An Unexpected Deamination Reaction after Hydrolysis of the Pyrimidine (6-4) Pyrimidone Photoproduct

Gengjie Lin,†∥ Yajun Jian,†∥ Hao Ouyang,§ and Lei Li,*†‡

†Department of Chemistry and Chemical Biology, Indiana University-Purdue University Indianapolis (IUPUI), 402 North Blackford Street, Indianapolis, Indiana 46202, United States
‡Department of Biochemistry and Molecular Biology & Department of Dermatology, Indiana University School of Medicine, Indianapolis, Indiana 46202, United States
§Preclinical Pharmacology, Johnson & Johnson, 199 Grandview Road, Skillman, New Jersey 08558, United States

Supporting Information

ABSTRACT: Pyrimidine (6-4) pyrimidone photoproduct (6-4PP), a common DNA photolesion formed under solar irradiation, was indicated to hydrolyze under strong basic conditions, breaking the N3−C4 bond at the 5′-thymine. The reanalysis of this reaction revealed that the resulting water adduct may not be stable as previously proposed; it readily undergoes an esterification reaction induced by the 5-OH group at 6-4PP to form a five-membered ring, eliminating a molecule of ammonia.

Pyrimidine (6-4) pyrimidone photoproduct (6-4PP) is a common DNA photodamage product resulting from the UV component of sunlight. Its presence leads to a sharp kink, which significantly distorts the DNA helical structure. In contrast, another common pyrimidine photolesion, the cyclobutane pyrimidine photoproduct (CPD), causes relatively minor structural disturbance, e.g. a 30° helical bend toward the major groove. 6-4PP is much more mutagenic than CPD. It arrests the replication forks of general replicative DNA polymerases. When bypassed by the human Y-polymerase pol η, there is a 7-fold tendency for a G instead of an A to be inserted into the position opposite to the 3′-T of the 6-4PP lesion. Due to the resulting drastic DNA structural change, 6-4PP is readily recognized by the DNA repair machinery, resulting in a quick removal in living cells (half-life of 6-4PP = 2.3 h). The unrepaired 6-4PP is suggested to be the primary cause of UVB-induced cell apoptosis especially in NER-deficient cells.

It is thus of significance to understand the physical properties of 6-4PP. 6-4PP is shown to be alkaline labile, inducing DNA strand scission upon hot alkaline treatment. Research from the Iwai laboratory suggests that the alkaline treatment first ruptures the N3−C4 bond on the 5′-thymine of 6-4PP to yield a hydrolysis product (Scheme 1), which is followed by a deglycosylation reaction at the 3′-thymine, leading to an ultimate DNA strand scission. Recently, our group proved that the third naturally occurring thymine dimer, i.e. 5-thyminyl-5,6-dihydridothymine which is commonly referred to as the spore photoproduct (SP), and 5,6-dihydro-2′-deoxuryridine (dHdU) resulting from the ionizing radiation damage to cytosine under anoxic conditions also undergo the N3−C4 bond rupture via a hemiaminal intermediate, suggesting that such a bond cleavage is probably a common feature possessed by a saturated pyrimidine residue. Our re-examination of the 6-4PP decomposition reaction however implies that the resulting water adduct is not stable as previously suggested; it undergoes an additional deamination reaction to yield 2-oxazolidinone (5-4) pyrimidone, eliminating the N3 as an ammonia. This report brings new insight into the reactivity of 6-4PP, which is important toward ongoing synthetic, analytical, and biological studies, i.e., handling and generating DNA containing 6-4PP and the quantitation of 6-4PP in DNA, and

Received: August 16, 2014
Published: September 24, 2014
thus may provide a fundamental understanding of the biological consequences of this common DNA photolesion.

We obtained 6-4PP via irradiation of the dinucleotide TpT in acetonitrile under 254 nm UV light using a published protocol. The 6-4PP was isolated by HPLC and redissolved in 50 mM KOH to a final concentration of 0.75 mM. The resulting solution was heated to 60 °C for various times and analyzed by HPLC. Such a reaction condition is similar to that adopted in the previous 6-4PP hydrolysis studies. As shown in Figure 1, compound 1 was isolated after a 5-h reaction as the dominant product (~70% yield). However, to our surprise, ESI-MS analysis under the negative ion mode found that the species of 1 exhibits an m/z signal of 546.12 amu, which is +1 amu higher than that of 6-4PP, but is −17 amu smaller than the predicted 6-4PP water adduct shown in Scheme 1 (563.14). None of the minor reaction products exhibits an m/z signal of 563.14 either. If the reaction was conducted in 0.2 M KOH at room temperature, a cleaner formation of 1 was observed while the reaction pattern remained the same.

The formation of 1 was observed from the very beginning of the reaction; no reaction intermediate en route to 1 was detected in our hands. Because the previous studies reported that 6-4PP was hydrolyzed under strong basic conditions, we wonder whether 1 derives from this hydrolysis reaction. We therefore carried out the reaction in 0.2 M KOH dissolved in 97% 18O-labeled water. The same product was generated as indicated by the HPLC analysis. ESI-MS analysis reveals an m/z signal of 546.12 in the [M − H]− form, which has +2 amu relative to that of 1 produced in unlabeled water. This observation is in line with the above hypothesis, indicating that 1 indeed results from the 6-4PP hydrolysis reaction. However, again, the mass is reduced by 17 amu compared with the predicted mass for the 6-4PP water adduct with one 18O atom incorporated.

Typically, a mass of 17 amu corresponds to an ammonia molecule; we thus wonder whether the amino moiety attached to the C2=O at the 5′-thymine of the hydrolysis product has been removed. To test this hypothesis, we first incorporated a 15N label at the N3 position via a N-nitration reaction of the uridine residue and synthesized a dinucleotide TpT with the 5′-thymine containing the 15N label. The corresponding 6-4PP was then prepared photochemically as described above. The labeled 6-4PP exhibits an [M − H]− signal of 546.13, and a single peak at 191.7 ppm in the 15N NMR spectrum (Figure 2).

![Figure 1. HPLC chromatograph of the 6-4PP hydrolysis reaction in 50 mM KOH at 60 °C monitored by UV detector at 310 nm. The reaction affords 1 as the major product. None of the minor products exhibits a molecular mass corresponding to that of the 6-4PP water adduct as indicated by the ESI-MS analysis.](image1)

![Figure 2. 15N NMR spectra describing the deamination reaction using 15N labeled 6-4PP in 0.30 M KOD at ambient temperature in D2O. The 15N peak (191.7 ppm), from the 15N moiety in 6-4PP, decreased over time as the reaction proceeded. A new 15N peak at 0 ppm, corresponding to 15NH3, increased correspondingly, indicating that a molecule of ammonia was released during the reaction.](image2)

![Scheme 2. Two Possible Deamination Mechanisms](image3)

Although we were unable to observe this product during our 6-4PP hydrolysis study, its formation is highly likely as indicated by the fully characterized similar hydrolysis reactions in alkaline treated SP and dHdU. It is thus of interest to reveal how this 6-4PP water adduct deaminates. Two possible mechanisms, via an anhydride (Scheme 2A) and via an esterification reaction involving the 5-OH group at the 5′-thymine of 6-4PP (Scheme 2B), can be proposed. Although...
similar anhydride species were observed in the MS/MS analyses of the SP hydrolysis reaction,24 and of the oxanosine deamination process,24 such a compound is not stable in aqueous solution, as indicated by the slow decomposition of uracil anhydride in ethanol at room temperature.25 Moreover, considering the similar structures among the hydrolysis products of SP, 6-4PP, and dHdU, if route A is possible, deamination processes should be observed during the hydrolysis of SP and dHdU. The lack of anhydride formation in these cases20 indicates that the esterification mechanism shown in route B is more likely.

This conclusion is further supported by the NMR characterizations of 1 as detailed below:

(1) The −CH$_3$ and H6 atom on the 5′-thymine of 6-4PP adopt a cis configuration, resulting in a strong ROE signal.22 Such an interaction is lost in the newly formed 1, which is consistent with the formation of a five-membered ester ring via route B, where the −CH$_3$ is trans to the H6.

(2) Comparing the $^{13}$C NMR spectrum of 6-4PP with that of 1 indicates that very minor changes on chemical shift were observed in both 2′-deoxyribose and the 3′-thymine ring, while relatively large changes were observed on carbons at the 5′-thymine ring (Table S1).22 Particularly, chemical shifts of −6.8 and 10.9 ppm were observed for its methyl group and C5 respectively, indicating drastic structural changes associated with these groups. Such changes are obvious in the ester product, but not so in the putative anhydride species.

(3) $^{18}$O isotope shifts in $^{13}$C NMR spectroscopy have been utilized to facilitate chemical structure determination.24,26−30 We therefore mixed $^{18}$O labeled 1, prepared by 6-4PP hydrolysis in $^{18}$O-labeled water,22 with unlabeled 1. Using ESI-MS analysis, the ratio between the unlabeled and $^{18}$O labeled 1 was found to be 1:1.8.22 We then acquired the $^{13}$C NMR spectrum for this compound mixture. In the $^{13}$C NMR spectrum, the C4 signal exhibits an upfield shift of 29 ppm which is in line with the isotope shifts observed in other $^{18}$O substituted carboxylates;31 the C2 signal still exists as a single peak (Figure 3). This observation suggests that only C4 is connected to an $^{18}$O (route B). If the anhydride is formed, both C2 and C4 signals are expected to exhibit an isotope shift.

Taken together, our data suggest that the 6-4PP hydrolysis product may not be stable as previously assumed; it undergoes an additional deamination process driven by esterification with the 5-OH at 5′-thymine to form a five-membered ring, resulting in 2-oxazolidinone (5-4) pyrimidone. Rupture of the N3–C4 bond was also observed in our recent SP and dHdU hydrolysis studies,20 resulting in stable water adducts. Different from SP and dHdU, 6-4PP has an extra 5-OH moiety leading to the unexpected deamination reaction reported here. The five-membered ring is considered to be the most kinetically favorable configuration in ring closing reactions due to the lowest entropic cost and transition state strain energy.12,35 This kinetically favorable ring closure process determines that the formed 6-4PP water adduct must be very short-lived. Considering the heterocyclic ring structures of nucleobases, similar deamination reactions may occur after ring opening in certain nucleobase modifications, which await further studies in the future.

Figure 3. $^{13}$C NMR spectrum using a mixture of 1 generated from hydrolysis of 6-4PP at the presence of 0.2 M KOH in unlabeled and $^{18}$O labeled water, respectively. The ratio between unlabeled ($^{16}$O) and $^{18}$O labeled 1 is suggested to be 1:1.8 by the ESI-MS analysis. An upfield isotope shift of 29 ppb was observed for C4a, and no shift was observed for C2a in the $^{13}$C NMR spectrum. This result suggests that the $^{18}$O atom is attached to C4a, but not to C2a, as indicated by the Chemical structure included. Integration of the two C4a signals reveals a ratio of 1:1.71, agreeing with the ratio revealed by the ESI-MS analysis and supporting the signal assignment shown in the figure.

ASSOCIATED CONTENT

5 Supporting Information

Synthesis of the 6-4PP, NMR characterization of the reaction products. This material is available free of charge via the Internet at http://pubs.acs.org.

AUTHOR INFORMATION

Corresponding Author

*E-mail: lilei@iupui.edu.

Author Contributions

G.L. and Y.J. contributed equally.

Notes

The authors declare no competing financial interest.

ACKNOWLEDGMENTS

This work is supported by a NIH grant (ES077177) and the IUPUI startup fund. We thank Professor Steven Rokita at the Johns Hopkins University for helpful discussions and suggestions and Dr. Xin Feng Gao at the Indiana University at Bloomington for the help on our structural studies using $^{13}$C NMR spectroscopy.

REFERENCES

(1) Lukin, M.; de los Santos, C. Chem. Rev. 2006, 106, 607−686.
(2) Park, H.; Zhang, K.; Ren, Y.; Nadjî, S.; Sinha, N.; Taylor, J.-S.; Kang, C. Proc. Natl. Acad. Sci. U.S.A. 2002, 99, 15965−15970.
(3) LeClerc, J. E.; Borden, A.; Lawrence, C. W. Proc. Natl. Acad. Sci. U.S.A. 1991, 88, 9685−9.
(4) Smith, C. A.; Wang, M.; JIang, N.; Che, L.; Zhao, X.; Taylor, J. S. Biochemistry 1996, 35, 4146−54.
(5) Kamiya, H.; Iwai, S.; Kasai, H. Nucleic Acids Res. 1998, 26, 2611−7.
(6) Wellinger, R.-E.; Thoma, F. Nucleic Acids Res. 1996, 24, 1578−1579.
(7) Johnson, R. E.; Haracska, L.; Prakash, S.; Prakash, L. Mol. Cell. Biol. 2001, 21, 3558−3563.
(8) Yamamoto, J.; Loakes, D.; Masutani, C.; Simmony, S.; Urabe, K.; Hanaoka, F.; Holliger, P.; Iwai, S. Nucleic Acids Symp. Ser. 2008, 52, 339−340.
(9) Young, A. R.; Chadwick, C. A.; Harrison, G. J.; Hawk, J. L. M.; Nikaido, O.; Potten, C. S. J. Invest. Dermatol. 1996, 106, 1307−1313.

dx.doi.org/10.1021/ol502433h | Org. Lett. 2014, 16, 5076−5079
Organic Letters

(10) Lima-Bessa, K. M. d.; Armelini, M. G.; Chiganças, V.; Jacysyn, J. F.; Amarante-Mendes, G. P.; Sarasin, A.; Menck, C. F. M. DNA Repair 2008, 7, 303−312.
(11) Lo, H. L.; Nakajima, S.; Ma, L.; Walter, B.; Yasui, A.; Ethell, D. W.; Owen, L. B. BMC Cancer 2005, 5, 135.
(12) Lippka, J. A.; Gordon, L. K.; Brash, D. E.; Haseltine, W. A. Proc. Natl. Acad. Sci. U.S.A. 1981, 78, 3388−3392.
(13) Pfeifer, G. P.; Drouin, R.; Riggs, A. D.; Holmquist, G. P. Proc. Natl. Acad. Sci. U.S.A. 1991, 88, 1374−1378.
(14) Yoon, J.-H.; Lee, C.-S.; Connor, T. R.; Yasui, A.; Pfeifer, G. P. J. Mol. Biol. 2000, 299, 681−693.
(15) Arichi, N.; Inase, A.; Eto, S.; Mizukoshi, T.; Yamamoto, J.; Iwai, S. Org. Biomol. Chem. 2012, 10, 2318−25.
(16) Higurashi, M.; Ohhtsuki, T.; Inase, A.; Kusumoto, R.; Masutani, C.; Hanaoka, F.; Iwai, S. J. Biol. Chem. 2003, 278, 51968−73.
(17) Arichi, N.; Yamamoto, J.; Takahata, C.; Sano, E.; Masuda, Y.; Kurooka, I.; Iwai, S. Org. Biomol. Chem. 2013, 11, 3526−3534.
(18) Villanueva, J. M.; Pohl, J.; Doetsch, P. W.; Marzilli, L. G. J. Am. Chem. Soc. 1999, 121, 10652−10653.
(19) Geacintov, N. E.; Broyde, S. The chemical biology of DNA damage; Wiley-VCH: Weinheim, 2010; p xxii, 449 p.
(20) Lin, G.; Jian, Y.; Dria, K. J.; Long, E. C.; Li, L. J. Am. Chem. Soc. 2014, 136, 12938−12946.
(21) Mizukoshi, T.; Hitomi, K.; Todo, T.; Iwai, S. J. Am. Chem. Soc. 1998, 120, 10634−10642.
(22) See Supporting Information.
(23) Ariza, X.; Bou, V.; Vilarrasa, J. J. Am. Chem. Soc. 1995, 117, 3665−3673.
(24) Majumdar, P.; Wu, H.; Tipton, P.; Glaser, R. Chem. Res. Toxicol. 2005, 18, 1830−1841.
(25) Chwang, T.-L.; Wood, W. F.; Parkhurst, J. R.; Nesnow, S.; Danenberg, P. V.; Heidelberger, C. J. Med. Chem. 1976, 19, 643−647.
(26) Vederas, J. C. J. Am. Chem. Soc. 1980, 102, 374−376.
(27) Dessingles, A.; Castillon, S.; Olesker, A.; Ton That, T.; Lukacs, G. J. Am. Chem. Soc. 1984, 106, 450−451.
(28) Albrecht, L.; Jiang, H.; Dickmeiss, G.; Gschwend, B.; Hansen, S. G.; Jørgensen, K. A. J. Am. Chem. Soc. 2010, 132, 9188−9196.
(29) Mega, T. L.; Van Etten, R. L. J. Am. Chem. Soc. 1993, 115, 12056−12059.
(30) Boebel, T. A.; Gin, D. Y. J. Org. Chem. 2005, 70, 5818−5826.
(31) Berger, S., Ed. Isotope effects in NMR spectroscopy; Springer-Verlag: Berlin; 1990.
(32) Aumiller, J. C.; Whittle, J. A. J. Org. Chem. 1976, 41, 2955−2959.
(33) Casadei, M. A.; Galli, C.; Mandolini, L. J. Am. Chem. Soc. 1984, 106, 1051−1056.