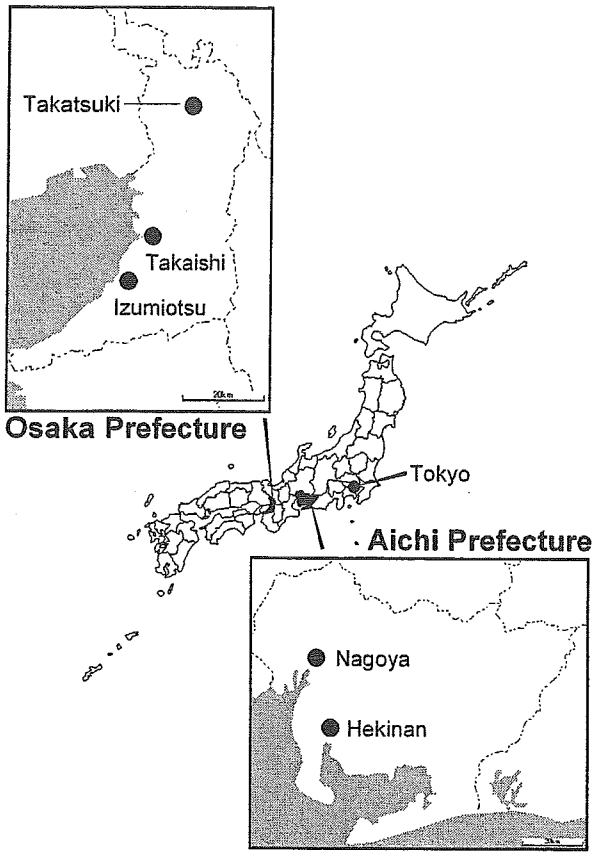

Figure 1. Geographic locations of the surface soil sampling sites in Osaka and Aichi Prefectures, Japan.

the samples induced 2-fold increases over the average yield of spontaneous revertants and showed well-behaved concentration–response patterns, the samples were judged to be positive. The percentage contributions of 3,6-DNBeP to the total mutagenicity of the organic extracts from soil samples were calculated based on the mutagenic activities of 3,6-DNBeP and the soil extracts.

Results

Detection and Isolation of Mutagens in Surface Soil in Takatsuki. A surface soil sample was collected at a park in a residential area in Takatsuki, Osaka Prefecture (Figure 1). The organic material (1.8 g) was extracted from 2.2 kg of the soil sample with a Soxhlet apparatus. The organic extract showed extremely high mutagenicity in *S. typhimurium* TA98 in the absence of S9 mix and induced 233 000 000 revertants/g of organic extract.

To identify the major mutagens in the organic extracts of the soil sample from Takatsuki, the soil extracts were fractionated by various rounds of column chromatography with monitoring of the mutagenicity of the fractions in TA98 without S9 mix. First, the soil extract was separated by column chromatography using Sephadex LH-20 resin. Several fractions from the soil extract showed mutagenicity, and the most potent mutagenic activity was observed in the fraction with elution volumes of 240–330 mL. This mutagenic fraction was applied to a silica gel column for LPLC. Mutagenicity was detected in several fractions, and the most potent mutagenic activity was observed in the fraction with elution volumes of 630–750 mL. The fraction with elution volumes of 630–750 mL accounted for 82% of the total mutagenicity

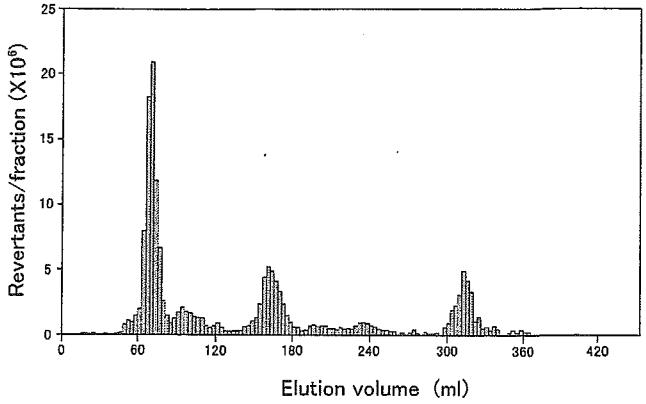

Figure 2. LPLC profile of mutagens in the soil sample from Takatsuki. LPLC was performed on an Ultra pack ODS column. The mutagenicity of each 3 mL fraction was tested in *S. typhimurium* TA98 without S9 mix.

of the organic extract from the soil sample. This fraction was further separated by LPLC on an Ultra pack ODS column. The LPLC profile of soil mutagens on the Ultra pack ODS column is shown in Figure 2. Many fractions showed mutagenicity in TA98. The fraction with elution volumes of 48–84 mL showed the most potent activity, followed by the fraction with elution volumes of 147–186 mL.

The mutagenic fraction with elution volumes of 48–84 mL, which accounted for 45% of the total mutagenicity of the soil extract, was separated by HPLC on a COSMOSIL $5C_{18}$ AR-II column. Potent mutagenicity was observed in fractions with retention times of 33–36 and 43–47 min, which corresponded to 1,6-DNP and 1,8-DNP, respectively. To confirm the participation of 1,6-DNP and 1,8-DNP in the mutagenicity of these fractions, these two mutagenic fractions were further purified by HPLC on a Luna 5 μ Phenyl-Hexyl column. Mutagenic activities were detected in fractions with retention times of 39–42 and 42–45 min, which corresponded to 1,8-DNP and 1,6-DNP, respectively. Moreover, UV absorption spectra of the peak fractions with retention times of 39–42 and 42–45 min were consistent with those of 1,8-DNP and 1,6-DNP. Mutagenic activities of the fractions corresponding to 1,8-DNP and 1,6-DNP accounted for 24 and 12% of the total mutagenicity of the soil extracts, respectively. These results indicate that most of the mutagenicity of the fractions with elution volume of 48–84 mL on the Ultra pack ODS column could be attributed to 1,6-DNP and 1,8-DNP.

The mutagenic fraction with elution volumes of 147–186 mL on the Ultra pack ODS column, which accounted for 20% of the total mutagenicity of the soil extract, was separated by HPLC on a COSMOSIL $5C_{18}$ AR-II column. Potent mutagenicity was observed in the fraction with retention times of 31–35 min. This mutagenic fraction was further purified by HPLC on a Luna 5 μ Phenyl-Hexyl column. A single UV absorption peak fraction was detected in the fraction with retention times of 44–48 min, and this fraction exhibited potent mutagenicity, in that it accounted for 15% of the total mutagenicity of the soil extract. To confirm the purity of this mutagenic fraction, an aliquot of this fraction was analyzed by HPLC on an Inertsil ODS-3 column. As shown in Figure 3A, a single UV absorption peak was observed at a retention time of 31 min.

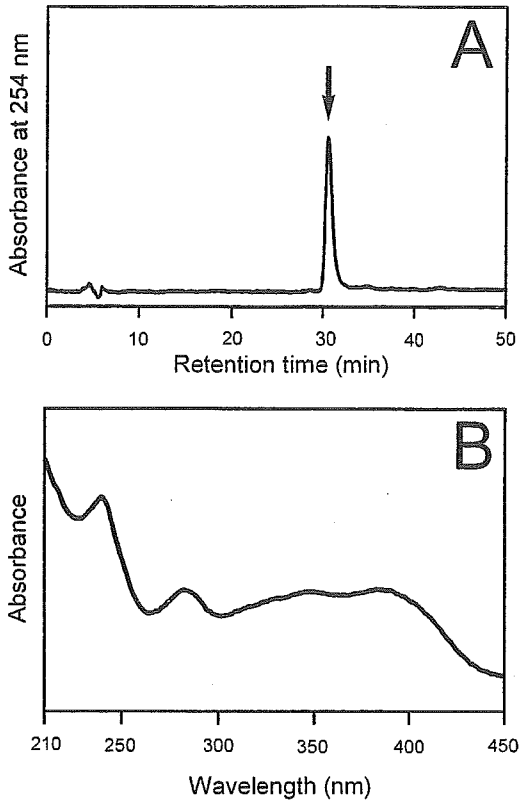

Figure 3. HPLC profile (A) of a mutagen isolated from the surface soil in Takatsuki. HPLC was performed on an Inertsil ODS-3 column, eluting with 75% acetonitrile at a flow rate of 0.7 mL/min. The UV absorption spectrum (B) of the mutagen, which is indicated by the arrow in the HPLC profile (A), was measured with a photodiode array detector.

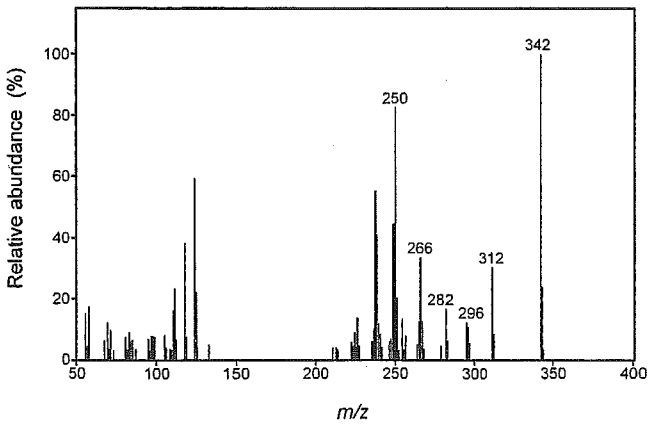

Figure 4. Electron impact mass spectrum of a mutagen isolated from surface soil in Takatsuki.

Structural Analysis of the Mutagen. The UV absorption spectrum of the new mutagen isolated from the soil sample collected at Takatsuki is shown in Figure 3B. UV absorption maxima were observed at 238, 281, 344, and 381 nm. As shown in Figure 4, a molecular ion peak of this mutagen was observed at *m/z* 342 $[M]^+$, and its mass spectra exhibited a fragmentation pattern typical of dinitrated PAH, such as *m/z* 312 $[M - NO]^+$, 296 $[M - NO_2]^+$, 282 $[M - 2 \times NO]^+$, and 250 $[M - 2 \times NO_2]^+$. These results indicate that this mutagen is a dinitrated PAH and the nonnitrated PAH has a molecular weight of *m/z* 252. A few PAHs with a molecular weight of *m/z* 252, such as BeP, have been detected in ambient air and surface soil (*40*, *41*). To determine the

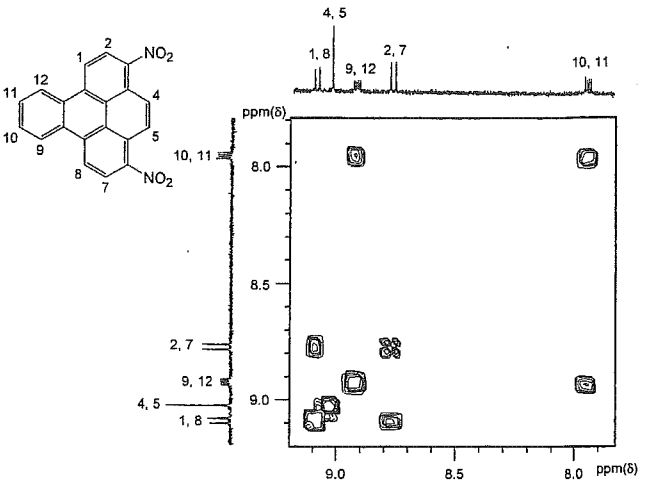

Figure 5. $^1H-^1H$ COSY spectrum of nitrated BeP in $CDCl_3$ and its chemical structure.

chemical structure of this mutagen, several authentic dinitrated PAHs with molecular weights of *m/z* 342 were synthesized and their chemical features were compared to those of the new mutagen isolated from the soil sample. Consequently, we found that one of the dinitrated BeP isomers had the same mass spectrum, UV absorption spectrum, and retention time on the Inertsil ODS-3 column as the isolated mutagen. The ^1H NMR spectrum of the dinitrated BeP in $CDCl_3$ indicated the presence of five pairs of protons in the molecule, implying that the dinitrated BeP has a symmetrical chemical structure. A subsequent $^1H-^1H$ COSY spectrum of this dinitrated BeP isomer is shown in Figure 5. A sextet at 7.95 ppm was correlated with a sextet at 8.93 ppm. The former was assigned to the protons at the 10- and 11-positions of BeP, and the latter was assigned to protons at the 9- and 12-positions. Doublets at 8.77 and 9.09 ppm were also correlated with each other; the former was attributed to protons at the 2- and 7-positions, and the latter was attributed to protons at the 1- and 8-positions. A singlet at 9.03 was assigned to the protons at the 4- and 5-positions, and nitro groups were deduced to be substituted at the 3- and 6- positions of BeP. On the basis of these results, the chemical structure of the mutagen isolated from the surface soil from Takatsuki was determined to be 3,6-DNBeP (Figure 5).

Mutagenicity of 3,6-DNBeP. The mutagenicity of 3,6-DNBeP was examined by using *S. typhimurium* TA98 and TA100, their *O*-acetyltransferase overproducing derivatives, i.e., YG1024 and YG1029 (*38*), and an *O*-acetyltransferase deficient strain, i.e., TA98/1,8-DNP_6 (*39*). Table 1 summarizes the mutagenicity of 3,6-DNBeP in the five strains with and without S9 mix in the Ames assay. 3,6-DNBeP was mutagenic in these five strains, and the activities in each strain without S9 mix were higher than those with S9 mix. The mutagenic potency of 3,6-DNBeP was extremely high in TA98 and YG1024 without S9 mix, in that it induced 285 000 revertants/nmol and 955 000 revertants/nmol, respectively. The potency in TA98 was comparable to those of 1,8-DNP and 1,6-DNP, which are the most potent mutagens reported to date (*29*), as shown in Table 1. Without S9 mix, 3,6-DNBeP was more mutagenic in TA98 than in TA100 and a similar tendency was observed in its activities toward YG1024 and YG1029. These findings indicate that 3,6-DNBeP induces more frameshift than base substitution

Table 1. Mutagenicity of DNBeP in *Salmonella* Tester Strains with and without S9 Mix

	mutagenicity (revertants/nmol)									
	TA98		TA100		YG1024		YG1029		TA98/1,8-DNP$_6$	
compound	−S9 mix	+S9 mix	−S9 mix	+S9 mix	−S9 mix	+S9 mix	−S9 mix	+S9 mix	−S9 mix	+S9 mix
3,6-DNBeP	285 000	15,000	14 000	10 900	955 000	161 000	110 000	45 800	404	112
1,8-DNP	257 000[a]		55 400[a]		2 150 000[b]		500 000[c]		639[d]	
1,6-DNP	175 000		21 600[a]							

[a] From ref *29*. [b] From ref *45*. [c] From ref *38*. [d] From ref *39*.

Table 2. Amounts and Mutagenicities of Organic Extracts from Surface Soil Samples

sampling site		amount (g)	mutagenicity[a] (revertants/g)
Izumiotsu	soil	1200	
	extract	1.4	5 280 000
Takaishi	soil	2100	
	extract	1.9	6 980 000
Nagoya	soil	2500	
	extract	3.3	14 460 000
Hekinan	soil	2500	
	extract	6.9	1 240 000

[a] Mutagenicity was examined by the Ames/*Salmonella* assay using TA98 without S9 mix.

mutagenic activity. The mutagenic activity of 3,6-DNBeP in YG1024 was about 3-fold higher than that in TA98. On the other hand, TA98/1,8-DNP$_6$ showed a markedly lower sensitivity toward 3,6-DNBeP than the parent strain TA98, as shown in Table 1. These results suggest that *O*-acetyltransferase is required for the mutagenicity of 3,6-DNBeP.

Detection of 3,6-DNBeP in Surface Soil Collected in Other Cities. To investigate the distribution of 3,6-DNBeP in surface soil at other sites, surface soil samples were collected at parks in four cities, i.e., Izumiotsu and Takaishi in Osaka Prefecture and Nagoya and Hekinan in Aichi Prefecture (Figure 1). During the assessment of the mutagenicity of surface soil in Japan (*17*, *22*, *27*), we found that organic extracts from these four soil samples showed potent mutagenicity in TA98 without S9 mix. Table 2 summarizes the amounts of organic extracts from the four soil samples and their mutagenicity in TA98 without S9 mix. From 1.4 to 6.9 g of organic extracts was acquired from the soil samples, and their mutagenic potencies ranged from 1 240 000 to 14 460 000 revertants/g of organic extract. These soil extracts were fractionated by the same method that was used for the soil sample from Takatsuki, with monitoring of the mutagenicity of the fractions in TA98 without S9 mix. The soil extracts were separated by chromatography using Sephadex LH-20 and silica gel columns. The mutagenic activity was detected in the fraction with elution volumes similar to those of the mutagenic fractions for the sample from Takatsuki. These active fractions were subsequently separated by an Ultra pack ODS column. The fractions that corresponded to 3,6-DNBeP showed potent mutagenicity, and these fractions were further separated by HPLC using COSMOSIL 5C$_{18}$ AR-II and then Luna 5 μ Phenyl-Hexyl columns. The retention times of the mutagens on both columns were identical to those of 3,6-DNBeP, i.e., 33 min for the COSMOSIL 5C$_{18}$ AR-II column and 46 min for the Luna 5 μ Phenyl-Hexyl column. A single UV absorption peak fraction was detected at the retention time of the mutagenic fraction on a Luna 5 μ Phenyl-Hexyl column for each soil sample. The UV absorption spectra of each peak fraction coincided with that of 3,6-DNBeP. The percent contributions of 3,6-DNBeP to the total mutagenicity of the organic extracts from these four soil samples in TA98 without S9 mix were as follows: 24% for Izumiotsu, 27% for Takaishi, 22% for Nagoya, and 29% for Hekinan. These results indicate that 3,6-DNBeP was present in the surface soil from all four cities and the percent contributions of 3,6-DNBeP to the mutagenicity of the surface soil from the four cities were higher than that in the sample from Takatsuki (15%).

Discussion

We previously identified DNP isomers as major mutagens in surface soil in three metropolitan areas of Japan, i.e., the Kanto, Chubu, and Kinki regions (*17*). In the present study, 1,6- and 1,8-DNP isomers and 3,6-DNBeP were detected in surface soil from Takatsuki and 12, 24, and 15% of the total mutagenicity of the soil extract was attributed to 1,6-DNP, 1,8-DNP, and 3,6-DNBeP, respectively. As shown in Table 1, 1,6- and 1,8-DNP isomers and 3,6-DNBeP exhibited extremely high mutagenicity in TA98. Many studies demonstrated that 1,6-DNP and 1,8-DNP were carcinogenic in experimental animals (*30*−*32*). 1,6-DNP was shown to induce lung carcinoma after intratracheal administration into Syrian golden hamsters (*30*) and direct injection into the lung of F334/DuCrj rats (*31*). IARC listed 1,6-DNP and 1,8-DNP as possible human carcinogens (group 2B) in IARC Monographs (*34*). The *Eighth Report on Carcinogens* published by the National Toxicology Program also listed 1,6-DNP and 1,8-DNP as "reasonably anticipated to be a human carcinogen" (*42*). This is the first report on the biological activity of 3,6-DNBeP and its detection in environmental samples. Because 3,6-DNBeP is an extremely potent bacterial mutagen, other biological activities of 3,6-DNBeP, including carcinogenicity, should be elucidated.

3,6-DNBeP was detected in the other four soil samples, which were collected in Osaka and Aichi Prefectures, and its percent contribution to the total mutagenicity of the soil extracts was 22−29%. These results indicate that 3,6-DNBeP is a major mutagen in surface soil and may largely contaminate the surface soil in these two regions in Japan. Nitrated PAHs, including DNP isomers, are produced by the incomplete combustion of organic compounds such as fossil fuels and are emitted into the ambient air (*7*, *29*). Therefore, motor vehicles are thought to be one of the major sources of nitrated PAHs in the environment (*28*, *34*, *43*). In addition, some nitroarenes have been shown to be formed by the atmospheric reaction of parent PAHs and nitrogen oxides (*6*, *7*, *44*). There has been no previous report on the formation of 3,6-DNBeP via atmospheric reactions. To clarify the source of 3,6-DNBeP in surface soil, the quantification of 3,6-DNBeP in airborne particles over extensive areas

and in exhaust particles from motor vehicles is required. Moreover, studies on the formation of 3,6-DNBeP under various environmental conditions are necessary. The exposure levels of inhabitants to 3,6-DNBeP should also be assessed to estimate its impact on the ecosystem and human health.

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References

(1) Kamiya, A., and Ose, Y. (1988) Isolation of dinitropyrene in emission gas from a municipal incinerator and its formation by a photochemical reaction. *Sci. Total Environ. 72*, 1–9.

(2) Henderson, T. R., Sun, J. D., Royer, R. E., Clark, C. R., Li, A. P., Harvey, T. M., Hunt, D. H., Fulford, J. E., Lovette, A. M., and Davidson, W. R. (1983) Triple-quadrupole mass spectrometry studies of nitroaromatic emissions from different diesel engines. *Environ. Sci. Technol. 17*, 443–449.

(3) Handa, T., Yamauchi, T., Sawai, K., Yamamura, T., Koseki, Y., and Ishii, T. (1984) In situ emission levels of carcinogenic and mutagenic compounds from diesel and gasoline engine vehicles on an expressway. *Environ. Sci. Technol. 18*, 895–902.

(4) Höner, A., Arnold, M., Hüsers, N., and Kleiböhmer, W. (1995) Monitoring polycyclic aromatic hydrocarbons in waste gases. *J. Chromatogr. A 710*, 129–137.

(5) Grimmer, G. (1983) *Environmental Carcinogens: Polycyclic Aromatic Hydrocarbons*, CRC Press, Boca Raton, FL.

(6) Pitts, J. N., Jr. (1983) Formation and fate of gaseous and particulate mutagens and carcinogens in real and simulated atmospheres. *Environ. Health Perspect. 47*, 115–140.

(7) Enya, T., Suzuki, H., Watanabe, T., Hirayama, T., and Hisamatsu, Y. (1997) 3-Nitrobenzanthrone, a powerful bacterial mutagen and suspected human carcinogen found in diesel exhaust and airborne particulates. *Environ. Sci. Technol. 31*, 2772–2776.

(8) Cohen, A. J. (2000) Outdoor air pollution and lung cancer. *Environ. Health Perspect. 47* (Suppl. 4), 115–140.

(9) IARC (1987) *IARC Monographs on the Evaluation of Carcinogenic Risks to Humans. Supplement 7: Overall Evaluations of Carcinogenicity, an Updating of IARC Monographs*, International Agency for Research on Cancer, Lyons, France.

(10) Archer, V. E. (1990) Air pollution and fatal lung disease in three Utah counties. *Arch. Environ. Health 45*, 325–334.

(11) Dockery, D. W., Pope, C. A., III, Xu, X., Spengler, J. D., Ware, J. H., Fay, M. E., Ferris, B. G., Jr., and Speizer, F. E. (1993) An association between air pollution and mortality in six U. S. cities. *N. Engl. J. Med. 329*, 1753–1759.

(12) Pope, C. A., III, Thun, M. J., Namboodiri, M. M., Dockery, D. W., Evans, J. S., Speizer, F. E., and Heath, C. W., Jr. (1995) Particulate air pollution as a predictor of mortality in a prospective study of U. S. adults. *Am. J. Respir. Crit. Care Med. 151*, 669–674.

(13) Barbone, F., Bovenzi, M., Cavalleri, F., and Stanta G. (1995) Air pollution and lung cancer in Trieste, Italy. *Am. J. Epidemiol. 141*, 1161–1169.

(14) Pope, C. A., III, Burnett, R. T., Thun, M. J., Calle, E. E., Krewski, D., Ito, K., and Thurston, G. D. (2002) Lung cancer, cardiopulmonary mortality, and long-term exposure to fine particulate air pollution. *J. Am. Med. Assoc. 287*, 1132–1141.

(15) Pope, C. A., III, Burnett, R. T., Thurston, G. D., Thun, M. J., Calle, E. E., Krewski, D., and Goldleski, J. J. (2004) Cardiovascular mortality and long-term exposure to particulate air pollution, epidemiological evidence of general pathophysiological pathways of disease. *Circulation 109*, 71–77.

(16) Edenharder, R., Ortseifen, M., Koch, M., and Wesp, H. F. (2000) Soil mutagens are airborne mutagens: Variation of mutagenic activities induced in *Salmonella typhimurium* TA98 and TA100 by organic extracts of agricultural and forest soils in dependence on location and season. *Mutat. Res. 472*, 23–36.

(17) Watanabe, T., Goto, S., Matsumoto, Y., Asanoma, M., Hirayama, T., Sera, N., Takahashi, Y., Endo, O., Sakai, S., and Wakabayashi, K. (2000) Mutagenic activity of surface soil and quantification of 1,3-, 1,6-, and 1,8-dinitropyrene isomers in soil in Japan. *Chem. Res. Toxicol. 13*, 281–286.

(18) Nishimura, T., Goto, S., Kata, Y., Okunuki, M., and Matsushita, H. (1984) Mutagenicity and benzo[*a*]pyrene contents in soils in Tokyo. *J. Jpn. Soc. Air Pollut. 19*, 228–238.

(19) Arashidani, K., Someya, T., Yoshikawa, M., and Kodama, Y. (1992) Polynuclear aromatic hydrocarbon concentration and mutagenic activity in soils sampled at the roadsides. *J. Jpn. Soc. Air Pollut. 27*, 190–197.

(20) Tamakawa, K., Takahashi, Y., Mishima, Y., Seki, T., and Tsunoda, A. (1985) Mutagenicity and benzo[*a*]pyrene contents in soils in Sendai City; Influences of particulate substances produced by studded tires of automobiles. *Eisei Kagaku 31*, 329–333.

(21) Wesp, H. F., Tang, X., and Edenharder, R. (2000) The influence of automobile exhausts on mutagenicity of soils: Contamination with, fractionation, separation, and preliminary identification of mutagens in the *Salmonella*/reversion assay and effects of solvent fractions on the sister-chromatid exchanges in human lymphocyte cultures and in the in vivo mouse bone marrow micronucleus. *Mutat. Res. 472*, 23–36.

(22) Watanabe, T., Hasei, T., Takahashi, Y., Otake, S., Murahashi, T., Takamura, T., Hirayama, T., and Wakabayashi, K. (2003) Mutagenic activity and quantification of nitroarenes in surface soil in the Kinki region of Japan. *Mutat. Res. 538*, 121–131.

(23) Goggleman, W., and Spitzauer, P. (1982) Mutagenic activity in agricultural soils. In *Carcinogens and Mutagens in the Environment* (Stich, H., Ed.), Vol. 3, pp 178–183, CRC Press, Orlando, FL.

(24) Brown, K. W., Donnelly, K. C., Thomas, J. C., Davol, P., and Scott, B. R. (1985) Mutagenicity of three agricultural soils. *Sci. Total Environ. 41*, 173–186.

(25) Knize, M. G., Takemoto, B. T., Lewis, P. R., and Felton, J. S. (1984) The characterization of the mutagenic activity of soil. *Mutat. Res. 192*, 23–30.

(26) Misik, M., and Micieta, K. (2002) *Tradescantia* micronucleus and *Vicia* chromosome anatelophase assay in monitoring of genotoxicity of urban soil. *Water Air Soil Pollut. 141*, 181–187.

(27) Endo, O., Goto, S., Matsumoto, Y., Sakai, S., Akutagawa, T., Asanoma, M., Hirayama, T., Watanabe, T., Tsukatani, H., Sera, N., Tada, A., and Wakabayashi, K. (2004) Mutagenicity of airborne particles, river waters and soils in Japan from 1996 to 2003. *Environ. Mutagen. Res. 26*, 9–22.

(28) The Editorial Board of the Cancer Statistics in Japan, Ed. (2001) *Cancer Statistics in Japan—2001*, Foundation for Promotion of Cancer Research, Tokyo.

(29) Tokiwa, H., and Onishi, Y. (1986) Mutagenicity and carcinogenicity of nitroarenes and their sources in the environment. *CRC Crit. Rev. Toxicol. 17*, 23–60.

(30) Takayama, S., Ishikawa, T., Nakajima, H., and Sato, S. (1985) Lung carcinoma induction in Syrian golden hamsters by intratracheal instillation of 1,6-dinitropyrene. *Jpn. J. Cancer Res. 76*, 457–461.

(31) Maeda, T., Izumi, K., Otsuka, H., Manabe, Y., Kinouchi, T., and Onishi, Y. (1986) Induction of squamous cell carcinoma in the rat lung by 1,6-dinitropyrene. *J. Natl. Cancer Inst. 76*, 693–701.

(32) Imaida, K., Lee, M. S., Wang, C. Y., and King, C. M. (1991) Carcinogenicities of dinitropyrenes in the female weanling CD rat. *Carcinogenesis 12*, 1187–1191.

(33) Adachi, S., Kawamura, K., Takemoto, K., Suzuki, H., and Hisamatsu, Y. (2000) Carcinogenicity of 3-nitrobenzanthrone, a potent mutagen in diesel exhaust-preliminary results in F344 rats after intratracheal administration. In *Relationships Between Respiratory Disease and Exposure to Air Pollution* (Heinrich, U., and Mohr, U., Eds.) pp 315–319, ILSI Press, Washington, DC.

(34) IARC (1989) *International Agency for Research on Cancer Monograph 46*, International Agency for Research on Cancer, Lyons, France.

(35) Watanabe, T., Ishida, S., Minami, H., Kasai, T., Ogawa, S., Wakabayashi, K., and Hirayama, T. (1998) Identification of 1,6- and 1,8-dinitropyrene isomers as major mutagens in organic extracts of soil from Osaka, Japan. *Chem. Res. Toxicol. 11*, 1501–1507.

(36) Yahagi, T., Nagao, M., Seino, Y., Matsushima, T., Sugimura, T., and Okada, M. (1977) Mutagenicity of *N*-nitrosamines on Salmonella. *Mutat. Res. 48*, 121–130.

(37) Maron, D. M., and Ames, B. N. (1983) Revised methods for the *Salmonella* mutagenicity test. *Mutat. Res. 113*, 173–215.

(38) Watanabe, M., Ishidate, M., Jr., and Nohmi, T. (1990) Sensitive method for detection of mutagenic nitroarenes and aromatic amines: New derivatives of *Salmonella typhimurium* tester strains possessing elevated *O*-acetyltransferase levels. *Mutat. Res. 234*, 337–348.

(39) McCoy, E. C., Anders, M., and Rosenkranz, H. S. (1983) The basis of the insensitivity of *Salmonella typhimurium* strain TA98/1,8-DNP6 to the mutagenic action of nitroarenes. *Mutat. Res. 121*, 17−23.

(40) Spitzer, T. (1993) Selective cleanup for polynuclear aromatic compounds in airborne particles and soil. *J. Chromatogr. 643*, 43−49.

(41) Spitzer, T., and Kuwatsuka, S. (1986) Simple method for determination of polynuclear aromatic hydrocarbons in soil by cleanup on XAD-2. *J. Chromatogr. 358*, 434−437.

(42) U.S. Department of Health and Human Services (1998) Eighth report on carcinogens. *Full Report*, National Toxicology Program, U.S. Department of Health and Human Services, Washington, DC.

(43) Hayakawa, K., Murahashi, T., Butoh, M., and Miyazaki, M. (1995) Determination of 1,3-, 1,6- and 1,8-dinitropyrenes and 1-nitropyrene in urban air by high-performance liquid chromatography using chemiluminescence detection. *Environ. Sci. Technol. 29*, 25−29.

(44) Hisamatsu, Y., Nishimura, T., Tanabe, K., and Matsushita, H. (1986) Mutagenicity of the photochemical reaction products of pyrene with nitrogen dioxide. *Mutat. Res. 172*, 19−27.

(45) Einisto, P., Watanabe, M., Ishidate, M., Jr., and Nomi, T. (1991) Mutagenicity of 30 chemicals in *Salmonella typhimurium* strains possessing different nitroreductase or *O*-acetyltransferase activities. *Mutat. Res. 259*, 95−102.

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Structures and Biological Properties of DNA Adducts Derived from *N*-Nitroso Bile Acid Conjugates

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A kind of *N*-nitrosobile acid conjugate, *N*-nitrosotaurocholic acid (NO-TCA), was incubated with calf thymus DNA, and formation of an adduct was detected by the ^{32}P-postlabeling method under nuclease P1 conditions. To examine the nucleotides containing the adduct from NO-TCA, each of 2′-deoxyribonucleotide 3′-monophosphates (3′-dAp, 3′-dGp, 3′-dCp, or 3′-Tp) was incubated with NO-TCA. The same adduct spot was detected in the reaction of NO-TCA with 3′-dCp. The structure of this adduct was determined to be 3-ethanesulfonic acid-dC by several spectrometry techniques. Moreover, bulky adducts containing bile acid moiety were also produced from the reaction of NO-TCA with 3′-dCp and 3′-dAp. From comparison with spectral data for authentic compounds, these adducts were concluded to be N^4-cholyl-dC and N^6-cholyl-dA. N^4-Cholyl-dC and N^6-cholyl-dA were also detected in calf thymus DNA treated with NO-TCA. In addition, 3-ethanesulfonic acid-dC and N^4-deoxycholyl-dC were found to be produced from *N*-nitrosotaurodeoxycholic acid (NO-TDCA) with dC. NO-TCA and NO-TDCA induced mutations in *Salmonella typhimurium* TA100 but not in TA98. Mutational spectrum analysis revealed that NO-TCA induced G to A transitions predominantly. When NO-TCA (250 mg/kg) was singly administered to male Wistar rats by gavage, both ethanesulfonic acid-dC and N^4-cholyl-dC could be detected in the glandular stomach and colon. The levels of ethanesulfonic acid-dC were 0.22−0.29 per 10^6 nucleotides, but values for N^4-cholyl-dC were about 500-fold lower. These observations suggest that *N*-nitroso bile acid conjugates, NO-TCA and NO-TDCA, may induce G to A base substitutions in genes via DNA adduct formation, producing ethanesulfonic acid- and/or (deoxy)cholic acid-DNA and, therefore, may be related to human carcinogenesis as endogenous mutagens.

Introduction

Epidemiological studies have indicated an association between bile acids and colorectal cancer (*1*, *2*), and high levels of secondary bile acids are present in the feces of populations at high risk of colorectal cancer (*3*, *4*). It has further been reported that deoxycholic acid (DCA)[1] stimulates proliferation of colonic epithelium in vitro and in vivo (*5*, *6*) and suppresses not only spontaneous but also butyrate-induced apoptosis in human colonic adenoma cells in vitro (*7*). In addition, bile acids or bile acid conjugates, such as cholic acid (CA), chenodeoxycholic acid (CDCA), litocholic acid (LCA), and taurodeoxycholic acid (TDCA), promote development of colorectal adenomas and adenocarcinomas in rats (*8*, *9*). There is some evidence of formation of DNA adducts with bile acids in vitro (*10*−*13*).

N-Nitroso bile acid conjugates, such as *N*-nitrosoglycocholic acid (NO-GCA) and *N*-nitrosotaurocholic acid (NO-TCA), have already been demonstrated to exert mutagenic activity in both bacterial and mammalian assay systems (*14*, *15*). Moreover, these compounds also can induce liver and stomach cancers in F344 rats (*16*). *N*-Nitroso bile acid conjugates are also thought to be among the presumed carcinogens of stomach and esophageal tumor development in rat duodenogastoric reflux models (*17*−*19*). For this purpose, rats are subjected to surgery in order to induce duodenal reflux into the stomach or the esophagus, thus being chronically exposed to the mixture of duodenal and gastric juice. After 50 weeks of such exposure, adenocarcinomas are found in the digestive tract without exogenous carcinogens, at reported incidences of 31−41% for stomach and 44% for esophagus. In general, patients undergoing distal gastrectomy have been reported to be at increased risk of gastric carcinoma (*20*, *21*). It has been shown that the risk is higher after Billroth II rather than Billroth I resection (*20*, *22*, *23*), the difference reflecting levels of duodenogastric reflux. The amounts of bile acid conjugates in humans are reported to be 16−40 mg/mL in bile juice, 0.08−6.77 mg/mL in gastric juices from patients

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[1] Abbreviations: DCA, deoxycholic acid; CA, cholic acid; CDCA, chenodeoxycholic acid; LCA, litocholic acid; NO-GCA, *N*-nitrosoglycocholic acid; NO-TCA, *N*-nitrosotaurocholic acid; NO-TDCA, *N*-nitrosotaurodeoxycholic acid; 3′-dAp, 2′-deoxyadenosine 3′-monophosphate; 3′-dGp, 2′-deoxyguanosine 3′-monophosphate; 3′-dCp, 2′-deoxycitidine 3′-monophosphate; 3′-Tp, thymidine 3′-monophosphate; dA, 2′-deoxyadenosine; dG, 2′-deoxyguanosine; dC, 2′-deoxycitidine; dT, 2′-deoxytymidine; N^4-cholyl-dC, N^4-cholyl-2′-deoxycytidine; N^6-cholyl-dA, N^6-cholyl-2′-deoxyadenosine; PEI, polyethyleneimine; T4-PNK, T4-polynucleotide kinase.

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with bile reflux gastritis, and 0.29−3.45 μg/mL in serum (*24*, *25*). Because intragastric formation of nitrosamides could be mediated by acid-catalyzed reaction of amides with nitrite, *N*-nitroso compounds have been suggested as plausible etiological factors in development of gastric cancer in humans (*26*, *27*). In fact, it has also been reported that taurocholic acid (TCA) is nitrosated in simulated gastric juice (*14*). Moreover, thioproline, an effective nitrite-trapping agent, could inhibit the development of esophageal adenocarcinoma induced by gastroduodenal reflux in rats (*19*). Such nitrosation is also suggested to be mediated by activated macrophages in infected and inflamed organs. Therefore, it is likely that nitrosated bile acid conjugates could contribute to cancer development as endogenous mutagens.

It is reported that O^6-carboxymethyl-guanine, 7-carboxymethyl-guanine, and 3-carboxymethyl-adenine are produced in vitro when *N*-nitrosoglycocholic acid (NO-GCA) is incubated with calf thymus DNA under neutral conditions (*28*). Similarly, it is suggested that *N*-nitrosotauroursodeoxycholic acid would form ethane sulfonic acid-DNA; however, the structure is not fully known yet (*27*). Moreover, little is known about the formation of bulky adducts containing bile acid moieties; we recently found that bile acid-DNA adducts could be formed by the reaction of DNA with cholyl adenylate, a reactive intermediate in the production of bile acid-amino acid conjugates. Structural analyses determined these bile acid-DNA adducts to be N^4-cholyl-2′-deoxycytidine (N^4-cholyl-dC) and N^6-cholyl-2′-deoxyadenosine (N^6-cholyl-dA) (*29*). Therefore, it is possible that similar bile acid-DNA adducts might be formed from *N*-nitroso bile acid conjugates. In the present study, we investigated the formation and chemical structures of DNA adducts containing ethane sulfonic acid and bile acid moieties, produced by the reaction of *N*-nitroso bile acid conjugates, NO-TCA, and *N*-nitrosotaurodeoxycholic acid (NO-TDCA), with DNA. Moreover, mutagenic activities of *N*-nitroso bile acid conjugates and their mutation spectra in *Salmonella typhimurium* TA100 were also examined. We also report here the formation of DNA adducts in some tissues of rats administered NO-TCA. From these results, the biological significance of NO-TCA and NO-TDCA as endogenous mutagens is discussed.

Materials and Methods

Materials. NO-TCA and NO-TDCA were obtained from the Nard Institute (Osaka, Japan). 2′-Deoxyribonucleotide 3′-monophosphate (3′-dAp, 3′-dGp, 3′-dCp, and 3′-Tp) and 2′-deoxyribonucleoside (dA, dG, dC, and T) were from Sigma Chemical Co. (St. Louis, MO). Micrococcal nuclease and phosphodiesterase II were purchased from Worthington Biochemical Co. (Freehold, NJ). [γ-^{32}P]ATP, T4 polynucleotidekinase (T4-PNK), nuclease P1, and phosphodiesterase I were obtained from ICN Biochemicals (Irvine, CA), Takara Shuzo Co. (Kyoto, Japan), Yamasa Shoyu Co. (Choshi, Japan), and Worthington Biochemical Co., respectively. All other chemicals used were of analytical grade.

Spectral Measurements. ^{1}H NMR spectra were recorded with JEOLGX-α600 or α800 instruments using microprobe FT-NMR spectrometers. Electronspray ionization mass spectrometry (ESI-MS) was performed using a Waters ZQ 2000 mass spectrometer equipped with an Agilent 1100 HPLC system. UV absorbance spectra were measured on a PD-8020 photodiode array detector (Tosoh, Tokyo, Japan).

Chemical Synthesis of N^4-Cholyl-dC and N^6-Cholyl-dA. Authentic N^4-cholyl-dC and N^6-cholyl-dA were synthesized according to the procedure reported previously (*29*). Briefly, 3′,5′-bis-*O*-*tert*-butyldimethylsilyl-2′-deoxycitiydine (0.1 mmol) or 3′,5′-bis-*O*-*tert*-butyldimethylsilyl-2′-deoxyadenosine (0.1 mmol) was stirred with CA (0.1 mmol) and dicyclohexylcarbodiimide (0.12 mmol) in pyridine at 70 °C for 4 h. The organic phase was evaporated, and then, these residues were separated on column chromatography, followed by deprotection.

Reaction of *N*-Nitrosobile Acid Conjugates and Nucleotide and/or DNA. Ten micromole aliquots of each *N*-nitrosobile acid conjugate (NO-TCA or NO-TDCA) were incubated with 650 nmol of calf thymus DNA in 1 mL of 100 mM phosphate buffer (pH 7.4) at 37 °C for 24 h. Then, DNA was extracted with phenol/chloroform:isoamyl alcohol (24:1 v/v) and precipitated with ethanol. DNA concentrations were measured spectrophotometrically at 260 nm and adjusted to 2 mg/mL in 0.01× SSC buffer. With reactions between *N*-nitrosobile acid conjugates and mononucleotides, 650 nmol of each 2′-deoxyribonucleotide 3′-monophosphate (3′-dAp, 3′-dGp, 3′-dCp, and 3′-Tp) was used instead of calf thymus DNA. Adduct formation was analyzed by the ^{32}P-postlabeling method under nuclease P1 conditions as described below.

Structural Analysis of Ethane Sulfonic Acid-dC Adducts. NO-TCA or NO-TDCA was incubated with dC in 100 mM phosphate buffer (pH 7.4) at 37 °C for 24 h, and the resulting solution was separated by HPLC as follows. An aliquot of the solution was applied to a semipreparative Capcell pack C18 column (5 μm particle size, 10 mm × 250 mm; Shiseido, Tokyo, Japan), and a mobile phase of 2% acetonitrile in 0.25% triethylamine-acetic acid (pH 6.3) was pumped in isocratically at a flow rate of 3 mL/min. In addition to a few peaks detected in the solution without dC, a peak, eluting at a retention time of 9.3 min, was newly observed in the reaction mixture of NO-TCA and dC. The peak was collected and injected into an analytical grade TSKgel ODS-80Ts column (5 μm particle size, 4.6 mm × 250 mm; Tosoh) for further purification. The applied material was eluted at a flow rate of 1 mL/min with a gradient system of 2.5% methanol in 0.1% heptafluorobutylic acid (pH 3.0) for 10 min and then a linear gradient to 80% methanol in 0.1% heptafluorobutylic acid. All of the above HPLC procedures were performed several times at an ambient temperature with monitoring of eluate at 254 nm. The compound was collected for determination of UV absorption, mass, and ^{1}H and ^{13}C NMR spectra. ^{1}H and ^{13}C NMR spectrum assignment of the adduct was performed with DQF-COSY, HMQC, and HMBC. The following data were obtained.

^{1}H NMR (DMSO-d_6): δ 8.09 [d, J = 7.2, H-6 (dC), 1H], 6.11 [d, J = 7.2, H-5 (dC), 1H], 6.07 [t, J = 6.4, H-1′ (dR), 1H], 5.29 [d, J = 4.0, H-3′-OH (dR), 1H], 5.07 [t, J = 4.8, H-5′-OH (dR), 1H], 4.21−4.19 [m, H-3′ (dR), 1H], 4.15 (dd, J = 7.2, 6.4, H-C$\underline{H_2}$CH$_2$SO$_3$H, 2H), 3.83 [dd, J = 7.2, 3.2, H-4′ (dR), 1H], 3.61−3.59 [m, H-5″ (dR), 1H], 3.57−3.54 [m, H-5′ (dR), 1H], 2.86 (t, J = 6.4, H-CH$_2$C$\underline{H_2}$SO$_3$H, 2H), 2.23−2.21 [m, H-2″ (dR), 1H], 2.13−2.10 [m, H-2′ (dR), 1H]. ^{13}C NMR (DMSO-d_6) δ 159.1 [C-4 (dC)], 147.6 [C-2 (dC)], 140.6 [C-6 (dC)], 95.2 [C-5 (dC)], 88.0 [C-4′ (dR)], 86.6 [C-1′ (dR)], 69.6 [C-3′ (dR)], 60.7 [C-5′ (dR)], 47.1 (C-CH$_2$C$\underline{H_2}$SO$_3$H), 41.1 (C-$\underline{C}$H$_2$CH$_2$SO$_3$H). ESI-MS *m/z* 334, 218. UV λ_{max} 219, 280 nm.

Structural Analysis of CA-dC Adducts. NO-TCA was incubated with dC, and the solution was subjected to HPLC under the following conditions. An aliquot was applied to an analytical grade TSKgel ODS-80Ts column (5 μm particle size, 4.6 mm × 250 mm; Tosoh) and eluted at a flow rate of 1 mL/min with a linear gradient of acetonitrile (from 20 to 80%) in 0.1% diethylamine-acetic acid (pH 6.0) over 30 min. In addition to a few peaks detected in the solution without dC, a peak, eluted at a retention time of 21.3 min, was observed in the reaction mixture of NO-TCA and dC. UV absorption and mass spectra showed absorption maxima at 245 and 298 nm and molecular ion peaks at *m/z* 502 and 618. From comparison with the UV and mass spectra for authentic N^4-cholyl-dC, the compound at a peak of 21.3 min was suggested to be a CA-dC adduct. The peak fraction was therefore collected and applied to ^{1}H NMR spectral analyses. The following data were obtained.

^{1}H NMR (DMSO-d_6): δ 10.81 (s, 1H, NHCO), 8.30 [d, J = 7.2, 1H, H-6 (dC)], 7.21 [d, J = 7.2, 1H, H-5 (dC)], 6.09 [t, J = 6.4, 1H, H-1′ (dR)], 5.27 [brs, 1H, 3′-OH (dR)], 5.04 [brs, 1H, 5′-OH (dR)], 4.31 [s, 1H, 3-OH (CA)], 4.20 [brs, 1H, H-3′ (dR)], 4.11 [d, J = 3.2, 1H, 12-OH (CA)], 4.00 [d, J = 3.2, 1H, 7-OH (CA)], 3.84 [dd, J = 7.2, 3.6, 1H, H-4′ (dR)], 3.77 [d, J = 2.8, 1H, H-12 (CA)], 3.60−3.54 [m, 3H, H-7 (CA), H-5′, 5″ (dR)], 3.50−3.17 [m, 1H, H-3 (CA) overlapped with the absorbance signal of H_2O], 2.30−2.13 [m, 5H, H-4α, 9, 23α, 23β (CA), H-2′ (dR)], 2.01−1.98 [m, 2H, H-14 (CA), H-2″ (dR)], 1.79−1.62 [m, 7H, H-17, 20, 22α, 16α, 1α, 6β, 15β (CA)], 1.45−1.14 [m, 12H, H-2α, 2β, 4β, 5, 6α, 8, 11α, 11β, 15α, 16β, 22β (CA)], 0.98−0.92 [m, 4H, H-1β, CH_3-21 (CA)], 0.80 [s, 3H, CH_3-19 (CA)], 0.57 [s, 3H, CH_3-18 (CA)]. ESI-MS *m/z* 618, 502. UV λ_{max} 245, 298 nm.

Structural Analysis of CA-dA Adducts. NO-TCA was incubated with dA as described above and separated by HPLC under the same conditions as for structural analysis of CA-dC adducts. A peak eluting at a retention time of 15.4 min was observed in the reaction mixture, and the UV absorption pattern showed absorption maximum at 273 nm. Because the HPLC eluting position and UV spectrum of the compound were similar to those of the authentic N^6-cholyl-dA, this compound was deduced to be a CA-dA adduct. LC-ESI/MS analysis demonstrated this compound to exhibit a molecular ion at *m/z* 642 and fragment at *m/z* 526, the latter deriving from loss of deoxyribose. Therefore, the chemical structure of the compound was concluded to be N^6-cholyl-dA.

Structural Analysis of DCA-dC Adducts. NO-TDCA and dC were incubated in 100 mM phosphate buffer (pH 7.4) at 37 °C for 24 h and separated by HPLC according to the same procedure as for CA-dC or dA adducts. A peak showing a similar UV absorption pattern to N^4-cholyl-dC was observed at a retention time of 23.0 min and collected for UV, mass, and ^{1}H and ^{13}C NMR spectral analyses. ^{1}H and ^{13}C NMR spectrum assignment of the adduct was conducted with DQF-COSY, HMQC, and HMBC. The following data were obtained.

^{1}H NMR (DMSO-d_6): δ 10.81 (s, 1H, NHCO), 8.30 [d, J = 7.2, 1H, H-6 (dC)], 7.20 [d, J = 7.2, 1H, H-5 (dC)], 6.09 [t, J = 6.2, 1H, H-1′ (dR)], 5.27 [brs, 1H, 3′-OH (dR)], 5.04 [brs, 1H, 5′-OH (dR)], 4.45 [s, 1H, 3-OH (DCA)], 4.20 [brs, 1H, H-3′ (dR)], 3.84 [dd, J = 6.6, 2.4, 1H, H-4′ (dR)], 3.77 [brs, 1H, H-12 (DCA)], 3.59−3.56 [m, 3H, H-7 (DCA), H-5′, 5″ (dR)], 3.50−3.17 [m, 1H, H-3 (DCA)], 2.40−2.25 [m, 5H, H-4α, 9, 23α, 23β (DCA), H-2′ (dR)], 2.02−1.96 [m, 2H, H-14 (DCA), H-2″ (dR)], 1.80−1.44 [m, 8H, H-17, 20, 22α, 16α, 1α, 6β, 7β, 15β (DCA)], 1.35−1.15 [m, 13H, H-2α, 2β, 4β, 5, 6α, 7α, 8, 11α, 11β, 15α, 16β, 22β (DCA)], 1.09−0.91 [m, 4H, H-1β, CH_3-21 (DCA)], 0.83 [s, 3H, CH_3-19 (DCA)], 0.58 [s, 3H, CH_3-18 (DCA)]. ^{13}C NMR (DMSO-d_6): δ 175.0 [C=O (DCA)], 162.9 [C-4 (dC)], 155.0 [C-2 (dC, C=O)], 145.5 [C-6 (dC)], 95.8 [C-5 (dC)], 88.5 [C-4′ (dR)], 86.7 [C-1′ (dR)], 71.6 [C-12 (DCA)], 70.5 [C-3 (DCA), C-3′ (dR)], 61.5 [C-5′ (dR)], 47.4 [C-14 (DCA)], 46.3 [C-17 (DCA)], 46.0 [C-13 (DCA)], 41.6 [C-5 (DCA)], 40.9 [C-2′ (dR)], 40−39 [C-4, 8 (DCA) overlapped with absorbance signal of DMSO], 36.3 [C-23 (DCA)], 35.1 [C-1 (DCA)], 35.0 [C-20 (DCA)], 33.8 [C-10 (DCA)], 32.9 [C-9 (DCA)], 30.9 [C-22 (DCA)], 30.2 [C-2 (DCA)], 28.6 [C-11 (DCA)], 27.1 [C-16 (DCA)], 27.0 [C-6 (DCA)], 26.1 [C-7 (DCA)], 23.5 [C-15 (DCA)], 23.1 [C-19 (DCA)], 17.0 [C-21 (DCA)], 12.5 [C-18 (DCA)]. ESI-MS *m/z* 602, 486. UV λ_{max} 246, 298 nm.

Analysis of DNA Adducts Derived from Bile Acid Conjugates in the Organs of Rats Administered NO-TCA. Five male Wistar rats, purchased from Charles River Japan, Inc. (Atsugi, Japan), were provided with food (CE-2 pellet diet, CLEA Japan) and tap water ad libitum. NO-TCA was dissolved in water and administered to three rats as a single oral dose by gavage at level of 250 mg per kg body wt. The two rats in the control group received the solvent alone. At 24 h after administration of NO-TCA, both chemically treated and control groups of rats were euthanized under ether anesthesia. The major organs, such as the liver, glandular stomach, and colon mucosa, were excised and stored at −80 °C until DNA extraction by a standard procedure with enzymatic digestion of protein and RNA followed by extraction with phenol and chloroform/isoamyl alcohol (24:1, v/v). The experiments were conducted according to the Guidelines for Animal Experiments in National Cancer Center of the Committee for Ethics of Animal Experimentation of the National Cancer Center.

Enzymatic digests of 100 μg aliquots of DNA were subjected to HPLC using the heptafulorobutylic acid-methanol system as described in the structure analysis of 3-ethane sulfonic acid-dC, and the eluents were collected at retention times of 3−4 min for analysis of 3-ethanesulfonic acid-dC and 46−47 min for analysis of N^4-cholyl-dC. The retention times for 3-ethanesulfonic acid-3′-dCp and N^4-cholyl-3′-dCp were confirmed using LC-ESI/MS analysis. After lyophilization of the fractioned samples, residues were dissolved in 10 μL of distilled water and then subjected to ^{32}P-postlabeling analysis under nuclease P1 conditions, as described below.

^{32}P-Postlabeling Method. DNA obtained from the in vitro experiments using calf thymus DNA or the in vivo experiments was digested with micrococcal nuclease and phosphodiesterase II and then ^{32}P-postlabeled using the nuclease P1 enrichment method as reported previously (*30*). For the in vitro experiments using deoxyribonucleotide 3′-monophosphate as a DNA source, ^{32}P-postlabeling was performed under nuclease P1 conditions without DNA digestion.

For the separation of ethanesulfonic acid-DNA adduct, the resulting ^{32}P-postlabeled samples were applied to a polyethyleneimine (PEI) cellulose TLC sheet (POLYGRAM CEL 300 PEI; Macherey-Nagel, Duren, Germany), attached to 10 cm filter paper at the top, and developed with 0.1 M lithium chloride, 3 M acetic acid, and 3 M urea to remove [^{32}P]phosphate and [γ-^{32}P]ATP, as previously reported (*31*).

For the analysis of CA-DNA adducts, the ^{32}P-postlabeled samples were applied to a PEI-cellulose sheet, attached to 10 cm filter paper at the top, and developed with 2.3 M sodium phosphate buffer (pH 6.0) to remove normal nucleotides. The modified nucleotides remaining at the origin were contact-transferred to another PEI-cellulose sheet and then subjected to two-dimensional TLC. The solvent system for development consisted of buffer A (2.07 M lithium formate and 7.65 M urea, pH 3.5) from bottom to top and buffer B (0.90 M lithium chloride, 0.45 M Tris-HCl, and 7.65 M urea, pH 8.0) from left to right, followed by 1.7 M sodium phosphate buffer, pH 6.0, from left to right, with 3.5 cm filter paper.

Adducts were detected with a Bio-Image Analyzer (BAS 2000; Fuji Photo Film Co., Tokyo, Japan) after exposing the TLC sheets to Fuji imaging plates or autoradiography on Kodak XAR-5 film with intensifying screens. Relative adduct labeling was determined by the methods of Gupta et al. (*32*) and Reddy et al. (*30*), and values were calculated as averages using data from three assays.

Mutagenicity Assay. The preincubation method (*33*) was carried out for testing the mutagenicity of *N*-nitrosobile acid conjugates to *S. typhimurium* TA98 and TA100. Briefly, bacterial cells were incubated with the test chemical without S9 mix for 20 min at 37 °C in a total volume of 0.7 mL. The mixture was poured onto agar plates with 2 mL of soft agar and incubated for 2 days at 37 °C. The numbers of His$^+$ revertants per plate were then determined.

Sequencing of *hisG46* Mutations. A 201 bp DNA fragment containing the *hisG46* site was amplified by colony PCR (forward primer: 5′-GAT TGA TAT CCT GCG CGT GCG TG-3′; reverse primer: 5′-TCG TCA ACC GGT GTT GCC AGC G-3′). DNA sequencing was performed with a BigDye Terminater Cycle Sequencing Kit (Applied Biosystems, Foster City, CA) on an ABI PRISM 310 Genetic Analyzer (Applied Biosystems).

Results

Formation of Ethane Sulfonic Acid-dC Adduct by NO-TCA. In reactions of NO-GCA with DNA, 7-carboxymethylguanine and O^6-methylguanine are reported

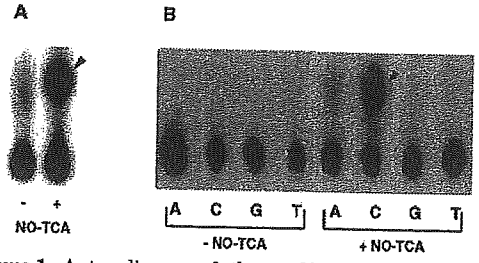


Figure 1. Autoradiogram of ethanesulfonic acid-DNA adducts derived from NO-TCA. Calf thymus DNA (A) or each of 2′-deoxyribonucleotide 3′-monophosphate (B) was incubated with or without NO-TCA, and DNA adduct formation was analyzed by ^{32}P-postlabeling under nuclease P1 conditions. The adduct spots are indicated by the arrowheads.

to be produced, thus taurine-modified DNA adduct is expected to be formed from NO-TCA (*27*, *28*). Therefore, NO-TCA was incubated with 650 nmol of calf thymus DNA under neutral conditions at 37 °C for 24 h and adduct formation was analyzed by ^{32}P-postlabeling under nuclease P1 conditions. TLC development was carried out with 0.1 M lithium chloride, 3 M acetic acid, and 3 M urea, as previously reported, being different from the TLC development systems for bulky adduct (*30*). As shown in Figure 1A, radioactive spots were detected at the origin in the mixture without NO-TCA, but one spot, in an upper position from the origin, was clearly observed in the reaction mixture of NO-TCA and calf thymus DNA. The adduct level was estimated to be 30.2 ± 6.21 per 10^5 nucleotides. To examine the nucleotides involved in the adducts with NO-TCA treatment, the compound was incubated with separate 2′-deoxyribonucleotide 3′-monophosphate (3′-dAp, 3′-dGp, 3′-dCp, and 3′-dTp), and adduct formation was analyzed. As shown in Figure 1B, the same adduct was also detected in the reaction of NO-TCA with 3′-dCp.

To analyze its chemical structure, NO-TCA was incubated with dC and the resulting solution was separated by HPLC. An aliquot of the solution was applied to a semipreparative ODS column, and a peak, eluting at a retention time of 9.3 min, was observed. This peak fraction was then further purified using an analytical grade ODS column, and a peak eluting at a retention time of 9.5 min was collected for analyses of UV absorption, mass, and ^{1}H and ^{13}C NMR spectra. The yield of this adduct from dC was about 1%. Its UV/vis absorption spectrum showed absorption maxima at 280 nm (Figure 2), and mass spectrum analysis revealed a molecular ion at *m/z* 334 corresponding to a protonated derivative of the conjugated product of ethanesulfonic acid and deoxycytidine and a fragmentation ion at *m/z* 218, consistent with loss of the deoxyribose moiety. Figure 3 shows the ^{1}H NMR spectrum of ethanesulfonic acid-dC compound measured in DMSO-d_6. The 15 protons were assigned with DQF-COSY, HMQC, and HMBC. The proton signals corresponding to the 2′-deoxycytidine moiety [H-1′, 2′, 2″, 3′, 4′, 5′, 5″, 3′-OH, 5′-OH (dR), H-5, -6 (dC)] were observed from 2.10 to 8.09 ppm. Moreover, proton signals at 4.15 and 2.86 ppm were assigned to the ethanesulfonic acid moiety. Although no proton signal was observed corresponding to the exocyclic amino group, the N4 position of the cytidine, HMBC analysis showed the signal at 4.15 ppm on ^{1}H NMR spectroscopy of the ethanesulfonic acid moiety to clearly correlate with signal peaks at 159.1 and 147.6 ppm in the ^{13}C NMR spectrum

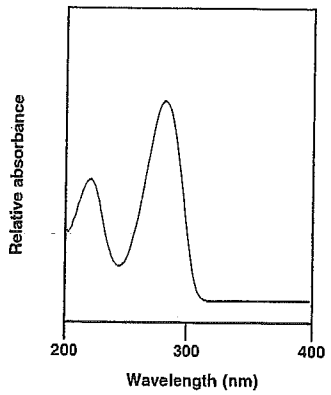


Figure 2. UV/vis absorption spectrum of the ethansulfonic acid-dC adduct. An aliquot of the reaction mixture of NO-TCA and dC was applied to HPLC on an analytical ODS column with a linear gradient of methanol (from 2.5 to 80%) in 0.1% heptafluorobutylic acid (pH 3.0), and the UV/vis absorption spectrum of the ethanesulfonic acid-dC was measured with a photodiode array detector.

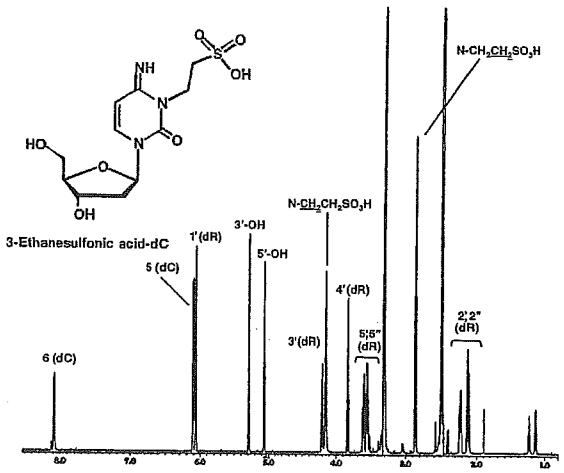


Figure 3. ^{1}H NMR spectrum in DMSO-d_6 and chemical structure of the ethanesulfonic acid-dC adduct.

of the cytidine moiety (see Supporting Information). Therefore, the structure of this compound was concluded to be 3-ethanesulfonic acid-dC (Figure 3).

Formation of CA-dC and -dA Adducts by NO-TCA. As mentioned above, the DNA adduct-containing taurine moiety was clearly formed by NO-TCA. Then, the formation of bile acid-DNA adduct was analyzed by the ^{32}P-postlabeling method, under the different condition from the case of 3-ethanesulfonic acid-dC adduct.

As shown in Figure 4, two major adduct spots (Figure 4A) were clearly detected on reaction of NO-TCA with calf thymus DNA. Levels of these adduct spots were estimated to be 3.40 ± 0.16 (Figure 4A, spot 1) and 1.69 ± 0.20 (Figure 4A, spot 2) per 10^8 nucleotides. In addition, one minor adduct spot (Figure 4A, spot 3) was also observed and the adduct level was almost 10 times lower than those of major spots.

To confirm the nucleotides involved in these adducts, this nitroso compound was incubated with 2′-deoxyribonucleotide 3′-monophosphates, and adduct formation was analyzed by the ^{32}P-postlabeling method under nuclease P1 conditions. As shown in Figure 4B, the major adduct

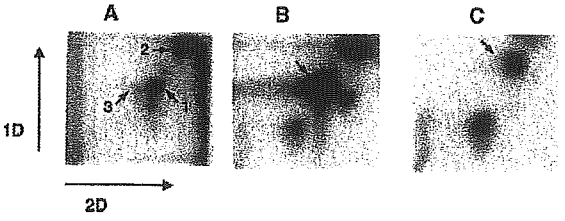


Figure 4. Autoradiograms of bile acid-DNA adducts derived from NO-TCA. NO-TCA was incubated with calf thymus DNA (A), 3′-dCp (B), and 3′-dAp (C) in 100 mM phosphate buffer (pH 7.4) at 37 °C for 24 h. Then adduct formations were analyzed by the ^{32}P-postlabeling method and developed with different conditions from the case of ethanesulfonic acid-dC adduct described in the Materials and Methods. Adducts are indicated by arrows.

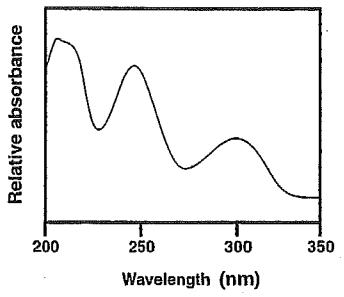


Figure 5. UV/vis absorption spectrum of the CA-dC adduct eluting at a retention time of 21.3 min on an ODS column and measured with a photodiode array detector.

spot (Figure 4A, spot 1) observed with the calf thymus DNA corresponded to the dC adduct, and another spot (Figure 4A, spot 2) corresponded to adduct spot detected in the reaction mixture of NO-TCA and 3′-dAp (Figure 4C). Moreover, the minor adduct spot (Figure 4A, spot 3) was deduced to be a dG adduct (data not shown). From these findings, DNA adducts detected in the NO-TCA treated calf thymus DNA were concluded to be NO-TCA-dC, NO-TCA-dA, and NO-TCA-dG. To analyze the chemical structure of the major adduct, NO-TCA was incubated with dC and the adduct was isolated by HPLC on an ODS column using a linear gradient of an acetonitrile in diethylamine-acetic acid (pH 6.0) solvent system. A peak eluting at a retention time of 21.3 min was observed in the reaction mixture of NO-TCA and dC. Thus, this peak fraction was collected and the UV/vis absorption spectrum of the responsible compound was measured. As shown in Figure 5, its absorption maxima were at 245 and 298 nm. Mass spectrometry demonstrated a molecular ion at *m/z* 618, which corresponded to a protonation product of a CA and deoxycytidine conjugate. A prominent fragment peak observed at *m/z* 502 was consistent with loss of the deoxyribose moiety from the protonated molecule. The authentic sample of N^4-cholyl-dC showed UV absorption maxima at 243 and 295 nm and exhibited a molecular ion of *m/z* 618. Therefore, the dC adduct formed from NO-TCA was deduced to be CA-dC adduct. By repeating the HPLC fractionation, about 400 μg of the CA-dC adduct could be collected for measurement of the ^{1}H NMR spectrum. The yield of CA-dC adduct from dC was about 0.06%. Figure 6A shows the ^{1}H NMR spectrum for the CA-dC adduct measured in DMSO-d_6, indicating the presence of 52 protons, which were assigned by two-dimensional NMR and comparison with NMR spectral data obtained from the authentic sample of N^4-cholyl-dC (Figure 6B). Forty upfield peaks from 0.57 to 4.27 ppm were assigned to the CA moiety. Proton signals corresponding to the 2′-deoxycytidine moiety [H-1′, 2′, 2″, 3′, 4′, 5′, 5″, 3′-OH, 5′-OH (dR), H-5, -6 (dC)] were also observed from 1.98 to 8.30 ppm. Thus, all proton signals of the product formed from NO-TCA and dC agreed with those of authentic N^4-cholyl-dC. Its structure is shown in Figure 7.

From the results of ^{32}P-postlabeling analysis, another major DNA adduct produced by NO-TCA with calf thymus DNA was suggested to contain a dA moiety. Speculating from the results of the dC adduct described above, the structure of the dA adduct derived from NO-TCA was suggested to be N^6-cholyl-dA. To confirm the structure of the NO-TCA-dA adduct, NO-TCA was incubated with dA in 100 mM phosphate buffer (pH 7.4) at 37 °C for 24 h and separated by HPLC under the same conditions for structural analysis of the CA-dC adduct. A peak eluting at a retention time of 15.4 min, the same as for authentic N^6-cholyl-dA, was observed in the reaction mixture. Moreover, this peak fraction had a similar UV absorption spectrum, with an absorption maximum at 273 nm, to that of the authentic sample of N^6-cholyl-dA (absorption maximum at 272 nm). LC-ESI/MS analysis revealed a molecular ion at *m/z* 642, corresponding to a protonated product of a CA and deoxyadenosine conjugate, and a fragmention ion at *m/z* 526, derived from the loss of the deoxyribose moiety from the original compound (Figure 8). From these observations, the chemical structure of this compound was concluded to be N^6-cholyl-2′-dA (Figure 8).

Structures of Adducts Formed by NO-TDCA. The other *N*-nitroso bile acid conjugate, NO-TDCA, would be expected to form similar dC adducts, including ethane sulfonic acid or deoxycholyc acid moiety. Therefore, NO-TDCA was incubated with dC, and the adduct formation was analyzed by LC-ESI/MS. A peak revealing a molecular ion at *m/z* 334 and its fragmentation ion at *m/z* 218 was eluted at the same retention time of 3-ethanesulfonic acid-dC (data not shown). Moreover, the UV/vis absorption pattern of this compound was identical to that of 3-ethanesulfonic acid-dC. Thus, 3-ethanesulfonic acid-dC was also produced from NO-TDCA and dC, and its yield was about the same levels as those of NO-TCA.

To confirm the formation of dC adduct including DCA moiety, NO-TDCA was incubated with dC and the adduct was isolated by HPLC on an ODS column according to the same procedures of N^4-cholyl-dC. The UV absorption pattern of this purified compound was similar to that of N^4-cholyl-dC, and its absorption maxima were at 246 and 298 nm (see Supporting Information). Like the N^4-cholyl-dC adduct, this compound exhibited a molecular ion at *m/z* 602, corresponding to a protonated product of DCA and dC conjugate, and a fragmentation ion at *m/z* 486, consistent with loss of the deoxyribose moiety from the protonated molecule (see Supporting Information). By repeating the HPLC procedures, around 1000 μg of the DCA-dC adduct could be collected for ^{1}H and ^{13}C NMR spectrum analysis. The yield of DCA-dC adduct from dC was 0.02%, and the ^{1}H and ^{13}C NMR spectra were assigned with DQF-COSY, HMQC, and HMBC (see Supporting Information). On the basis of the UV/vis, mass, and NMR spectral data, the structure of the adduct formed from NO-TDCA and dC was concluded to be N^4-deoxycholyl-dC (see Supporting Information).

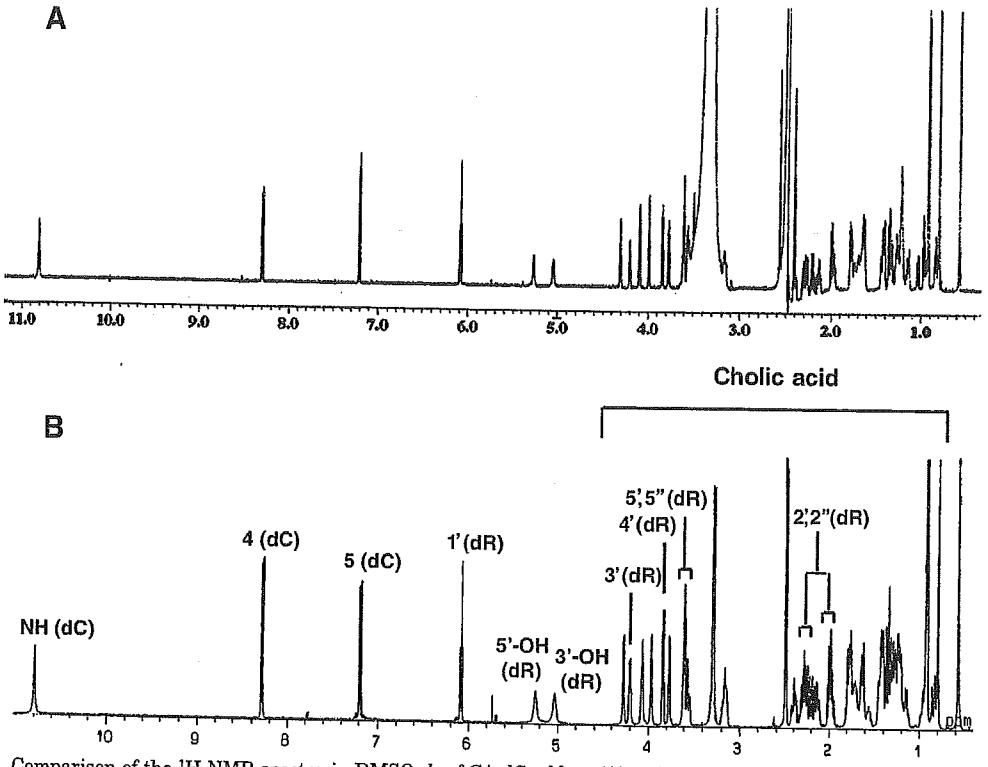


Figure 6. Comparison of the ^{1}H NMR spectra in DMSO-d_6 of CA-dC adduct (A) and authentic N^4-cholyl-dC (B).

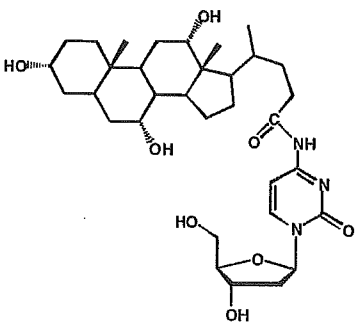


Figure 7. Chemical structure of N^4-cholyl-dC.

Mutagenic Activity of *N*-Nitroso Bile Acid Conjugates in *Salmonella* Strains. Mutagenic responses of *N*-nitroso bile acid conjugates in TA98 and TA100 were examined without metabolic activation. NO-TCA and NO-TDCA showed mutagenicity to TA100 in a dose-dependent manner but did not induce revertants in TA98. Their mutagenic activities were 9488 revertants for NO-TCA and 20800 revertants for NO-TDCA per μmol. Analysis of reverse mutations occurring at the *hisG46* site of TA100 with NO-TCA by DNA sequencing (Table 1) revealed G:C to A:T transitions dominantly. Among these, the pattern of CCC to CTC was almost four times higher than those of spontaneous mutations.

In Vivo Formation of Ethanesulfonic Acid-dC and N^4-Cholyl-dC. To confirm the formation of DNA adducts derived from bile acid conjugates in vivo, NO-TCA was singly injected at a dose of 250 mg/kg into Wistar rats by gavage, and then, 3-ethanesulfonic acid-dC and N^4-cholyl-dC were analyzed in the liver, glandular stomach, and colon mucosa, by a combination of HPLC separation and ^{32}P-postlabeling analysis. One hundred microgram aliquots of DNA digests were subjected to HPLC, and eluates were collected at 1 min intervals at retention times of 3−4 and 46−47 min, for the analysis of 3-ethanesulfonic acid-dC and N^4-cholyl-dC, respectively. The fractions were lyophilized and then analyzed by the ^{32}P-postlabeling method under nuclease P1 conditions. The adduct spot corresponding to the 3-ethanesulfonic acid-dC could be detected in the stomach and colon mucosa but not in the liver of rats treated with NO-TCA; no adduct spots were detected in these organs of control rats (Figure 9A). Adduct levels were estimated to be 0.22 ± 0.05 for stomach and 0.29 ± 0.09 for colon per 10^6 nucleotides. As the case of 3-ethanesulfonic acid-dC adduct, N^4-cholyl-dC adduct was seen in the stomach and colon mucosa of NO-TCA treated animals at estimated levels of 0.51 ± 0.33 and 0.67 ± 0.42 per 10^9 nucleotides, respectively (Figure 9B), although no adduct spot corresponding to N^4-cholyl-dC could be detected in fraction samples at a retention time of 46−47 min from control rats.

Discussion

The present study revealed formation and chemical structures of DNA adducts derived from *N*-nitroso bile acid conjugates using the ^{32}P-postlabeling method and various spectrometry techniques. Ethanesulfonic acid-dC adduct was formed from NO-TCA or NO-TDCA with calf thymus DNA or 2′-deoxynucleotides as a major adduct, and its chemical structure was concluded to be 3-ethanesulfonic acid-dC. In the case of reaction between NO-GCA with DNA, the major adducts were earlier demonstrated to be 7-carboxymethylguanine, 3-carboxymethyladenine,

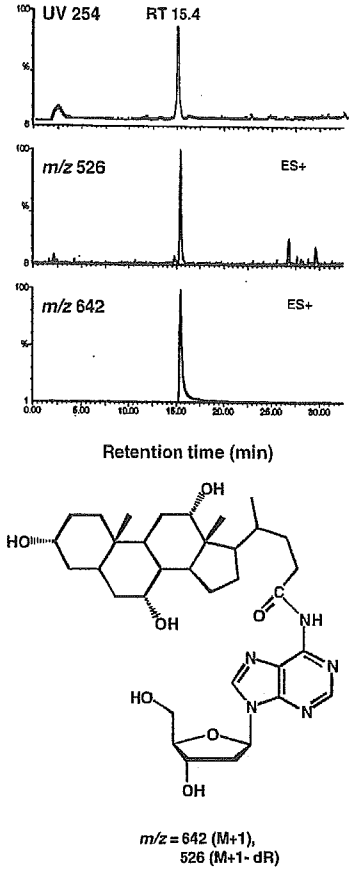


Figure 8. LC-ESI/MS analysis and chemical structure of the CA-dA adduct. An aliquot of purified CA-dA adduct was subjected on an analytical grade ODS column under the same conditions as used in CA-dC adduct and detected by UV (254 nm) absorption and ESI/MS (*m/z* 642 and 526, M + 1). Proposed mass of *m/z* = 642 corresponds to the $[M + H]^+$ of the N^6-cholyl-dA. A fragmentation at *m/z* 526 corresponded to the loss of the deoxyribose moiety from the original compound.

Table 1. Mutation Spectra for *hisG46* Reversions of TA100 Treated with NO-TCA

	TA100	
	control	NO-TCA
	G:C to A:T	
TCC (Ser)	11	11
CTC (Leu)	21	76
	G:C to T:A	
ACC (Thr)	37	13
CAC (His)	26	0
	G:C to C:G	
GCC (Ala)	5	0
%	100	100
no. of mutants	(19)	(53)

and O^6-carboxymethylguanine (*28*). As with formation of carboxymethyl-DNA adducts, ethanesulfonic acid adducts with other nucleotides than dC might be formed from NO-TCA and DNA. However, only a single spot could be detected in the reaction mixture of NO-TCA and 3′-dCp under the ^{32}P-postlabeling conditions used in the present study. On the other hand, several adducts containing a bile acid moieties were also formed from NO-TCA or NO-TDCA with calf thymus DNA or 2′-deoxyribonucleotides. The chemical structures of these major adducts were

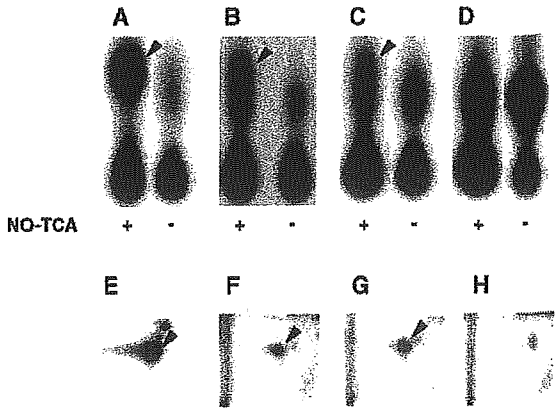
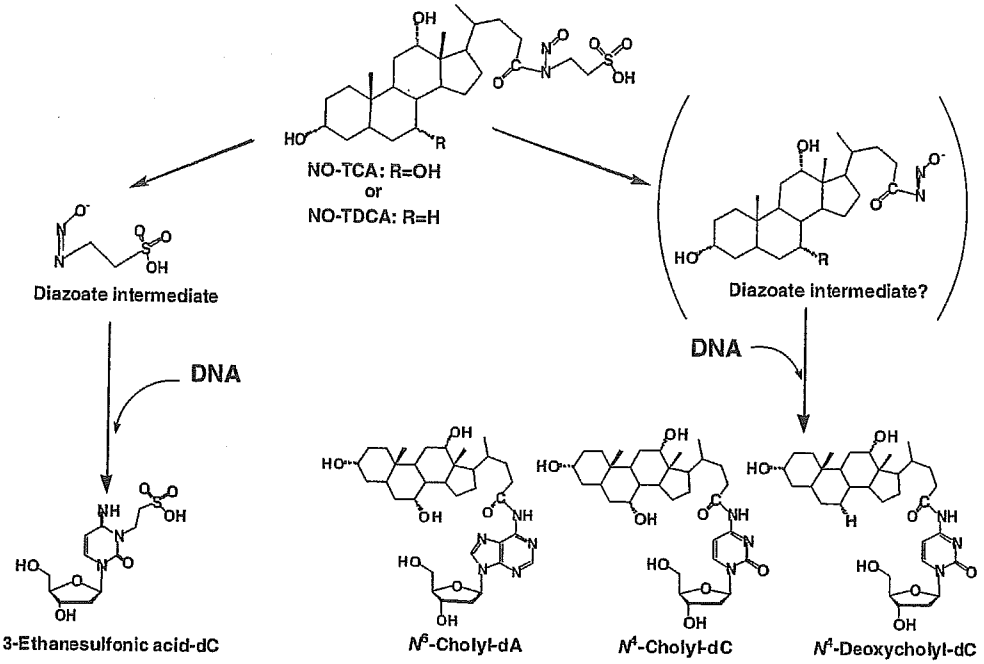


Figure 9. Autoradiograms of 3-ethanesulfonic acid-dC (A–D) and N^4-cholyl-dC (E–H) in calf thymus DNA or some organs of Wistar rats treated with or without NO-TCA. Adducts were analyzed by a combination of HPLC separation and ^{32}P-post-labeling analysis, as described in the Materials and Methods. Calf thymus DNA treated with NO-TCA (A, E) was used as a positive control. DNA samples were isolated from glandular stomach (B, F), colon mucosa (C, G), and liver (D, H) of rats with or without NO-TCA treatment. Adducts are indicated by the arrowheads.

concluded to be N^4-cholyl-dC, N^4-deoxycholyl-dC, and N^6-cholyl-dA, respectively, by spectrometry analysis. In addition, one minor adduct formed from NO-TCA and calf thymus DNA was suggested to be a dG adduct, although its chemical structure has yet to be clarified. Because the carbonyl moiety of CA was found to bind to N^4- or N^6-exocyclic amino groups of dC or dA in the present study, it is possible that the structure of CA-dG is N^2-cholyl-dG. In general, the positions of N7-, O6-dG, and N3-dA are preferentially attacked by alkylating agents, and 7-dG and 3-dA adducts are well-known to be unstable; thus, these adducts undergo spontaneous cleavage of the glycosidic bond, to produce abasic sites in DNA (*34*). Therefore, if 7-dG- or 3-dA-ethanesulfonic acid or bile acid adducts are produced from NO-TCA or NO-TDCA, they would be impossible to be detected by the ^{32}P-postlabeling methods used in the present study. As shown in Scheme 1, possible mechanisms for the formation of 3-ethanesulfonic acid-dC might be involved in the decomposition products of *N*-nitroso tauro bile acid conjugates. Dayal et al. have reported that *N*-nitroso bile acid conjugates decompose between pH 6 and pH 9 in aqueous buffered solutions, to generate several carcinogenic species, such as alkyl diazoate and its protonated form (*27*, *34*). Such electrophilic species might be implicated in the alkylation of DNA. In fact, the existence of electrophilic decomposition products of *N*-nitrosotauroursodeoxycholic acid, such as isethionic acid, diazoisethionic acid, and the protonated diazoate intermediate, has already been demonstrated by mass spectrum analysis (*27*). Meanwhile, there are no data regarding how bile acid-DNA adducts are formed at present; if the same mechanisms are involved in the generation of (deoxy)CA-DNA and ethanesulfonic acid-DNA, production of bile acid diazoate from NO-TCA or NO-TDCA would be necessary (Scheme 1). It has been well-known that the majority of the bulky-DNA adducts derived from exogenous mutagenic/carcinogenic compounds such as polycyclic aromatic hydrocarbons and heterocyclic amines have been identified as dG-C8 or dG-N2 adducts (*35*–*37*). However, in the present study, *N*-NO-bile acid conjugates preferentially

Scheme 1. Possible Reaction Mechanisms of *N*-Nitroso Bile Acid Conjugates and DNA



attacked the amino group at the N^4-position of dC and N^6-position of dA. Because the most reactive exocyclic amino group is known to be the N^4-position of dC, followed by the N^6-position of dA and the N^2-position of dG, it is likely that *N*-NO-bile acid conjugates could form N^4-cholyl-dC or N^6-cholyl-dA, rather than a dG adduct. Moreover, there are some reports describing the reaction between alkylating agents and cytidine or adenosine, and the N^3- and N^4-positions of dC and N^6-position of dA have been alkylated predominantly (*38*–*42*). In addition, the rearrangement of N^3-(deoxy)cholyl-dC or N^1-cholyl-dA to N^4-cholyl-dC or N^6-cholyl-dA is not ruled out. The mechanisms underlying formation of CA- or DCA-DNA adducts remain unclear. To understand the mechanisms for the formation of bile acid-DNA adducts, further study is needed.

Furthermore, we here demonstrated *N*-nitroso bile acid conjugates to induce mutations and their mutation spectrum in *S. typhimurium*. NO-TCA and NO-TDCA induced revertants to TA100, which detects base pair change mutations, but not in TA98, a detector of frameshift mutations. Moreover, NO-TCA induced G:C to A:T transitions predominantly in TA100, and most of them were CCC to CTC. This suggests that 3-ethanesulfonic acid- and N^4-cholyl-dC lead to G:C to A:T transitions. At present, there are no data to explain which adduct, ethanesulfonic acid- or CA-dC, is mainly responsible in TA100, and further studies are clearly warranted to clarify the mechanisms of mutagenicity.

When NO-TCA was injected to Wistar rats at a dose of 250 mg/kg, ethanesulfonic acid- and CA-DNA were detected in the glandular stomach and colon mucosa but not the liver, under the conditions used in the present study. While levels of these two kinds of adducts greatly differed, there was little variation between the two organ sites. Because the yield of CA-DNA in vitro was about 90 times lower than that of 3-ethanesulfonic acid-dC, it is reasonable that the yield of CA-DNA in vivo was also much lower than that of ethanesulfonic acid-DNA. Moreover, it has been reported that NO-TCA is not stable under physiological conditions and thus might easily produce the chemically active compound, alkyl diazoate, and inactive compounds, such as free bile acids (*27*). In the present study, NO-TCA was singly administered to rats by intragastric intubation and the stomach and colon mucosa would have been exposed to decomposition products. However, active compound apparently did not reach to the hepatic cells or only at very low levels. Alternatively, NO-TCA was efficiently inactivated by enzymes in the liver. It should be borne in mind that Busby et al. previously demonstrated NO-TCA to induce liver and stomach tumors in rats (*16*), with administration by gavage for 6 consecutive weeks with a total of 300 mg compound per rat. To clarify the tissue distribution of ethanesulfonic acid-dC and CA-DNA, an increase in the amount of NO-TCA and/or differing administration routes need to be explored.

Nitrosation of bile acid conjugates can be mediated by the acid-catalyzed reaction of amides with nitrite (*14*) and activated macrophages in infected and inflamed organs also believed to be involved in such reactions. Thus, nitrosated bile acid conjugates might contribute to human cancer development as endogenous mutagens. It is well-documented that G to A base substitutions are frequently observed in cancer related genes, such as *ras* family members and *p53* (*43*–*45*). Chemical instability might explain why nitroso bile acid derivatives have not been detected in any biological samples so far tested (*46*). Targeting specific DNA adducts, such as 3-ethanesulfonic acid-dC, N^4-cholyl-dC, N^4-deoxycholyl-dC, and N^6-cholyl-dA, therefore appears to be a useful way of monitoring endogenous formation of nitrosated bile acid conjugates. To clarify involvement of these nitroso bile acid conjugates in human carcinogenesis, it is clearly necessary to evaluate exposure levels.

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Supporting Information Available: HMBC spectrum of ethanesulfonic acid-dC adduct and various spectral data of DCA-dC. This material is available free of charge via the Internet at http://pubs.acs.org.

References

(1) Debruyne, P. R., Bruyneel, E. A., Li, X., Zimber, A., Gespach, C., and Mareel, M. M. (2001) The role of bile acids in carcinogenesis. *Mutat. Res. 480–481*, 359–369.

(2) de Kok, T. M., and van Maanen, J. M. (2000) Evaluation of fecal mutagenicity and colorectal cancer risk. *Mutat. Res. 463*, 53–101.

(3) Owen, R. W. (1997) Faecal steroids and colorectal carcinogenesis. *Scand. J. Gastroenterol. 222* (Suppl.), 76–82.

(4) Kishida, T., Taguchi, F., Feng, L., Tatsuguchi, A., Sato, J., Fujimori, S., Tachikawa, H., Tamagawa, T., Yoshida, Y., and Kobayashi, M. (1997) Analysis of bile acids in colon residual liquid or fecal material in patients with cololectal neoplasia and control subjects. *J. Gastroenterol. 32*, 306–311.

(5) Deschner, E. E., Cohen, B. I., and Raicht, R. F. (1981) Acute and chronic effect of dietary cholic acids on colonic epithelial cell proliferation. *Digestion 21*, 290–296.

(6) Bartram, H. P., Scheppach, W., Schmid, H., Hofmann, A., Dusel, G., Richter, F., Richter, A., and Kasper, H. (1993) Proliferation of human colonic mucosa as an intermediate biomarker of carcinogenesis: Effects of butyrate, deoxycholate, calcium, ammonia, and pH. *Cancer Res. 53*, 3283–3288.

(7) Mcmillan, L., Butcher, S., Wallis, Y., Neoptolemos, J. P., and Lord, J. M. (2000) Bile acids reduce the apoptosis-inducing effects of sodium butyrate on human colon adenoma (AA/CC) cells: Implication for colon carcinogenesis. *Biochem. Biophys. Res. Commun. 273*, 45–49.

(8) Narisawa, T., Magadia, N. E., Weisburger, J. H., and Wynder, E. L. (1974) Promoting effect of bile acids on colon carcinogenesis after intrarectal instillation of *N*-methyl-*N'*-nitrosoguanidine in rats. *J. Natl. Cancer Inst. 53*, 1093–1097.

(9) Reddy, B. S., Watanabe, K., Weisburger, J. H., and Wynder, E. L. (1977) Promoting effect of bile acids in colon carcinogenesis in germ-free and conventional F344 rats. *Cancer Res. 37*, 3228–3242.

(10) Hamada, K., Umemoto, A., Kajikawa, A., Seraj, M. J., and Monden, Y. (1994) *In vitro* formation of DNA adducts with bile acids. *Carcinogenesis 15*, 1911–1915.

(11) Scates, D. K., Spigelman, A. D., and Venitt, S. (1994) Bile acids do not form adducts when incubated with DNA in vitro. *Carcinogenesis 15*, 2945–2948.

(12) Scates, D. K., Spigelman, A. D., and Venitt, S. (1995) Appearance of artifacts when using ^{32}P-postlabeling to investigate DNA adduct formation by bile acids in vitro: Lack of evidence for covalent binding. *Carcinogenesis 16*, 1489–1491.

(13) Scates, D. K., Spigelman, A. D., Phillips, R. K. S., and Venitt, S. (1997) The use of ^{32}P-postlabeling in studies of the nature and origin of DNA adducts formed by bile from patients with familial adenomatous polyposis and from normal patients. *Mutat. Res. 378*, 113–125.

(14) Shuker, D. E. G., Tannenbaum, S. R., and Wishnok, J. S. (1981) *N*-Nitroso bile acid conjugates. 1. Synthesis, chemical reactivity, and mutagenic activity. *J. Org. Chem., 46*, 2092–2096.

(15) Puju, S., Shuker, D. E. G., Bishop, W. W., Falchuk, K. R., Tannenbaum, S. R., and Thilly, W. G. (1982). Mutagenicity of *N*-nitroso bile acid conjugates in *Salmonella typhimurium* and diploid human lymphoblasts. *Cancer Res. 42*, 2601–2604.

(16) Busby, W. F., Shuker, D. E. G., Charley, G., Newberne, P. M., Tannenbaum, S. R., and Wogan, G. N. (1985) Carcinogenicity in rats of the nitrosated bile acid conjugates: *N*-Nitrosoglycocholic acid and *N*-nitrosotaurocholic acid. *Cancer Res. 45*, 1367–1371.

(17) Miwa, K., Hasegawa, H., Fujimura, T., Matsumoto, H., Miyata, R., Kosaka, T., Miyazaki, I., and Hattori, T. (1992) Duodenal reflux through the pylorus induces gastric adenocarcinoma in the rat. *Carcinogenesis 13*, 2313–2316.

(18) Miwa, K., Kinami, S., Miyazaki, I., and Hattori, T. (1996) Positive association between dietary fat intake and risk of gastric stump carcinoma in rats. *Carcinogenesis 17*, 1885–1889.

(19) Kumagai, H., Mukaisyo, K., Sugihara, H., Miwa, K., Yamamoto, G., and Hattori, T. (2004) Thioproline inhibits development of esophageal adenocarcinoma induced by gastroduodenal reflux in rats. *Carcinogenesis 25*, 723–727.

(20) Caygill, C. P., Hill, M. J., Kirkham, J. S., and Northfield, T. C. (1986) Mortality from gastric cancer following gastric surgery for peptic ulcer. *Lancet 1*, 929–931.

(21) Viste, A., Bjornestad, E., Opheim, P., Skarstein, A., Thunold, J., Hartveit, F., Eide, G. E., Eide, T. J., and Soreide, O. (1986) Risk of carcinoma following gastric operations for benign disease: A historical cohort study of 3470 patients. *Lancet 2*, 502–504.

(22) Lundegardh, G., Adami, H. O., Helmick, C., Zack, M., and Meirik, O. (1988) Stomach cancer after partial gastrectomy for benign ulcer disease. *N. Engl. J. Med. 319*, 195–200.

(23) Toftgaard, C. (1989) Gastric cancer after peptic ulcer surgery. A historic prospective cohort investigation. *Ann. Surg. 210*, 159–164.

(24) Street J. M., Trafford, D. J. H., and Makin H. L. J. (1983) The quantitative estimation of bile acids and their conjugates in human biological fluids. *J. Lipid Res. 24*, 491–511.

(25) Scalia, S. (1988) Simultaneous determination of free and conjugated bile acids in human gastric juice by high-performance liquid chromatography. *J. Chromatogr. 431*, 259–269.

(26) Mirvish, S. S. (1995) Role of *N*-nitroso compounds (NOC) and *N*-nitrosation in etiology of gastric, esophageal, nasopharyngeal and bladder cancer and contribution to cancer of known exposures to NOC. *Cancer Lett. 93*, 17–48.

(27) Dayal, B., and Ertel, N. H. (1997) Studies on *N*-nitroso bile acid amides in relation to their possible role in gastrointestinal cancer. *Lipids 32*, 1331–1340.

(28) Shuker, D. E. G., and Margison, G. P. (1997) Nitrosated glycine derivatives as a potential source of O^6-methylguanine in DNA. *Cancer Res. 57*, 366–369.

(29) Takamura, T., Sugimura, T., and Wakabayashi, K. (2004) Formation of DNA adducts by reactive intermediates, bile acid-adenylate. Proceedings of 63rd Annual Meeting of the Japanese Cancer Association, p 85.

(30) Reddy, M. V., and Randerath, K. (1986) Nuclease P1-mediated enhancement of sensitivity of ^{32}P-postlabeling test for structurally diverse DNA adducts. *Carcinogenesis 7*, 1543–1551.

(31) Takamura-Enya, T., Watanabe, M., Totsuka, Y., Kanazawa, T., Matsushima-Hibiya, Y., Koyama, K., Sugimura, T., and Wakabayashi, K. (2001) Mono(ADP-ribosyl)ation of 2′-deoxyguanosine residue in DNA by an apoptosis-inducing protein, pierisin-1, from cabbage butterfly. *Proc. Natl. Acad. Sci. U.S.A. 98*, 12414–12419.

(32) Gupta, R. C. (1985) Enhanced sensitivity of ^{32}P-post-labeling analysis of aromatic carcinogen: DNA adducts. *Cancer Res. 45*, 5656–5662.

(33) Yahagi, T., Nagao, M., Seino, Y., Matsushima, T., Sugimuta, T., and Okada, M. (1977) Mutagenicities of *N*-nitrosoamines on *Salmonella*. *Mutat. Res. 48*, 121–130.

(34) Dayal, B., Bhojawala, J., Rapole, K. R., Pramanik, B. N., Ertel, N. H., Shefer, S., and Salen, G. (1996) Chemical synthesis, structural analysis, and decomposition of *N*-nitroso bile acid conjugates. *Bioorg. Med. Chem. 4*, 885–890.

(35) Chen, S. C., Hilton, B. D., Roman, J. M., and Dipple, A. (1989) DNA adducts from carcinogenic and noncarcinogenic enantiomers of benzo[*a*]pyrene dihydrodiol epoxide. *Chem. Res. Toxicol. 2*, 334–340.

(36) Sayer, J. M., Chadha, A., Agarwal, H. S. K., Yeh, H. J. C., Yagi, H., and Jerina, D. M. (1991) Covalent nucleoside adducts of benzo[*a*]pyrene 7,8-diol 9,10-epoxides: Structural reinvestigation and characterization of a novel adenosine adduct on the ribose moiety. *J. Org. Chem. 56*, 20–29.

(37) Snyderwine, E. G., and Turteltaub, K. W. (2000) Interactions with cellular macromolecules. In *Food Borne Carcinogenic Heterocyclic Amines* (Nagao, M., and Sugimura, T., Eds.) pp 131–161, John Wiley & Sons Ltd., West Sussex.

(38) Brookes, P., and Lawley, P. D. (1962) The methylation of cytosine and cytidine. *J. Chem. Soc.* 1348.

(39) Sun, L., and Singer, B. (1974) Reaction of cytidine with ethylating agents. *Biochemistry 13*, 1905–1913.

(40) Singer, B., Sun, L., and Fraenkel-Conrat, H. (1974) Reaction of adenosine with ethylating agents. *Biochemistry 13*, 1913–1920.

(41) Singer, B. (1976) O^2-Alkylcytidine—A new major product of neutral, aqueous reaction of cytidine with carcinogens. *FEBS Lett. 63*, 85–88.
(42) Segal, A., Solomon, J. J., Mignano, J., and Dino, J. (1981) The isolation and characterization of 3-(2-carboxyethyl)cytosine following in vitro reaction of b-propiolactone with calf thymus DNA. *Chem. Biol. Interact. 35*, 349–361.
(43) Smith, G., Crey, F. A., Beattie, J., Wilkie, M. J., Lightfoot, T. J., Coxhead, J., Garner, R. C., Steele, R. J. C., and Wolf, R. (2002) Mutations in APC, Kirsten-ras, and p53-alternative genetic pathways to colorectal cancer. *Proc. Natl. Acad. Sci. U.S.A. 99*, 9433–9438.
(44) Gealy, R., Zhang, L., Siegfried, J. M., Luketich, J. D., and Keohavong, P. (1999) Comparison of mutations in the p53 and K-ras genes in lung carcinomas from smoking and nonsmoking women. *Cancer Epidemiol., Biomarkers Prev. 8*, 297–302.
(45) Olivier, M., Hussain, S. P., de Fromentel, C. C., Hainaut, P., and Harris, C. C. (2004) TP53 mutation spectra and load: A tool for generating hypotheses on the etiology of cancer. In *Mechanisms of Carcinogenesis: Contributions of Molecular Epidemiology* (Buffler, P., Rice, J., Baan, R., Bird, M., and Boffetta, P., Eds.) IARC Scientific Publications No. 157, pp 247–270, IARC, Lyon.
(46) Fein, M., Fuchs, K.-H., Stopper, H., Diem, S., and Herderich, M. (2000) Duodenogastric reflux and foregut carcinogenesis: Analysis of duodenal juice in a rodent model of cancer. *Carcinogenesis 21*, 2079–2083.

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Formation of DNA Adducts with Cholyl Adenylate, a Putative Intermediate for Biosynthesis of Cholyl-CoA

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Cholyl adenylate is a putative intermediate for biosynthesis of cholic acid-coenzyme A (CoA) thioester conjugates by acyl-CoA synthetase. Early studies showed the conjugated acid anhydride moiety of cholyl adenylate to be reactive, attacking proteins to form protein-cholic acid adducts. In the present study, to clarify reactions of cholyl adenylate with DNA under physiological conditions, products with nucleosides were analyzed. HPLC-MS analyses indicated cholyl adenylate to primarily attack hydroxy groups of ribose moieties of nucleosides. Moreover, as speculated from UV and MS studies, exocyclic amino groups of 2′-deoxycytidine and 2′-deoxyadenosine were found to serve as targets of cholyl adenylate; the corresponding cholic amides, N^4-cholyl-2′-deoxycytidine and N^6-cholyl-2′-deoxyadenosine, were formed at yields of 0.32 and 0.06%, respectively. Structures of these base modified adducts were confirmed by direct comparison with synthetic compounds obtained from coupling reactions of cholic acid with each nucleoside in the presence of dicyclohexylcarbodiimide in pyridine at 70 °C. N^4-Cholyl-2′-deoxycytidine was also obtained at a level of 1.6 adducts per 10^5 nucleosides from enzymatic hydrolysates of calf thymus DNA reacted with cholyl adenylate. These results suggest that cholyl adenylate, released from CoA synthetase, may have some possibility as a DNA modifier in vivo.

Introduction

Bile acids are synthesized de novo in the liver from cholesterol and conjugated with glycine or taurine. They are stored in the gall bladder and excreted into the upper intestinal tract to promote absorption of dietary lipid materials by micellar formation. Most are reabsorbed from the ileum-proximal colon and returned to the liver. Remaining bile acids are changed by intestinal bacteria to secondary bile acids, which are absorbed in the colon to join in the enterohepatic circulation (*1*, *2*).

It has been postulated that bile acids play some role in carcinogenesis, especially contributing to the risk of colon cancer (*3*, *4*). Unconjugated deoxycholic acid and chenodeoxycholic acid have promoting effects on colon cancer development in rodent carcinogenesis models (*5*, *6*), and concentrations of bile acids have been reported to be elevated in patients with adenomatous polyps or colon cancer (*7*, *8*). Bile from familial adenomatous polyposis patients was further shown to contain constituents that form DNA adducts (*9*). In vitro direct modification of DNA with some bile acids was also demonstrated with ^{32}P-postlabeling methods (*10*). Moreover, mutagenicity of polycyclic aromatic hydrocarbons is reported to be enhanced with glycocholic acid and taurodeoxycholic acid (*11*), and comet assays have revealed DNA damage in cells treated with deoxycholic acid (*12*). However, there have been no reports of determination of chemical structures of DNA adducts modified with bile acids.

Cholyl adenylate is a putative intermediate in the biosynthesis of CoA thioester conjugates of cholic acid, one of the primary bile acids synthesized in the liver and excreted as a taurine or glycine conjugate (*13*−*16*) (Scheme 1). During synthesis of the amino acid conjugates, cholic acid is converted to a corresponding coenzyme A (CoA)[1] thioester by the catalytic action of acyl-CoA synthetase, followed by amide formation with amino acids catalyzed by *N*-acyltransferase (*1*, *2*, *17*−*19*). Generally, CoA thioester formation in acyl-CoA synthetases affords an acyl adenylate as a highly active intermediate where the adenosine monophosphate (AMP) moiety of ATP is covalently transferred to a carboxyl group of substrates (*20*, *21*). Cholyl adenylate is, thus, a putative intermediate during the synthesis of CoA thioesters. Formation of this intermediate is well-documented from in vitro experiments where cholic acid was converted to cholyl adenylate by cell extracts of intestinal *Eubacterium* and also by a rat liver microsomal fraction (*13*). It has also been shown that cholyl adenylate is sufficiently

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[1] Abbreviations: CoA, coenzyme A; dA, 2′-deoxyadenosine; dC, 2′-deoxycytidine; dG, 2′-deoxyguanosine; T, thymidine; DCC, dicyclohexylcarbodiimide; THF, tetrahydrofuran; TBAF, tetrabutylammonium fluoride.

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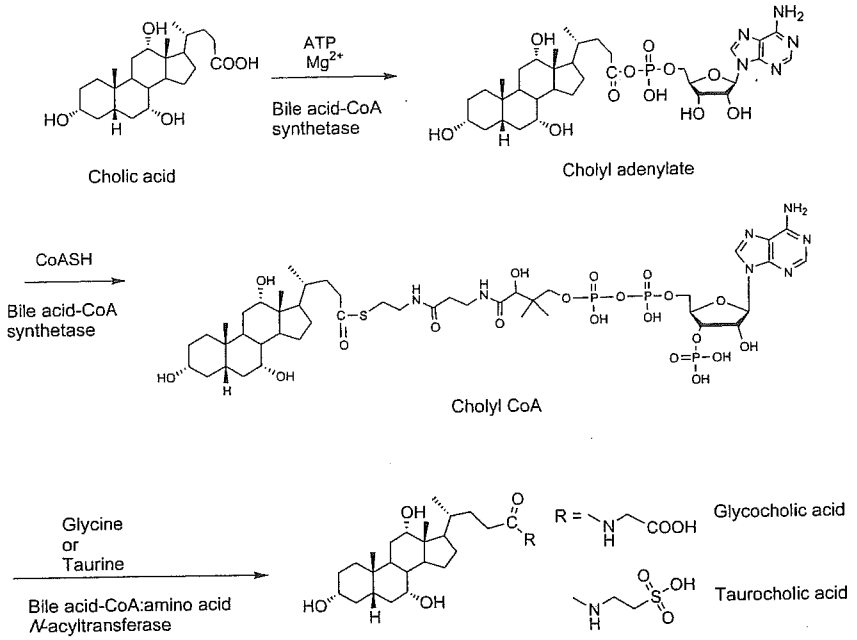
Scheme 1. In Vivo Pathway of Formation of Amino Acid Conjugates of Cholic Acid



reactive to attack taurine nonenzymatically (*13*). Moreover, reactions of cholyl adenylate with lysozyme clearly yield protein-bound cholic acids involving lysine residues at codons 1, 33, 97, and 116 (*15*). As an analogous bile acid-adenylate compound, deoxycholyl adenylate has also been demonstrated to form protein adducts with histone H3 (*16*). Bile acid-protein adducts were already found in in vivo samples, implicating the probable presence of a corresponding bile acid-adenylate intermediate in vivo (*22*).

Considering its reactivity and acid anhydride nature, cholyl adenylate would be expected to attack cellular DNA to form DNA adducts. In the present study, we analyzed reaction products of nucleosides and found cholyl adenylate to attack exocyclic amino groups of 2′-deoxycytidine (dC) and 2′-deoxyadenosine (dA). The structures of the resulting nucleoside adducts were further confirmed by direct comparison of independent synthetic adducts. In addition, dC-cholic acid adducts were found to be formed by reactions of calf thymus DNA with cholyl adenylate. On the basis of these results, possible in vivo formation of DNA adducts with cholic acid is discussed.

Experimental Procedures

Materials. Cholyl adenylate was synthesized as described earlier (*13*), and its purity was determined to be 98% by LC-MS analysis. The main impurities were cholic acid and AMP, formed by decomposition. Calf thymus DNA, dC, dA, 2′-deoxyguanosine (dG), thymidine (T), and bovine intestinal and bacterial alkaline phosphatases were all from Sigma Chemical Co. (St. Louis, MO). ^{1}H NMR, ^{13}C NMR, ^{1}H−^{1}H-correlated spectroscopy (2D-COSY), heteronuclear multiple quantum coherence (HMQC), and heteronuclear multiple bond correlations (HMBCs) were recorded on a JEOL α-600 (600 MHz) or a JEOL α-500 (500 MHz) spectrometer. Chemical shifts are reported in parts per million (ppm), and J values are given in Hz. HPLC-ESI-MS was performed with a Waters ZQ 2000 mass single stage spectrometer equipped with an Agilent 1100 HPLC system.

Reaction of Deoxynucleosides and Cholyl Adenylate. Aliquots of dC, dG, dA, or T (40 mM) were incubated with cholyl adenylate (20 mM) at 37 °C, pH 7.0, in 100 mM phosphate buffer. The overnight reaction mixture was then applied to LC-ESI-MS on an ODS column (Imtakt Cadenza CD-C18, 4.6 mm × 75 mm, Kyoto, Japan) with a linear gradient during the course of 30 min from 0 to 80% acetonitrile in 50 mM HCOOH−NH_4-OH buffer solution at pH 7.0 with a flow rate of 1.0 mL/mim. ESI-MS analysis of total current chromatographs was performed with positive ion detection with a cone voltage of 30 V. Selective molecular ions of *m/z* 618 for dC modified with cholic acid, *m/z* 641 for dA modified with cholic acid, *m/z* 658 for dG modified with cholic acid, and *m/z* 632 for T modified with cholic acid were monitored at a cone voltage of 30 V with positive ion detection. Molecular ions arising from loss of the deoxyribose moiety from the parent compounds (*m/z* 116 minus from mass of the parent compounds) were monitored with sequential rapid exchange of the initial cone voltage of 30−50 V. UV chromatograms were also recorded at a wavelength of 260 nm.

Reaction of Calf Thymus DNA with Cholyl Adenylate. Calf thymus DNA (2 mg) was reacted with cholyl adenylate (20 mM) at 37 °C in 1 mL of 100 mM phosphate buffer (pH 7.0) for 24 h. After the reaction, DNA was recovered by ethanol precipitation and digested enzymatically to nucleosides with deoxyribonuclease I, followed by alkaline phosphatase and phosphodiesterase I (*23*). The reaction mixture was then extracted with water-saturated butanol, and the organic layer was recovered and evaporated. Residues were redissolved in 20% acetonitrile and subjected to HPLC conditioned as for the analysis of nucleosides with cholyl adenylate.

Synthesis of N^4-Cholyl-2′-deoxycytidine. A mixture of 0.1 mmol of 3′,5′-bis-*O*-*tert*-butyldimethylsilyl-2′-deoxycitiydine, obtained with a published method (*24*), cholic acid (0.1 mmol), and dicyclohexylcarbodiimide (DCC, 0.12 mmol) in pyridine (10 mL) was stirred at 70 °C for 4 h. After the precipitate was filtered off, the solvent was evaporated. The residue was applied to silica gel column chromatography (35 mm × 250 mm) with a stepwise gradient of methanol/chloroform (0−20%) to give silyl-protected N^4-cholyl-dC as a white solid. This material was dissolved in 5 mL of tetrahydrofuran (THF) and further treated with 1 M tetrabutylammonium fluoride (TBAF) in THF (500 μL). After the reaction mixture was stirred overnight at room temperature and then evaporated, the residue was further purified by column chromatography using ODS (26 mm × 300 mm) with methanol/water (10−80%) to give the desired compound as a white solid with a yield of 20%. ^{1}H NMR (500 MHz, DMSO-d_6): δ 10.79 (s, 1H, NHCO), 8.29 [d, J = 7.5 Hz, 1H, H-6 (dC)], 7.19 [d, J = 7.5 Hz, 1H,H-5 (dC)], 6.09 [t, J = 6.2 Hz, 1H, H-1′(dR)], 5.24 [brs, 1H, 3′-OH (dR)], 5.06 [brs, 1H, 5′-OH (dR)], 4.27 [d, J = 4.5 Hz,

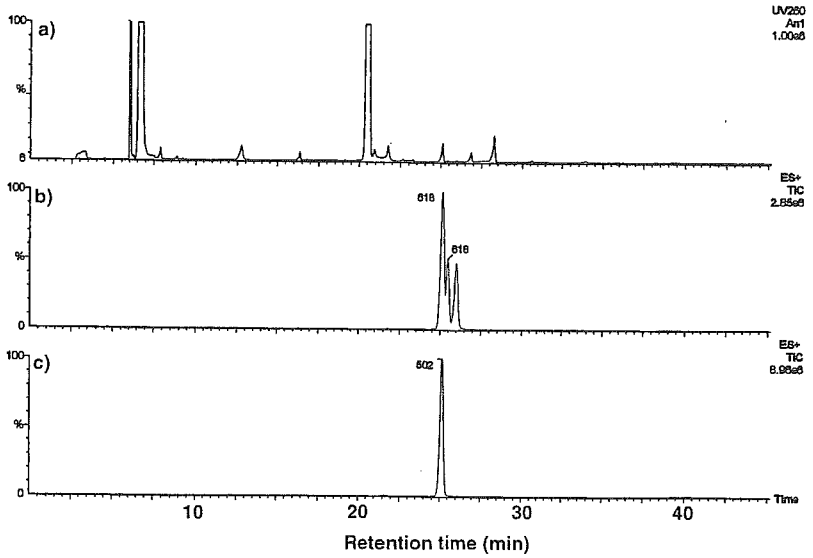


Figure 1. LC-ESI-MS chromatogram of reaction products of cholyl adenylate with dC. Cholyl adenylate (20 mM) was incubated with dC (20 mM) at 37 °C, pH 7.0, in 100 mM phosphate buffer. The overnight reaction mixture was then applied to LC-ESI-MS. HPLC conditions are shown in the Experimental Procedures. (a) UV chromatogram at 260 nm; (b) selected ion monitoring at *m/z* 618, which is estimated to be a molecular ion of dC modified with cholic acid; and (c) selected ion monitoring at *m/z* 502 derived from loss of the deoxyribose moiety from the parent dC-cholic acid adduct with a molecular ion of *m/z* 618.

1H, 3-OH(CA)], 4.20 [dt, J = 3.5, 5.5 Hz, 1H, H-3′(dR)], 4.07 [d, J = 3.0 Hz, 1H, 12-OH (CA)], 3.97 [d, J = 3.0 Hz, 1H, 7-OH (CA)], 3.84 [dt, J = 3.5, 4.0 Hz, 1H, H-4′(dR)], 3.77 [brs, 1H, H-12(CA)], 3.65−3.53 [m, 3H, H-7(CA), H-5′, 5″(dR)], 3.25−3.13 [m, 1H, H-3(CA)], 2.45−2.14 [m, 5H, H-4α, −9, −23α, −23β-(CA), H-2′(dR)], 2.03−1.94 [m, 2H, H-14(CA), H-2″(dR)], 1.86−1.52 (m, 7H, H-17, 20, 22α, 16α, H-1α, H-6β, H-15β), 1.48−1.25 (m, 12H, H-2α, −2β, 4β, 5, 6α, 8, 11α, 11β, 15α, 16β, 22β), 1.0−0.92 (m, 1H, H-1β), 0.91 [d, J = 6.0 Hz, 3H, CH_3-21(CA)], 0.79 [s, 3H, CH_3-19(CA)], 0.57 [s, 3H, CH_3-18(CA)]. ^{13}C NMR (125 MHz, DMSO-d_6): δ 174.4 [C=O(CA)], 162.3 [C-2(dC, C=O)], 154.4 [C-4(dC)], 144.8 [C-6(dC)], 95.2 [C-5(dC)], 87.6 [C-4′(dR)], 86.1 [C-1′(dR)], 70.9 [C-12(CA)], 70.4 [C-3(CA)], 69.9 [C-3′(dR)], 66.2 [C-7(CA)], 60.9 [C-5′(dR)], 46.2 [C-17(CA)], 45.7 [C-13(CA)], 41.5 [C-5(CA)], 41.3 [C-14(CA)], 40.8 [C-2′(dR)], 40−39 [C-4, −8 (CA) overlapped with the absorbance signal of DMSO], 35.3 [C-1(CA)], 35.1 [C-20(CA)], 34.8 [C-6(CA)], 34.3 [C-10(CA)], 33.7 [C-23(CA)], 30.9 [C-22(CA)], 30.4 [C-2(CA)], 28.5 [C-11(CA)], 27.2 [C-16(CA)], 26.2 [C-9(CA)], 22.7 [C-15(CA)], 22.6 [C-19(CA)], 13.4 [C-21(CA)], 12.3 [C-18(CA)]. ESI-MS *m/z* 618, 502. UV λ_{max} 243, 295 nm.

Synthesis of N^6-Cholyl-2′-deoxyadenosine. The same procedures described for N^4-cholyl-2′-deoxycytidine were performed with 3′,5′-bis-*O*-*tert*-butyldimethylsilyl-2′-deoxyadenosine (*25*) instead using 3′,5′-bis-*O*-*tert*-butyldimethylsilyl-2′-deoxycytidine. The desired compound was obtained at a yield of 7%. ^{1}H NMR (600 MHz, DMSO-d_6): δ 10.58 (s, 1H, NHCO), 8.62 [s, H-8(dA)], 8.61 [s, H-2(dA)], 6.43 [t, J = 6.6 Hz, 1H, H-1′-(dR)], 5.32 [brs, 1H, 3′-OH (dR)], 4.98 [brs, 1H, 5′-OH (dR)], 4.42 [brs, 1H, H-3′(dR)], 4.27 [d, J = 4.8 Hz, 1H, 3-OH(CA)], 4.07 [d, J = 3.6 Hz, 1H, 12-OH (CA)], 3.97 [d, J = 3.6 Hz, 1H, 7-OH (CA)], 3.88 [dt, J = 3.0, 4.2 Hz, 1H, H-4′(dR)], 3.79 [brs, 1H, H-12(CA)], 3.55−3.41 [m, 3H, H-7(CA), H-5′, 5″(dR)], 3.25−3.13 [m, 1H, H-3(CA)], 2.75 [dt, J = 6.0, 13.2, 1H, H-2′(dR)], 2.59 [ddd, J = 4.8, 9.6, 14.4 Hz, 1H, H-23α(CA)], 2.48−2.45 [m, 1H, H-23β(CA)], 2.32 [ddd, J = 4.8, 6.0, 13.2 Hz, 1H, H-2″(dR)], 2.21 (dt, J = 12.0, 14.2 Hz, 1H, H-4α), 2.14 [dt, J = 4.8, 12.0 Hz, 1H, H-9(CA)], 1.99 (ddd, J = 7.2, 12.1, 12.1 Hz, 1H, H-14), 1.86−1.52 (m, 7H, H-17, 20, 22α, 16α, H-1α, H-6β, H-15β), 1.48−1.25 (m, 12H, H-2α, −2β, 4β, 5, 6α, 8, 11α, 11β, 15α, 16β, 22β), 0.96 [d, J = 7.2 Hz, 3H, CH_3-21(CA)], 0.96−0.92 (m, 1H, H-1β), 0.80 [s, 3H, CH_3-19(CA)], 0.59 [s, 3H, CH_3-18(CA)]. ^{13}C NMR (150 MHz, DMSO-d_6): δ 172.0 [C=O(CA)], 162.3 [C-2(dC, C=O)], 151.5 [(C-2(dA)], 151.4 [C-4(dA)], 149.6 [C-6(dA)], 142.5 [C-8(dA)], 123.8 [C-5(dA)], 87.9 [C-4′(dR)], 83.7 [C-1′(dR)], 71.0 [C-12(CA)], 70.6 [C-3(CA)], 70.4 [C-3′(dR)], 66.2 [C-7(CA)], 61.6 [C-5′(dR)], 46.2 [C-17(CA)], 45.7 [C-13(CA)], 41.5 [C-5(CA)], 41.3 [C-14(CA)], 40−39 [C-4, −8(CA), C-2′(dR) overlapped with the absorbance signal of DMSO], 35.3 [C-1(CA)], 35.1 [C-20(CA)], 34.8 [C-6(CA)], 34.3 [C-10(CA)], 33.4 [C-23(CA)], 31.0 [C-22(CA)], 30.4 [C-2(CA)], 28.5 [C-11(CA)], 27.2 [C-16(CA)], 26.2 [C-9(CA)], 22.7 [C-15(CA)], 22.6 [C-19(CA)], 17.1 [C-21(CA)], 12.3 [C-18(CA)]. ESI-MS *m/z* 642, 526. UV λ_{max} 272 nm.

Results and Discussion

When cholyl adenylate was incubated with dG, dA, dC, and T at 37 °C for 24 h, in each reaction mixture, two (for dG and T) or three new peaks (for dC and dA) were observed in HPLC chromatograms detected at a wavelength of 260 nm after a retention time of 15 min; these peaks were not observed in control reaction mixtures without nucleosides. Each of these peaks had a molecular ion corresponding to dehydrated condensation products between the cholic acid and the nucleobase (*m/z* = cholic acid + nucleobase − H_2O). Judging from integrated knowledge of the reactivity of nucleobases with acid anhydride, the reaction sites of nucleobases toward cholyl adenylate are probably the hydroxy groups of ribose moieties and amino groups of the base moieties, generating esters or amides of cholic acid, respectively. Among the peaks that appeared in the HPLC chromatograms of the reaction mixture, our current interest is modification of the base rather than the deoxyribose moiety, because hydroxy groups in the latter are generally fully protected with phosphorous acid in DNA molecules. With the aid of LC-ESI-MS analyses, we were able to select base modification peaks, which always accompanied the molecular ion peaks derived from the elimination of the deoxyribose moiety from the parent compounds. With reaction mixtures of dG or T with cholyl adenylate, the desired peaks were not obtained, indicating that only the deoxyribose moiety was modified with cholyl adenylate. On the other hand, in the reaction mixtures with dC or dA, base modification peaks were clearly observed. The yield of formation of the base-modified dC adduct was about 0.32%, which was about five times higher than that

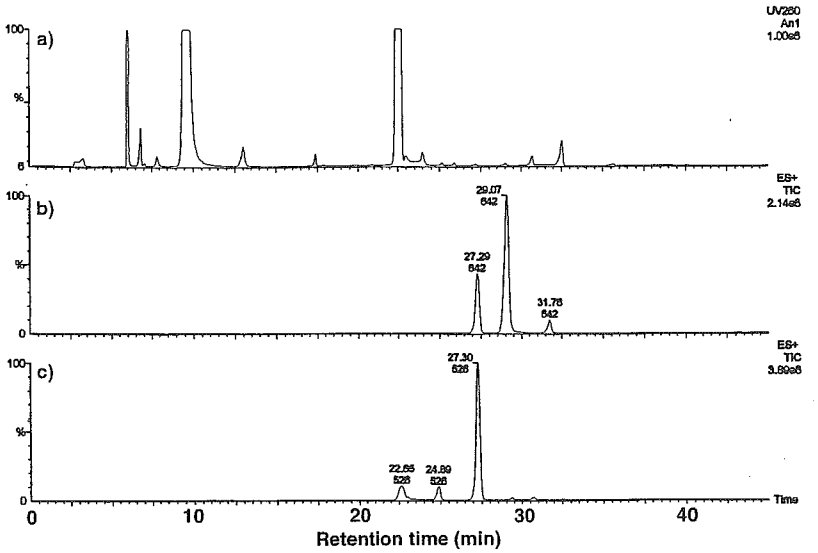


Figure 2. LC-ESI-MS chromatogram of reaction of cholyl adenylate with dA. Cholyl adenylate (20 mM) was incubated with dA (20 mM) at 37 °C, pH 7.0, in 100 mM phosphate buffer. The overnight reaction mixture was then applied to LC-ESI-MS. HPLC conditions are shown in the Experimental Procedures. (a) UV chromatogram at 260 nm; (b) selected ion monitoring at *m/z* 642, which is estimated to be a molecular ion of dA modified with cholic acid; and (c) selected ion monitoring at *m/z* 526, derived from loss of the deoxyribose moiety from the parent dA-cholic acid adduct with a molecular ion of *m/z* 642.

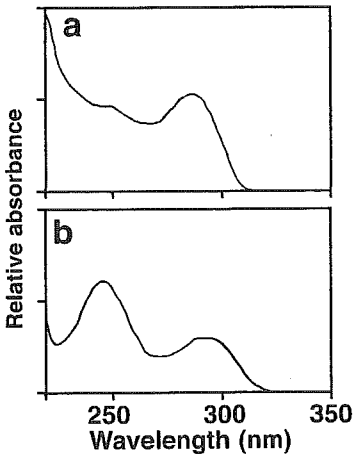


Figure 3. UV spectra of dC (a) and of an adduct from dC with cholyl adenylate (b) recorded on a Shimadzu VPD photoodiode array detector.

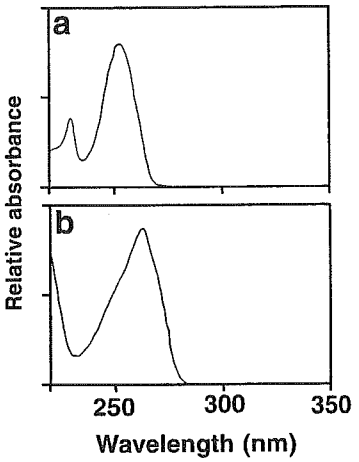
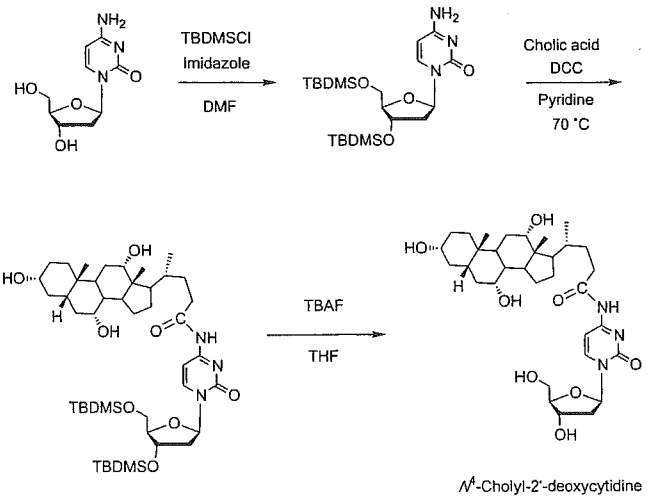


Figure 4. UV spectra of dA (a) and of an adduct from dA with cholyl adenylate (b) recorded on a Shimadzu VPD photoodiode array detector.

of the dA adduct (0.06%), as estimated from chromatograms of UV absorbance at 260 nm. The reaction of dC with cholyl adenylate yielded a base modified compound at a retention time of 25.0 min on HPLC chromatograms, with a molecular ion peak of *m/z* 618 accompanied with 502 (*m/z* 618-dR) (Figure 1). In the case of dA with cholyl adenylate, a peak was detected at a retention time in 27.3 min with molecular ion peaks of *m/z* 642 and 526 (*m/z* 642-dR) (Figure 2). The UV spectra of these adducts were drastically changed from those of the original compounds and were similar to the patterns of acyl amides of dC and dA, indicating cholyl adenylate to attack exocyclic amino groups of dC and dA to form N^4-cholyl-dC and N^6-cholyl-dA, respectively (Figures 3 and 4).

To confirm chemical structures of these adducts, the deduced compounds, dC- and dA-cholamide, were synthesized as outlined in Schemes 2 and 3, respectively. Because of the difficulty of fully protecting hydroxy groups, cholic acid was applied without protection. Silyl-protected dC was reacted with cholic acid under the conditions of DCC in pyridine at a reaction temperature of 70 °C. Half the amount of dC remained intact on TLC

Scheme 2. Synthesis of N^4-Cholyl-dC



analyses after 4 h of reaction, and with a more prolonged reaction time, coupling did not proceed further. The desired product was easily separated from silyl-protected