ± 145 nM for G•C [71]). The original hOGG1 LRC structure was obtained from a catalytically inactive Lys249Gln mutant bound to DNA containing an 8oxoG•C base pair [52], which revealed how hOGG1 utilizes the HhH architecture to kink the DNA duplex, disrupt the 8oxoG•C base pair, and extrude the 8oxoG out of the helix and into a base binding pocket [52]. Of the multiple contacts to the extrahelical 8oxoG, only one—between the carbonyl oxygen of Gly42 and the N7 hydrogen of 8oxoG—is specific to 8oxoG (Fig. 4B) and was thus proposed to account for OGG1's ability to distinguish 8oxoG from G. However, the position of the backbone and the integrity of the 8oxoG-specific hydrogen bond are not dependent on glycine in this position, as a Gly42Ala substitution did not alter the protein backbone conformation, disrupt the hydrogen bond, or affect the K_d (~15 nM) of the interaction with 8oxoG–DNA [70].

In the hOGG1 IC structure, which used a disulfide crosslinking strategy to trap the enzyme bound to a G•C base pair, the extrahelical guanine was situated in a pocket adjacent to the active site that the authors termed the 'exo' site [68]. In a subsequent IC structure, in which the enzyme was forcibly presented with a G•C base pair adjacent to 8oxoG, the extrahelical guanine was not observed in the active or exo sites, likely as a result of steric and electrostatic clashes imposed by the 8oxoG [69]. In both of these ICs, the protein (Asn149Cys) was crosslinked to the cytosine opposite the extrahelical G. In a more recent structure of a catalytically active hOGG1/G•C–DNA complex that was crosslinked at a more remote location from the lesion (Ser292Cys), the target guanine was fully engaged inside the active site in a virtually identical position as 8oxoG in the LRC. In the IC, however, the guanine remained uncleaved, presumably because it lacks the N7 hydrogen present in 8oxoG that forms a specific hydrogen bond with the carbonyl of Gly42 [72]. The alignment of active site residues other than Gly42 are also important for catalysis, as observed in a phototrapped, uncleaved hOGG1/8oxoG–DNA complex that showed an intact 8oxoG–Gly42 interaction amidst a collection of

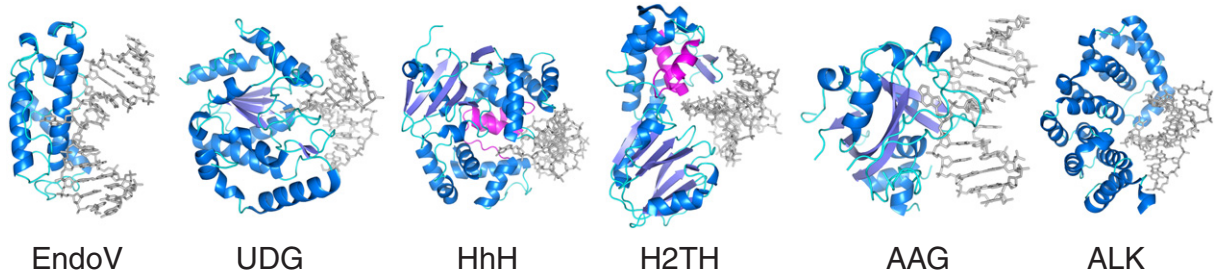

Fig. 3. DNA glycosylase structural superfamilies. Representative crystal structures from each class shown are: EndoV, T4 pyrimidine dimer DNA glycosylase EndoV (PDB ID 1VAS); UDG, human uracil-DNA glycosylase UDG (1EMH); Helix–hairpin–Helix (HhH), human 8-oxoguanine DNA glycosylase OGG1 (1YQK); Helix-two turn-helix (H2TH), *Bacillus stearothermophilus* 8-oxoguanine DNA glycosylase MutM (1L1T); AAG, human alkyladenine DNA glycosylase AAG/MPG (1EWN); ALK, *Bacillus cereus* alkylpurine DNA glycosylase AlkD (3JXZ). Proteins are colored according to secondary structure with the HhH and H2TH domains magenta. DNA is shown as gray sticks.

Table 1
DNA glycosylases specific for oxidized, alkylated, mismatched, uracil, and 5-methylcytosine bases.

Eukaryotes	Archaea	Prokaryotes	Protein fold	Substrates	PDB entries
Oxidation					
OGG1	OGG	Ogg	HhH	8oxoG•C, FapyG, FapyA	1KO9 (hOGG1) 1EBM (K249Q/8oxoG-DNA) 1FN7 (THF-DNA) 1HU0, 1LWV, 1LWW, 1LWY (NaBH4-trapped DNA complex) 1M3H (D268E/nicked-DNA) 1M3Q (D268E/abasic-DNA/8-aminoG) 1N39 (D268E/THF-DNA) 1N3A (D268Q/THF-DNA) 1N3C (D268N/THF-DNA) 1YQK (N149C/G•C-DNA XL) 1YQL (N149C,K249Q/7-deaza-8-azaguanine-DNA XL) 1YQM (N149C,K249Q/7-deazaguanine-DNA XL) 1YQR (N149C,K249Q/8oxoG•C-DNA XL) 2I5W (N149C/8oxoG•G-DNA XL) 2NOB (N149C,K249Q,H270A/8oxoG•C-DNA XL) 2NOE (G42A,K249Q/8oxoG•C-DNA) 2NOF (N149C,Q315F/8oxoG•C/DNA XL) 2NOH (K249Q,Q315A/8oxoG•C-DNA) 2NOI (G42A,N149C,K249Q/8oxoG•C-DNA XL) 2NOL (K249Q,S292C/8oxoG•C-DNA) 2NOZ (S292C,Q315F/8oxoG•C-DNA) 2XHI (K249C,C253K,D268N/8oxoG•C-DNA) 3KTU (2′F-8oxoG-DNA)
	OGG2		HhH	8oxoG (paired with any base)	3FHF (MjOGG) 3KNT (MjOGG K129G/8oxoG•C-DNA) 3FHG (SsOGG)
	AGOG		HhH	8oxoG (ssDNA, dsDNA)	1XQO 1XQP (free 8oxoG)
		MutM/Fpg	H2TH	8oxoG, FaPy, 7mFapyG, Sp, Gh, Tg, Ug, DHT, DHU, 5-OHU, 5-OHC, FU, urea, oxazolone, oxaluric acid, oxidized εA derivatives, sulfur mustard guanine N7-adduct, ring-opened oxidized aminofluorene guanine C8-adduct, 5-hydroxy-5-methylhydantoin, 3-[(aminocarbonyl) amino]-(2R)-hydroxy-2-methylpropanoic acid	1EES TtMutM 2F5Q, 2F5S (E3Q GsMutM/8oxoG•C-DNA XL) 1R2Y (E3Q GsMutM/8oxoG•C-DNA) 1R2Z (E3Q GsMutM/DHU-DNA) 1LIT (GsMutM/reduced abasic site-DNA) 1LIZ (GsMutM/NaBH4-trapped DNA complex) 1L2B (GsMutM/nicked DNA complex) 1L2C (GsMutM/HPD•T-DNA) 1L2D (GsMutM/HPD•G-DNA) 2F5N, 2F5P (Q166C GsMutM/A•T-DNA XL) 2F5O (Q166C GsMutM/G•C-DNA XL) 2F5Q, 2F5S (E3Q,Q166C GsMutM/8oxoG•C-DNA XL) 3JR4, 3JR5 (N174C GsMutM/G•C-DNA XL) 3GP1 (Q166C,V222P GsMutM/8oxoG•C-DNA XL) 3GPP (Q166C,T224P GsMutM/8oxoG•C-DNA XL) 3GPU, 3GQ3, 3GO8 (Δ220-235,Q166C GsMutM/8oxoG•C-DNA XL) 3GPX (Δ220-235,Q166C GsMutM/G•C-DNA XL) 3GPY (Q166C GsMutM/8oxoG•C-DNA XL) 3GQ4 (GsMutM/8oxoG•C-DNA XL) 3GQ5 (Q166C,T224P GsMutM/G•C-DNA XL) 3SAR, 3SAU (Q166C GsMutM/A•T-DNA XL) 3SAS, 3SAT (Q166C,R112A GsMutM/G•C-DNA XL) 3SAV (A149S,Q166C, GsMutM/G•C-DNA XL) 3SAW (GsMutM/G•C-DNA XL) 3SBJ (Q166C,V222P GsMutM/A•T-DNA XL) 1XC8, 1TDZ (LlMutM/FapyG•C-DNA) 1PJI, 1NNJ (LlMutM/PDI•C-DNA) 1PJJ, 1PM5 (LlMutM/THF•C-DNA) 1KFV(P1G LlMutM/PDI•C-DNA) 3C58 (LlMutM/7bFapyG•C-DNA) 2XZF (LlMutM-HC-DNA) 2XZU (LlMutM-HC-DNA XL)
NTH1	EndoIII	Nth/EndoIII	HhH/FeS[2]	Tg, Ug, DHU, 5-OHU, 5-OHC, urea	2ABK (EcEndoIII) 1ORN, 1ORP (GsEndoIII/NaBH4-trapped DNA complexes) 1P59 (GsEndoIII/THF-DNA)

Table 1 (*continued*)

Eukaryotes	Archaea	Prokaryotes	Protein fold	Substrates	PDB entries
NEIL1		Nei/Endo VIII	H2TH	Tg, DHT, DHU, 5-OHU, 5-OHC, 5fU, 5hmU, FapyG, FapyA, urea, 8oxoA, Gh, Sp, Ia; (Nei only: Ug, 8oxoG, 7mFapyG, 5,6dhC, 5-OHT)	1Q39 (EcEndoVIII) 1Q3B, 1Q3C (EcEndoVIII R252 and E2A mutants) 1K3W, 1K3X (EcEndoVIII/NaBH4-trapped DNA complexes) 2EA0, 2OPF, 2OQ4 (EcEndoVIII/PED-DNA) 1TDH (NEIL1) 3A45 (MvNei1) 3A46 (MvNei1/THF-DNA) 3VK8, 3VK7 (MvNei1/Tg-DNA, MvNei1/5-OHU-DNA)
NEIL2			H2TH	Gh/Ia, 5-OHU, FapyG	[Refs. 115, 124]
NEIL3			H2TH	Sp, Gh, FapyG, FapyA	[Refs. 115, 129]
Alkylation					
AAG			AAG	3mA, 7mG, εA, Hx, A, G	1EWN (εA-DNA) 1BNK (pyrrolidine-DNA) 3QI5 (εC-DNA)
MAG, Mag1			HhH	3mA, 3mG, 7mG, 7-CEG, 7-HEG, εA, Hx, G	3S6I (SpMag1/THF-DNA)
	AfAlkA		HhH	3mA, 7mG, εA, 1mA, 3mC	2JHN (AfAlkA)
	MpgII		HhH	3mA, 7mG	[Ref. 182]
		AlkA	HhH	3mA, 3mG, 7mG, 7-CEG, 7-HEG, 7-EG, O^2-mT, O^2-mC, εA, Hx, A, G, T, C, Xa	1MPG (EcAlkA) 1DIZ (EcAlkA/1-azaribose-DNA) 1PVS (EcAlkA/Hx base) 3OGD, 3OH6, 3OH9 (EcAlkA/undamaged DNA XL) 2H56 (BhAlkA) 2YG9 (DrAlkA)
		MagIII	HhH	3mA, mispaired 7mG	1PU6 1PU7 (MagIII/3,9-dimethylA) 1PU8 (MagIII/εA)
		TAG	HhH[1]	3mA, 3mG	2OFK (StTAG) 2OFI (StTAG/THF-DNA/3mA) 1NKU, 1LMZ (EcTAG NMR) 1P7M (EcTAG/3mA NMR) 4AIA (SaTAG)
		AlkC, AlkD	ALK/ HEAT	3mA, 3mG, 7mG, 7-POB-G, O^2-POB-C	3BVS (AlkD) 3JX7 (AlkD/3d3mA-DNA) 3JXY (AlkD/G•T-DNA) 3JXZ (AlkD/THF•T-DNA) 3JY1 (AlkD/THF•C-DNA)
Adenine					
MUTYH	MutY	MutY	HhH/FeS[2]	A•8oxoG, A•G,	1MUY, 1KG2, 1KG3 (EcMutY CD) 1RRQ (GsMutY/A•8oxoG-DNA XL) 1RRS, IVRL (GsMutY/HPD•8oxoG-DNA XL/adenine) 3N5N (HsMUTYH) 1WEF (K20A EcMutY CD) 1WEG, 1KG4 (K142A EcMutY CD) 1WEI (K20A EcMutY CD/adenine) 1KG5 (K142Q EcMutY CD) 1KG6 (K142R EcMutY CD) 1KG7 (E161A EcMutY CD) 1KQJ (C199H EcMutY CD) 1MUD (D138N EcMutY CD/adenine) 1MUN (D138N EcMutY CD)
U/T/5mC					
UDG		Ung	UDG-1	U•G	1AKZ (HsUDG) 1SSP (HsUDG/U-DNA) 1LAU, 1UDG (HSV1 UDG) 1UDH (HSV1 UDG/uracil) 1EUG, 2EUG, 3EUG, 5EUG (EcUng/U, EcUng/glycerol)

(*continued on next page*)

Table 1 (*continued*)

Eukaryotes	Archaea	Prokaryotes	Protein fold	Substrates	PDB entries
SMUG			UDG-3	U (ssDNA), U•G, U•A, 5hmU, 5-OHU, 5fU	1OE4 (XlSMUG/THF-DNA) 1OE5 (XlSMUG/THF-DNA/U) 1OE6 (XlSMUG/THF-DNA/5hmU)
TDG		MUG	UDG-2	T•G, U•G, U•A, 5fC, 5caC, 5FU•G, 5FU•A, 5BrU•G, 5BrU•A, 5hmU•G 5-OHU•G, Tg•G, εC•G, εC•A, Hx•G, 8hmεC, εG, Xa	2D07 (HsTDG/SUMO3) 1WYW (HsTDG/SUMO1) 2RBA (HsTDG/THF-DNA) 3UO7 (HsTDG/5caC-DNA) 3UFJ (HsTDG/dU analog) 1MWJ (EcMUG/U-DNA) 1MTL, 1MWI (EcMUG/AP-DNA) 1MUG (EcMUG)
	UDG		UDG-4	U (ssDNA), U•G	1UI0 (TtUDG)
MBD4			HhH	T•G, U•G, 5FU•G, εC, 5mC	1NGN (MmMBD4 CD) 3IHO (HsMBD4 CD) 4DK9 (HsMBD4/THF-DNA) 4EVV (MmMBD4/T•G-DNA) 4EW0 (MmMBD4/5hmU•G-DNA) 4EW4 (MmMBD4/AP-DNA)
	MIG		HhH/FeS[2]	T•G	1KEA
DME, ROS1, DML2, DML3			HhH/FeS[2]	5mC, T•G	Plants only; [Refs. 308, 311, 315, 323]

Abbreviations: AP, abasic site; THF, tetrahydrofuran; HPD, 1-hydroxypentane-3,4-diol; PDI, 3-hydroxypropyl; PED, pentane-3,4-diol; HC; hydantoin carbanucleoside; 8oxoG, 8-oxo-7,8-dihydroguanine; FapyG, 2,6-diamino-4-hydroxy-5-formamidopyrimidine; FapyA, 4,6-diamino-5-formamidopyrimidine; 7mFapyG, *N*7-methylFapyG; 7bFapyG; *N*7-benzylFapyG;Tg, thymine glycol; DHT, dihydrothymine; DHU, dihydrouracil; 5-OHC, 5-hydroxycytosine; 5-OHT, 5-hydroxythymine; 5,6dhC, 5,6-dihydroxycytosine; 5-OHU, 5-hydroxyuracil; 5mC, 5-methylcytosine; 5hmC, 5-hydroxymethylcytosine; 5fC, 5-formylcytosine; 5caC, 5-carboxylcytosine; 5hmU, 5-hydroxymethyluracil; 5fU, 5-formyluracil; 5FU, 5-fluorouracil; 5BrU, 5-bromouracil; Gh, guanidinohydantoin; Ia, iminoallantion; Sp, spiroiminodihydantoin; 3mA, *N*3-methyladenine; 3mG, *N*3-methylguanine, 7mG, *N*7-methylguanine; 7-CEG, 7-(2-chloroethyl)guanine; 7-HEG, 7-(2-hydroxyethyl)guanine; 7-POB-G, *N*7- pyridyloxobutylguanine; O^2-POB-C, O^2-pyridyloxobutylcytosine; εA, 1, N^6-ethenoadenine; εG, 1,N^2-ethenoguanine; εC, 3,N^4-ethenocytosine; 8hmεC, 8-(hydroxymethyl)-3,N^4-ethenocytosine; 3d3mA, 3-deaza-*N*3-methyladenine; Hx, hypoxanthine; Xa, xanthine; ssDNA, single-stranded DNA; XL, covalent cross-linked protein-DNA; CD, catalytic domain; O^2-mT, *O2*-methylthymine;O^2-mC, *O2*-methylcytosine.
Organisms: Hs, *Homo Sapeiens*; Mm, *Mus musculus*; Xl, *Xenopus laevis*; Mv, mimivirus; HSV1, Herpes simplex virus 1; Sc, *Saccharomyces cerevisiae*; Sp, *Schizosaccharomyces pombe*; Mt, *Methanothermobacter thermautotrophicus*; Af, *Archaeoglobus fulgidus*; Ss, *Sulfolobus solfataricus*; Tt, *Thermus thermophilus*; Gs, *Geobacillus stearothermophilus*; Mj, Methanocaldococcus jannaschii; Ec, *Escherichia coli*; Ll, *Lactobacillus lacti*; St, *Salmonella* typhi; Sa, *Staphylococcus aureus*; Bh, *Bacillus halodurans*; Dr, *Deinococcus radiodurans*.
[1] TAG adopts the HhH architecture, but lacks the conserved catalytic aspartate and lysine residues present in mono- and bifunctional HhH glycosylases.
[2] EndoIII, MutY, MIG, and DME/ROS incorporate Fe_4S_4-type iron sulfur clusters (FeS) into their HhH architecture.

side chain conformers that differed from their position in the LRC [73]. Taken together, these data demonstrated that hOGG1 recognition of 8oxoG within DNA occurs in multiple steps, and that 8oxoG excision relies on precise chemical compatibility within the base binding pocket.

hOGG1 has been regarded as a bifunctional DNA glycosylase involving two key catalytic residues, Asp268 and Lys249 [52,74–76]. The proposed catalytic mechanism involves Asp268-dependent deprotonation of the Lys249 ε-amino group, which forms a Schiff base with ribose C1′ of the 8oxoG nucleotide, resulting in β-elimination. However, various groups have reported monofunctional glycosylase activity for hOGG1 *in vivo* [71,77–80]. Recently, Dalhus and colleagues used structural and mutational analysis to show that the weak AP lyase activity in hOGG1 is an artifact of the proximity of Lys249 to the C1′ and may not reflect a physiological role [53]. A double Lys ↔ Cys swap mutant (Lys249Cys/Cys253Lys) abrogated AP lyase activity while maintaining 8oxoG excision activity, and a Lys249Cys/Cys253Lys/Asp268Asn triple mutant also eliminated the base excision activity. A crystal structure of the triple mutant revealed that Lys253 was too far (4.7 Å) away from the incoming C1′ to form the Schiff base, whereas Asn268 was in the same position as Asp268 in the wild-type enzyme. These results provided additional evidence for hOGG1 acting as monofunctional enzyme, in which Asp268 stabilizes an oxocarbenium intermediate during base hydrolysis [76,81] and Lys249 helps to position 8oxoG in the active site.

In addition to discrimination of 8oxoG from G, OGG1 shows a preference for the nucleobase opposite the lesion [56]. K_m values of mOGG1 are 42.7 ± 14.6 nM (8oxoG•C), 114 ± 28 nM (8oxoG•T), 233 ± 9.5 nM (8oxoG•G), and 2164 ± 502 nM (8oxoG•A) [71]. Specificity of hOGG1 for 8oxoG•C base pairs likely results from the five hydrogen bonds between the enzyme (Arg204, Asn149 and Arg154) and the opposing C, and substitution of Arg154 with histidine eliminates the specificity [52] (Fig. 4C). Structures of an OGG ortholog from the bacterium *C. acetobutylicum* CaOGG provided additional insight into specificity for the opposing base [61–63]. Whereas OGG1 has high preference for 8oxoG opposite C [56,71], CaOGG can excise 8oxoG opposite any base [61]. Structures of CaOGG in complex with DNA containing 8oxoG•C and 8oxoG•A showed that the bacterial protein maintains the fold and general DNA interactions as hOGG1, but lacks two of the five hydrogen bonds with the opposing nucleobase as a result of Met132 residing in place of the Arg154 in hOGG1 (Fig. 4C) [52,62]. In addition, the Asn149-cytosine hydrogen bond in hOGG1 is stabilized by Asn149's interaction with the hydroxyl group of Tyr203, which is missing in CaOGG (Phe179 at this position). A CaOGG Phe179Tyr mutant was 14-fold less efficient than the wild-type enzyme at excising 8oxoG•A, but did not affect 8oxoG•C excision. Moreover, the double mutant Phe179Tyr/Met132Arg, which mimics two of the critical interactions in hOGG1, was 50-fold less efficient at excising 8oxoG•A compared to the wild-type protein [61]. Thus, the fewer number of stabilizing contacts with

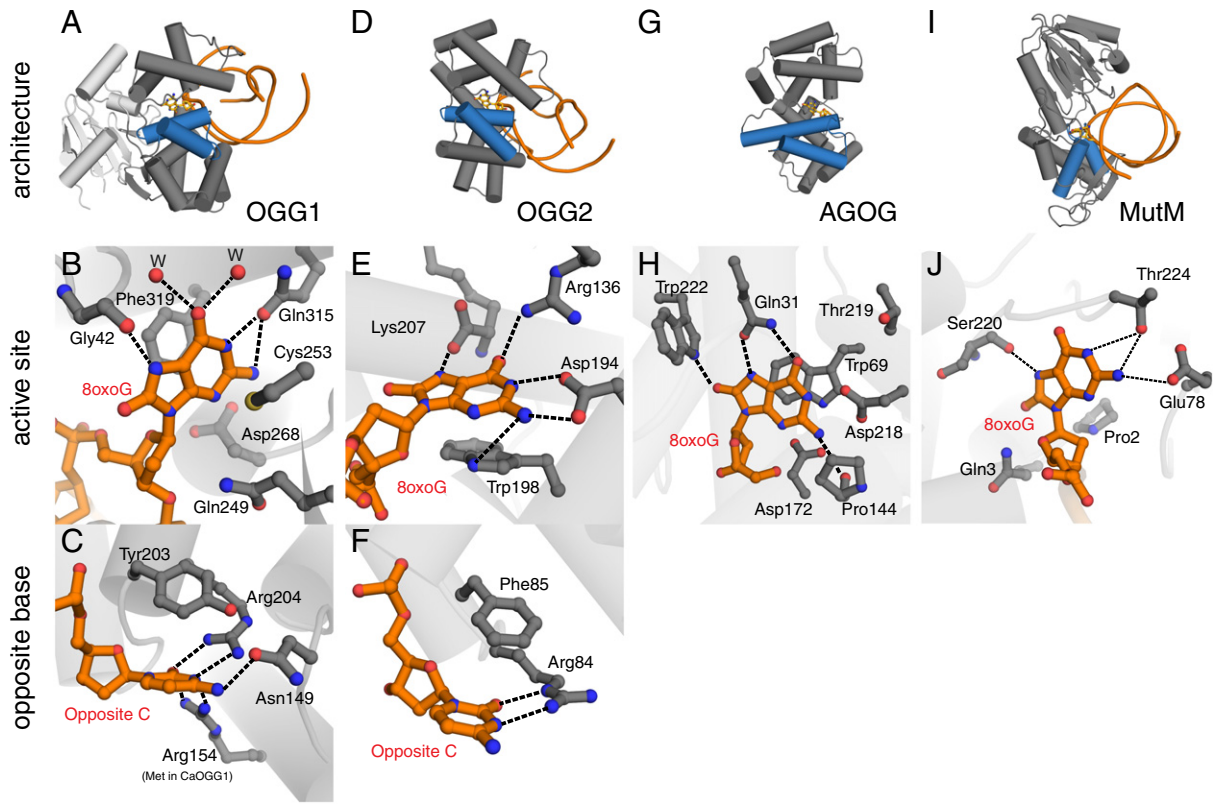

Fig. 4. Oxidative DNA glycosylases. (A–C) OGG1, represented by human OGG1 (PDB ID 1EBM), (D–F) OGG2, represented by MjOGG (3KNT), (G–H) *Pyrobaculum aerophilum* AGOG (1XQP) and (I–J) *Geobacillus stearothermophilus* MutM. The overall folds of each enzyme are shown on the top row (blue HhH motif), active sites on the second row, and opposing base on the bottom row. In the close-up views, the protein side-chains are grey and the DNA orange. Water molecules are represented by red spheres and hydrogen bonds are shown as dashed lines. (B) The human OGG1 8oxoG recognition pocket. The only 8oxoG specific contact is the hydrogen bond from the carbonyl group of Gly42 to the protonated N7 of 8oxoG. (C) The high specificity of hOGG1 for 8oxoG•C base pairs can be rationalized by the 5 hydrogen bonds between the opposite cytosine and 3 side chains. (E) In MjOGG, the 8oxoG N7 donates a hydrogen bond to the C-terminal Lys207 carboxylate. (F) The opposite cytosine in MjOGG is contacted by only one side chain. (H) 8oxoG nucleoside bound inside the AGOG active site, with a unique 8oxoG-specific contact to Trp222. (J) Active site of MutM (1R2Y) shows multiple contacts to 8oxoG but lacks the aromatic residues seen in the OGG1, OGG2, and AGOG enzymes.

and around the opposite base in CaOGG creates an environment that can accommodate other nucleobases at this position [52,62,63].

2.1.2. OGG2

The OGG2 family of DNA glycosylases consists of enzymes from various archaeal species that were predicted to be structurally similar to the OGG1 catalytic domain [64,82,83]. Despite very low sequence identity with hOGG1, structures of OGG2 from *Methanocaldococcus janischii* (MjOGG) and *Sulfolobus solfataricus* (SsOGG) confirmed that these enzymes adopt the HhH fold and contain the catalytic lysine and aspartate residues present in OGG1, but lack the N-terminal β-sheet domain [65] (Fig. 4D). The structure of MjOGG in complex with 8oxoG–DNA illustrated that the OGG2 family of enzymes utilize a distinct mechanism for identification of 8oxoG in the active site, in which the C-terminal carboxylate group of Lys207, as opposed to the Gly42 backbone carbonyl interaction in hOGG1, interacts with the N7 of 8oxoG [84] (Fig. 4E). Deletion of the three C-terminal residues abolished 8oxoG excision activity in MgOGG, but did not significantly affect enzyme integrity since the truncation only slightly diminished lyase activity [65]. Similar to CaOGG1, the OGG2 enzymes do not significantly discriminate against the base opposite 8oxoG [64,82,83], and this lack of specificity in OGG2 can be explained by the fewer contacts to the orphaned base relative to hOGG1; OGG2 forms two hydrogen bonds from a single residue, Arg84 (Fig. 4F), compared to the hydrogen bond pentad observed in hOGG1 [52] (Fig. 4C).

2.1.3. AGOG

AGOG is a recently discovered 8oxoG-specific DNA glycosylase from the aerobic hyperthermophillic archaeon, *P. aerophilum*, that removes 8oxoG from both ssDNA and dsDNA [66,85]. Like OGG2, AGOG has similar overall HhH fold and active site composition as that of hOGG1, but the specific residues contacting 8oxoG are not conserved in the two enzymes [86] (Fig. 4G). An 8oxoG base soaked into the crystal shows that the 8oxoG-specific hydrogen bond from N7 of the nucleobase (to Gly42 main chain carbonyl in OGG1) is mediated by the Gln31 side chain in AGOG. Substitution of Gln31 to serine caused a 180-fold reduction in catalytic activity (Gln31Ser $k_{cat} = 0.011 \pm 0.0004\ min^{-1}$) [87]. Unlike other 8oxoG glycosylases, AGOG also forms a direct hydrogen bond to the 8oxo moiety via the indole nitrogen of Trp69 (Fig. 4H), although this interaction may be dispensable since a Trp69Phe mutant did not significantly reduce activity [86,87]. Mutational analysis confirmed the roles of residues Trp222, Gln31 and Lys147 in substrate recognition and Asp172 and Lys140 in catalysis [87]. Like CaOGG and OGG2, AGOG shows no significant preference for the nucleobase paired with 8oxoG, with single turnover rates of 8oxoG excision of $3.15 \pm 0.03\ min^{-1}$ (8oxoG•C), $3.12 \pm 0.06\ min^{-1}$ (8oxoG•A), and $6.8 \pm 0.6\ min^{-1}$ (8oxoG•G) [87]. The basis for this cannot be determined from the current structure, although the robust activity for 8oxoG in ssDNA, which occurs at a rate of $5.4 \pm 0.4\ min^{-1}$ (nearly two-fold faster than 8oxoG•C), argues that the enzyme primarily contacts only the lesion-containing strand within the duplex [87].

2.1.4. MutM/Fpg

MutM/Fpg excises a number of oxidized nucleobases in addition to 8oxoG, including FapyG, hydantoins, Tg, DHU, and 5-OHU [49,88–91]. The crystal structure of *Thermus thermophilus* MutM/Fpg defined the

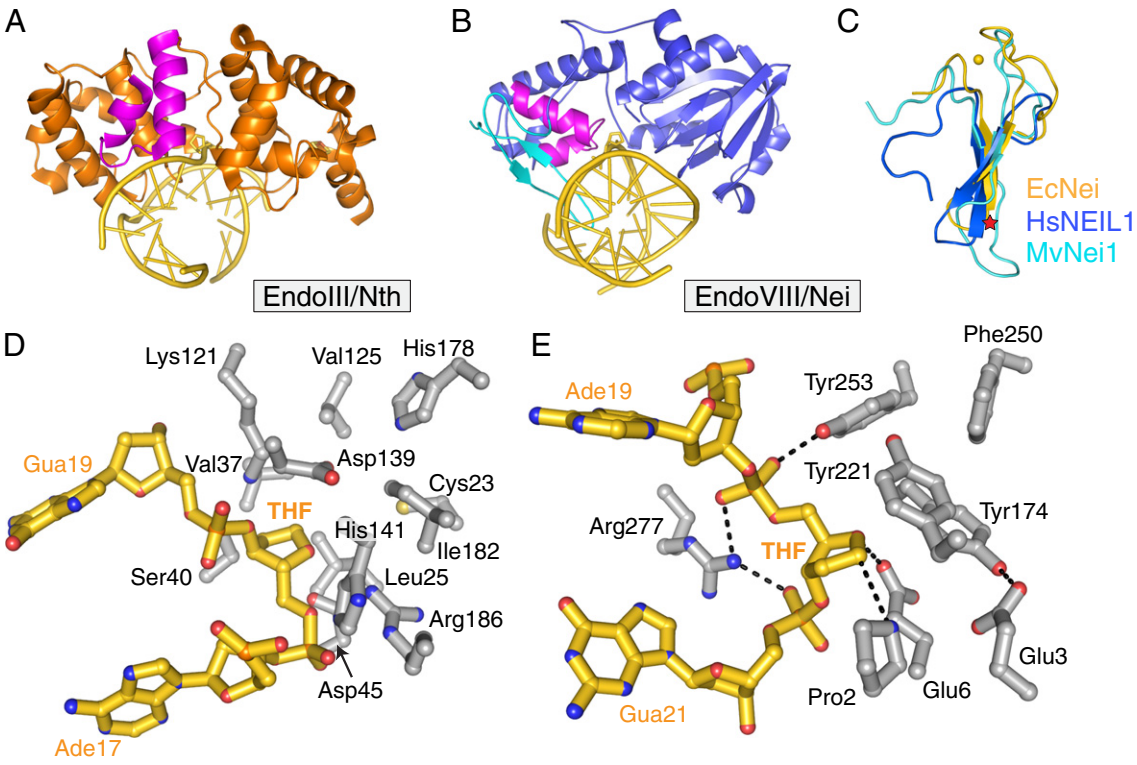

Fig. 5. EndoIII/Nth and EndoVIII/Nei. (A) *Bacillus stearothermophilus* Endonuclease III (BsEndoIII) (PDB ID 1P59) bound to THF-DNA (gold). The THF moiety and iron-sulfur cluster are shown as sticks in the center and right side of the figure, respectively. The HhH DNA binding motif is magenta. (B) Mimivirus NEIL1 ortholog MvNei1 (3A46) bound to THF-DNA (gold). The H2TH motif is colored magenta and the zincless finger is cyan. (C) Overlay of the zinc finger from *Escherichia coli* Endonuclease VIII (EcNei) (1K3W) (gold) with zincless finger motifs from human NEIL1 (1TDH, blue) and MvNei1 (cyan). The zinc ion in EcNei is depicted as a gold sphere. A red star denotes the general location of arginine residues that contact the DNA. (D) Active site of BsEndoIII (grey) bound to THF-DNA (gold). (E) Active site of MvNei1 bound to THF-DNA.

structural architecture as distinct N- and C-terminal domains separated by a flexible hinge [47] (Fig. 4I). The N-terminal domain is comprised of a two layer β-sandwich flanked by α-helices on either side and contains the catalytically important N-terminal proline and glutamate residues. The predominantly α-helical C-terminal domain contains the hallmark H2TH motif essential for DNA binding [49]. DNA-bound structures of MutM/Fpg from *Lactococcus lactis* [92] and *Geobacillus stearothermophilus* [93] revealed that the DNA was severely kinked by ~75° with the lesion flipped into the active site similar to other DNA glycosylases (Fig. 4J). Subsequent structures detailed the interactions of the enzyme with various substrates and abasic analogs, including 8oxoG, FapyG, DHU, tetrahydrofuran (THF), 1,3-propanediol (Pr), hydroxy propanediol and hydantoin carbanucleoside [94–97]. These structures illustrated that even though specific amino acids contacting the base in the active site may differ, the orientation of the backbone deoxyribose remains relatively unchanged, suggesting that catalysis proceeds by properly positioning the deoxyribose ring [97]. In addition, the MutM/abasic-DNA complexes suggested that β-elimination occurs concurrently with depurination, as opposed to sequential depurination-β-elimination reactions proposed previously for hOGG1, based on the fact that the enzyme sterically clashes with the cyclic, but not the ring-opened form of the deoxyribose [97,98].

More recently, a series of crystal structures of *G. stearothermophilus* MutM/Fpg from the Verdine laboratory provided detailed snapshots along the reaction pathway, illustrating how the enzyme actively interrogates the DNA duplex to differentiate between 8oxoG and guanine in the context of duplex DNA [4,20,93,95]. MutM ICs crosslinked with DNA containing normal A•T or G•C base pairs showed Phe114 probing the minor groove, with the interrogated base pairs severely buckled but remaining intrahelical [93,99]. In the LRC structure of MutM crosslinked to 8oxoG–DNA, the Phe114 residue fully penetrates the base stack and helps to induce a severe kink in the DNA that allows the target 8oxoG to become extrahelical [20]. In *E. coli* MutM, mutation of this phenylalanine to alanine (Phe111Ala) resulted in significantly reduced activity for 8oxoG excision and altered diffusion along DNA in single molecule studies [99]. The side chains of Met77 and Arg112 fill the space vacated by the flipped 8oxoG, with the Arg112 guanidinium moiety interacting with the Watson–Crick face of the estranged cytosine [20]. A third set of so-called encounter complexes (ECs) with 8oxoG–DNA or Gua–DNA were determined using a variant form of *G. stearothermophilus* MutM that has an altered or absent 8oxoG capping loop, which normally interacts with 8oxoG in the active site [100,101]. These complexes showed that MutM can detect the presence of intrahelical 8oxoG in the duplex based on local steric effects that influence the surrounding phosphate backbone. Recent data from the *E. coli* enzyme showed that the interaction with the 8oxoG capping loop is specific for 8oxoG, since an EcMutM/Fpg variant lacking the tip of the capping loop can efficiently excise mFapyG, DHU, Sp, and Gh but not 8oxoG [102]. Furthermore, a recent study showed that hydrophobic isosteres of 8oxoG are good, and in some cases better, substrates for Fpg, demonstrating that hydrogen bonding to the base is not important for efficient excision by Fpg [103].

2.2. Repair of oxidized pyrimidines and hydantoins

2.2.1. EndoIII/Nth/NTH1

Bacterial EndoIII/Nth and human NTH1 are bifunctional DNA glycosylases that use an aspartate/lysine catalytic pair to excise a variety of oxidized pyrimidine lesions (Table 1) and nick the backbone at the resulting AP site [104]. Tg, the preferred substrate with a K_m of 10 nM, is excised three- to four-fold faster than DHT, which is preferred over 5-OHC and 5-OHU [91,105,106]. The structure of the *E. coli* enzyme was the first to describe the HhH architecture for any

glycosylase and the inclusion of a [4Fe–4S]-type iron-sulfur cluster in any DNA binding protein [14]. More recently, the crystal structure of *G. stearothermophilus* EndoIII covalently tethered to DNA, along with subsequent modeling experiments, suggested that the broad substrate specificity is a consequence of the highly polar nature of the active site, and that the enzyme may recognize its substrates on the basis of glycosidic bond stability [107]. EndoIII binds DNA in the minor groove, bends the DNA at the site of the lesion, and extrudes the modified nucleobase into an active site pocket (Fig. 5A). A unique feature of this particular HhH enzyme is the extensive contacts made to the DNA backbone of the strand opposite the lesion. A glutamine side chain plugs the DNA gap and an intercalated leucine stabilizes the estranged base [107] (Fig. 5D). There are currently no known structures of human NTH1, although sequence and substrate similarities [108] suggest the current bacterial and archaeal EndoIII/Nth structures are accurate representations of the human ortholog. An outstanding question remains regarding the significance of the iron–sulfur cluster in these and other DNA repair enzymes, although studies point to a possible role in DNA damage detection based on the iron–sulfur redox potential [109–111].

2.2.2. EndoVIII/Nei

Like Nth, bacterial Nei (EndoVIII) is a bifunctional DNA glycosylase specific for oxidized pyrimidines [112]. Whereas Nth is a member of the HhH superfamily, Nei is structurally similar to MutM/Fpg and contains tandem H2TH/antiparallel β-hairpin zinc finger motifs that bind and stabilize the kinked DNA substrate, and a catalytic N-terminal proline [48]. The disparate substrate specificities of Nei and MutM/Fpg are reflected in the fact that residues involved in substrate recognition differ between these enzymes [48]. The structure of a covalently trapped DNA complex of *E. coli* Nei revealed that the protein undergoes a significant interdomain conformational change upon DNA binding [113]. This conformational switch between free (open) and DNA bound (closed) states, similar to that observed in other DNA binding proteins (e.g., *lac* repressor bound to target site [114]), has not been observed in other DNA glycosylases, although it has been proposed that the MutM/Fpg proteins may have some degree of conformational flexibility in solution [47,48,113]. For a more in depth review of EndoVIII/Nei, see reference [112].

2.2.3. NEIL1

Three eukaryotic Nei-like orthologs, NEIL1-3, have been discovered in humans [115–119] and have been characterized to some extent both functionally and structurally [recently reviewed in 120]. Like Nei, the NEIL orthologs are bifunctional H2TH glycosylase/AP lyases that excise a broad spectrum of oxidized pyrimidines and ring-opened purines (Table 1). Specifically, NEIL1 has a preference for Gh, Sp, Tg, 5-OHU, DHU, and Fapy, but also has been shown to have activity toward DHT, 5fU, 5hmU, 5-OHC, urea, and even abasic sites within a variety of structural contexts, including ssDNA, dsDNA, bulges, and bubbles [106,116–118,121–128]. NEIL2 primarily cleaves 5-OHU, but has not been shown to excise Tg or 8oxoG [115]. NEIL3 has a preference for various oxidized purines and pyrimidines in ssDNA and bubble structures [129] and has been reported to remove Gh and Sp hydantoins from both ss- and dsDNA [129]. In contrast, NEIL2 removes Gh and Ia from ss- and dsDNA, but Sp from ssDNA only [130]. NEIL1 has weak activity for 8oxoG in dsDNA, but unlike NTH1 and OGG1, NEIL1 can excise 8oxoG located near the 3′ end of single-strand breaks, suggesting that NEIL1 is not simply a back-up glycosylase for NTH1 and OGG1 but instead reinforces its unique substrate specificity [131,132]. The ability of NEIL enzymes to remove ssDNA lesions and their interactions with several replication proteins implicates them in DNA repair during S-phase [116,121,127]. Interestingly, NEIL1 was shown to remove psoralen-induced monoadducts and interstrand crosslinks (ICLs) in dsDNA, implicating it in nucleotide excision repair (NER) [133].

The NEIL enzymes are proposed to operate by a mechanism similar to Nei despite a few differences among them. Most notably, NEIL1 lacks the zinc finger motif present in Nei, NEIL2 and NEIL3 (Fig. 5C). This so-called zincless finger retains many aspects of the antiparallel β-hairpin zinc finger motif, but the loops that coordinate a zinc ion are missing [134]. The crystal structure of NEIL1 also revealed the position of a conserved arginine in the zincless finger that was confirmed by mutagenesis to be critical for glycosylase activity [134]. Structures of a viral NEIL1 ortholog (MvNei1) bound to THF-DNA illustrated how this longer β-hairpin loop of the zincless finger interacts with the strand opposite the lesion [135] (Fig. 5B). Like in other glycosylases, the DNA bound to MvNei1 is kinked at the site of the lesion and the THF moiety is flipped out of the duplex [135]. Both DNA-bound and free MvNei1 structures superimpose on the closed conformation of Nei, demonstrating that the large-scale domain movements notable in EcNei are not observed in MvNei1, although small-scale movements in the catalytic proline and the zincless finger to accommodate the DNA are evident upon DNA binding [48,113,135]. A tyrosine residue (Tyr221) in the proposed lesion recognition loop stacks against the abasic site (Fig. 5E) and most likely is in an alternate conformation in the presence of substrate, although the side chain could not be discerned in structures of MvNei1 bound to Tg- and 5-OHU–DNA [135,136]. Recognition of the pyrimidine ring takes place through a hydrogen bond interaction between the main chain amide of Tyr221 and the O4 of Tg and 5-OHU [136]. Two other residues (Glu6 and Tyr253) are within hydrogen-bonding distance, and mutation of these residues decreased the rate of Tg excision 7-fold (Tyr253) and 4-fold (Glu6), but had no significant effect on 5-OHU activity [136].

The plasticity of the active site to accommodate different oxidative lesions while discriminating against 8oxoG has been illustrated by homology modeling and molecular dynamics simulations [137]. Interestingly, A

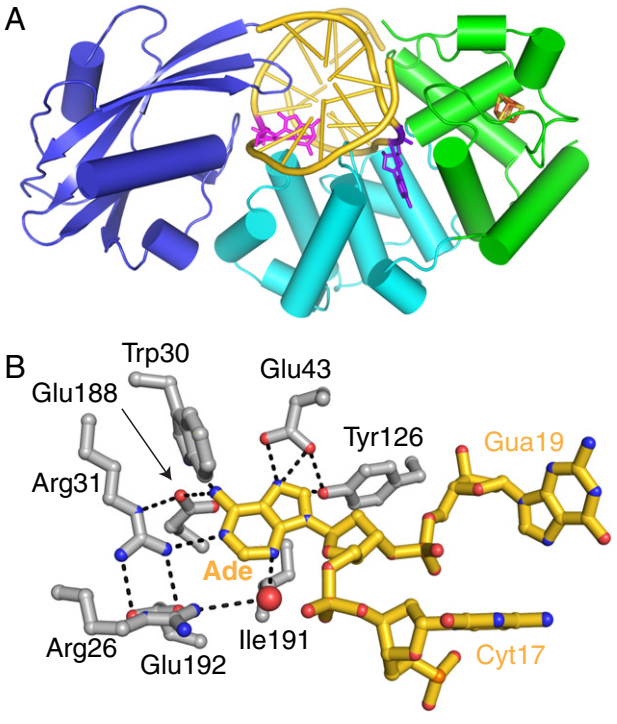

Fig. 6. Crystal structure of *Bacillus stearothermophilus* MutY. (A) Overall structure of BsMutY (PDB ID 1RRQ) colored by domain (green, iron-sulfur cluster domain; cyan, catalytic domain; blue, C-terminal (8oxoG recognition) domain). The DNA is colored gold with adenine substrate in purple and opposite 8oxoG in magenta. (B) Active site details of the MutY fluorinated lesion recognition complex (FLRC) bound to adenine-DNA (3G0Q). Protein (silver) and nucleic acid (gold) atoms are shown as sticks, water molecules are shown as red spheres. Hydrogen bonds are shown as dashed lines.

to I editing by adenosine deamination on dsRNA leads to NEIL1 variants that contain either an Arg and Lys at position 242 in the lesion recognition loop and have different substrate specificities, implying that the substrate specificity of NEIL1 changes in response to cellular conditions and may even be modulated by protein binding partners [138–141]. For example, an interaction between the C-terminal domain of NEIL1 and flap endonuclease 1 (FEN-1) ($K_d = 0.2$ μM) stimulates 5-OHU excision activity by 5-fold [138]. NEIL1 also interacts with BER enzymes pol β and DNA ligase IIIα through the C-terminal region of NEIL1 [142].

2.3. Repair of A•8oxoG mismatches by MutY/MUTYH

Failure of MutM/OGG1 to excise 8oxoG prior to replication results in 8oxoG•A mispairs, the adenine of which is the substrate for MutY/MUTYH glycosylase [143,144]. BER of the resulting AP site restores the 8oxoG•C pair, providing another chance for MutM/OGG1 to eliminate the 8oxoG from the DNA [reviewed in 145]. Structures of the catalytic domain of *E. coli* (Ec) MutY bound to adenine base revealed a HhH–FeS architecture similar to EndoIII and provided details of the active site and a proposed catalytic mechanism for adenine excision [146,147]. Transition state analysis from kinetic isotope effect measurements confirmed a stepwise, dissociative (S_N1) reaction mechanism whereby Glu43 acts as a general acid to protonate adenine N7, creating a positive charge on the nucleobase that facilitates cleavage of the *N*-glycosidic bond. The resulting oxocarbenium ion in the DNA is likely stabilized by nearby Asp144 and converted to the product AP site upon nucleophilic attack by water [148]. A high-resolution crystal structure of EcMutY bound to adenine provided evidence that MutY-catalyzed β-elimination, involving Lys142, Lys20 and possibly Glu161, is an activity secondary to and separable from the depurination reaction, similar to that observed in hOGG1 (see Section 2.1.1) [147].

A similar disulfide crosslinking strategy employed in the OGG1 and MutM structures was used to obtain structures of the full-length *B. stearothermophilus* homolog (BsMutY) anchored to 8oxoG•A-DNA [149,150] (Fig. 6A). In this structure, the adenine is flipped into the glycosylase active site but remains uncleaved as a result of mutation of the catalytic aspartate (Asp144Asn) [149]. Surprisingly, no direct hydrogen bonds were observed between the catalytic domain and the extrahelical adenine substrate. A subsequent structure of a catalytically proficient (Asp144) BsMutY crosslinked to DNA containing a non-hydrolyzable 2′-fluorinated deoxyadenosine showed adenine deeper into the active site and directly hydrogen bonded to Gln43, Tyr126, Arg31, Glu188, and Trp30 [150] (Fig. 6B). Mutation of the Glu188 residue in EcMutY (Gln182) decreased binding and activity for 8oxoG•A

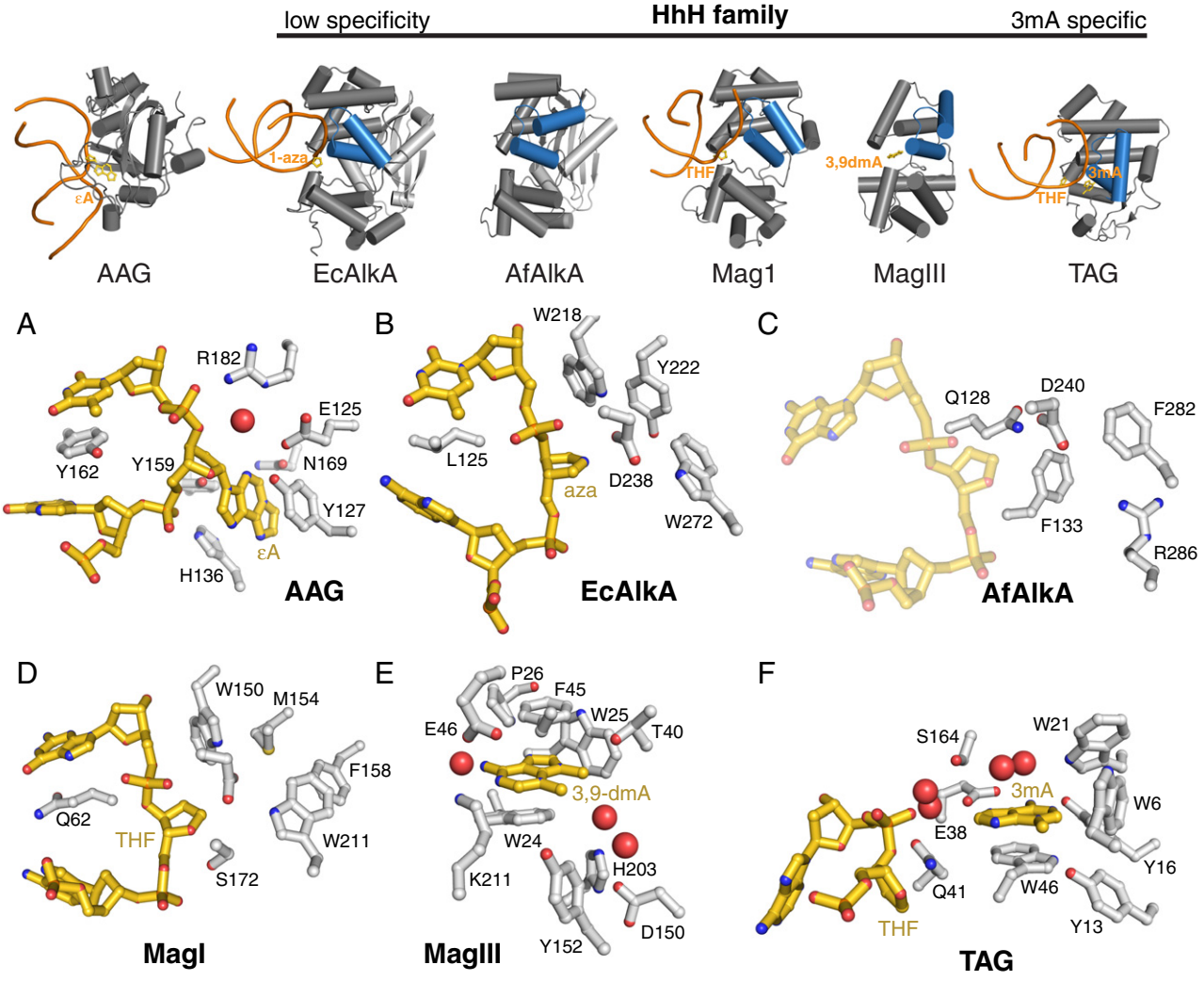

Fig. 7. Alkylpurine DNA glycosylases. Overall architectures are shown on the top row, with HhH enzymes arranged in order of increasing specificity for 3mA. (A–F) Active sites. Protein and nucleic acid atoms are shaded grey and gold, respectively, and waters are shown as red spheres. (A) Human AAG/εA-DNA substrate complex (PDB ID 1EWN). (B) *E. coli* AlkA bound to 1-azaribose-DNA (1DIZ). (C) *A. fulgidus* AlkA (2JHJ) with THF-DNA modeled from the *S. pombe* MagI/DNA complex (3S6I). (D) *S. pombe* Mag1/THF-DNA (3S6I). (E) *H. pylori* MagIII/3,9-dimethyladenine (1PU7). (F) *E. coli* TAG/THF-DNA/3mA product complex (2OFI).

and G•A mismatches but increased binding affinity toward 8oxoG•T and G•T mismatches, which are not normal substrates for MutY [151]. Cellular repair assays on the *E. coli* enzyme confirmed the importance of Asp138 (BsMutY144) and Glu37 (BsMutY Glu43) for the excision of adenine opposite 8oxoG [152].

The C-terminal domain contributes specific contacts to the stacked 8oxoG lesion that are functionally important for lesion recognition and enzyme activity. Tyr88 intercalates the duplex and stacks against the 8oxoG nucleobase, and Gly260 contacts the phosphate 5′ to 8oxoG [149]. Inherited mutations at these positions in MUTYH (Tyr165Cys and Gly382Asp) have been implicated in the development of colorectal cancer [153]. Substitution of the analogous residues in EcMutY (Tyr82Cys and Gly253Asp) reduce the DNA binding and base excision activities relative to the wild-type enzyme and the glycine has been implicated in discrimination of 8oxoG from G [154,155]. Furthermore, enzymatic studies with modified substrates *in vivo* demonstrated that MutY cannot effectively process adenine paired with guanine or modified forms of 8oxoG, whereas changes made to the target adenine are tolerated [156], implying that recognition of the 8oxoG by the C-terminal domain is necessary for locating the misincorporated adenine.

A crystal structure of a human MUTYH consisting of the catalytic domain and the interdomain connector (IDC) that tethers the catalytic and C-terminal domains was recently determined [157]. The human IDC sequence, which is not conserved in prokaryotic MutY, has been reported to recruit the Rad9, Rad1, Hus1 (9-1-1) complex involved in genome maintenance in eukaryotes [158–160]. Mutations in the IDC disrupted the MUTYH-9-1-1 interaction and decreased DNA repair of oxidative lesions *in vivo*, suggesting that structural studies of the human enzyme will reveal insights into its broader role in maintaining genome integrity [157,160].

3. Alkylation damage

A diverse array of alkylated DNA adducts are produced by environmental mutagens, cellular metabolites, and chemotherapeutic agents (Fig. 1B) [1,161–163]. The major and minor groove-exposed N7 and N3 positions of purines make them susceptible to reaction with electrophiles, with guanine N7 being the most nucleophilic [164]. Whereas *N*7-methylguanine (7mG) is relatively innocuous compared to larger *N*7-alkyl substituents, the positive charge generated from *N*7-substitution destabilizes the base and leads to spontaneous depurination and ring decomposition to produce, for example, 5-*N*-methyl-2,6-diamino-4-hydroxyformamidopyrimidine (mFapyG). The glycosidic linkage of *N*3-methyladenine (3mA) is especially unstable, with a half-life for 3mA depurination as short as 24 h at 37 °C [165]. Reactive aldehydes and epoxides generated from lipid peroxidation produce a number of ethenoadducts with A, G, and C, including 1,N^6-ethenoadenine (εA), 1,N^2- and N^2,3-ethenoguanine (1,N^2-εG and N^2,3-εG), and 3, N^4-ethenocytosine (εC) [1,166,167]. In general, these lesions cause genomic instability through mutations and strand breaks [1]. 3mA is cytotoxic, likely as a result of inhibition of DNA synthesis caused by disruption of the contacts between DNA polymerase and the adenine N3 position in the minor groove [168–171].

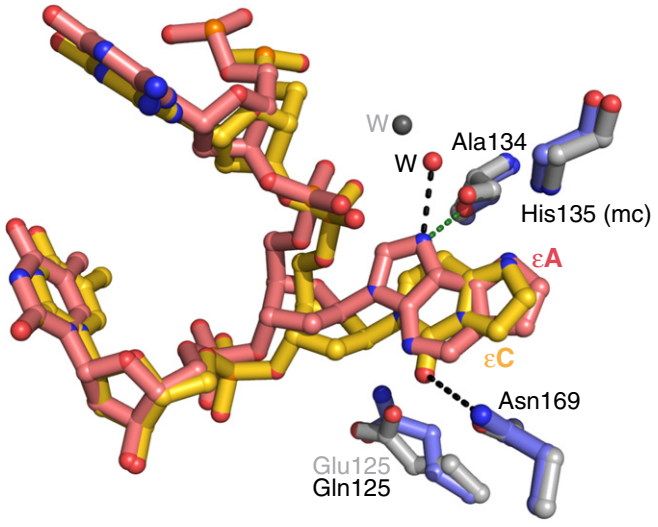

Fig. 8. Binding of εA and εC to AAG. The εA complex (PDB ID 1EWN) is colored blue (protein) and salmon (DNA), and the εC complex (3QI5) is silver and gold. Hydrogen bonds are depicted as dashed lines. A water molecule (red sphere) is in position to protonate N7 of εA, and protonated N7 would donate a hydrogen bond to Ala134 (green dashed line). εC does not have an ionizable group at this position.

DNA glycosylases specific for alkylation damage have been characterized from eukaryotes, archaea, and bacteria. These include human AAG/MPG/ANPG [172,173], *Saccharomyces cerevisiae* MAG and *Schizosaccharomyces pombe* Mag1 [174–176], *E. coli* 3mA DNA glycosylase I (TAG) and II (AlkA) [177,178], *Archaeoglobus fulgidus* AlkA (AfAlkA) [179,180], *Deinococcus radiodurans* AlkA (DrAlkA) [181], *Thermotoga maritima* MpgII [182], *Helicobacter pylori* MagIII [183], and *Bacillus cereus* AlkC and AlkD [184]. AAG, AlkA, and MAG/Mag1 excise a broad range of alkylated and deaminated bases [179,185–191]. Interestingly, AfAlkA has robust activity toward *N*1-methyladenine (1mA) and *N*3-methylcytosine (3mC), which are normally repaired by oxidative demethylation [179,180,188]. In contrast, TAG is highly specific for *N*3-methylpurines 3mA and 3mG [192], and MagIII, MpgII and AlkC/D are selective for positively charged lesions (e.g., 3mA and 7mG) [182–184]. The alkylpurine DNA glycosylases can be grouped into three structural classes: 1) AAG, defined by the human enzyme, 2) ALK, including AlkC and AlkD, and 3) HhH, comprising all others (Fig. 3) [193]. Despite their different architectures, AAG and the HhH enzymes have similar active sites that contain aromatic, electron-rich side chains that stack against the extrahelical alkylpurine substrate (Fig. 7) [193–196], whereas the ALK family is distinct structurally and mechanistically from the canonical base-flipping enzymes [12].

3.1. AAG

Human AAG, also known as MPG and ANPG, excises a variety of alkylated purines, including 3mA, 7mG, and εA, as well as hypoxanthine (Hx), the oxidative deamination product of adenine (Fig. 1B) [197,198]. The exceptional rate enhancement of Hx excision relative to alkylated substrates suggests that Hx is the predominant biological substrate [199]. AAG has also been shown to excise *N*1-methylguanine and 1,N^2-εG [200,201]. Crystal structures of a catalytic fragment of AAG bound to oligonucleotides containing either a pyrrolidine transition-state analog or an εA nucleobase showed that AAG is a single domain protein with a mixed α/β structure and a positively charged DNA binding surface [195,202]. The flipped εA base is stacked between two tyrosine residues (Tyr127 and Tyr159) and His136 inside the active site cavity, while Tyr162 on the tip of a β-hairpin plugs the gap in the DNA left by the flipped nucleotide (Fig. 7A). These structures provided a framework for a number of recent kinetic and thermodynamic studies aimed at dissecting AAG's mechanism, substrate specificity, and collaboration with other BER enzymes. These studies are described in detail and referenced in the following sections.

3.1.1. Mechanism of base flipping and substrate discrimination

A series of careful biochemical examinations of substrate binding, flipping, and excision by AAG has recently been reported. As is typically true for other glycosylases, substrates that decrease the stability of the DNA increase the efficiency of excision by AAG, with bulged nucleotides excised more efficiently than mismatched base pairs [199,203]. Interestingly, the strength of AAG binding to bulges correlates with

increased spontaneous frameshift mutations upon overexpression of the enzyme, which may be a result of AAG shielding bulged bases from mismatch repair [204]. Discrimination of nucleobases on the basis of their stability within the DNA duplex can be rationalized by the barrier to base flipping. Kinetic analysis using intrinsic εA fluorescence revealed that εA flipping by AAG is highly favorable, which helps to explain discrimination of this lesion from undamaged bases [205]. These experiments also generated a two-step binding regime in which distinct DNA-bound and base flipped complexes form on the millisecond to second time scale, whereas *N*-glycosidic bond cleavage takes place on the minute time scale [205,206]. Thus, destabilized base pairing allows AAG to selectively excise DNA lesions. More stringent selection takes place inside the active site, in which side chains create steric clashes with unmodified A and G bases [199,202,207].

Excision of neutral substrates by AAG has been shown by pH-activity profiles to employ both a general acid and general base [208]. The general acid acts to protonate the nucleobase, facilitating its dissociation, and the general base would deprotonate a catalytic water molecule to attack C1′ [208]. Consistent with such a mechanism, excision of positively charged lesions (e.g., 7mG) does not require the general acid [208]. Although the identity of the general acid has not been determined, the necessity to protonate the base explains the specificity of AAG for purines versus pyrimidines [208]. Quantum mechanical modeling studies indicate that base excision by AAG is facilitated by π–π interactions between the enzyme and its substrate DNA, consistent with the structures, and suggest that the nucleobase is not fully protonated but rather hydrogen bond donation by a protein-bound water molecule lowers the catalytic barrier [209].

3.1.2. Structural basis of AAG inhibition by εC

In addition to εA, AAG has a modest activity toward 1,N^2-εG [200]. Although AAG binds εC with a 2-fold greater affinity than εA [210], AAG is incapable of excising εC, which is normally removed by the uracil/thymine DNA glycosylase family of enzymes (see Section 4) [211–213]. A recent structure of AAG in complex with εC–DNA showed εC to reside in the active site in a virtually identical position as εA [210] (Fig. 8). The hydrogen bond between His136 and εA (N^6) is preserved to εC (N^4), and as a consequence the εC nucleotide is pulled slightly farther into the binding pocket. The enhanced binding to εC may be explained by one additional hydrogen bond between the protein (Asn169) and O^2 of εC, which is not present in εA. Regarding inhibition, protonation of substrate purines likely occurs at the N7 nitrogen [208], and crystal structures suggest that a protonated N7 would be stabilized by a hydrogen bond to the backbone oxygen of Ala134 (Fig. 8). The AAG/εC-DNA structure proposes that inhibition by εC is due to the inability of AAG to protonate εC, which lacks a nitrogen at the position corresponding to N7 of εA. In addition, this structure also showed an octahedral coordinate Mn^{2+} ion bound to the guanine opposite the εC that perturbed the guanine sugar pucker. This was the first observation of bound divalent ion to AAG and suggested that inhibition of the enzyme by divalent ions might be a consequence of impaired base flipping or duplex opening to expose the substrate base [210]. AAG has also been trapped onto εC-DNA in a non-specific orientation, providing a structural basis for the enzyme's ability to bind single-base bulges [204,214].

3.1.3. Product release and diffusion along DNA

Single- and multiple-turnover kinetic experiments have shown that the rate-limiting step of hypoxanthine hydrolysis by AAG is the release of the abasic DNA product [215]. In fact, the tight binding of AAG to product DNA enables AAG to catalyze the reverse reaction to re-form the *N*-glycosidic bond [216]. Product release is promoted by APE1, the next enzyme in the BER pathway [217]. Displacement of the glycosylase by APE1 has also been observed for TDG and OGG1 [218–220]. The nonspecific binding of both AAG and APE1 to DNA suggests that these enzymes may bind DNA simultaneously and facilitate a handoff of the abasic site from AAG to APE1. Baldwin and O'Brien propose that APE1 displaces AAG from the AP site without a direct protein-protein interaction, and that AAG remains bound to the DNA upon AP dissociation [215]. The processivity of AAG along DNA is dependent on ionic strength, indicating a reliance on electrostatic interactions with the DNA backbone. Furthermore, the amino terminal 80 amino acids, which are not necessary for catalysis by AAG, contribute to the enzyme's ability to diffuse along DNA [221].

3.2. HhH superfamily

The majority of yeast, archaeal, and bacterial alkylpurine DNA glycosylases adopt the HhH protein fold, with the exception of AlkC/AlkD and bacterial orthologs of human AAG [222,223]. The HhH glycosylases contain two α-helical domains with the active site cleft located at their interface. The domain containing the HhH motif and DNA intercalating residues is formed from an internal region of the primary structure and has a relatively conserved tertiary structure. The HhH anchors the protein to the DNA through a series of hydrogen bonds between main-chain atoms of the hairpin and the phosphoribose backbone downstream of the lesion. At the damage site, bulky side chains from neighboring loops fill the void left by the extrahelical nucleobase target and wedge into the base stack opposite the flipped out nucleotide. Both plug and wedge residues are important for stabilizing the bent conformation of the DNA and have been implicated in probing the DNA helix during the search process [224]. The second domain, formed from the N- and C-termini, is more structurally divergent and often contains additional structural elements, such as a zinc ion (TAG), iron-sulfur cluster (MpgII), or carbamylated lysine (MagIII) [193].

Comparative analysis of the HhH alkylpurine glycosylases has been instrumental in deciphering the physical and chemical determinants of substrate recognition [225]. On one hand, we have learned that the HhH scaffold accommodates a diverse array of nucleobase binding pockets that discriminate between lesions on the basis of shape complementarity. For example, AlkA's nucleobase binding surface is a shallow cleft that can accommodate a variety of alkylpurines, whereas the active sites of TAG and MagIII are more constrained and perfectly shaped for 3mA. On the other hand, this steric selection is not the only determinant of specificity since some active sites can accommodate nucleobases for which they do not efficiently excise (e.g., Mag1) [191]. In addition, the catalytic requirements for excision of cationic lesions 3mA and 7mG differ from the uncharged alkylpurines (e.g., εA) by virtue of their weaker *N*-glycosidic bonds [9]. Hence, the inherent instability of these lesions render their excision highly dissociative, and recent reports suggest that cationic lesions may be removed and even detected within DNA differently than neutral lesions [191,193,226].

3.2.1. E. coli AlkA

Crystal structures of unliganded AlkA identified the enzyme as a member of the HhH superfamily and revealed a shallow nucleobase binding surface that can accommodate a variety of alkylpurines, a feature that helped to explain its broad specificity [194,227] (Fig. 7B). In addition to the two-domain HhH architecture, AlkA contains an amino-terminal β-sheet domain of unknown function that is also present in OGG1 (Figs. 4 and 7). A structure of AlkA bound to DNA containing 1-azaribose, which mimics the oxocarbenium reaction intermediate, has contributed greatly to our understanding of these enzymes [196,228]. The HhH anchors the protein to the DNA and does not directly participate in lesion recognition. The DNA is kinked by ~60° around the 1-azaribose, which is rotated 180° around the phosphoribose backbone and stabilized by the Leu125 plug in the gap left behind (Fig. 7B). Rotation of the 1-azaribose into the active site places the N1′ nitrogen directly adjacent to the carboxylate group of the catalytic Asp238, which is in a prime location to stabilize the oxocarbenium intermediate

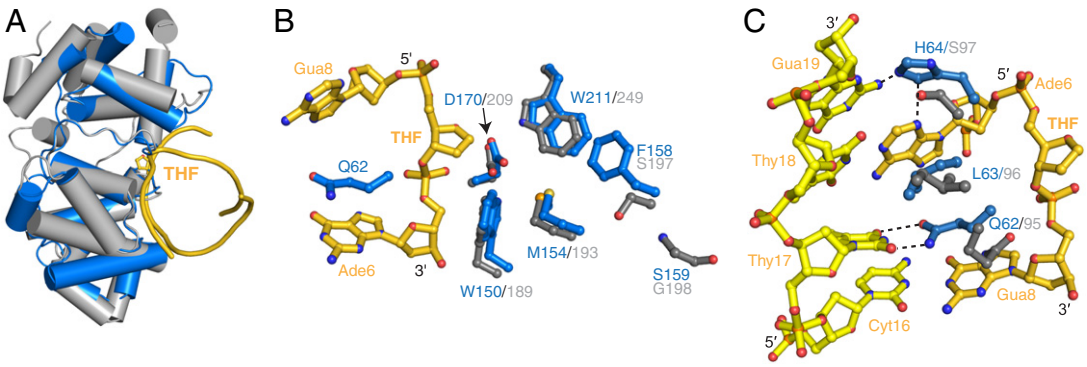

Fig. 9. Yeast 3-methyladenine DNA glycosylases MAG and Mag1. In all panels, the unbound *Saccharomyces cerevisiae* MAG (grey, unpublished) free enzyme is superimposed onto the *Schizosaccharomyces pombe* Mag1/THF-DNA complex (blue/gold, PDB ID 3S6I). (A) Overall structures. (B) Active sites. Mag1 residues Phe158 and Ser159 are the only two active site residues that differ between the two enzymes. (C) Close-up of Mag1-DNA contacts at the lesion. Swapping Mag1 His64 and MAG Ser97 between the two enzymes effectively swaps their respective abilities to remove εA (see text for details).

[196]. In addition to this lesion-specific binding mode, AlkA has the ability to bind to DNA ends [229], which may explain why a structure of AlkA bound to a substrate DNA has not been determined. Nonetheless, this feature was exploited to develop a host–guest crystallization strategy to determine structures of various lesions in DNA [230].

High resolution structures of AlkA crosslinked to undamaged DNA bases provided insight into how the enzyme detects damage within the context of unmodified DNA [224]. Not surprisingly, the most notable differences between these undamaged DNA complexes (UDCs) and the 1-azaribose lesion recognition complex (LRC) are centered around the lesion. The UDCs do not exhibit the kink present in the LRC DNA. The domain containing most of the catalytically important residues, including Asp238, is shifted 2.4 Å toward the lesion strand in the LRC compared to the UDCs. This movement, combined with a modest 1-Å shift of the Leu125 plug residue toward the lesion strand, clamps the lesion between the two domains and creates additional protein contacts that stabilize the LRC. In contrast, the HhH motif makes the same DNA contacts in LRC and UDC structures, providing additional evidence that the HhH motif is a non-specific DNA binding element and is not involved in distorting the DNA for catalysis. Leu125 in the UDCs does not interact with the DNA, although it is still present in the minor groove. The phosphate backbone in the LRC is significantly (~9 Å) closer to the protein, which allows the Leu125 side-chain to intercalate into the DNA base stack in that structure. A 3mA base modeled in place of a centrally located cytosine indicated that Leu125 likely makes van der Waals contacts to the *N*3-methyl group [224]. These observations suggest that AlkA employs a passive scanning mechanism along the minor groove and uses the Leu125 side chain to detect abnormal bases and flip them into the active site.

3.2.2. Archaeal AlkA

An AlkA ortholog from the archaeon *A. fulgidus* (AfAlkA), has been shown to excise 1mA and 3mC in addition to 3mA, 7mG, εA and Hx from DNA [179,180,188]. The crystal structure of this ortholog shows that the nucleobase binding pockets of AfAlkA and *E. coli* AlkA are strikingly different despite the similarity in their overall fold [180] (Fig. 7B, C). Mutation of the catalytic Asp240 (Asp238 in EcAlkA) completely eliminates base excision activity in AfAlkA. The substrate nucleobase is predicted to stack between Phe133 and Phe282, similar to stabilization of 3mA by MagIII (Section 3.2.4, Fig. 7E). In support of this, substitution of Phe133 or Phe282 with alanine diminishes εA and 1mA base excision, and the double mutant abrogates activity. Arg286 is predicted to orient εA in the active site through hydrogen bonding, but would potentially repel the protonated amine groups of 1mA and 3mC [180]. Thus, the AfAlkA structure is a nice example of how the versatility of the HhH scaffold allows for inclusion of various active sites that dramatically alters the enzyme-substrate specificity.

3.2.3. Yeast MAG/Mag1

S. cerevesiae MAG and *S. pombe* Mag1 are 42% and 47% similar in sequence to *E. coli* AlkA, respectively, but have a more restricted substrate specificity (Table 1) [225]. MAG excises 3mA, 7mG, εA, Hx, and guanine, but not oxidized substrates (e.g., O^2-methylthymine) from DNA, while Mag1 is more restricted to 3mA, 3mG, and 7mG and has only a modest activity toward εA [185,189–191,231–233]. These differences suggest that these proteins have different roles in protecting cells against alkylation damage [234,235]. For example, MAG deletion strains are more sensitive to alkylation agents than are *S. pombe mag1*, and MAG expression is induced to higher levels than Mag1 upon exposure to alkylation agents [235,236].

Our laboratory recently determined crystal structures of Mag1 bound to DNA containing a THF abasic analog [191] and of free MAG (unpublished results) (Fig. 9). Neither MAG nor Mag1 contain the mixed α/β domain present at the N-terminus of the AlkA orthologs (Fig. 9A). Nevertheless, Mag1 engages the THF–DNA similarly to AlkA, with the DNA bent by ~60° and the THF moiety rotated around the phosphate backbone toward the nucleobase binding pocket. Inside the active site, there are only two notable differences between MAG and Mag1. Mag1 residues Phe158 and Ser159 at the back of the binding cleft are occupied by Ser197 and Gly198 in MAG (Figs. 7D and 9B). Swapping these residues (Mag1 FS→SG and MAG SG→FS double mutants) did not affect their relative εA activities, providing evidence that the bulky Phe residue in the binding pocket is not responsible for the lower εA excision activity of Mag1 [191]. Interestingly, substitution of the catalytic aspartate residues had dramatically different effects. MAG Asp209Asn completely abrogated εA and 7mG excision activities similar to that observed for AlkA Asp238 [194], while Mag1 Asp170Asn had a more modest effect, implying that this residue in MAG plays a more significant role in catalysis, possibly explaining the broader substrate preference of this enzyme [191].

Outside of the active site, there is a notable difference between MAG and Mag1 at the point of contact with the DNA minor groove flanking the damage site (Fig. 9C). In addition to the plug and wedge residues, His64 in Mag1 is in position to hydrogen bond with either the N3 of the adenine immediately 5′ to the lesion or to the exocyclic N2 of the guanine on the opposite strand [191]. MAG and AlkA orthologs, including those from *Bacillus halodurans* and *D. radiodurans*, which have broad substrate preferences and for which crystal structures are available, contain a serine residue at this position (Fig. 9C) [181,191,225]. Surprisingly, swapping histidine and serine between Mag1 and MAG led to dramatic increase in εA excision rate in Mag1 and a decrease in εA excision in MAG, whereas the 7mG excision rates in both enzymes

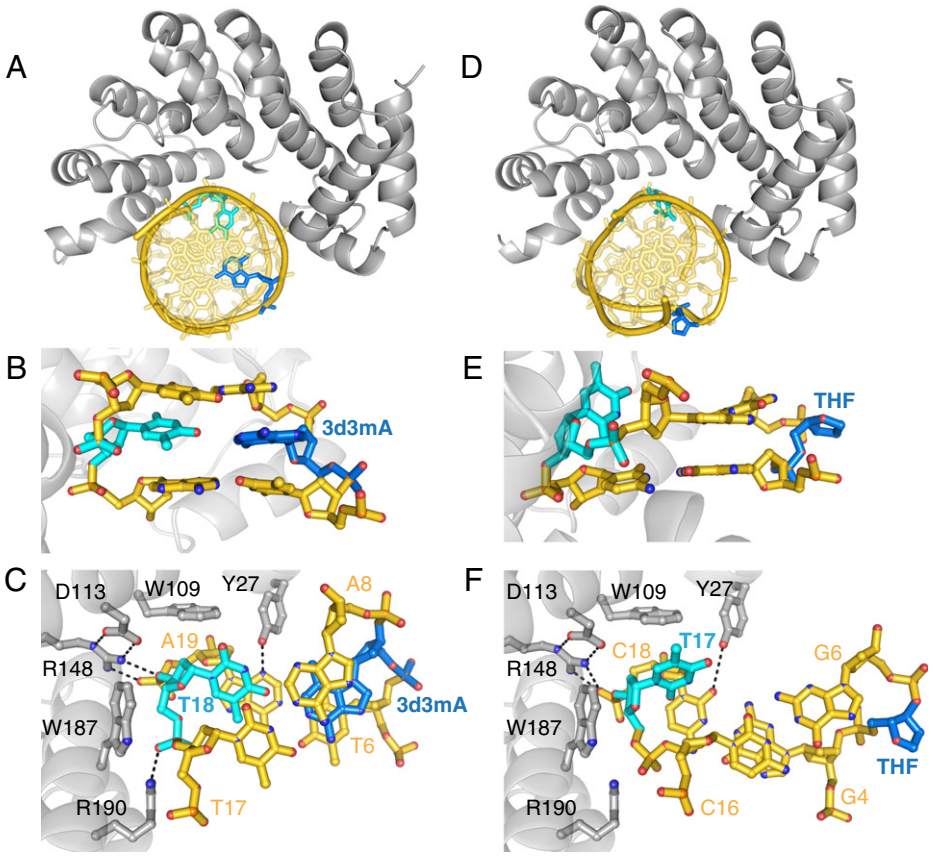

Fig. 10. Crystal structures of AlkD in complex with 3d3mA-DNA (PDB ID 3JX7) (A–C) and THF-DNA (3JXZ) (D–F). The protein is colored silver, DNA is gold, 3d3mA and THF lesions are blue and opposite thymines are cyan. (A,D) Overall structures showing DNA bound to the concave surface of the protein. (B,E) Side views of the base pairs flanking the lesions. (C,F) View rotated 90° with respect to panels A/D and B/E. Hydrogen bonds are shown as dashed lines.

remained the same [191]. Thus, contacts to the minor groove may be important for damage detection and/or stabilizing a specific enzyme-substrate complex for catalysis. These results also suggest that cationic and uncharged lesions may be detected or stabilized differently, although more work is required to test this hypothesis.

3.2.4. H. pylori MagIII and T. maritima MpgII

MagIII and MpgII are related alkylpurine glycosylases identified by their sequence similarity to EndoIII [182,183]. MagIII is highly specific for 3mA but can excise mispaired 7mG, whereas MpgII can excise both 3mA and 7mG [182,183]. The crystal structure of MagIII showed a unique feature in the N/C-terminal domain, which contains a carbamylated lysine (Lys205) that neutralizes an otherwise highly positively charged region of the protein [237]. MagIII's preference for 3mA can be explained by the snug fit of 3mA inside the active site, which partially excludes *N*7-substituted purines. Structures of MagIII bound to positively charged 3,9-dimethyladenine (3,9-dmA) and uncharged εA bases showed the nucleobases stacked between Phe45 and Trp24 and bounded on three sides by Trp25, Pro26, and Lys211 (Fig. 7E). Other than these van der Waals and π-stacking interactions, there were no specific hydrogen bonding or polar contacts to the adenine ring like those observed in TAG (see Section 3.2.5). Similar to Mag1, mutation of the putative catalytic aspartate Asp150 in MagIII did not completely abrogate base excision activity, again suggesting that the catalytic power of this residue determines the ability of the HhH enzymes to remove more stable, neutral nucleobases from DNA, and that little catalytic assistance is required for hydrolysis of the labile 3mA glycosidic bond [9,237].

Unlike MagIII, MpgII contains an iron-sulfur cluster and has robust activity toward 7mG, which is intriguing given the sequence similarity between MagIII and MpgII [182,225]. Although there is no structure for MpgII, sequence comparison predicts that only two residues differ within the active site: MpgII Trp52 and Lys53 are occupied by Phe45 and Glu46 in MagIII, respectively. The MagIII active site is constrained by a salt bridge between Glu46 and Lys211. Substitution of Glu46 with the corresponding lysine residue (Lys53) in MpgII should relieve this constraint from electrostatic repulsion. Indeed, a MagIII Glu46Lys mutant resulted in an 8-fold increase in 7mG•T activity, suggesting that steric exclusion of 7mG partially accounts for MagIII's low activity toward methylguanine bases [237].

3.2.5. E. coli TAG

TAG substrate preference is strictly limited to *N*3-substituted purines 3mA and 3mG [192]. NMR studies of *E. coli* TAG showed it to be a structurally divergent member of the HhH family, containing a zinc ion in the N/C-terminal domain and lacking the catalytic aspartate residue present in other 3mA DNA glycosylases [238–240]. Similar to MagIII, TAG's specificity can be partially attributed to the fact that the 3mA binding pocket would sterically exclude all other nucleobases (Fig. 7F). Binding studies and NMR investigation of 3mA in the active site led to the suggestion that TAG enhances the rate of 3mA depurination by binding tightly to the nucleobase, thereby destabilizing the ground state of the enzyme–substrate complex [240]. This idea was illustrated by crystal structures of a TAG/abasic-DNA/3mA product complex using the *Salmonella typhi* ortholog, which is 82% identical and 92% conserved overall with *E. coli* TAG [226]. In that structure, the bound DNA is more B-form when compared to the highly distorted 1-azaribose DNA bound to AlkA, and there was a large (7 Å) separation between the THF, which is not fully engaged inside the active site, and 3mA, which is buried deep inside the cleft. These observations indicated

that the DNA undergoes significant relaxation upon breakage of the *N*-glycosidic bond, suggesting that steric strain may contribute to bond cleavage [226]. A recent structure of *Staphylococcus aureus* TAG recapitulates the structural features observed in the *E. coli* and *S. typhi* structures, and the authors suggested that tautomerization of 3mA contributes to its recognition by TAG [241].

3.3. AlkC and AlkD

Recently, AlkC and AlkD were identified in *B. cereus* as two related alkylpurine glycosylases to be highly specific for 3mA and 7mG [184], and were predicted to represent a new structural superfamily of DNA glycosylases on the basis of their sequence similarity to an unpublished entry in the Protein DataBank (2B6C) [242]. The crystal structure of *B. cereus* AlkD confirmed this prediction [222]. AlkD is composed exclusively of HEAT repeats (Fig. 3)—tandem pairs of short α-helices that generate extended, non-enzymatic scaffolds that typically mediate protein but not nucleic acid interactions. To our knowledge, AlkD is the first HEAT repeat protein identified to interact with nucleic acids or to contain enzymatic activity [12]. AlkD's positively-charged, concave surface is perfectly suited to bind a DNA duplex, and is lined with highly conserved residues that are important for 7mG excision and DNA binding activities and for protection against bacterial sensitivity to alkylating agents [193,222,242].

High resolution crystal structures of AlkD in complex with DNAs resembling the substrate (3-deaza-3-methyladenine, 3d3mA) and product (THF) of 3mA excision confirmed that the DNA duplex is positioned via electrostatic interactions along AlkD's concave surface and revealed a novel lesion capture mechanism distinct from other glycosylases [193]. The 3d3mA and THF moieties are positioned on the side of the DNA facing away from the protein with no contact to the protein whatsoever (Fig. 10A,D) [193]. In the substrate structure, the 3d3mA•T base pair is sheared as a result of movement of the thymine into the minor groove toward the protein (Fig. 10B,C). In the product structures, both the abasic site and its opposing nucleobase are rotated out of the helix to create a single-base bulge with base stacking maintained by the flanking base pairs (Fig. 10E,F). The THF is flipped 180° around the phosphoribose backbone into a solvent exposed orientation, while the opposing base is tipped up and sandwiched between the minor groove and the protein.

Several distinguishing structural and biochemical features of AlkD indicate that it utilizes a unique mechanism to liberate positively charged bases from DNA [193]. Unlike the base-flipping glycosylases, AlkD lacks the plug residue universally used by DNA glycosylases to prevent the flipped substrate base from re-entering the DNA base stack. Second, AlkD is not inhibited by high concentrations of free nucleobase. Third, AlkD does not discriminate against the base opposite the lesion, and activity is dramatically *reduced* by a bulky pyrene opposite the lesion, counter to that found for the case of base flipping by UDG [243]. Fourth, AlkD liberates bulky, positively-charged pyridyloxobutyl (POB)-bases from DNA [193]. Thus, AlkD does not employ a specific nucleobase binding pocket to recognize or remove its substrates, suggesting that depurination of *N*3- or *N*7-alkylpurines can be facilitated without direct contact to the protein.

The 3d3mA and THF structures suggested that AlkD has the ability to detect and trap destabilized base pairs but would only excise modified nucleobases that contained weak *N*-glycosidic bonds. We therefore trapped AlkD in complex with a G•T mismatch in order to evaluate how the enzyme restructures DNA by comparing G•T–DNA in the free and AlkD-bound states. AlkD significantly resculpts the non-Watson–Crick base pair from the canonical wobble G•T structure in order to create an optimized protein–DNA binding surface by maximizing contacts between the phosphoribose backbone of the thymine strand and the concave cleft [193]. These specific protein–DNA contacts are identical to the 3d3mA•T structure (Fig. 10C), and substitution of the participating side chains either abolished or severely impaired 7mG excision, indicating that the specific DNA capture mechanism is a prerequisite for catalysis.

The specific structure of the DNA trapped in the AlkD complexes provides a rationale for the enzyme's specificity toward bases with a low threshold for depurination. As a result of the collapsed duplex, the phosphoribose backbone is highly kinked, which places the flipped THF in close proximity to a phosphate immediately 5′ to the lesion [193]. We have recently found that chemical perturbation of this phosphate to a methylphosphonate abolishes 7mG excision activity by AlkD (unpublished results), indicating that this phosphate participates in catalysis either directly, by stabilizing the oxocarbenium ion intermediate, or indirectly, by maintaining a specific kink in the duplex that weakens the *N*-glycosidic bond. Interestingly, a direct role of the DNA in catalysis of base excision has also been observed in uracil DNA glycosylase [244–246]. More work will be necessary to verify the structure of a bound 7mG–DNA that is activated for hydrolysis.

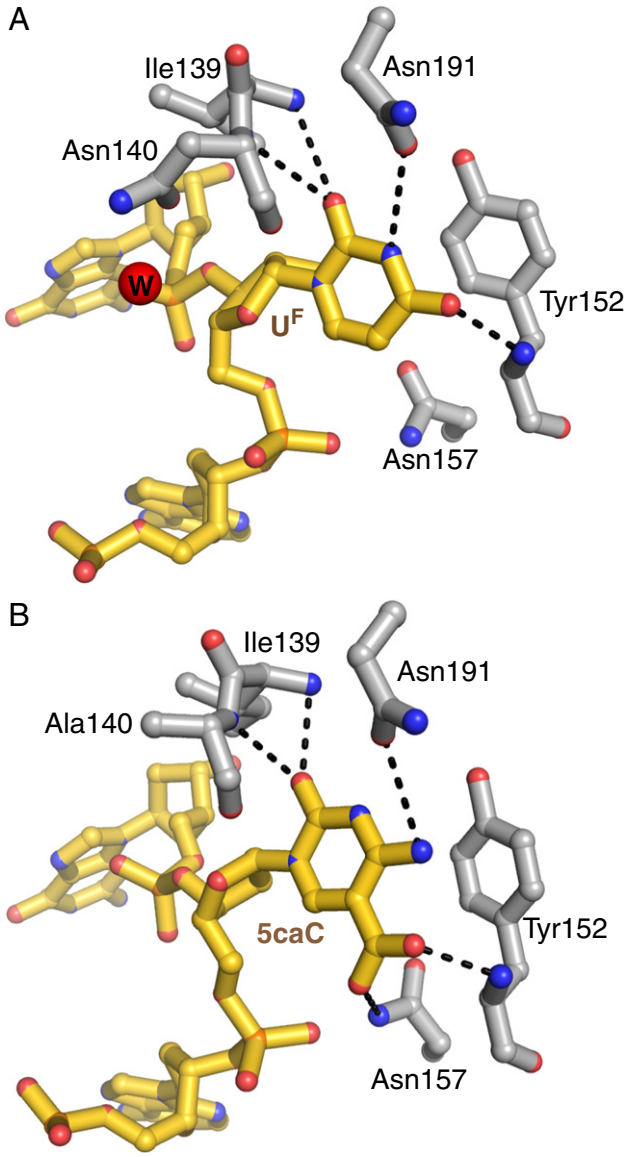

Fig. 11. Protein-DNA contacts within the TDG active site. (A) TDG bound to DNA containing 2′-deoxy-2′-fluoroarabinouridine (U^F) (PDB ID 3UFJ). Hydrogen bonds are shown as dashed lines and the putative catalytic water molecule is a red sphere. (B) TDG in complex with 5-carboxylcytosine (5caC)-DNA (3UO7).

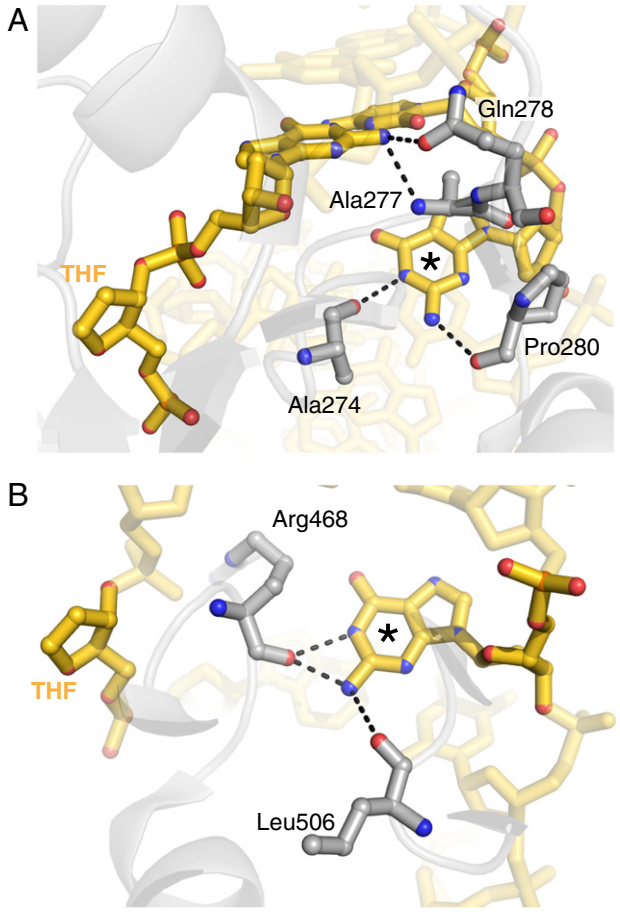

Fig. 12. Comparison of TDG (A) and MBD4 (B) contacts to the strand opposite the lesion. The guanine base opposite the THF abasic site is marked with an asterisk. (A) TDG/THF-DNA complex (PDB ID 2RBA). (B) MBD4/THF-DNA complex (4DK9).

4. Uracil/Thymine/5mC

G•U and G•T mismatches arise from deamination of cytosine and 5-methylcytosine (5mC), respectively, and lead to A•T transition mutations [247,248]. Uracil is excised in eukaryotes by uracil DNA glycosylase (UDG, also known as UNG), single-stranded monofunctional uracil glycosylase (SMUG), and to a lesser extent by thymine DNA glycosylase (TDG). In bacteria, uracil is removed by the UDG ortholog, Ung, and mispaired uracil glycosylase (MUG) [249–252]. Thymine is removed from G•T mismatches by TDG and methyl binding domain 4 (MBD4) in eukaryotes and by archaeal mismatch specific glycosylase (MIG) [253–255]. With the exception of MBD4 and MIG, which belong to the HhH superfamily, the UDG/TDG glycosylases adopt a highly conserved α/β fold (Fig. 3) and can be divided into 4 subfamilies on the basis of sequence similarity and substrate specificity [16,256,257] (Table 1). UDG family 1 contains UDG/UNG and is defined by the landmark structures of the human and viral enzymes in various states, which revealed mechanistic details about substrate recognition and catalysis common to the entire superfamily [16–19]. Family 2 is composed of thymine-specific TDG and MUG, which are homologous to UDG in structure but not sequence [258–261]. The third family is defined by SMUG, and the fourth by *T. thermophilus* TDG. The common α/β fold of the UDG superfamily contains a positively-charged groove approximately the width of a DNA duplex that is ideal for binding double-stranded DNA [16].

UDG has served as a model for understanding the structural and biochemical functions of DNA glycosylases in general, and recent work has focused on the mechanism by which the enzyme locates uracil amidst undamaged DNA. This collective body of work on UDG has been the subject of several recent reviews [5–7,10,262–264], and thus will not be discussed here. We instead focus on recent structural results for TDG in light of new evidence implicating this enzyme in active 5mC demethylation [265,266].

4.1. A possible role of BER in DNA demethylation

In addition to repair of thymine mismatches, the biological functions of TDG and MBD4 may extend beyond DNA repair as a defense against mutation to a potential role in regulating gene expression and DNA demethylation [265,267]. 5mC is an important marker for gene expression, X-chromosome inactivation, and transposon silencing among other developmental processes [268–270]. Whereas DNA methylation mechanisms are relatively well understood [271], the demethylation pathways are not. Demethylation can occur passively after replicative synthesis of unmethylated daughter strands or actively by demethylase enzymes. In plants, active demethylation takes place by the DME/ROS1 family of 5mC DNA glycosylases (see Section 4.4 below), but an analogous 5mC glycosylase has not been discovered in mammals. TDG and MBD4 have been implicated in active demethylation on the basis of their abilities to excise mispaired thymine produced from AID- or APOBEC-dependent 5mC deamination, and recent studies have shown TDG to be necessary for maintenance of epigenetic stability [265,272,273]. In addition, the recent discoveries that the ten-eleven translocation (TET) proteins oxidize 5mC to 5-hydroxymethylcytosine (5hmC), 5-formylcytosine (5fC), and 5-carboxylcytosine (5caC) (Fig. 1C), and that 5hmC is found at transcriptional start sites and within actively transcribed genes raises the distinct possibility that these 5mC derivatives and their deamination products are intermediates in a BER-dependent active demethylation pathway [274–283]. Indeed, TDG is capable of excising 5mC oxidation products 5fC and 5caC, with single-turnover rate constants (2.6 ± 0.1 min^{-1}, 5fC•G; 0.5 ± 0.01 min^{-1}, 5caC•G) comparable to T•G ($k_{cat} = 1.8 \pm 0.04$ min^{-1}) [284,285], further implicating the thymine glycosylases in active demethylation.

4.2. Structural insight into TDG function

Structures of the catalytic domain of TDG (residues 111–308) bound to substrate and product DNA and conjugated by the regulatory protein SUMO have provided a basis to understand TDG's sequence specificity and its mechanisms of base excision and product release [261,285–287]. In addition, we review an NMR study of the N-terminal regulatory domain of TDG (residues 1–111) that supports models for allosteric control of TDG activity [288,289].

4.2.1. TDG–DNA complexes

Structures of TDG bound to duplex DNA containing a THF product mimic provided the general features of DNA binding and the first glimpse into thymine recognition [261]. In this structure, two TDG molecules are bound to a single 22-nucleotide DNA duplex, with one protein anchored at the abasic site and the other positioned at an undamaged site, although biochemical analysis indicated that only one protein per lesion is required for catalysis [261,290]. The TDG complex is very similar to DNA-bound structures of UDG and *E. coli* MUG, with notable exceptions. Both TDG and UDG impose a ~43° bend in the substrate DNA at the abasic site, although MUG does not significantly bend the DNA [9,258,260,291]. TDG utilizes an arginine (Arg275) to plug the gap created by the flipped nucleotide, whereas UDG and MUG have leucine plugs. Substitution of Arg275 with either alanine or leucine results in a significant decrease in both the rate of thymine excision and substrate binding [292]. Another unique aspect of the TDG family are lysine residues Lys246 and Lys248, which make contacts to the DNA backbone of the non-damaged strand, 8 and 9 nucleotides away from the damage site,

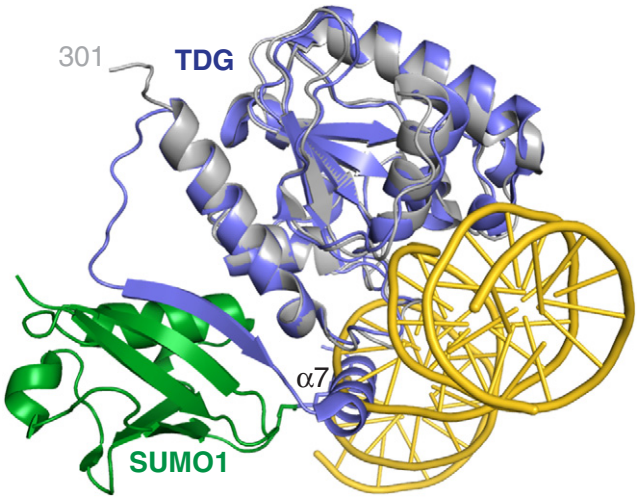

Fig. 13. SUMO1 modified TDG creates steric clash with DNA. The SUMO1-modified TDG structure (blue TDG, green SUMO1, PDB ID 1WYW) is superimposed onto the TDG/THF-DNA complex (silver/gold, 2RBA). SUMO1 modification holds helix α7 in a position that would presumably clash with the DNA.

providing an explanation for TDG's requirement for 9–10 bases 5′ to the lesion [261,293].

Two TDG–substrate–DNA complexes were recently determined that provided a snapshot of an uncleaved nucleobase in the active site [285,287]. One structure contained the wild-type enzyme bound to DNA containing a non-hydrolyzable dU mimetic (2′-deoxy-2′-fluoroarabinouridine, U^F) (Fig. 11A) and the other trapped 5caC in the active site by utilizing a variant TDG containing an Asn140Ala substitution (Fig. 11B), which had previously been shown to decrease the rate of thymine excision while having only marginal effect on substrate binding [261,292]. The overall structures of U^F and 5caC substrate complexes are similar to the THF product complex, and the active sites reveal common modes of recognition of the two substrates. A hydrogen bond was observed from Asn191 to N4 in the 5caC structure and to pyrimidine N3 in the U^F structure, and this contact is conserved in UNG and SMUG1 but not MUG. In UNG and SMUG1, this asparagine side chain forms an additional hydrogen bond to the pyrimidine O4 [260,291,294]. Maiti et al. suggest the differential orientation of this residue in UNG and SMUG1 prevents these enzymes from excising cytosine analogs such as 5fC and 5caC [287]. In addition, 5caC and likely thymine participate in van der Waals interactions with Ala145, and both U^F and 5caC form hydrogen bonds with main chain atoms of Tyr152 at either the O4 of uracil (and thymine) or the carboxyl group of 5caC. The 5caC forms an additional interaction with Asn157. Even though modeling a thymine base into the active site of the U^F structure shows a steric clash with Ala145, this residue is able to accommodate the 5-carboxyl group in the 5caC structure. Nevertheless, Ala145Gly and His151Ala mutants both increase TDG's thymine excision activity by 13-fold over the wild-type. Maiti et al. propose that His151 slows the cleavage reaction by destabilizing the partial negative charge that develops during the reaction [295]. The mutations showed an even greater increase in activity for thymine from normal A•T base pairs, suggesting that these highly conserved residues are needed to limit aberrant action on undamaged DNA [295]. Zhang, et al. found that TDG binds DNA containing 5caC with higher affinity than 5fC, U, and T, which they attribute to the hydrogen bonds to the electronegative 5caC carboxyl group from Asn157, His151, and Tyr152 [285]. The inability of other members of the UDG family, including SMUG1 and UDG, to bind 5caC and 5fC may result from the presence of residues that interfere with these interactions [285].

Regarding catalysis, stabilization of Asn140 and the β2-α4 catalytic loop, which encircles the active site, by Thr197 was found to be important for TDG function, as a Thr197Ala substitution resulted in a 32-fold reduction in base excision activity [295]. Interestingly, the U^F structure suggested that a putative water nucleophile, which is absent from other TDG structures and the enzyme-substrate complex for related MUG enzymes, is positioned by the side chain and main chain of Asn140 and Thr197, respectively. The low resolution and extensive merohedral twinning of the crystallographic data precludes an unambiguous assignment of such a water molecule. However, the presence of a water molecule at this position in TDG is consistent with those observed in the high-resolution structure of free MUG [258] and with the putative water nucleophiles located in high-resolution structures of UNG [291,296].

TDG makes several contacts to the guanine opposite the lesion that offer an explanation for the enzyme's specificity for thymine in certain sequence contexts. Specificity for G•T mispairs [297] is dictated by Ala274 and Pro280, which make guanine-specific hydrogen bonds from their backbone oxygen atoms to N1 and N2 of the G opposite the abasic site (Fig. 12A). The Ala274 contact is conserved by UDG enzymes, whereas the Pro280 interaction is unique to TDG. In addition, TDG has the greatest excision activity for thymine that is immediately 5′ to a guanine (i.e., in a *T*pG/CpG dinucleotide step) [298,299]. Interestingly, TDG does not contact the 5′ cytosine on the non-lesion strand. The specificity for *T*pG/CpG is likely explained by contacts to the TpG guanine from the conserved Gln278 side chain and Ala277 main chain, as well as the Ala274/Pro280 contacts to the G•T guanine described above (Fig. 12A) [261,297,299].

4.2.2. TDG–SUMO interaction

Posttranslational modification of TDG by Small Ubiquitin-like Modifier (SUMO) proteins occurs at the C-terminal end of the catalytic domain (Lys330). TDG sumoylation facilitates dissociation of TDG from AP–DNA and modulates enzymatic activity through a mechanism involving conformational changes of the N-terminal regulatory

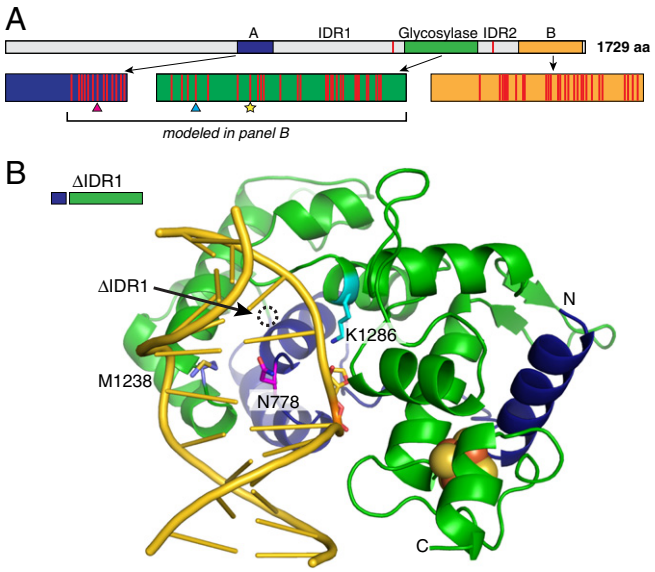

Fig. 14. DME domain structure and distribution of critical residues. (A) Schematic of *A. thaliana* DEMETER (DME), showing the location of the glycosylase domain (green) and domains A (blue) and B (gold) of unknown function. IDR1, inter-domain region 1; IDR2, inter-domain region 2. Red bars mark the locations of residues identified by random mutagenesis to be critical for DME function. Putative residues important for DNA binding and catalysis and conserved in other HhH enzymes are marked by symbols below the schematic (Asn778 DNA plug, magenta triangle; Met1238 DNA wedge, blue triangle; catalytic Lys1286, yellow star). (B) Homology model of the C-terminal half of domain A and the glycosylase domain with DNA superimposed from the structure of *Bacillus stearothermophilus* EndoIII bound to abasic DNA (PDB ID 1P59). The putative catalytic Lys1286 (cyan) Met1238 wedge (slate) residues are contributed by the glycosylase domain (green), the putative Asn778 plug residue (magenta) is from domain A.

domain [289,300,301]. In addition, sumoylation of TDG is essential for activation of CREB-binding protein (CBP)-dependent transcription and localization to promyelocytic leukemia protein oncogenic domains (PODs) [302]. The crystal structure of human TDG conjugated with SUMO1 was determined in 2005 and provided insight into how SUMO1 modulates TDG–DNA binding [286]. C-terminal residues of TDG (307–330) form a crossover β-strand that extends a β-sheet with SUMO1. In addition, TDG and SUMO1 interact through a series of main chain hydrogen bonds and side chain hydrophobic and polar interactions at its SUMO-interacting site (SIM). A mutational analysis revealed that these noncovalent bonds are necessary for SUMO1-induced disruption of DNA binding by TDG, confirming an earlier study [286,300]. The covalent tethering of SUMO1 to the C-terminus of TDG places helix α7 in an outwardly extended conformation from the rest of the protein. Superposition of sumoylated and DNA-bound forms of TDG predict that helix α7 in this orientation collides with the DNA strand immediately opposite the lesion (Fig. 13). The nature of the conformational change in the C-terminal end of the TDG glycosylase domain is uncertain since helix α7 was not present in the DNA-bound structures. Nevertheless, the SUMO1–TDG structure suggests that sumoylation of TDG locks helix α7 in an orientation that promotes DNA release. A structure of SUMO3 conjugated to TDG shows very similar binding between the two proteins as with SUMO1 [303].

4.2.3. TDG regulatory domain

TDG contains at its N-terminus a lysine-rich regulatory domain (RD) that interacts with DNA and a number of proteins involved in genome maintenance. The RD binds to DNA methyltransferase DNMT3a [304] and is a target for acetylation by transcriptional co-activators CBP/p300, which aids in recruitment of APE1 [305]. In addition, the RD is important for TDG specificity for G•T mismatches [252]. This regulatory domain is highly flexible and contains a non-specific DNA binding function that is modulated by sumoylation of the catalytic domain, thereby affecting its enzymatic activity [289]. A recent NMR study of this domain revealed residues 1–50 to be unstructured even in the context of the full protein. Residues 51–111, on the other hand, showed a modest degree of structure and an extended conformation that contacts the catalytic domain in the context of the full-length protein [288]. The authors proposed that an electrostatic interaction between the regulatory and catalytic domains modulate rates of thymine and uracil excision, supporting previous models for allosteric control of G•T specificity [288,289].

4.3. MBD4

Mismatch specific thymine glycosylase MBD4 contains a methyl-CpG-binding domain (MBD) and a C-terminal glycosylase domain that preferentially excises T mispaired with G [254]. MBD4 excises thymine from G•T and G•U mispairs at rates (k_{cat}) of 0.012 s^{-1} and 0.05 s^{-1}, respectively [306]. Crystal structures of the glycosylase domains of mouse and human MBD4 showed that the enzyme belongs to the helix–hairpin–helix structural superfamily [256,307]. A very recent structure of MBD4's glycosylase domain bound to THF-DNA showed the overall DNA binding regime characteristic of HhH glycosylases, including a 57° bend in the DNA, an extrahelical THF moiety, the opposite base (guanine) stacked in the duplex, and plug (Leu506) and wedge (Arg468) residues that stabilize the flipped nucleotide and distorted duplex [295]. Like TDG, MBD4 makes several guanine-specific contacts to the base opposite the thymine, and thus the structure provides a rationale for discrimination of G•T over A•T base pairs [306] (Fig. 12B). Specifically, Leu506 and Arg468 mainchain oxygens participate in hydrogen bonds or polar interactions with the N1 and exocyclic N2 of guanine, contacts which cannot be made to adenine [261,295] (Fig. 12B). Finally, although MBD4 has been proposed to process G•T mispairs created by active demethylation [273], MBD4 is not able to excise 5fC or 5caC [283,295]. Manvilla et al. attribute this lack of activity to incompatibility between the MBD4 active site and the negative charges that develop on 5fC and 5caC upon dissociation [295].

4.4. DME/ROS1

Plants contain a family of 5mC glycosylases, represented by the *Arabidopsis* proteins DEMETER (DME), Repressor of Silencing 1 (ROS1), DME like 2 (DML2) and DML3, that are responsible for active demethylation via BER. DME is responsible for endosperm gene imprinting and is necessary for seed viability [308]. The DML enzymes, including ROS1, likely function as genome wide demethylases, particularly at sites 5′ and 3′ to genes to regulate transcription, protecting plants from erroneous gene silencing [309–312]. DME, ROS1, and DML3 excise 5mC in CpG, CpNpG, and CpNpN contexts, and DML2 was shown to have 5mC excision activity in a CpG context [313–315].

The DME/ROS1 family enzymes utilize a bifunctional glycosylase-lyase mechanism and, although there are no crystal structures available, sequence analysis predicts an iron-sulfur-containing HhH glycosylase domain similar to EndoIII and MutY [308,313,315,316] (Fig. 14A). A homology model of ROS1 using BsEndoIII as a template helped to identify several residues conserved among the HhH superfamily that are important for ROS1 function [317]. Tyr606 and Asp611 are predicted to reside near the base recognition pocket and are necessary for the excision of both 5mC and thymine from G•T mismatches. A Tyr606Leu mutant slightly reduced the DNA binding activity, whereas Asp611Val mutation increased DNA binding with respect to wild-type ROS1 [317]. The homology model also revealed that two aromatic residues (Phe589 and Tyr1028) conserved in the DME family are positioned to interact with the lesion. Interestingly, Phe589Ala and Tyr1028Ser mutations changed the preference of ROS1 from 5mC to T•G mismatches, indicating that they are important for substrate recognition [317]. A similar homology model for the glycosylase domain of DME predicts analogous residues that may be necessary for catalysis (S. Brooks, B.F. Eichman, unpublished) (Fig. 14).

Unlike other glycosylases, DME/ROS1 proteins contain two additional domains flanking the glycosylase domain and that are essential for 5mC excision (Fig. 14A) [314,318,319]. The C-terminal domain lacks any identifiable sequence homology to proteins outside of the DME/ROS1 family, and thus additional structural and functional studies will be necessary to ascertain its function. The N-terminal domain, on the other hand, contains a conserved lysine-rich region required for ROS1 binding to non-methylated DNA and enhances ROS1 specificity for 5mC over T. Deletion of this domain caused ROS1 to process long DNA substrates less effectively [319]. On its own, the N-terminal region binds DNA with a high affinity. Deletion of the N-terminal domain reduces the ability of ROS1 to bind DNA and excise 5mC, but does not affect the ability of DME to bind methylated and non-methylated DNA [318,319]. The N-terminal and glycosylase domains are separated by ~240 and 400 residues in ROS1 and DME, respectively. Deletion of this interdomain region (IDR1) in DME does not affect 5mC excision activity [318]. The ROS1-EndoIII homology model predicted that this linker region is an inserted sequence within the glycosylase domain and that the lesion-intercalating plug residue is N-terminal to the IDR1 insertion [317]. Interestingly, although ROS1 Asn608 aligns with the Gln42 plug in BsEndoIII, elimination of the Asn608 side chain did not affect base excision, whereas Gln607 was shown to be essential for both 5mC and T excision and binding of both methylated and non-methylated DNA [317]. In contrast, a DME homology model (Fig. 14B), predicts the plug residue to be Asn778, which was shown by a random mutagenesis study to be critical for 5mC excision, whereas Gln777 was not identified as an important residue in that assay [318]. A better understanding of these 5mC glycosylases awaits structural information for DME and ROS1.

5. Summary and perspectives

Recent structural and enzymological studies on previously known and newly discovered DNA glycosylases that process a wide variety of oxidized, alkylated, and deaminated nucleobases have dramatically advanced our understanding of the inner workings of these amazing DNA repair machines. One of the most significant questions has focused on how each glycosylase imparts specificity for a particular type of damage. On the most basic level, the specificity of each glycosylase can be considered to be a function of the chemical complementarity of the modified nucleobase within the active site. That is, glycosylases are a collection of specifically evolved active sites situated among a variety of scaffolds. On the other hand, recent work on several enzymes within the oxidation (e.g., OGG1, MutM/Fpg, EndoVIII/Nei) and alkylation (e.g., AlkA, Mag1) classes indicates that some aspect of substrate recognition takes place within the DNA duplex, before the modified nucleobase has been extruded into the active site [68,100,136,191,224]. A series of structures of OGG1 and MutM crosslinked to unmodified DNA have detailed this search process for the oxidative enzymes, and together with a crosslinked AlkA-DNA structure highlight the role of the intercalating plug residue in probing the minor groove. What follows from this notion of substrate discrimination prior to base flipping is that the intrinsic structure of the DNA lesion itself certainly plays a large role in enzymatic recognition and excision. Related to this, work on UDG and alkylation damage specific glycosylases (e.g., TAG, AlkD) has provided several recent examples for how the DNA conformation and the intrinsic stability of the lesion contributes to substrate-assisted catalysis of base excision [193,226,244–246,320].

Most of the work to date has focused on their catalytic domains, but many glycosylases contain extra domains, interact with other proteins, or undergo post-translational modification, all of which may serve to regulate base excision activity or localize the protein to specific locations on the chromosome. The structure of the full-length MutY protein in complex with an 8oxoG•A mispair is an excellent example of how an extra domain serves to provide enhanced specificity for the base opposite the lesion [149], and work has begun to address the interaction between human MutY and the 9-1-1 complex involved in genome maintenance [157–159]. Sumoylation of TDG and its modulation of DNA affinity through conformational changes involving the N-terminal, regulatory domain is a more complex example involving both covalent modification and a non-catalytic domain [286,289,300,301]. The 5mC glycosylase, DME, contains N- and C-terminal domains flanking the HhH glycosylase domain, but in that case, the extra domains seem to be part of the catalytic core as their mutation or deletion renders the protein inactive [S. Brooks and B.F.Eichman, unpublished results and ref. 318]. Finally, it is becoming increasingly evident that glycosylases do not perform their duties in isolation, but are part of larger complexes that exist to efficiently shuttle DNA damage through the BER pathway possibly by molecular handoff of intermediates, or even to shunt damage into alternative repair pathways. For example, physical interactions of the BER scaffolding protein XRCC1 with AAG and hNTH1 and hNEIL2 have been shown to stimulate their base excision repair activity [321,322], and APE1 has been shown to stimulate the activities of AAG and TDG, effectively coordinating the first 2 steps of BER [217,219]. In order to fully understand the role of DNA glycosylases in the context of BER, work in the future will need to more closely explore how these protein interactions and modifications modulate glycosylase behavior.

Acknowledgements

The authors thank Sylvie Doublié, Sheila David, and Alex Drohat for critique of the manuscript. Work on DNA glycosylases in the author's laboratory is supported by grants from the National Institutes of Health (R01 ES019625) and the National Science Foundation (MCB-1122098). S.C.B. is supported by a National Science Foundation Graduate Research Fellowship under Grant No. 0909667. E.H.R. was supported in part by the Vanderbilt Training Program in Environmental Toxicology (T32 ES07028).

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